District-Wide Watershed Management Plan

Standards and Methodologies
for Surface Water Quality Monitoring

Metropolitan North Georgia
Water Planning District

March 2007

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March 2007
Modified April 6, 2007

Metropolitan North Georgia Water Planning District
40 Courtland Street NE
Atlanta, Georgia 30303

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Preface

The legislation that created the Metropolitan North Georgia Water Planning District (Official Code of Georgia Annotated §12-5-574) charges the District with promoting regional coordination and cooperation through the *"development and coordination of an effective regional and watershed specific water quality monitoring program and development and maintenance of a corresponding data base reflecting available monitoring data."* Furthermore, §12-5-582 requires that *"appropriate standards and methodologies for monitoring water quality and maintaining and organizing an inventory of collected water quality data"* be included as part of a District-wide Watershed Management Plan.

This document contains the standards and methodologies for surface water quality monitoring for communities in the Metropolitan North Georgia Water Planning District. These monitoring protocols have been developed in cooperation with the Georgia Environmental Protection Division and member local governments through the Stormwater Subcommittee of the District's Technical Coordinating Committee.

Part 1 of the document is an overview of the District's water quality monitoring plan and requirements for local governments that are contained in the District-wide Watershed Management Plan. Parts 2 through 5 contain the monitoring protocols for each element of the water quality monitoring program.

Please periodically check the District website at www.northgeorgiawater.org for updates to this document.

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Contents

PART 1
Introduction

This document has been prepared by the Metropolitan North Georgia Water Planning District (District) to serve as a technical reference for conducting surface water quality monitoring. It is intended to provide consistency in how water quality and biological data are collected, analyzed and managed by local governments and other entities. This guidance was assembled with input from the District Technical Coordinating Committee (TCC) and the Georgia Environmental Protection Division (EPD).

1.1 Monitoring Plan Overview

The District-wide Watershed Management Plan includes a water quality monitoring plan which provides for comprehensive and consistent watershed-based water quality monitoring across the District. The purpose of the monitoring is to assess water quality and watershed conditions, as well as to evaluate the effectiveness of the District-wide Watershed Management Plan (WMP).

Specific objectives of this monitoring include:

- **Documenting Water Quality Improvements and Effectiveness of the Watershed Management Program –** The goal of the District-wide WMP is to maintain or improve water quality and watershed conditions within and downstream of the District. Monitoring will allow decision-makers to determine the extent to which the recommended management strategies are helping to meet this goal.
- **Identifying Streams Requiring Further Action –** A number of streams within the District have been identified as not meeting their designated use. Monitoring is needed on a local scale to evaluate overall water quality and watershed conditions and provide information for local watershed improvement and restoration efforts. In addition, verification and delisting sampling is recommended for stream segments on the 303(d) list.

The Water Quality Monitoring Plan includes a number of different program elements to be undertaken by District local governments, the U.S. Geological Survey (USGS), and the District. Table 1.1 provides a summary of the various monitoring plan elements and responsible entities.

The District monitoring plan is intended to help local governments meet their existing regulatory monitoring requirements, including those of the National Pollutant Discharge Elimination System (NPDES) Municipal Separate Storm Sewer System (MS4) program, Total Maximum Daily Load (TMDL) program, and Georgia EPD watershed assessment plans (as shown in Table 1.2).

TABLE 1.1
Summary of Water Quality Monitoring Plan Elements and Responsibilities
Metropolitan North Georgia Water Planning District Watershed Management Plan

Responsible Entity	Program Element	Frequency	Methodology/Approach
Local Government			
	Long-Term Ambient Trend Monitoring	Eight grab or composite samples per year at each station. 1 baseflow and 3 wet weather samples collected during each season: summer (May–October) and winter (November–April). 16 grab samples per year for bacteria (fecal coliform & E. Coli) at each station.	1 Manual grab sampling -or- 2 EWI/EDI[1] composite-grab sampling -or- 3 Automated sampling -- Composite hydrograph sampling triggered by data loggers
	Dry Weather Illicit Discharge Screening[2]	Annual Inspections	Rotate sites as necessary based on data collected through water quality sampling
	Commercial/Industrial Inspection Program	Annual Inspections	Inspect a minimum of 5% of relevant industries/commercial operations each year
	Monitoring for Assessing TMDL Implementation and Delisting	As specified in the local TMDL Implementation Plan(s)	Sample for 303(d)/305(b) listed constituents
	Biological and Habitat Assessments	Every 5 years on a rotating basis	GAEPD/Wildlife Resources Division (WRD) methodology
Regional / Interjurisdictional			
US Geological Survey (USGS)	Regional Network/Mainstem Monitoring	Continuous	Real-time flow and water quality gages
		Approximately Monthly / Rain event-driven	Composite hydrograph sampling triggered by data loggers at real-time gages
District	Best Management Practice (BMP)/Restoration Project Effectiveness	Project dependent[3]	Project dependent[3]
	Database Development and Management	Ongoing	

[1] EWI/EDI – Equal Width Integrated or Equal Depth Integrated
[2] Streamwalk surveys can be substituted for dry weather outfall screenings
[3] Project-specific monitoring plans will be prepared to evaluate effectiveness of BMPs or restoration projects

It is the intent of the District Water Quality Monitoring Plan to consolidate the many current local monitoring requirements into a larger, comprehensive program that provides consistency in sampling methodologies and effort across the District, as was forseen in the legislation creating the District. This will provide the ability to better compare and analyze data across jurisdictional and watershed boundaries, as well as provide for reduced redundancies and the more effective use of financial resources. Accordingly, this document was designed to provide local governments with the necessary sampling and analysis standards and methodologies required to meet these objectives.

The elements of the District Water Quality Monitoring Plan are discussed in more detail in the remainder of this introduction.

TABLE 1.2
Water Quality Monitoring Plan Summary and Comparison with Requirements
Metropolitan North Georgia Water Planning District Watershed Management Plan

Program Element	Monitoring Requirements			
	District-wide WMP Effectiveness	NPDES MS4 Program	TMDL Implementation[1]	Watershed Assessment Plans[2]
Local Government-Based Monitoring				
Long-Term Ambient Trend Monitoring	X	X	X	X
Dry Weather Illicit Discharge Screening	X	X	X	
Commercial/Industrial Inspection Program	X	X	X	
Monitoring for Assessing TMDL Implementaton and Delisting Streams	X		X	
Biological/Habitat Assessment	X		X	X
Regional/Interjurisdictional Monitoring Activities				
Regional Network/Mainstem Monitoring	X			X
BMP Practice/Restoration Project Effectiveness	X		X	
Database Development and Management	X			X

[1] Varies depending on the TMDL pollutant and the local TMDL implementation plan

[2] Varies depending on the monitoring requirements in the local plan

1.2 Local Government-Based Monitoring

1.2.1 Long-Term Ambient Trend Monitoring

The overall goal of the District-wide Watershed Management Plan is to meet and maintain water quality standards and designated uses of streams and other water bodies within the District. Monitoring of local streams for long-term ambient trends provides a means of demonstrating progress toward water quality goals as the Plan is implemented. These data will also be used to help refine future management strategies to address watershed management and water quality protection within the District.

Long-term trend monitoring involves both wet weather and dry weather water quality sampling as well as bacteria (fecal coliform and E. Coli) monitoring at permanent stream sampling locations throughout the District. At each stream sampling location, a minimum of three wet weather samples and one dry weather (baseflow) sample will be collected during each of the two sampling seasons (summer and winter)—for a total of eight samples annually. Samples may be collected using simple grab sampling, time-weighted composite sampling, or flow-weighted composite sampling. The composite samples may be collected either manually or with automatic sampling equipment. The local government may use EWI/EDI grab sampling if they so choose. In addition, 16 bacteria grab samples (4 sets of 4 samples within a 30-day period) will be collected at each permanent station.

1.2.1.1 Distribution of Sampling Stations

Long-term trend monitoring will be performed by local governments at permanent in-stream sampling locations across the District. The number of permanent sampling stations is based on population. Within each county, one long-term trend station will be required for every 50,000 persons, with a minimum of two stations per county. Table 1.3 lists the number of sampling stations required by county based on 2000 population. It will be up to each county government and the municipalities within each county to work out the responsibilities and financial obligations for this monitoring.

TABLE 1.3
Long-Term Ambient Trend Water Quality Sampling Stations Based on 2000 Population Estimates
Metropolitan North Georgia Water Planning District Watershed Management Plan

County	2000 Population	Number of Stations
Bartow	76,019	2
Cherokee	141,903	3
Clayton	236,517	5
Cobb	607,751	13
Coweta	89,215	2
DeKalb	665,865	14
Douglas	92,174	2
Fayette	91,263	2
Forsyth	98,407	2
Fulton	816,006	17
Gwinnett	588,448	12
Hall	139,277	3
Henry	119,341	3
Paulding	81,678	2
Rockdale	70,111	2
Walton	60,687	2

It should be noted that many of the jurisdictions in the District have already submitted monitoring plans to the GAEPD as part of their NPDES permitting process for existing or new wastewater discharges or their MS4 stormwater permit. In many cases, the stream monitoring being performed under these programs will fulfill or partially fulfill a jurisdiction's portion of the District Long-term ambient trend monitoring program.

1.2.1.2 Selection of Sampling Locations

Strategic sampling station location will serve to provide information on long-term ambient trends in waters affected by both point source discharges and nonpoint source impacts. To promote consistency in sampling across the District, the following guidelines are recommended for siting long-term water quality stations:

- Long-term trend monitoring location siting should take into consideration a number of factors including: watershed size; watershed land use; existing water quality conditions; stream characteristics; riparian ownership; accessibility; and proximity to point source and stormwater discharges, water supply intakes, solid waste facilities (landfills), land application systems, and septic service areas.

- Ideally, a monitoring site should be located on a stream with a medium-size watershed (approximately 5-30 square miles). Maximum watershed size should be limited to 12-digit Hydrologic Unit Codes (HUCs).
- Watersheds and streams should be selected that are representative of the land use and water quality conditions of the local jurisdiction. Special consideration should be given to impaired stream reaches.
- Major streams should be monitored near the entry to or exit from important land use areas, jurisdictional boundaries, or at critical stream reaches.
- Existing sites should be used (where appropriate) to preserve the "historical record" and facilitate trend evaluation.
- If possible, sites should be located near USGS gauging stations.
- Monitoring sites and equipment should be safely accessible from nearby roads, particularly during wet weather conditions.
- Sites should be located at least 50 feet below any major stormwater outfall, and 200 feet below a point source discharge, to ensure that well-mixed samples are collected.
- Sites should not be influenced by unnatural physical conditions associated with a stream crossing such as large pools or road runoff.

1.2.1.3 Sampling Frequency

To provide consistency in sampling within the District, the following guidelines will be used for long-term ambient water quality sampling:

- Wet Weather Monitoring: Flow weighted composite, time-weighted composite, or single grab samples will be collected six times per year for wet weather events. Three of these samples are to be collected in the summer (May-October) season and three in the winter (November-April) season. Representative wet weather events require a minimum precipitation of 0.3 inch. Additionally, a minimum period of 72 hours is required between each wet weather event sampled to ensure that the events are discrete and the water quality parameters are associated with the event sampled.
- Dry Weather Monitoring: Baseflow samples will be collected twice per year during dry weather. One of these samples is to be collected in the summer (May-October) season and one in the winter (November-April) season. Dry weather is defined as a period prior to sampling of at least 72 hours (3 days) receiving less than 0.1 inches of precipitation per day. Dry weather baseflow sampling is to be conducted by either manually operating an automated sampler or collecting a grab sample at the sampling station.
- Bacteria Monitoring: Grab samples for bacteria (fecal coliform and E. Coli) analysis will be collected 16 times per year. Four samples are to be collected over a 30-day period (at intervals not less than 24 hours) during each of the following quarters: May-July, August-October, November-January, and February-April. Each set of four samples will be used to calculate a geometric mean, per State guidelines.

1.2.1.4 Sample Collection Methods

Manual grab sampling methods may be used for small and medium streams and where samples during storm flows can be collected without danger of personal harm or injury. Manual grab samples may consist of either a single sample or composite samples. Manual grab sampling should be conducted on the rising limb of the hydrograph and as close to the peak as possible to more accurately estimate pollutant loadings during wet weather events. The sampling procedures and protocols for manual grab samples (simple and composite) are provided in Part 2B.

Equal Width-Integrated and Equal Depth-Integrated (EWI and EDI) grab sampling methods can be used to collect discharge-weighted, depth-integrated, isokinetic samples. The EWI/EDI methods and protocols are presented in Part 2C.

Automated sampling methods are designed to collect a flow-weighted composite, time-weighted composite, or single grab sample from a fixed location in the stream. The use of automated samplers is recommended for larger streams that cannot be safely accessed, especially during storm flows. However, they may also be installed on smaller streams if there is enough flow during a typical storm event to trigger the device. The sampling procedures and protocols for using automatic samplers are presented in Part 2D.

Measurement of stream flow during sampling events is important for estimating total pollutant loadings for that stream or tributary. Therefore, a stage-discharge rating curve should be developed at each station and flow should be recorded when water quality samples are taken.

1.2.1.5 Sampling Parameters

Both wet weather and dry weather baseflow samples will be analyzed for the following parameters:

- BOD_5
- COD
- TSS
- TDS
- Nitrate + Nitrite Nitrogen
- TKN
- Ammonia
- Hardness
- Total Phosphorus
- Orthophosphate (dissolved reactive phosphorus)
- Total / Dissolved Recoverable Metals (cadmium, copper, lead, and zinc)

Bacteria grab samples will be analyzed for both fecal coliform and E. Coli bacteria.

1.2.2 Illicit Discharge and Illegal Connection Screening

Illicit discharges are unpermitted non-stormwater flows to the stormwater drainage system that contain pollutants or pathogens. Illicit discharges can be direct discharges or dumping to the stormwater system, or can occur through upstream activities that eventually flow to storm drain or drainage channel. Illegal connections are physical connections such as pipes that allow illicit discharges to the stormwater system on an ongoing basis.

Illicit discharges are a major source of water quality impairment, and an illicit discharge detection and elimination (IDDE) program is a required activity under both Phase I and II NPDES MS4 stormwater permits.

To address this potential source of water quality problems, local governments within the District will be required to perform screenings in their jurisdiction to look for dry weather flows and potential illicit discharges. Two alternative approaches may be used for this screening: dry weather stormwater outfall screening or streamwalk surveys. In addition, local governments will be required to conduct inspections of commercial and industrial sites to inspect for illicit discharges and illegal connections. These activities are designed to help meet the local government requirements under their MS4 stormwater permits.

1.2.2.1 Dry Weather Outfall Screening

Screening of stormwater outfalls for illicit discharges is performed during periods of dry weather, which is defined as rainfall of less than 0.1 inch per day for at least 72 hours. This criterion avoids the screening of flows that may have resulted from wet weather (stormwater) events.

The number of outfall screening sites required per county is based on population rate of 1 site per 5,000 persons. Table 1.4 lists the minimum number of outfalls that should be examined annually in each county based on 2000 population estimates. It is recommended that communities screen more outfalls that the minimum if possible, especially where there are problem areas or known water quality violations due to illicit discharges. In order to provide a comprehensive screening of outfalls within a community, sites should be rotated on an annual basis.

TABLE 1.4
Minimum Number of Outfall Screening Sites Based on 2000 Population Estimates
Metropolitan North Georgia Water Planning District Watershed Management Plan

County	2000 Population	Number of Screening Sites
Bartow	76,019	16
Cherokee	141,903	29
Clayton	236,517	48
Cobb	607,751	122
Coweta	89,215	18
DeKalb	665,865	134
Douglas	92,174	19
Fayette	91,263	19
Forsyth	98,407	20
Fulton	816,006	164
Gwinnett	588,448	118
Hall	139,277	28
Henry	119,341	24
Paulding	81,678	17
Rockdale	70,111	15
Walton	60,687	13

Local governments should select screening locations based on the potential for illicit discharges. The following guidelines should be used to prioritize stormwater outfalls within a jurisdiction for dry weather screening of potential illicit connections:

- Utilize an up-to-date inventory of the city or county separate storm sewer system outfalls;
- Review records of previously screened outfalls to identify any subset of outfalls that have previously, and consistently, had illicit dry weather flows;
- Identify any new outfalls, or outfalls not previously screened, or outfalls identified by citizen complaints;
- Identify outfalls that drain into 303(d) listed waters, or have significant industrial land use, or discharge to streams with water quality concerns without obvious point sources;
- Rank previously screened outfalls by quarter since last screening; and
- Prioritize the set of outfalls for quarterly screening by adding the number of problem outfalls to the number of previously unscreened outfalls.

Outfalls are to be inspected at least quarterly (four times per year) for flow. When a dry weather flow is observed at an outfall, the following are to be performed on the flow:

1. Field observations and measurements – Site descriptions and qualitative observations of physical conditions of the outfall and flow, as well as measurement of several in-situ water quality parameters.
2. Water Quality Sampling – Collection of water quality samples for field analysis or laboratory analysis when indicated by the field observations & measurements.

The detection of one or more abnormal parameters may warrant a more detailed pollutant source identification investigation.

Part 3 contains detailed guidance on the dry weather outfall screening method and procedures.

1.2.2.2 Streamwalk Surveys

As an alternative to the dry weather outfall screening described above, local jurisdictions may develop a streamwalk survey program that involve an annual pedestrian reconnaissance of at least 10 percent of the stream miles in a jurisdiction. The streamwalk will be conducted during dry weather (at least 72 hours since last rainfall event) baseflow conditions. During the annual streamwalk, an inventory of all the pipes, outfalls, and ditches will be compiled. Field screening observations include a site description and a series of qualitative (mainly visual) observations of physical and biological conditions at the site.

If an illicit discharge or illegal connection is located or suspected, subsequent field screening observations and monitoring, and/or grab sampling would be performed similar to the dry weather outfall screening.

1.2.2.3 Commercial/Industrial Inspections

In addition to the outfall screening or streamwalk surveys, local governments will also be required to perform commercial and industrial inspections for illicit discharges and illegal connections. Illicit discharges from businesses and industry may come from a variety of sources, including exposed materials, wash waters, process wastewater and sanitary wastewater. To ensure that only stormwater is being discharged into the local stormwater system from a commercial/industrial site, local governments will perform site inspections and follow-up with sampling as necessary. A visual inspection will include locating discharges to the MS4 or local waters using visual observation and drainage schematics. If deemed necessary, field testing, sample collection, and laboratory analysis of any flows can be performed. The process of identifying, testing, and eliminating commercial and industrial illicit connections should be adequately documented, including recording the location of the connection, the date of testing, and the method used to remove the connection. At least 5 percent of relevant businesses or Standard Industrial Classification (SIC) codes should be inspected annually.

1.2.3 Monitoring for Assessing TMDL Implementation

The Clean Water Act requires that each state establish TMDLs for the pollutants of concern (33 USC §1313), in accordance with a priority ranking, for impaired waterbodies as listed on the 303(d) list. Specifically, for waterbodies on the 303(d) list, states, territories, and authorized tribes must develop TMDLs that will achieve water quality standards, allowing for seasonal variations and an appropriate margin of safety.

A water quality monitoring plan is an important component of the overall TMDL process because it provides the information necessary to make adjustments to the overall assessment and numeric targets and to assess progress towards attainment of the desired future conditions as expressed by the numeric targets. Sampling protocols and frequency of sampling will vary depending on the pollutant and watershed conditions. The objectives of TMDL monitoring include:

- Determine compliance with regulations. Identify how much higher the actual loads are than the target loads (sometimes referred to as "tolerable loads").
- Identify the sources of major loadings, if the waterbody is not in compliance.
- Determine if BMPs are needed and if existing BMPs are working to bring the waterbody back into compliance.

Stream segment-specific TMDL implementation plans are being developed by EPD with assistance from the Regional Development Centers (RDCs) and local stakeholders. Local governments are involved in developing these implementation plans and will be responsible for initiating the recommended water quality monitoring activities.

1.2.4 Monitoring for Delisting Impaired Waterbodies

Georgia EPD has developed sampling data guidance for listing and delisting streams that will be followed to assure consistency and quality of the data used in evaluating compliance with State water quality standards. This guidance can be found in Part 4 of this document.

1.2.5 Biological/Habitat Assessment

Biological monitoring involves collecting and evaluating biological data using standard metrics in order to identify trends in the integrity of the stream and watershed. In counties that have conducted biological and habitat assessments as part of watershed assessment studies, this sampling will be a continuation of those programs in support of their watershed assessment requirements. In counties that have not conducted a watershed assessment, these studies may be used in meeting future watershed assessment requirements.

Biological monitoring is to be performed at the 12-digit HUC watershed level. A minimum of one station per 12-digit HUC is to be identified within each county using criteria established by the District. Sampling at these stations should be conducted so that all of the stations will be assessed at least once every 5 years.

Each station is to be evaluated for both benthic macroinvertebrates and fish communities to detect trends in biotic integrity.

Biological monitoring and analysis for benthic macroinvertebrates and habitat will follow Georgia EPD's Standard Operating Procedures for Macroinvertebrate Biological Assessment of Wadeable Streams in Georgia. Fish sampling and analysis will follow Georgia DNR Wildlife Resources Division's Standard Operating Procedures for Conducting Biomonitoring on Fish Communities in Wadeable Streams in Georgia. See Parts 5A and 5B for these protocols, respectively.

1.3 Data Transfer and Database Management

Data management and reporting is an integral part of any monitoring program. To efficiently utilize available data, it is essential that a framework be in place that facilitates data storage, retrieval, and analysis. One component of the District-wide Water Quality Monitoring Plan is managing long-term monitoring data.

The District data management efforts will conform with Georgia EPD's reporting and data collection requirements, and will be designed to used with EPD's database. The District will require local jurisdictions to submit their data using Excel and Access templates provided by the District and/or EPD.

References

Metropolitan North Georgia Water Planning District. 2003. District-wide Watershed Management Plan.

PART 2A

Long-Term Ambient Trend Monitoring

Monitoring of local streams for long-term ambient trends provides a means of demonstrating progress toward water quality goals as watershed management efforts are implemented. The objective of the District Long-Term Ambient Trend Monitoring is to establish the extent of the progress toward the goal of maintaining or improving water quality and watershed conditions within and downstream of the District. The data will also be used to help refine future management strategies to address watershed management and water quality protection within the District.

2A.1 Monitoring Overview

The long-term ambient trend monitoring involves both wet weather and dry weather water quality sampling at permanent stream sampling locations throughout the District. The long-term ambient trend monitoring consists of three components:

1. **Wet Weather Monitoring –** A minimum of three wet weather samples will be required during both the summer and winter seasons (May-October and November-April)—for a total of six wet weather samples annually.
2. **Dry Weather (Baseflow) Monitoring –** A minimum of one dry weather (baseflow) sample will be required during both the summer and winter seasons (May-October and November-April)—for a total of two samples annually.
3. **Bacteria Monitoring –** A minimum of four geometric means of bacteria grab sampling for fecal coliform and E. Coli during each of the following quarters: May-July, August-October, November-January, and February-April. A geometric mean consists of a set of 4 samples within a 30-day period regardless of weather conditions. This will require a minimum total of 16 samples annually.

2A.2 Sampling Locations

Long-term trend monitoring is performed at permanent stream sampling locations. Strategic sampling station location will serve to provide information on long-term ambient trends in waters affected by both point source discharges and nonpoint source impacts. To promote consistency in sampling across the District, the following guidelines are recommended for siting long-term water quality stations:

- Long-term trend monitoring location siting should take into consideration a number of factors including: watershed size; watershed land use; existing water quality conditions; stream characteristics; riparian ownership; accessibility; and proximity to point source and stormwater discharges, water supply intakes, solid waste facilities (landfills), land application systems, and septic service areas.

- Ideally, a monitoring site should be located on a stream with a medium-size watershed (approximately 5-30 square miles). Maximum watershed size should be limited to 12-digit Hydrologic Unit Codes (HUCs).
- Watersheds and streams should be selected that are representative of the land use and water quality conditions of the local jurisdiction. Special consideration should be given to impaired stream reaches.
- Major streams should be monitored near the entry to or exit from important land use areas, jurisdictional boundaries, or at critical stream reaches.
- Existing sites should be used (where appropriate) to preserve the "historical record" and facilitate trend evaluation.
- If possible, sites should be located near USGS gauging stations.
- Monitoring sites and equipment should be safely accessible from nearby roads, particularly during wet weather conditions.
- Sites should be located at least 50 feet below any major stormwater outfall, and 200 feet below a point source discharge, to ensure that well-mixed samples are collected.
- Sites should not be influenced by unnatural physical conditions associated with a stream crossing such as large pools or road runoff.

2A.3 Sampling Requirements (Frequency and Criteria)

2A.3.1 Wet Weather Monitoring

Flow weighted composite, time-weighted composite, or single grab samples are to be collected six times per year for wet weather events at each sampling location. Three of these samples are to be collected in the summer (May-October) season and three in the winter (November-April) season.

Representative wet weather events require a minimum precipitation of 0.3 inch to ensure adequate runoff. Wet weather event monitoring at each site should be preceded by at least 3 days of dry weather (<0.1 inch of rainfall each day). Additionally, a minimum period of 72 hours is required between each wet weather event sampled to ensure that the events are discrete and the water quality parameters are associated with the event sampled.

Measurement of stream flow during sampling events is important for estimating total pollutant loadings for that stream or tributary. Therefore, a stage-discharge rating curve should be developed at each station and flow should be recorded when water quality samples are taken. Appendix 2A-4 provides information on establishing the rainfall-runoff relationship for a stream sampling location.

A minimum of three aliquots should be collected with an adequate sample volume to analyze the required parameters (note that approximately 4.25 liters of composite sample is required to meet the minimum volume requirements).

2A.3.2 Dry Weather (Baseflow) Monitoring

Baseflow samples will be collected twice per year during dry weather at each sampling location. One of these samples is to be collected in the summer (May-October) season and one in the winter (November-April) season.

Dry weather is defined as a period prior to sampling of at least 72 hours (3 days) receiving less than 0.1 inches of precipitation per day.

Dry weather baseflow sampling may be conducted by either manually operating an automated sampler or collecting a grab sample at the sampling station.

A minimum of three aliquots should be collected with an adequate sample volume to analyze the required parameters (note that approximately 4.25 liters of composite sample is required to meet the minimum volume requirements).

2A.3.3 Bacteria Monitoring

Grab samples for fecal coliform and E. Coli analysis will be collected 16 times per year at each sampling location. Four samples are to be collected over a 30-day period (at intervals not less than 24 hours) during each of the following quarters: May-July, August-October, November-January, and February-April.

Each set of four samples will be used to calculate a geometric mean, per State guidelines.

Two samples should be collected with an adequate sample volume to analyze for both fecal coliform and E. Coli bacteria.

2A.4 Sample Collection Methods

2A.4.1 Wet and Dry Weather Monitoring

Manual grab sampling methods may be used for small and medium streams and where samples during storm flows can be collected without danger of personal harm or injury. Manual grab samples may consist of either a single sample or composite samples. The sampling procedures and protocols for manual grab samples (simple and composite) are provided in Part 2B.

Equal Width-Integrated and Equal Depth-Integrated (EWI and EDI) are grab sampling methods that can be used to collect discharge-weighted, depth-integrated, isokinetic samples. The EWI/EDI methods and protocols are presented in Part 2C.

Automated sampling methods are designed to collect a flow-weighted composite, time-weighted composite, or single grab sample from a fixed location in the stream. The use of automated samplers is recommended for larger streams that cannot be safely accessed, especially during storm flows. However, they may also be installed on smaller streams if there is enough flow during a typical storm event to trigger the device. The sampling procedures and protocols for using automatic samplers are presented in Part 2D.

A discussion of manual versus automated sampling is provided in Appendix 2A-2.

2A.4.2 Bacteria Monitoring

Manual grab sampling methods are recommended for fecal coliform and E. Coli monitoring due to time sensitive nature of laboratory analysis for bacteria. The sampling procedures and protocols for manual grab sampling are provided in Part 2B.

2A.5 Sample Handling

2A.5.1 Sample Preservation

Proper preservation and conformity to established holding times for each parameter listed in section 2A.6 is essential for the integrity of monitoring results. The following guidance should be observed:

- Filtration and acid preservation for grab samples should be performed in the field or at a remote staging area as soon as possible after sample retrieval. If possible, samples shall be taken directly to the laboratory where filtration and preservation can be conducted. The preferred filtration procedure is included in Appendix 2A-4.
- Due to the variability in preservation techniques, sample preservation cannot be done in the field during composite sample collection, therefore samples should be preserved on ice.
- Approximately 1 liter of the sample must be filtered prior to preservation for orthophosphate.
- Proper preservation and maintenance of the holding time for bacteria (fecal coliform and E. Coli) samples is essential. Fecal coliform and E. Coli samples have a short holding time of six hours and must be delivered to the laboratory for analysis within this time or the results may be unrepresentative of stream conditions.

Bottle type, preservation requirements, and holding time limits for all the parameters being analyzed are provided in Appendix 2A-5.

2A.5.2 Sample Identification and Labeling

A sample numbering system should be used to ensure that each sample is uniquely identified in the field and tracked on field data collection forms. The following sample numbering system is recommended:

###-MMDDYY-HH:MM

where:

- ### = A unique alphanumeric designation for each sample location;
- MMDDYY = Month, day, and year; and
- HH:MM = Time in military units.

All of the samples collected at the site should be placed in the appropriate sample containers for preservation and shipment to the designated laboratory. Each sample should be identified with a separate identification label. A waterproof, gummed label should be attached to each sampling container. Information to be recorded on the label should include:

- Site name;
- Sample number;
- Sample type;
- Date and time of collection;
- Analysis to be performed;
- Preservation used and any other field preparation of the sample; and
- Initials of field crew collecting the sample.

2A.5.3 Sample Documentation

A chain-of-custody (COC) form should accompany all samples. A sample COC form is found in Appendix 2A-1. The COC form shall include all of the information provided on the sample label discussed in the preceding section.

The primary purpose of the COC form is to provide a mechanism for tracking each sample submitted for laboratory analysis and documenting sample collection information. The information on the COC form must be identical to the information of the sample label. A separate COC form shall be prepared for each monitoring site. If samples are shipped, the form should be placed in a re-sealable plastic bag (to keep the form dry) and sealed inside each sample cooler. When transferring possession of the samples, the individual relinquishing, and the individual receiving samples should both sign, date, and note the time on the COC form. This record documents transfer of custody from the sampler to another person, to/from a secure storage area, and to the laboratory. Copies of the COC forms should be kept in the project file.

In addition to the COC form, all wet weather and dry weather samples must have a completed Wet Weather/Dry Weather Sampling Form found in Appendix 2A-1. The Wet Weather/Dry Weather Sampling Form is used to document the following data and observations:

- Sampling information including location, sampling dates and times, personnel names, sample identification numbers;
- Receiving stream observations (presence of scum, foam, floatables, etc)
- Results of in-situ water quality observations/measurements performed in the field, including conductivity, pH, and temperature;
- Peak Stream Flow [wet weather only];
- Lab parameters ordered; and
- Lab results.

For bacteria (fecal coliform and E. Coli) samples, the Bacteria Sampling Form in Appendix 2A-1 must be completed. The following sample observations should be noted on the form:

- Sampling information including location, sampling dates and times, personnel names, sample identification numbers;
- Receiving stream observations ;
- Turbidity (light, medium, heavy);
- Sample Temperature;
- Weather conditions at the time of sampling, including air temp and cloud cover;
- Other comments (e.g. foam, oil sheen, etc).

2A.5.4 Sample Packaging and Analytical Laboratory Coordination

The samples should be packed in coolers with ice and some water to ensure they maintain the required temperature of less than or equal to 4°C during transport to the designated laboratory. Contact the laboratory prior to sampling to assure that the samples will be analyzed within their holding time. Samples may be placed in individual one-gallon re-sealable bags as a precaution to avoiding spilling the sample. All glass bottles should be individually bagged and bubble-wrapped to prevent breakage on the way to the lab. Samples may be placed in a large trash bag inside a cooler (to ensure against the sample leaking) with ice completely covering the samples.

2A.6 Analysis Parameters

Both wet weather and dry weather baseflow samples will be analyzed for the following parameters using EPA approved laboratory analysis methods:

- BOD_5
- COD
- TSS
- TDS
- Nitrate + Nitrite Nitrogen
- TKN
- Ammonia
- Hardness
- Total Phosphorus
- Orthophosphate (dissolved reactive phosphorus)
- Total / Dissolved Recoverable Metals (cadmium, copper, lead, and zinc)

The direct measurement of dissolved metals in wet weather is not required due to limitations involved with wet weather sampling. For wet weather the dissolved metal fraction may be calculated using total recoverable metals, total suspended solids, and hardness measurements. Dissolved metal fraction can be calculated using Georgia EPD's guidelines provided in EPD Water Quality Controls 391-3-6, Amended April 2000.

If samples show results for metals that do not meet water quality standards, Georgia EPD recommends that two samples annually (one wet and one dry) be collected and analyzed using U.S. Environmental Protection Agency (EPA) clean sampling techniques (Method 1669, Sampling Ambient Water for Trace Metals at EPA Water Quality Criteria Levels). Although use of automated samplers does not strictly comply with the "clean sampling" procedures in the EPA methods, water quality samples collected with automated samplers should be analyzed using the "clean metals" analytical procedures to assure the most accurate estimate of actual metals concentrations.

Bacteria grab samples will be analyzed for both fecal coliform and E. Coli bacteria using EPA-approved methods.

2A.7 Analytical Procedures

Any EPA-approved analysis method (40 CFR Part 136) can be used for analyzing long-term ambient trend monitoring samples.

Some recommended methods and detection limits for the water quality trend monitoring program are provided in Table 2A-1.

TABLE 2A-1
Recommended Analytical Methods

Parameter	Method	Detection Limit*
BOD5	EPA 405.1	2.0 mg/L
COD	EPA 410.1 or	50.0 mg/L or
	EPA 410.2 or	5.0 mg/L or
	HACH 41100-05	10 mg/L
Hardness	EPA 130.1 or EPA 130.2	10 mg/L
TSS	EPA 160.2	4.0 mg/L
TDS	EPA 160.1	10.0 mg/L
Total Phosphorus / Orthophosphate	EPA 365.2 or EPA 365.3	0.01 mg/L
Ammonia	EPA 350.1 or	0.01 mg/L
	EPA 350.3	0.1 mg/L
TKN	EPA 351.1 or	0.05 mg/L
	EPA 351.2 or	0.1 mg/L
	EPA 351.3 or	0.05 mg/L
	EPA 351.4	0.03 mg/L
Nitrate + Nitrite	EPA 353.1 or	0.1 mg/L or
	EPA 353.2 or	0.05 mg/L or
	EPA 353.3	0.01 mg/L
Total Recoverable Metals (cadmium, copper, lead, zinc)	EPA 200.8 or EPA 200.7 or EPA 200.9	cadmium: 10 μg/L copper: 20 μg/L lead: 25 μg/L zinc: 20 μg/L
Fecal Coliform	Standard Methods 9221E or	2 MPN/100 ml
	Standard Methods 9222D	1 colony/100 ml
E. Coli	Standard Methods 9221B.1 or	2 MPN/100 ml
	Standard Methods 9221F	2 MPN/100 ml

* The recommended methods have different detection limits, some lower than those provided here. However these detection limits are considered achievable for all laboratories.

2A.8 Quality Assurance/Quality Control

This section describes the elements of the field quality assurance/quality control (QA/QC) program. The overall QA/QC objective for the monitoring program is to ensure that the data collected are of good quality.

2A.8.1 Field QA/QC

Field quality assurance procedures include equipment cleaning, collection of field blanks and field duplicates, and proper sample preservation methods. When using automated collection methods, calibration of the automatic composite sampler using the manufacturer's procedures is required.

2A.8.1.1 Field Blanks

Field blanks should be collected to determine potential sample contamination during sample collection, handling, shipment, storage, or laboratory handling and analysis. Reagent grade water should be used for the field blanks.

Field blanks for nutrients (total phosphorus, orthophosphate, TKN, and nitrite plus nitrate nitrogen) and total recoverable metals (cadmium, copper, lead, and zinc) shall be collected once a year at each sampling location during a dry weather grab sampling event by filling pre-labeled sample bottles with reagent-grade water in the field. Field blank samples for orthophosphate should be filtered prior to sample preservation. For bacteria (fecal coliform and E. Coli) sampling, a minimum of one field blank should be collected for each geometric mean.

For automatic composite samplers, an equipment blank shall be collected once a year at each location and analyzed for nutrients and total recoverable metals. At a minimum, an equipment blank shall be collected at each sampling location prior to the initiation of monitoring activities using the following procedure:

1. Pour approximately 500 mL of reagent-grade water into a designated 1-L HDPE pre-cleaned sample bottle (labeled QA/QC).
2. Flush the composite sampler tubing once by inserting the sample collection tubing into the QA/QC sample bottle with approximately 500 ml of reagent-grade water, drawing water through the sample tubing via manual mode and flushing the water back out.
3. Refill the QA/QC sample bottle with 1L of reagent-grade water. Collect the equipment blank by inserting the tubing/intake line into the QA/QC bottle with reagent water. In manual mode, draw the required sample volume through the composite sampler and collect the equipment blank in a clean composite jar (refilling the QA/QC bottle as necessary). Pour the collected equipment blank into the pre-labeled sample collection bottles.
4. Preserve the samples according to Appendix 2A-5. The equipment blank sample for orthophosphate should be filtered prior to sample preservation.

2A.8.1.2 Field Duplicates

Field duplicates shall be collected once a year at each sampling location during a dry weather sampling event for all parameters. Field duplicates are collected to assess the representativeness of sampling procedures in addition to the normal uncertainty associated with analysis. The sample is to be split in two after the sample collection.

2A.8.2 Laboratory QA/QC

The laboratory selected for compliance monitoring should follow the Georgia EPD-approved methods, and routinely perform quality control checks during laboratory analysis, including calibration standards, blanks, laboratory control samples, laboratory control duplicate samples, matrix spikes, and matrix spike duplicates. Spikes and duplicates should be performed on a minimum of 10 percent of the samples and should meet data quality objectives established by the laboratory.

2A.9 Data Management, Analysis and Reporting

This section describes the procedures for reviewing, managing, interpreting, and reporting the data collected for the long-term ambient trend monitoring program. Detailed descriptions of data evaluation and statistical testing procedures are presented in Appendix 2A-7.

2A.9.1 Data Quality Review

The laboratory should conduct quality assurance reviews of all analytical data. The data reported to Georgia EPD under the stormwater monitoring program should be limited to useable data that has passed all data quality objectives established by the laboratory.

The following checks should be conducted, upon receipt of data from the laboratory:

- Check that all requested analyses were performed and reported, including quality control samples.
- Check the dates of analysis to ensure that samples were analyzed within the required holding times.
- Check that the laboratory's performance objectives for accuracy and precision were achieved. This includes a check of method blanks, detection limits, laboratory duplicates, matrix spikes, matrix spike duplicates, laboratory control samples, and standard reference materials.
- Check that field QA/QC was acceptable. This includes a check of field blanks, field duplicates, and chain of custody procedures.
- Assign data qualifiers as needed to alert potential data users in local governments of any uncertainties that should be considered during data interpretation (e.g., field blank showed trace amount of copper).

2A.9.2 Data Storage and Retrieval

Hard copies of all Wet Weather/Dry Weather Sampling and Bacteria Sampling forms, as well as all laboratory reports, should be retained in local files.

Sample and field data that is collected will eventually be stored in a long-term regional database. Initially, the data may be maintained in a local database or Excel spreadsheet and should include appropriate fields for each sampling outfall location.

2A.9.3 Data Calculations and Analysis

Many different types of data are generated during a stormwater monitoring program, including rainfall intensities and depths, discharge rates and flow volumes, the concentrations of chemical parameters, and the measurement of physical parameters. The tools of statistics and data evaluation can be applied to assess the major factors affecting stormwater quality, and to infer conditions or trends over time from the limited information obtained from during this monitoring program. A detailed description on these calculations and analyses, and the appropriate use of statistical tools is presented in Appendix 2A-7.

2A.9.3.1 Preliminary Data Evaluation and Summary Statistics

A preliminary data evaluation can be conducted to help determine whether it will be useful to statistically test hypotheses and further identify trends. The objective of the initial statistical evaluation is to obtain an overview of the results and to determine whether rigorous statistical evaluation will be useful to answer key questions. The preliminary data evaluation should consist of summary statistics to indicate how well the sample results represent stormwater quality at a given site. Typically used statistical measures include sample mean, variance, median, standard deviation, coefficient of variation, coefficient of skewness, and kurtosis. To assist in the interpretation of the analytical results, the flow and rainfall data should be plotted over time to produce a storm hydrograph. It is helpful to indicate the times of the composite samples collected on the hydrograph as well.

2A.9.3.2 Nonparametric Statistical Analysis

In many cases, the data may not follow a normal distribution or concentration may fall above or below a threshold value. In these cases, nonparametric (non-normal) statistics must be used to compute summary statistics, t-tests (Mann-Whitney, for example) for comparing two datasets, or Analysis of Variance (Kruskal-Wallace) for comparing three or more datasets.

2A.9.3.3 Geometric Mean Analysis

The geometric mean, used for bacteria (fecal coliform and E. Coli) analysis reporting, is calculated using the following equation:

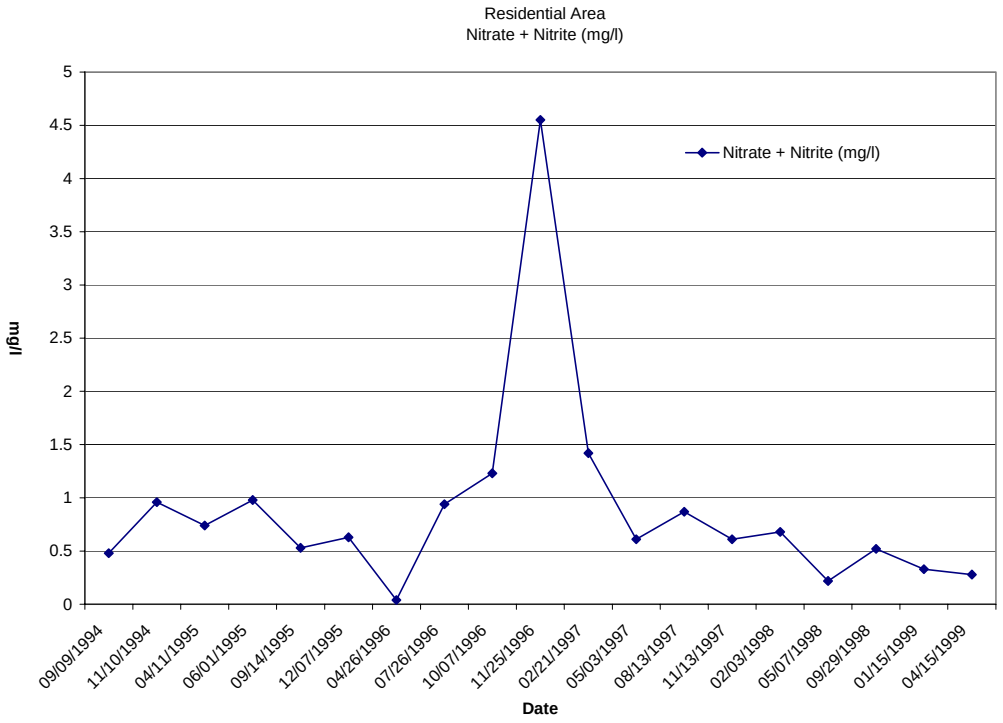
$$\text{Geometric mean} = e^{[(\ln \text{“data point 1”} + \ln \text{“data point 2”} + \ldots \ln \text{“data point n”})/n]}$$

The Georgia EPD fecal coliform standards are based on geometric mean of four samples taken with a 30-day period.

2A.9.3.4 Trend Analysis

Methods of trend analysis include both parametric and nonparametric methods of analysis. Parametric methods, including simple linear and multiple linear regression, are used to identify trends in data regardless of the normality of the data. However, the residuals must be normally distributed for valid trend analysis. Nonparametric methods of trend analysis include smoothing locally-weighted scatterplots by computing a running median. These are often effective when data sets contain extreme or censured values. Computing the residuals from a smoothed line can then be analyzed for trend while accounting for seasonality by using a Seasonal Kendall test. Figure 2A-1 is an example of how data can be graphically represented to provide a trend analysis.

FIGURE 2A-1
Example of Data Representation



2A.9.4 Data Presentation and Reporting

Analytical reports should be prepared and submitted to the project manager in compliance with the requirements of the quality assurance project plan. The laboratory report should contain the following information:

- Project identification;
- Field sample number(s);
- Laboratory sample number(s);
- Sample matrix description;
- Date and time of sample collection;

- Analytical method number;
- Individual parameter results;
- If sample results are below the detection limit (BDL), sample result should be reported as BDL. The detection limit should be provided, as the resulting sample concentration of one half the BDL is required for the purpose of calculations.
- Date and time of analysis;
- Detection limits achieved;
- Precision, accuracy, and completeness data for each parameter; and
- Identification of problem conditions and corrective measures (if applicable).

The laboratory quality assurance officer, as part of the validation process, confirms that documentation is complete and legible; qualitative identifications are accurate; calculations are accurate; results are expressed in the appropriate units and number of significant figures; and the required quality control checks were run and met acceptance criteria.

Appendix 2A-1
Long-Term Ambient Trend Monitoring
Data Collection Forms

Distict Long-Term Ambient Trend Monitoring Program – Wet Weather/Dry Weather Monitoring Form			
Name of City or County:			
Data Sheet Number:			
Permanent Station Name/Location:		Stream Name:	
Receiving stream observations (e.g. odor, foam, floatables, oil sheen, etc.):			
Type of Sampling: (minimum 3 wet and 1 dry samples during Nov-Apr and May-Oct) ❒ Wet weather ❒ Dry weather			
Sampling Method: ❒ Manual Grab (single point) ❒ EDI/EWI Method ❒ Automated Sampler			
Date (MM/DD/YY) and time (HH:MM) of sampling: / / @ : am / pm			
Sampling performed/ collected by:			
Field sample preservation method:			
Approximate start time and length of storm (applicable to wet-weather sampling):			
Field pH:		pH meter calibrated? Y / N	
Field Conductivity (umho/cm):		Conductivity meter calibrated? Y / N	
Sample Temperature (ºF):		Peak Stream Flow (cfs):	

Lab Analysis	
Sample ID:	Name of analytical lab:
Parameter	Lab Results
BOD_5 (mg/L)	
COD (mg/L)	
TSS (mg/L)	
TDS (mg/L)	
TKN (mg/L)	
Nitrite + Nitrate (mg/L)	
Ammonia (mg/L)	
Hardness (mg/L)	
Total Phosphorus (mg/L)	
Orthophosphate (mg/L)	
Total Recoverable Cadmium (mg/L)	
Total Recoverable Copper (mg/L)	
Total Recoverable Lead (mg/L)	
Total Recoverable Zinc (mg/L)	
Other _____	
Comments:	

District Long-Term Ambient Trend Monitoring Program – Bacteria Monitoring Form	
Name of City or County:	
Data Sheet Number:	
Permanent Station Name/Location:	Stream Name:
Date (MM/DD/YY) and time (HH:MM) of sampling: / / @ : am / pm	
Sampling performed by:	
Receiving stream observations (e.g. odor, foam, floatables, oil sheen, etc.):	
Observed Turbidity (light, medium, heavy):	
Sample Temp (ºC):	Sample Temp (ºC) (dup.):
Weather conditions at time of sampling:	
Description of Sample and Other comments (e.g. odor, foam, etc.):	

Lab Analysis
Name of Analytical Lab
Fecal Coliform Grab Sample ID:
Fecal Coliform Concentration (MPN/100ml or colony/100ml):
E. Coli Grab Sample ID:
E. Coli Concentration (colony/100ml):
Comments (holding time, etc.):

Collect 16 samples annually – 4 grab samples within a 30-day period with four 30-day periods per year, one each quarter: May-July, Aug-Oct, Nov-Jan, and Feb-Apr. Samples will be collected once a week on same day without regard to wet or dry conditions.

Note: Fecal Coliform / E. Coli sample holding time is 6 hours

District Long-Term Ambient Trend Monitoring Program – Bacteria Monitoring Annual Form

Name of City or County:

Permanent Station Name/Location:

Stream Name:

Date	Time	Data Sheet Number	Sampling Performed By	Weather Conditions	Sample Temp (ºC)	Sample Temp (ºC) dup.	Fecal Coliform Grab Sample ID	Fecal Coliform Concentration	E. Coli Grab Sample ID	E. Coli Concentration
							Geometric Mean		Geometric Mean	
							Geometric Mean		Geometric Mean	
							Geometric Mean		Geometric Mean	
							Geometric Mean		Geometric Mean	

Sample Chain-of-Custody (COC) Form

APPENDIX 2A-2
Manual versus Automated Sampling Methods

Manual monitoring involves sample collection and flow measurement by personnel using hand-operated equipment (e.g., bailer, bottle). For a monitoring program that is modest in scope (i.e., relatively few sampling sites and storm events), manual methods for obtaining grab and composite samples may be preferable to those employing automated equipment. Also, if your program requires monitoring large streams, you may need to use manual methods in order to collect cross-section composites. The principal advantages to manual sampling are its relatively low capital cost and high degree of flexibility. In addition to the capital outlay required for the purchase of automated samplers, other costs, such as installation, training personnel to use the samplers correctly, and field maintenance and operations (replacing batteries, interrogating data loggers, retrieving and cleaning sample jars) can be substantial. However, manual monitoring may not be feasible if:

- Monitoring personnel are not available after normal working hours
- Monitoring personnel have strict job descriptions that do not include sampling
- The organization's insurance policy doesn't cover stormwater monitoring activities
- Managers and monitoring personnel are not able to deal with sick days, vacations, and competing priorities

Manual sampling is generally less practical than automated monitoring for large-scale programs (e.g., monitoring programs involving large numbers of sites or sampling events over multiple years). It is difficult to collect true flow-weighted composites using manual methods. Under these circumstances, labor costs and logistical problems can far outstrip those associated with automated equipment. For the same reason, manual sampling is seldom practiced if specific program objectives require that samples by composited over the entire duration of a storm (EPA's Municipal NPDES Stormwater Permit Application monitoring programs require compositing over only the extent of the storm or the first three hours, whichever comes first).

Automated monitoring involves sample collection using electronic or mechanical devices that do not require an operator to be on-site during actual stormwater sample collection. It is the preferred method for collecting flow-weighted composite samples. Automated monitoring is generally a better choice than manual monitoring at locations where workers could be exposed to inadequate oxygen, toxic or explosive gases, storm waves, and/or hazardous traffic conditions. Also, automated methods are better than manual methods if you are unable to accurately predict storm event starting times. Automated samplers can be set so that sampling operations are triggered when a pre-determined flow rate of storm runoff is detected. Conversely, manual monitoring relies on weather forecasts) and considerable judgment and good luck) to decide when to send crews to their monitoring stations. It is very difficult to predict when stormwater runoff is likely to begin; consequently, manual monitoring crews may arrive too early and spend considerable time waiting for a storm that begins later than predicted, or they may arrive too late and miss the "first flush" from a storm that began earlier than predicted. If the automated equipment is set up to collect flow-weighted composite samples using the constant volume-time proportional to flow method, it reduces the need to measure samples for compositing.

If you have determined that field-measured "indicator" parameters (e.g., turbidity, conductivity, dissolved oxygen, pH) are sufficient for your monitoring objectives, consider using electronic sensors and data loggers. Using electronic sensors and data loggers, you can obtain near-continuous measurements of indicator parameters at reasonable cost.

References

Woodward-Clyde. November 1995. Stormwater Quality Monitoring Guidance Manual.

APPENDIX 2A-3
Flow and Time Composite Sampling

The two basic approaches for obtaining composite samples are referred to as time-proportional and flow-proportional. A time-proportional composite sample is prepared by collecting individual sample "aliquots" of equal volume at equal increments of time (say, every 20 minutes) during a storm event, and mixing the aliquots to form a single sample for laboratory analysis. Time-proportional samples do not account for variations in flow; pollutant concentrations in sample aliquots collected during the portion of the storm with lower flows are given the same "weight" as sample aliquots collected during higher flows. Consequently, time-proportional composite samples generally do not provide reliable estimates of event mean concentrations or pollutant loads, unless the interval between sample aliquots is very brief and flow rates are relatively constant.

Flow-weighted composite samples are more suitable for estimating event mean concentrations and pollutant loads. The event mean concentration (EMC) is a statistical parameter used to represent the average flow-proportional concentration of a given parameter during a storm event. It is defined as the total constituent mass divided by the total runoff volume. When combined with flow measurement data, the EMC can be used to estimate the pollutant loading from a given storm. A flow-weighted composite sample is prepared by collecting sample aliquots and combining them based on the proportion of flow represented by each. This can be accomplished in several ways (USEPA 1992):

Constant Time – Volume Proportional to Flow Rate – Sample aliquots are collected at equal increments of time during a storm event, and varying amounts of each aliquot are combined to form a single composite sample. The amount of water removed from each aliquot is proportional to the flow rate at the time the aliquot was collected. This type of composite sample can be collected using either manual or automated techniques.

Constant Time – Volume Proportional to Flow Volume Increment – Sample aliquots are collected at equal increments of time during a storm event, and varying amounts from aliquot are combined to form a single composite sample. The amount of water removed from each aliquot is proportional to the volume of flow since the preceding aliquot was collected. This type of compositing is generally used in conjunction with an automated monitoring system that includes a continuous flow measurement device. It can be used with manual sampling in conjunction with a continuous flow measurement device, but this combination is uncommon.

Constant Volume – Time Proportional to Flow volume Increment – Sample aliquots of equal volume are taken at equal increments of flow volume (regardless of time) and combined to form a single composite sample. This type of compositing is generally used in conjunction with an automated monitoring system that includes a continuous flow measurement device.

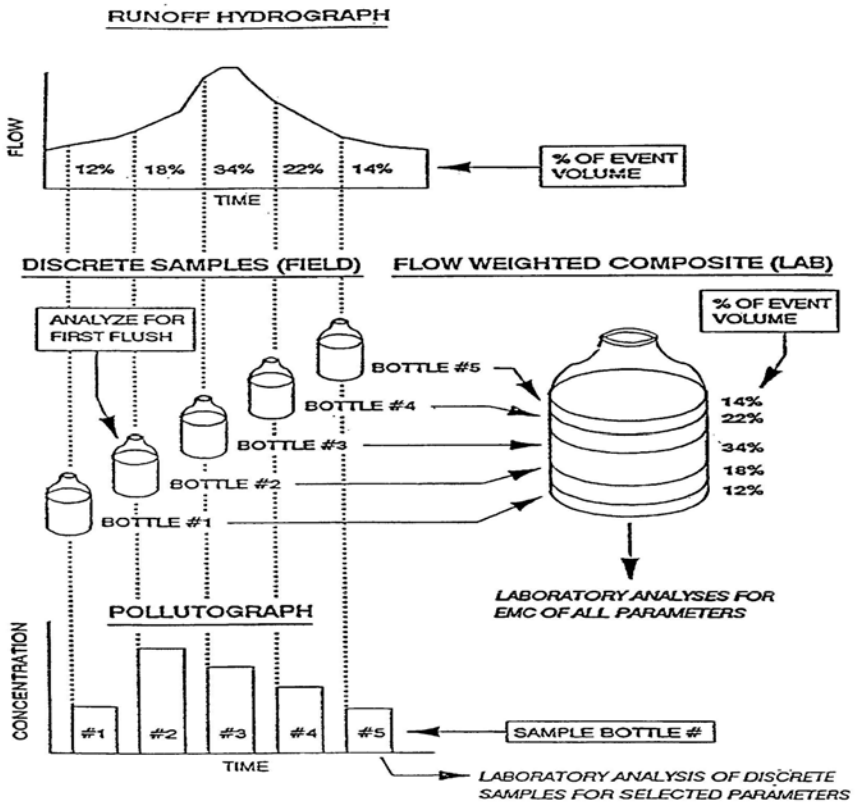
Select the flow-weighted compositing method most suitable for your program based on the monitoring technique (manual or automated) and equipment you plan to use. Compositing Methods 2 and 3 are more accurate than Method 1 because Methods 2 and 3 use the total volume of flow based on continuous flow measurement to scale the sample volume; in contrast, Method 1 uses a single instantaneous rate measurement to estimate the flow over

the entire sampling interval. However, if you intend to use manual methods, compositing Method 1 is generally the most practical choice. If automated equipment is to be used, Method 3 is generally preferred because it minimizes the need for measuring and splitting samples, activities which can increase the chance for sample contamination or errors. If you plan to use automated methods, review the equipment manufacturer's specifications and instructions to select the compositing method most appropriate for that particular make and model.

If you have determined that field-measured "indicator" parameters (e.g., turbidity, conductivity, dissolved oxygen, pH) are sufficient for your monitoring objectives, consider using electronic sensors and data loggers. Using electronic sensors and data loggers, you can obtain near-continuous measurements of indicator parameters at reasonable cost.

References

Woodward-Clyde. November 1995. Stormwater Quality Monitoring Guidance Manual.



APPENDIX 2A-4
Filtration of Samples

Background

Filtration of the samples for dissolved parameters may be done in the field or at a remote staging area prior to sample preservation and shipment to the lab. Filtration of samples sooner helps to maintain the integrity of the samples, therefore yielding the most accurate data.

Equipment

- 1 4-L bottle of reagent water
- Air diaphragm pump with tubing (one or more)
- Disposable filter units (i.e., Nalgene) with 0.45 micron cellulose acetate membranes and/or Filtration units, filter flasks, and 0.45 micron cellulose acetate membranes
- Glass fiber prefilters (i.e., Whatman GF/D or equivalent)
- Sample collection bottles

Procedure

1) Connect the tubing from the air diaphragm pump to a reusable or disposable filter unit. Turn on the pump and condition the filter by pouring approximately 50 ml of reagent-grade water into the filter unit. Turn the pump off, unscrew the top of the filter unit and discard this portion. Replace the top of the filter and pour an additional 500-ml aliquot of reagent-grade water into the filter unit and collect the filtrate. Collect the filtration blank into a 500-ml sample collection bottle. A filtration blank should be analyzed for each lot of filter units purchased.

2) Filter the first quality control blank samples (i.e., equipment or field blanks) using the same filter assembly.

3) Discard the filter unit and replace with a new one. Condition each new filter with approximately 50 ml of reagent-grade water and discard the filtrate. Continue to filter remaining quality control blank samples and storm water samples requiring dissolved parameters. Use a new filter unit for each sample.

4) For turbid samples, place a glass fiber prefilter directly on top of the cellulose acetate filter. Replace the glass fiber prefilter and cellulose acetate filter during the filtration process as necessary. (Note: the cellulose acetate filter cannot be replaced with the disposable units. If the filter clogs, a new unit will be required).

5) Label bottles with sample ID number, date, time, analysis required, type of preservative, and initials.

Cleaning

Reusable filter units and flasks should be cleaned according to Appendix 2A-6 prior to filtering each new sample. No cleaning is required for the disposable filtration units.

References

Standard Methods for the Examination of Water and Wastewater, 19th Edition, 1995.

Appendix 2A-6 - Glassware Cleaning

APPENDIX 2A-5
Preservation of Samples

Background

Samples must be preserved to ensure the integrity of the samples prior to analysis. Many laboratories provide sample containers with preservatives already included. If the preservatives are not already added, follow this procedure. (See Table below for appropriate preservative for each analyte.)

Modified Handling Requirements for Samples

Parameter	Bottle Type[1]	Sample Volume[2]	Preservation	Holding Time
BOD_5	P,G	1 L	Cool, 4^0C	48 hours
COD	P,G	250 ml	Cool, 4^0C, H_2SO_4 to pH<2	28 days
TSS	P,G	500 ml	Cool, 4^0C	7 days
TDS	P,G	500 ml	Cool, 4^0C	7 days
TKN	P,G	1 L	Cool, 4^0C, H_2SO_4 to pH<2	28 days
Nitrite + Nitrate	P,G	1 L	Cool, 4^0C, H_2SO_4 to pH<2	28 days
Ammonia	P,G	1 L	Cool, 4^0C, H_2SO_4 to pH<2	28 days
Hardness	P,G	125 ml	HNO_3 to pH <2 H_2SO_4 to pH<2	6 months
Total Phosphorus	P,G	250 ml	Cool, 4^0C, H_2SO_4 to pH<2	28 days
Orthophosphate	P,G	125 ml	Filter immediately, Cool, 4^0C	48 days
Total Recoverable Metals (Cadmium, Copper, Lead, Zinc)	P,G	500 ml	HNO_3 to pH<2	6 months
Fecal Colifom	PP,G	100 ml	Cool, 4^0C	6 hours
E. Coli	PP,G	100 ml	Cool, 4^0C	6 hours

[1] Polyethylene (P), Polypropylene (PP), Glass (G) – EPA-approved sample containers (40 CFR 136)

[2] Additional sample volume should be collected when possible, to allow for laboratory quality control samples. Check with your laboratory for optimum sample volume.

Equipment

- Preservative ampoules: H_2SO_4, HNO_3
- pH paper (i.e., Whatman)
- Filtration flask and filter funnel apparatus (three units)
- Glass fiber filters (i.e., Whatman GF/A, B, D or equivalent)
- Air diaphragm pump
- Coolers with ice
- Sample bottles with labels
- Disposable gloves and safety glasses

Procedure

1) Disposable gloves and safety glasses should be worn during handling of samples and preservatives. Refer to Material Safety Data Sheets (Appendix X – Health and Safety Plan) as needed.

2) Filter a portion of the composite sample following Appendix 2A-4 and dispense composite samples for total and dissolved parameters to appropriate sample bottles.

3) Preservation requirements are identified in the appropriate sections of the Monitoring Plan. For samples requiring acid preservation, sample pH will be measured using pH paper. Add the appropriate preservative dropwise, until sample pH reaches 2.0. Cap and shake sample bottle in between acid additions to completely mix sample prior to pH measurement.

4) Prepare sample bottles for packing in coolers. Bubble wrap should be taped around all glass bottles. One or more bottles should be packed in a re-sealable bag (glass bottles should always be packed individually). The re-sealable bags will be placed in a lined cooler. The samples should be packed with wet ice (packed in re-sealable bags) to maintain a sample temperature of 4 degrees C or less during sample shipment.

References

Material Safety Data Sheets for Sulfuric Acid and Nitric Acid

Standard Methods for the Examination of Water and Wastewater, 19th Edition, 1995.

APPENDIX 2A-6
Glassware Cleaning

Background

If any of the containers are re-used, follow the procedure listed below.

Equipment

- Non-phosphate detergent
- Diluted (10% by volume) hydrochloric acid
- Pesticide-grade acetone
- Distilled or deionized water
- Nitrile or latex gloves and safety glasses

Procedure

NOTE: Nitrile or latex gloves and safety glasses should be worn during glassware cleaning

1) Wash equipment with non-phosphate detergent and rinse twice with tap water.

2) Carefully rinse with fresh dilute hydrochloric or nitric acid.

3) Rinse twice with distilled or deionized water.

4) For glassware, rinse once with full-strength pesticide-grade acetone to remove organic compounds (DO NOT CONDUCT THIS STEP WITH PLASTIC CONTAINERS OR EQUIPMENT) and rinse three times with distilled or deionized water.

5) Allow to air dry.

Always follow the manufacturer's guidelines if they differ from the above procedure.

APPENDIX 2A-7
Data Evaluation and Statistical Testing

Introduction

Many different types of data are generated during a stormwater monitoring program, including rainfall intensities and depths, discharge rates and flow volumes, the concentrations of chemical parameters, and the measurement of physical parameters. We can examine these data for patterns and trends to evaluate impacts of watershed changes as well as the implementation of stormwater program activities.

However, many highly variable factors influence stormwater quality and receiving water impacts. These factors include storm intensity and duration, the length of the antecedent dry period, and the magnitude and frequency of pollution-causing activities within the catchment area. The tools of statistics and data evaluation are used to assess the major factors affecting stormwater quality, and to infer, with a predictable level of error, generalities about average conditions or trends over time and the variability from the limited information obtained from any monitoring program.

The first step to evaluate a stormwater data set is to validate the chemical data, qualifying those that do not meet the criteria established for quality assurance/quality control (QA/QC). After completing the data validation, conduct an initial evaluation using summary (univariate) statistics. The initial evaluation shows whether the data are suitable for statistical hypothesis testing. The type of hypothesis tested is determined by the program objectives. These objectives usually include one or more of the following:

- Characterize stormwater discharges (e.g., average conditions, variability, ranges, etc.)
- Compare stormwater discharge quality to state and federal water quality criteria
- Detecting trends in discharge quality over time and between different locations

The statistical testing techniques appropriate to each of these objectives are discussed in the following sections.

Data Editing, Validation, and Treatment

Prior to conducting a statistical test, screen data to eliminate biased or unrepresentative data. Biased and unrepresentative data may result from equipment malfunctions (for example, equipment does not function throughout the event), field or laboratory protocol errors, weather problems (for example, rainfall totals exceed predictions to a degree that prevents sampling from much of the storm), human errors, etc. If applicable to your data set, assess any data below laboratory detection values and estimate particulate fractions of metals. Finally, transform data to a normal distribution, if using statistical tests that assume normal data. Specific criteria to consider in eliminating biased or unrepresentative data and adjusting data for analysis are discussed below.

Percent Capture

If samples were taken using flow-weighted compositing techniques, estimate the percent of the total captured discharge for each sample (i.e., the total flow from which the equipment sampled during the time the equipment was operating, versus the total flow of the storm event). As a general rule, reject samples with less than 60% capture as not representative of the event. In some circumstances, samples with less than 60% capture may be used, depending on the objective of the analysis. For example, the 60% capture criterion may not be applicable to a sample collected to characterize the "first flush" of a storm event. In compiling data, it is suggested that data with less than 80% capture (but greater than 60%) be noted.

QA/QC Qualifiers

Based on the results of the QA/QC evaluation, qualify or reject laboratory data that is suspect due to the contamination of blanks, exceedences of holding times, or low surrogate recoveries. Ideally, statistical tests will be performed only on data that have passed this screening process. Although it is possible to use data that have been qualified as estimated values, a higher level of uncertainty is associated with the test results. It is up to the data user to make an educated decision whether to include estimated values, based on the objectives of the sampling program and the use of the data. The uncertainty associated with using estimated data may be more acceptable in a screening program than in a risk assessment context, for example.

Event Mean Concentrations

If using Event Mean Concentrations (EMCs) for characterizing stormwater quality, then either collect all data as an EMC by using a flow-weighted composite sample collection technique or, if analyzing individual samples, compute an EMC. This can be accomplished by integrating the hydrograph (plot of flow rate vs. time) and pollutograph (plot of concentration vs. time). Estimate pollutant mass by computing pollutant loads for a number of corresponding time segments of the hydrograph and the pollutograph. The product of the partial flow volume and associated concentration estimates the mass in that segment of the discharge. The sum of all such segment masses estimates the total mass discharged by the event. The estimation of the total area under the hydrograph provides the total volume of runoff. Total mass divided by the total runoff volume provides the desired value for the EMC. (Note: Composite samples must be collected over the whole hydrograph to determine the EMC. Samples collected during the first 3 hours of a longer storm cannot be used to determine EMC.)

Practical Quantification Unit and Method Detection Limit

The method detection limit (MDL) is defined as the “minimum concentration of an analyte that can be measured and reported with a 99 percent confidence that the analyte concentration is greater than zero" (40 CFR 136.2). The practical quantification limit (PQL) is the minimum concentration of an analyte that can be accurately and precisely quantified. In general, the PQL is five to ten times the MDL, depending on the analyte. In general, statistical tests will be more accurate if the reported concentrations are above the PQL. If a large amount of the data is between the MDL and PQL, statistical tests still can be performed. However, the confidence (power) of the test may be lower, due to increased uncertainty. Prior to conducting statistical tests, examine the data set to determine the

percentage of points that are below the MDL and PQL. If a large proportion of the data is below the MDL, statistical testing may not be appropriate.

Averaging Duplicates

Average data from duplicate samples (laboratory or field) prior to statistical analysis. That is, use the average value in place of either of the two duplicate values.

Calculating Metal Fractions

GAEPD water quality in-stream standards are written for dissolved fraction. Dissolved fraction must be calculated and incorporated into the database. Where total fractions of metals are measured, it is possible to estimate several other fractions from these numbers and the concentration of total suspended solids (TSS). Dissolved metal fraction can be calculated using GAEPD's guidelines provided in (GAEPD Water Quality Controls 391-3-6, Amended April 2000)

Distributional Tests

Many commonly-used statistical tests (e.g., parametric analysis of variance) are based on the assumption that the data were sampled at random from a population with a normal distribution. Therefore, another attribute of the data that should be investigated is its apparent probability distribution. It is important to determine whether the probability distribution can be assumed to be normal or lognormal (or potentially another distribution). Researchers have found that the lognormal distribution generally provides the best fit to stormwater quality data (EPA 1983; Driscoll et al. 1990). If the data are not normally distributed or lognormal, or if the data set contains a very high proportion of nondetects, use a nonparametric statistical procedure for testing trends. Non-parametric techniques examine the data based on rank rather than distribution.

Several methods can be used to determine the normality of a data set or of the transformed values. These include a graphical check of the data, the W-test, and the Probability Plot Correlation Coefficient (PPCC). All are useful for the analysis of stormwater quality data. It is recommended that the use of a lognormal distribution be examined first. The procedure employed for the graphical test of a lognormal distribution is to develop a log probability plot for visual assessment of the log-normal distribution. First, the data is transformed by finding the logarithm (base e) of each data point, then computing the mean and standard deviation of the transformed data. The theoretical distribution is constructed from the mean of the logs (U) and the standard deviation of the logs (W). When combined with the plotting position based on the normal distribution, this derived distribution indicates the expected value of a pollutant's concentration at any probability of occurrence, assuming that the data follow a lognormal distribution. Compare this expected probability distribution with the data by plotting the two on the same log probability plot. A visual check of the data using a log probability plot can be a very effective test, and is recommended.

Treatment of Nondetects

When stormwater data sets include some nondetects within the data, separate data analysis techniques are required to accurately estimate sample statistics. When below-detection-limit data exist in a data set, they will affect statistical parameters computed from that set. For example, when below-detection-limit data is set to the detection limit (often cited as a

conservative approach), it causes an overestimation of central tendency measures and an underestimation of dispersion measures, as opposed to what would have been obtained had the true values of the below-detection-limit data been known. The magnitude of the error made by failing to properly treat below-detection-limit data is a function of the size of the data set (i.e., the total number of events for which a concentration was reported [N], the percentage of the total set represented by detection limit data, and the value of the detection limit relative to the median of the data above the detection limit).

Treatment of below-detection-limit data varies among workers in the field and the objectives for which the data are being analyzed. One practice is to simply set all below-detection-limit data at their detection limit, the argument being that, since the actual values are likely to be lower, the average so calculated would be conservative for prediction of mean concentrations. However, prediction of values that are rarely exceeded (i.e., pollutant concentrations that are observed less than 5 percent of the time) may very likely be underpredicted significantly. Another practice is to set the values equal to one-half (or some other fraction) of the detection limit. When a significant percentage of a data set is at or below the detection limit, the treatment method can seriously affect analytical results and their interpretation.

Descriptive Statistics

The purpose for calculating general descriptive statistics is to gain an overview of the data and to prepare for more formal statistical hypothesis testing. The data can be displayed in a variety of ways and summary statistics are generated. These exploratory techniques can provide clues as to the presence of major treatment effects (e.g., station, year, land use type) that can be tested for statistical significance. Descriptive statistics may also indicate how groups of data (e.g., data from several residential land use catchments) can be combined, or pooled, prior to statistical testing. Pooling effectively increases the sample size and the power of the analysis to detect significant differences. For example, if data collected at two residential land use stations are demonstrated to not differ statistically from each other, the data could be pooled for further testing to compare to other land uses (or over time). The reverse may be demonstrated by the descriptive statistics as well.

Summary Statistics

First, calculate simple descriptive statistics, characterizing the central tendency, variability, and distribution of the data set. Central tendency is measured by the sample mean (if normal, the arithmetic average of the data), the median (the 50th percentile of the distribution), and the mode (the most probable value). The sample standard deviation and its squared value, the variance, representing the variability of the data set. For non-parametric tests, data variability is measured by the interquartile difference, the difference between the values of the first (25th percentile) and third quartile (75th percentile) values. Any statistical software program, and most hand calculators, can be used to calculate these parameters.

Descriptive Statistics Using the Lognormal Distribution

If a sample (a data set of N observations) is drawn from an underlying population that has a lognormal distribution, the following apply:

- Computing the mean and standard deviation of the log transforms of the sample data, rather than computing summary statistics on untransformed data, obtains a better estimate of the mean and variance of the population.
- Base the estimates of the arithmetic summary statistics of the population (mean, median, standard deviation, coefficient of variation) on their theoretical relationships (see Table 1) with the mean and standard deviation of the transformed data.
- The arithmetic mean, so computed, will not match that produced by a straight average of the data. Both provide an estimate of the population mean, but the approach utilizing the log-transformed data provides a better estimator. As the sample size increases, the two values converge. For the entire population, both approaches produce the same value.

Table 1 Relationships of Lognormal Distributions

$T = EXP(U)$	$S = M * CV$
$M = EXP(U + 0.5 * W^2)$	$W = SQRT(LN(1 + CV^2)$
$M = T * SQRT(1 + CV^2)$	$U = LN(M/EXP(O.5 * W^2))$
$CV = SQRT(EXP(W^2) - 1)$	$U = LN(M/SQRT(1 + CV^2)$

Note: Parameter designations are defined as:

	Arithmetic	Logarithmic
MEAN	M	U
STD DEVIATION	S	W
COEF OF VARIATION	CV	
MEDIAN	T	

LN(x) designates the base e logarithm of the value x

SQRT(x) designates the square root of the value x

EXP(x) designates e to the power x

A few mathematical formulas based on probability theory summarize the pertinent statistical relationships for lognormal probability distributions. These provide the basis for back-and-forth conversions between estimates of the arithmetic summary statistics of the untransformed data (in which concentrations, flows, and loads are reported) and summary statistics of the transformed data (in which probability and frequency characteristics are defined and computed). Table 1 presents the formulas that define these relationships, from which other values can be computed.

If data are better predicted by a lognormal distribution than a normal distribution, use the lognormal distribution to estimate population statistics and analysis of variance tests. This is typically the case with urban stormwater data.

Box and Whisker Plots

The Box and Whisker Plot is a graphic method of displaying the variability, spread, and distribution of the data set. The "box" shows the 25th, 50th, and 75th percentile. One method of assessing variability is the interquartile range, defined above. The "whiskers" are obtained by multiplying the interquartile range by 1.5, and show the spread of the data. Use these plots to display the degree of overlap between two data sets, which is an indication (but not proof) of whether the data sets are likely to be derived from the same populations. If data are lognormal, produce the plots using the log-transformed data.

Characterization of Stormwater Discharges

The characterization of runoff provides both qualitative and quantitative overviews of a storm event. The qualitative analysis for each monitored event should include a narrative, which describes the timing and nature of the field activities. The narrative should include, at a minimum:

- Station identification
- Date of storm event
- Names of field personnel
- Time precipitation started and ended (if known), times samples were taken, time monitoring ended
- Information regarding any problems encountered and changes to the sampling protocol that can affect the interpretation of the data

After writing the narrative, graph the hydrologic data (flow and precipitation). Examine the graphs for patterns in the timing and intensity of runoff relative to those of precipitation. After sampling a minimum of three or four storms, calculate summary statistics from the analytical results. Use these results to determine whether the data set is sufficiently robust to support statistical hypothesis testing. Do this by examining the confidence levels of the summary statistics and by performing selected hypothesis testing. If not, continue to monitor at selected locations in order to obtain more data.

Stormwater Discharge and Rainfall Information

When data is available from a flow measurement device, a hydrograph can be created for each storm, displaying storm duration on the horizontal axis and discharge rate on the vertical axis. Rainfall should be plotted on the same graph (or in a different graph on the same page). Note the times when subsamples for compositing were collected on the horizontal axis of each plot. Analysis of these graphs for data gaps and outlying (i.e., extreme) data points may provide some information about the functioning of the automated equipment during the storm. Reject outliers from the data set for the purpose of statistical analysis if the cause of their behavior can be identified (e.g., poor QA/QC of a particular data point, poor storm capture, etc.).

Typical Applications of Hypothesis Testing to Characterization Data

Typical applications of statistical testing procedures to discharge quality data include determining whether any of the following are significant:

- Differences between stations
- Differences between monitoring years
- Correlations between different water quality parameters

Assessing Potential for Stormwater Impact

Comparison to State and Federal Water Quality Objectives

Use state and federal water quality criteria as benchmarks for assessing the potential impacts of stormwater runoff on surface water quality. The water quality standards applicable to a given water body depend on the designated beneficial uses of that waterbody. Numerical standards are set for temperature, pH, turbidity, dissolved oxygen, fecal coliform, and the concentrations of toxic substances such as metals and organic chemicals in receiving waters.

Compare the validated analytical results for samples from piped or open channel drainage systems from an individual storm event to the state water quality criteria for the protection of aquatic life under acute (short-term) conditions. Although the pipe or open channel (in many cases) is not a receiving water body that supports beneficial uses, comparison to criteria can provide an indication of the potential for storm water impact on aquatic life. For parameters other than metals, this will entail a simple comparison of the observed grab or flow-weighted composite concentration and the corresponding criterion. The toxicity of several trace metals increases as hardness decreases. Consequently, the acute criteria for most metals must be calculated for each sample, based on the hardness measured in the sample. Dissolved metal fraction can be calculated using GAEPD's guidelines provided in (GAEPD Water Quality Controls 391-3-6, Amended April 2000).

Pollutant Loading Estimates

Pollutant loading estimates may provide an indication of the potential impact of a storm water discharge on a receiving water body. Calculating pollutant loads provides a direct quantitative measurement of the pollutants in storm water discharge to the receiving water.

Calculate pollutant loads using either an estimate of flow in an average year (annual load) or flow measured during a specific storm event.

Loadings can be estimated using Schueler's simple model (EPA 1992), statistical models, or one of several dynamic models. The simple model estimates the mean annual pollutant loading from a particular outfall or subbasin to a receiving water or is adapted to estimate average seasonal or storm event loadings. A statistical-based model, such as the Federal Highway Administration model (Driscoll et al. 1990), can be used to characterize the variability of pollutant loading and concentrations, including the expected frequency of exceeding water quality criteria. A dynamic model also can calculate the expected frequency of exceedences. In addition, a dynamic model can account for the variability inherent in stormwater discharge data, including variations in concentration, flow rate, and runoff volume. Thus, it can be used to calculate the entire frequency distribution for the concentration of a pollutant and the theoretical frequency distribution (i.e., the probability distribution) for loadings from the outfall or subbasin. This enables the modeler to describe the effects of observed discharges on receiving water quality in terms of the frequency at which water quality standards are likely to be exceeded. Dynamic models include EPA's Stormwater Management Model (SWMM) and Hydrologic Simulation Program (HSPF), the USACE Storage, Treatment, Overflow, Runoff Model (STORM), and Illinois State Water Survey's Model QILLUDAS (or Auto-QI) (EPA 1992).

Whatever method is used to estimate annual pollutant loadings, use an estimate of the EMC as input. Build-up/wash-off functions, which are available in SWMM and several other models, cannot accurately simulate all of the ways pollutants can enter stormwater; thus, the results should be interpreted with caution. The EMC is defined as the storm constituent mass discharge divided by the total storm flow volume. In stormwater monitoring programs, the EMC is estimated from the concentration of a constituent in a flow-weighted composite sample. Studies by Collins and Dickey (1992) demonstrate that the EMC derived from a flow-weighted composite sample does a good job of estimating the true event mean concentration for all but very short, intense storms. During short storms, the automated sampler cannot be programmed to collect a sufficient number of samples to ensure that the results are representative.

Assessing Trends in Stormwater Discharge Quality

Time Trends

Several statistical methods, both parametric and nonparametric, are available for detecting trends. They include graphical methods, regression methods, the Mann-Kendall test, Sen's non-parametric estimator of slope, the Seasonal Kendall test (Pitt 1994) and ANOVA. Preliminary evaluations of data correlations and seasonal effects should be made prior to trend analysis. Data auto-correlation is more likely if data are taken close together in time. Close data can be influenced by each other and do not provide unique information. Seasonal effects should also be removed, or a procedure that is unaffected by data cycles should be selected (seasonal Kendall test). The correlation between concentration and flow should also be checked by fitting a regression equation to a concentration versus flow plot. The effect of any such correlations should be eliminated from the data prior to the trend analysis.

Graphical Methods

Plots of trends in constituent concentrations over time can be examined for seasonal or annual patterns:

- Sort the data set by station and sampling date (i.e., first station and oldest sampling data are the first line of data
- For each station, select "date" as the x-variable and plot the parameter of interest on the y-axis
- Visually inspect the data for upward or downward trends and note any large "peaks" or "valleys"
- Compute annual or seasonal mean concentrations and plot these values versus the date, then inspect for upward or downward trends

Statistical Tests

Time trends can be assessed using a variety of statistical tests. Some of the more commonly used tests include:

- Regression
- Mann-Kendall Test
- Sen's Nonparametric Estimator of Slope
- Seasonal Kendall Tests
- Analysis of Variance

It is important that you use a test that is appropriate to your data and objectives. Refer to Gilbert (1987) or another statistics textbook for detailed information about these tests.

Regression

Traditional linear regression ($Y = a + bX$) applied to the data presented graphically. Linear regression demonstrates the direction and strength of the relationship.

Seasonal Kendall Tests Computation Procedure

Seasonal changes can be a major source of variation in the indicator value (Y) in many instances. Therefore, seasonal variations must be compensated for in order to better detect trends over time. Considering the seasonality of water quality and environmental indicators, the Seasonal Kendall Test computes the Kendall's test statistic on each of m seasons separately, and then combines the result to test for an annual trend. The "seasons" can be defined as months, wet or dry seasons, etc. Data for a given season are compared only with data from the same season from other years. The null hypothesis, H_0, and alternative hypothesis, H_1, are:

H_0: No trend exists, i.e. tau = 0, or X and Y are independent;

H_1: A trend exists, i.e. tau $\neq$ 0, or X and Y are dependent.

For this analysis, the null hypothesis means that the relative difference between observed values over time (y) is independent of time (X) and that no trend exists. The alternative hypothesis means that a trend exists and that the values observed have been changing over time. Guidance by way of a step-by-step description for applying Kendall's Tau is described below.

Step 1: Order the data from each site chronologically.

Step 2: Calculate Y:

Y = Water quality data at each site

Step 3: Calculate the statistic S.

The statistic S measures the direction of differences between serial observations over time. In Kendall's Tau Test, S is calculated by subtracting the number of discordant pairs, M (the number of x, y pairs where Y decreases as X increases), from the number of concordant pairs, P (the number of X, y pairs where Y increases with increasing X). For a data set with n pairs:

$$S = P - M$$

Where:

P = the number of times the Y's increase as the X's increase, or the number of $y_i < y_j$ for all $i < j$,

M = the number of times the Y's decrease as the X's increase, or the number of $y_i > y_j$, for $i < j$, for all I = 1, 2, … (n-1), j = I+1, n.

The annual or overall statistic S_k can be obtained by summing Kendall's S statistic (S_i) for each season:

$$S_k = \sum_{i=1}^{m} S_i$$

Where:

m = number of "seasons" in a year.

The expectation of the S_k, which is equal to the sum of expectations of individual S_i, is zero.

Step 4: Calculate the Statistic Tau (τ)

$$\tau = S / [n*(n+1)/2]$$

Among all possible comparisons, if all Y values increase over time, S = n* (n-1)/2, and the correlation statistic tau equals +1. When all Y values decrease over time, the statistic tau equals –1. The value of tau should always fall in the range of –1 to +1. Positive values of S and tau indicate an "upward trend" (Y increases with time), and negative values of S and tau indicate a "downward trend".

Step 5: Test the Significance of Tau

To test for the statistical significance of tau, S is compared to what would be expected when the null hypothesis is true (no trend exists). If it is further from zero than expected, the null hypothesis, H_0, is rejected (i.e., a trend exists).

Although it is recommended that records greater than ten years in length should be used, the Kendall's Tau Test can be applied to a shorter period of data. For $n \geq 10$, the test statistic can be modified to be closely approximated by a normal distribution. For $n < 10$, an exact test should be computed. A table of exact critical values is listed in Helsel and Hirsch (1992).

When the product of the number of seasons and the number of years is more than 25, the distribution of the annual or overall statistic, S_k, can be approximated by a normal distribution. The expected value of S_k equals zero, and its variance is equal to the sum of the variances of S_i. To test for significance, S_k is first standardized by subtracting its expected value and dividing by its standard deviation, σ_{sk}. The resulting Z_{sk} is evaluated against the table of the normal distribution:

$Z_{sk} = (S_k - 1) / \sigma_{sk}$ if $S_k > 0$

$Z_{sk} = 0$ if $S_k = 0$

$Z_{sk} = (S_k + 1) / \sigma_{sk}$ if $S_K < 0$

Where

$$\sigma_{sk} = \sqrt{\sum_{i=1}^{m} (n_i / 18)(n_i - 1)(2n_i + 5)}$$

n_i = number of Y values in the i^{th} season.

The null hypothesis is rejected at significance level α (indicating a significant trend exists) if $|Z_{sk}| > Z_{crit}$, where Z_{crit} is the value of the standard normal distribution with a probability of exceedence of $\alpha / 2$.

Dealing with Missing Data and Ties

Missing data are not unusual in water quality monitoring data analysis, and can be expected. Modification must be made to accommodate missing and tied data when applying the Kendall tests.

When missing values are encountered in the calculation of S, a value of zero is assigned. This protocol applies to either earlier (y_i) or later (y_j) missing values because it is impossible to tell whether a missing value is greater or less than the actual value. The Kendall test statistic S is unchanged where missing values occur. The σ_s is calculated as:

$$\sigma = \sqrt{(n_g / 18) * (n_g - 1) * (2n_g + 5)}$$

Where:

n_g = the number of non-missing data.

To compute τ when ties are present tied values of y_i and y_j produce a 0 instead of either +1 or –1. In this way, ties do not contribute to either P or M. S and τ are calculated exactly the same. For testing the significance level of a large sample approximation Z_s, σ_s is calculated as:

$$\sigma = \sqrt{1/18[(n-1)*(2n+5) - \sum_{i=1}^{n} t_i(i)(i-1)(2i-5)]}$$

Where:

t_i = the number of ties of extent i.

More detailed information on dealing with missing and tied data is presented by Hirsch et al, (1982), Hirsch and Slack (1984), and Helsel and Hirsh (1992).

Other Issues

Because tau depends only on the ranks of data as presented above, it can be implemented in cases where some data are censored, such as concentrations reported as "below the detection limit". Use of one-half of the detection level or other standard methods for approximating data reported as below the detection level can be employed, and the estimated values can be included in the ranking.

The tau value is generally lower than values of traditional correlation coefficients for linear relations of the same strength. These lower values simply mean that a different scale of correlation is being used. As such, it is improper to compare tau values with the traditional correlation coefficient from a parametric test to decide which method is more sensitive.

The Kendall Test can be computed manually, and computer programs are also available (Press et al., 1989).

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NOTE: This Appendix was summarized from the Washington State Department of Ecology Stormwater Quality Monitoring Guidance Manual, November 1995.

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PART 2B

Long-Term Ambient Trend Monitoring -- Manual Grab Sampling

This section contains the guidance for the District Long-Term Ambient Trend Monitoring using manual grab sampling.

2B.1 Overview

Grab samples are discrete water samples pulled from a surface water body. They may be collected as single samples at a given location or time, or may be composited across a stream's width or depth, or over the duration of a storm-event using time or flow weighting. This section cover procedures for simple manual grab samples and time- and flow-weighted manual composite sampling. Equal Width-Integrated and Equal Depth-Integrated grab sampling methods are presented in Part 2C.

Manual grab sampling methods may be used for wet weather monitoring for small and medium streams and where samples during storm flows can be collected without danger of personal harm or injury. Grab sampling is typically used for dry weather monitoring since chemical water quality parameters are more constant over time compared to wet weather events. Grab samples are recommended for bacteria (fecal coliform and E. Coli) monitoring due to the time sensitive nature of laboratory analysis for bacteria.

2B.2 Sampling Preparation

2B.2.1 Grab Sampling Equipment

Sampling station characterisics often dictate the equipment to be used. Direct dipping of the sample container into a stream is desirable. However, if the stream is too deep to wade, or the sample must be collected from a bridge or other access point due to safety considerations, then supplemental sampling equipment may be needed.

2B.2.2 Sampling Equipment Checklist

Before visiting a sampling station, the field team should ensure that all of the necessary equipment is present and in order. Table 2B-1 shows the required equipment needed for long-term ambient trend monitoring (*this checklist assumes that sample filtration and preservation occurs at the laboratory rather than in the field – see the next section for more details*).

TABLE 2B-1
List of Equipment and Supplies for Long-Term Ambient Trend Monitoring

Field Equipment	Function
Sample bottles with labels	For collection of wet/dry weather grab samples
Sealed, sterile sample bottles with labels	For collection of bacteria (fecal coliform and E. Coli) grab samples
Composite jar	For collection of wet weather composite samples only
Clear tape and applicator	To apply over label
Coolers	For transport of grab samples
Cloth or mesh grocery bag	For transport of composite jar
Ice/ ice packs	To keep samples preserved after collection and during transport from the site
Clipboard with data collection forms, COC forms, and pens	To document field data and activities
List of directions, protocols, and H&S plan.	For reference in the field
Field logbook and Sharpie (extra fine)	To record notes and label sample bottles
Cell phone	Communication in the field.
First Aid Kit	Health and Safety Plan
Disposable gloves, safety shoes, and safety glasses	Health and Safety Plan

2B.2.3 Wet Weather Event Considerations

Obtaining the most reliable and current information on the intensity and duration of forecasted precipitation events is critical to sampling wet weather events. Once it has been determined that a storm event is expected that is predicted to meet the 0.3 inch rainfall criteria and antecedent dry period, the field team should begin final preparation for the sampling event. The analytical laboratory should be contacted to alert them to the potential of an upcoming sampling event.

Manual grab sampling should be conducted on the rising limb of the hydrograph and as close to the peak as possible to more accurately estimate pollutant loadings during wet weather events.

2B.3 Sample Collection and Handling

The following procedures should be used for collecting manual grab samples.

2B.3.1 Grab Sampling Protocols

- Grab samples should be collected before any other water quality work is performed so that the sampling stream will not be disturbed.
- Never sample pooled/ponded (stagnant) or backwater flow.
- Always collect sample where water is freely flowing.
- Do not open sample bottle until sample is to be actually collected.

- Use gloves at all times when handling sampling bottles.
- A minimum of three aliquots should be collected with an adequate sample volume to analyze the required parameters for the wet weather and dry weather monitoring requirements.
- Bacteria (fecal coliform and E. Coli) grab samples must be collected directly into the sterile sample container.

2B.3.2 Grab Sample Collection

1. Locate the center of the stream current. If collecting the sample in-stream, wade out to this point. If collecting the sample from a boat or bridge, position sampling equipment above this point.
2. Remove the seal and carefully remove the lid of the sample container. To avoid possible contamination, make sure fingers do not come in contact with the inner surface of lid or inside of bottle. Hold the lid cupped inside the hand so that only the outside of the lid is touched or carefully set down the lid so the inside is facing up.
3. Face upstream and hold the samping container so the opening faces upstream.
4. Plunge the open container into the stream at mid-depth or at least 8-10 inches underwater (facing upstream) and fill the bottle to the required fill line. Some air space should remain in the bottle. Avoid stirring up bottom sediments and keep the sample free of uncharacteristic floating debris. If the water has been disturbed, wait a few minutes for water to clear or sample further upstream where water has been undisturbed.
5. After collecting enough samples, secure the lids tightly onto the containers. To avoid possible contamination, make sure fingers do not come in contact with the inner surface of lid or inside of bottle.
6. Preserve the sample(s) as necessary (see Appendix 2A-5).
7. Label the sample container(s) with the appropriate information.
8. Place the filled sample container(s) upright in the ice chest immediately.
9. Complete field measurements for ph, conductivity, and temperature.
10. Record sample collection information in the field logbook and complete the chain-of-custody form and field sheets.

2B.3.3 Manual Composite Sample Collection

Composite sampling involves the collection of discrete grab samples and pooling portions of each grab sample into a composite bottle in order to obtain an average sample over a storm event for wet weather monitoring. This may be done on a time-weighted or flow-weighted basis. Appendix 2A-3 provides a discussion of flow versus time composite sampling.

Consideration for manual composite sample collection:

- The aliquots for composite samples must be collected within the first three hours of the storm event (or during the entire event if the storm is less than three hours).
- For flow-weighted samples, equal aliquots may be collected at the time of sampling and then flow-proportioned and composited in the laboratory, or the aliquots taken may be based on the flow rate at the time of sample collection and composited in the field.
- A minimum of 15 minutes must separate the collection of each sample aliquot, and a minimum frequency of three sample aliquots within each hour of discharge must be maintained.

The following procedure should be used for manual flow-weighted composite sample collection:

1. After rain begins to fall, record the date and time rain started to produce stormwater runoff, and determine and record streamflow at this time. Determine appropriate volume for sample aliquot.
2. Locate the center of the stream current. If collecting the sample in-stream, wade out to this point. If collecting the sample from a boat or bridge, position sampling equipment above this point.
3. Face upstream and hold the sample container so the opening faces upstream.
4. Lower the sample container (for laboratory compositing) or stainless steel or PTFE sample aliquot measuring device (for onsite compositing) into the center of the stream current. Record the time and water depth. Avoid stirring up bottom sediments and keep sample free of uncharacteristic floating debris.
5. Allow the device to fill with appropriate volumes based on flow observed, or suggest a minimum of 1000 mL be acquired for each aliquot (for laboratory compositing). NOTE: The laboratory should also be consulted to determine the overall sample volumes necessary for the required analyses to ensure that sufficient volumes of individual sample aliquots are collected to support the composite sample.
6. Repeat the steps (354) for each aliquot of the composite sample, retaining each stormwater sample aliquot in separate, labeled sample containers. As stated above, a minimum frequency of three sample aliquots within each hour of the storm event for the first three hours or duration of the storm event, if less than three hours.
7. If field compositing is performed and after the sample aliquots have been collected, combine appropriate volumes of sample aliquots into stainless steel or PTFE bucket to create the flow-weighted composite sample. Fill appropriate sample containers with the composite sample mixture, using care not to overfill them.
8. Secure the lids tightly onto the containers. To avoid possible contamination, make sure fingers do not come in contact with the inner surface of lid or inside of bottle.
9. Preserve the sample(s) as necessary (see Appendix 2A-5).

10. Label the sample container(s) with the appropriate information.
11. Place the filled sample container(s) upright in the ice chest immediately.
12. Complete field measurements for ph, conductivity, and temperature.
13. Record sample collection information in the field logbook and complete the chain-of-custody form and field sheets.

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Atlanta Regional Commission (ARC). 2000. Atlanta Region NPDES MS4 Phase I Standard Operating Procedures for Stormwater Monitoring.

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PART 2C

Long-Term Ambient Trend Monitoring -- EWI/EDI Composite Grab Sampling

This section contains the guidance for the District Long-Term Ambient Trend Monitoring using the equal-width-increment (EWI)/equal-depth-increment (EDI) composite grab sampling method. This approach is considered an alternative to the use of automated composite samplers.

These procedures and protocols were taken directly from Chapter 4, Section 4.1.1 of the USGS National Field Manual.

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4.1.1 FLOWING-WATER SITES

Flowing streamwater is collected using either isokinetic, depth-integrating or nonisokinetic sampling methods. Isokinetic, depth-integrating methods are designed to produce a discharge-weighted (velocity-weighted) sample; that is, each unit of stream discharge is equally represented in the sample (Office of Water Quality Technical Memorandum 99.02). The analyte concentrations determined in a discharge-weighted sample are multiplied by the stream discharge to obtain the discharge of the analyte.

Collection of an isokinetic, depth-integrated, discharge-weighted sample is standard procedure; however, site characteristics, sampling-equipment limitations, or study objectives constrain how a sample is collected and could necessitate use of other methods. If the QC plan calls for collection of concurrent samples, then the relevant procedures must be reviewed and the appropriate equipment prepared (section 4.3).

Nonisokinetic sampling methods, such as those involving use of an automated point sampler, generally do not result in a discharge-weighted sample unless the stream is completely mixed laterally and vertically. Thus, the analytical results cannot be used to directly compute analyte discharges.

Document the sampling method used on the appropriate field form for each sample.

Isokinetic, Depth-Integrated Sampling Methods 4.1.1.A

Collection of isokinetic, depth-integrated samples involves using either an equal-width-increment (EWI) or equal-discharge-increment (EDI) sampling method. The EWI or EDI methods usually result in a composite sample that represents the discharge-weighted concentrations of the stream cross section being sampled. The EWI and EDI methods are used to divide a selected cross section of a stream into increments having a specified width. The term **vertical** refers to that location within the increment at which the sampler is lowered and raised through the water column. EWI verticals are located at the midpoint of each width increment. EDI verticals are located at the centroid, a point within each increment at which stream discharge is equal on either side of the vertical.

Isokinetic samplers usually are used to obtain a discharge-weighted sample along the stream cross section. When using an isokinetic sampler there should be no change in velocity (speed and direction) as the sample enters the intake (fig. 4-1). If properly implemented, EDI and EWI methods should yield identical results. The uses and advantages of each method are summarized below and in table 4-3.

- Collect isokinetic, depth-integrated samples by using a standard depth- and width-integrating method if analysis of a representative sample from a cross section of flowing water is required for discharge computations. Appendix A4-A and Edwards and Glysson (1998, figures 39-43), provide detailed information about isokinetic, depth-integrating transit rates for collecting samples.

- For isokinetic sampling, the mean velocity of the vertical that is sampled must exceed the minimum-velocity requirement of an isokinetic sampler—the minimum velocity requirement is either 1.5 ft/s for a bottle sampler or 3 ft/s for a bag sampler (Appendix A4-A; NFM 2).
  - The transit rate (the rate at which the sampler is lowered or raised) used to collect an isokinetic, depth-integrated sample is mainly a function of the nozzle diameter of the sampler, volume of the sampler container, stream velocity, and sampling depth (Appendix A4-A; NFM 2). Note that water temperature can affect isokinetic sampling. For example, bag samplers do not work isokinetically in water temperatures that are less than about 7 °C.
  - An error in concentrations of suspended particulates coarser than 62 mm can be significant when the velocity of the sample entering the nozzle and the stream velocity differ significantly. The velocity of the sample entering the nozzle also can be affected by the transit rate: too fast a transit rate will cause a sampler to undersample sand-sized particulates (Edwards and Glysson, 1998).
  - The transit rate must be kept constant during sampler descent through a vertical and also during sampler ascent through a vertical. Although not necessary, usually the same transit rate is used for raising the sampler as was used for lowering the sampler through a given vertical.

RULE OF THUMB: For isokinetic, depth-integrating sampling, do not exceed the designated maximum transit rate.

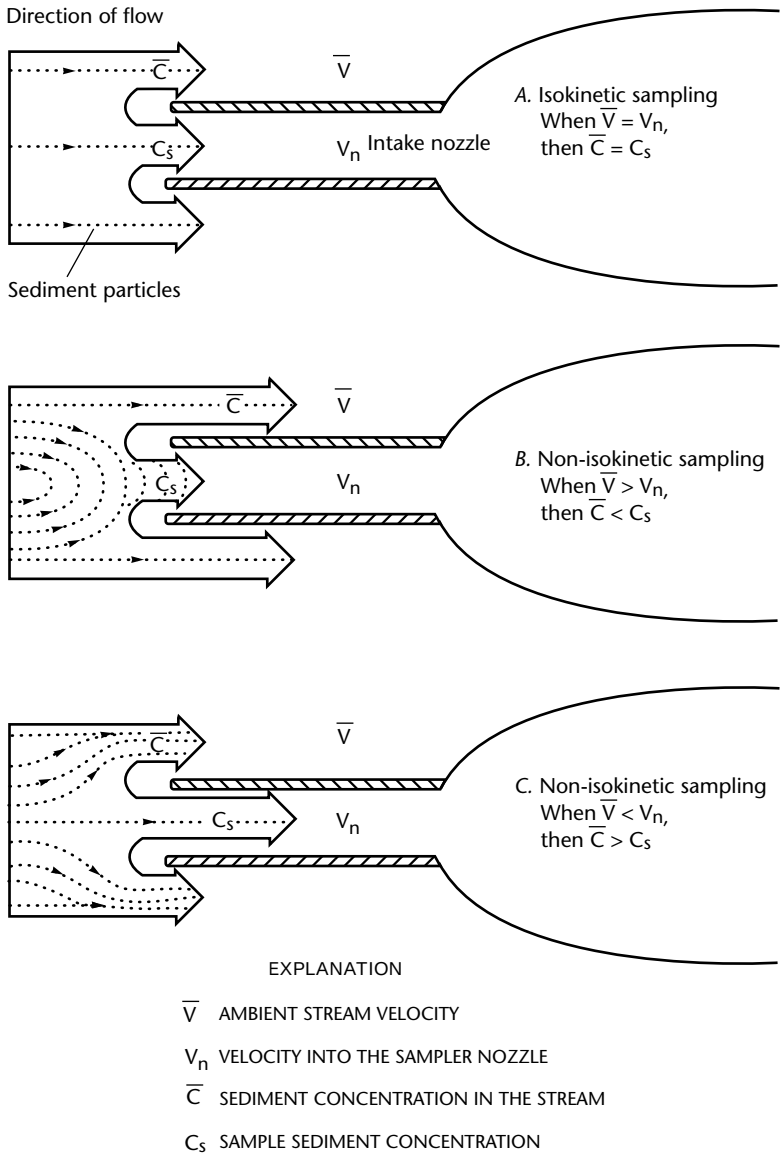


Figure 4-1. Relation between intake velocity and sediment concentration for isokinetic and nonisokinetic collection of water samples that contain particulates greater than 0.062 millimeters (modified from Edwards and Glysson, 1998, p. 13).

The number of increments needed in order to get a discharge-weighted sample at a site is related primarily to data objectives (for example, the accuracy needed) and how well-mixed or heterogeneous the stream is with respect to the physical, chemical, and biological characteristics of the cross section. The recommended number of increments for EWI and EDI methods are discussed in the sections to follow. Edwards and Glysson (1998) describe a statistical approach for selecting the number of increments to be used, based on sampling error and suspended-sediment characteristics.

Selecting the number of increments

- Examine the variation in field-measurement values (such as specific electrical conductance, pH, temperature, and dissolved oxygen) along the cross section (NFM 6).
- Consider the distribution of streamflow (discharge), suspended-materials concentration and particle-size distribution, and concentrations of other targeted analytes along the cross section. Consider whether the distribution or analyte concentrations will change during sample collection.
- Consider the type of sampler that will be used and the volume of sample that will have to be collected for the analysis of the target analytes.
- Avoid side-channel eddies. EDI and EWI methods cannot be used at locations with upstream eddy flow.

Table 4-3. Uses and advantages of equal-width-increment (EWI) and equal-discharge-increment (EDI) sampling methods

EWI method	Advantages of the EWI method
EWI is used when information required to determine locations of sampling verticals for the EDI method is not available, and (or) the stream cross section has relatively uniform depth and velocity. Use EWI whenever: • The location of EDI sampling verticals changes at the same discharge from one sampling time to another. This situation occurs frequently in streams with sand channels.	• EWI method is easily learned and implemented for sampling small streams. • Generally, less time is required onsite if the EWI method can be used and information required to determine locations of sampling verticals for the EDI method is not available.
EDI method	**Advantages of the EDI method**
EDI is used when information required to determine locations of sampling verticals for the EDI method is available. Use EDI whenever: • Small, nonhomogeneous increments need to be sampled separately from the rest of the cross section. The samples from those verticals can be analyzed separately or appropriately composited with the rest of the cross-sectional sample. (Have the sampling scheme approved.) or • Flow velocities are less than the isokinetic transit-rate range requirement. A discharge-weighted sample can be obtained, but the sample will not always be isokinetic. or • The EWI sampling method cannot be used. For example, isokinetic samples cannot be collected because stream velocities and depths vary so much that the isokinetic requirements of the sampler are not met at several sampling verticals. or • Stage is changing rapidly. (EDI requires less sampling time than EWI, provided the locations of the sampling verticals can be determined quickly.)	• Fewer increments are necessary, resulting in a shortened sampling time (provided the locations of sampling verticals can be determined quickly and constituents are adequately mixed in the increment). • Sampling during rapidly changing stages is facilitated by the shorter sampling time. • Subsamples making up a sample set may be analyzed separately or may be proportionally composited with the rest of the cross-sectional sample. • The cross-sectional variation in constituent discharge can be determined if subsample bottles are analyzed individually. • A greater range in velocity and depths can be sampled isokinetically at a cross section. • The total composite volume of the sample is known and can be adjusted before sampling begins.

Equal-width-increment (EWI) method

For the EWI sampling method, the stream cross section is divided into a number of equal-width increments (fig. 4-2). Samples are collected by lowering and raising a sampler through the water column at the center of each increment. (This sampling location is referred to as the vertical.) The combination of the same constant transit rate used to sample at each vertical and the isokinetic property of the sampler results in a discharge-weighted sample that is proportional to total streamflow.

- **Isokinetic sampling is required for the EWI method.** Use isokinetic, depth-integrating sampling equipment (NFM 2).
  - Use the same size sampler container (bottle or bag) and nozzle at each of the sampling verticals (fig. 4-2).
  - Collect samples using the same transit rate at each vertical during descent and ascent of the sampler. The transit rate must be constant and within the operational range of the sampler (Appendix A4-A).
- Composite the subsamples from all verticals in a churn splitter or process subsamples through the cone splitter (NFM 5).

Do not use EWI when stream velocities are less than the minimum velocity required for the isokinetic sampler selected:

- 1.5 ft/s for the bottle sampler
- 3 ft/s for the bag sampler

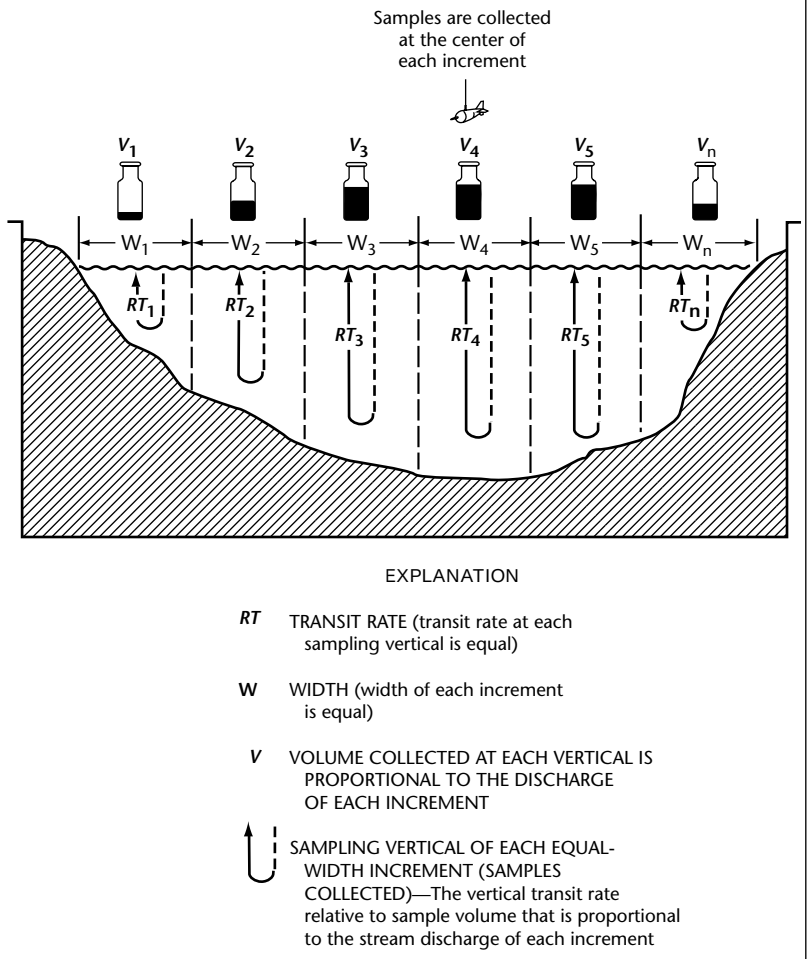
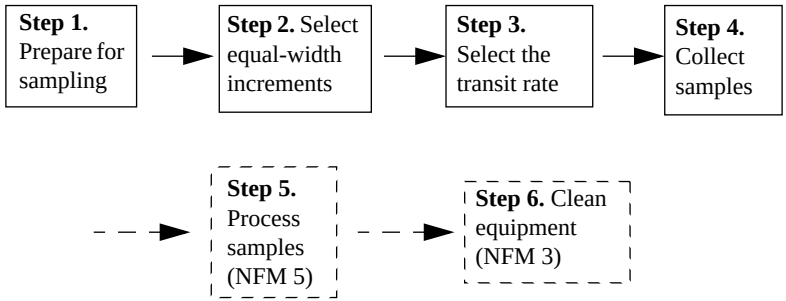


Figure 4-2. Equal-width-increment method for collection of water samples (modified from Edwards and Glysson, 1998).

Guidelines for the EWI sampling method



Be sure that the field effort is adequately staffed and equipped. Check QC requirements before departing—QC samples require additional equipment and supplies.

Step 1. Prepare for sampling[7]

a. Upon arrival at the field site, set out safety equipment such as traffic cones and signs. Park vehicle in a location and direction so as to prevent sample contamination from vehicle emissions.

b. Assemble sampling equipment and set up a clean work space.

- Organic compounds. Select equipment with fluorocarbon polymer, glass, or metal components if components will directly contact samples to be analyzed for organic compounds. Do not use plastics other than fluorocarbon polymers.
- Inorganic constituents. Select equipment with components made of fluorocarbon polymer or other relatively inert and uncolored plastics or glass if components will directly contact samples to be analyzed for inorganic constituents. Do not use metal or rubber components for trace-element sampling.
- Microbiological analyses. Collect samples for microbiological analyses using equipment and techniques described in NFM 7.

[7]Preparations for water sampling are described in NFM 1, 2, and 3. Consult NFM 5 for sample processing, NFM 6 for field measurements, NFM 7 for biological indicators, NFM 8 for bottom-material sampling and NFM 9 for field safety.

Step 2. Select the number and width of equal-width increments.

a. Visually inspect the stream from bank to bank and longitudinally, observing velocity, width, and depth distribution, and apparent distribution of sediment and aquatic biota along the cross section. Note and document the location of stagnant water, eddies, backwater, reverse flows, areas of faster than normal flow, and piers or other features along the cross section.

b. Determine stream width from a tagline or from distance markings on a bridge railing or cableway.

c. At sites with little sampling history, measure and record the cross-sectional variation of field measurements (such as specific electrical conductance, pH, temperature, and dissolved oxygen). Review the magnitude of the variations along the cross section.

d. Determine the width of the increment. To obtain the number of increments, divide the stream width by the increment width. The number of increments must be a whole number. Increment width is based on study objectives, variation in field measurements and flow, and stream-channel characteristics along the cross section.

   - Collect the subsample at the center of each equal-width increment (the vertical).
   - If the subsample does not represent the mean value for that increment, decrease the increment width until the mean value for the increment is represented. This will increase the number of increments sampled.

e. Locate the first sampling vertical at a distance of one-half of the selected increment width from the edge of the water. Locate all the other verticals at the center of each remaining equal-width increment along the cross section.

   Example:

   - If a stream 56 ft wide has been divided into 14 increments of 4 ft each, the first sampling vertical would be 2 ft from the water's edge and subsequent verticals would be at 6, 10, 14 ft from the water's edge, and so forth.
   - Even if streamflow is divided, as in a braided channel, equal-width increments must be identical from channel to channel, and the same constant transit rate must be used at each vertical.

f. Make slight adjustments to sampling locations, if necessary, to avoid sampling where the flow is affected by a pier or other obstruction.

TECHNICAL NOTE: Sampling near or downstream from large in-stream obstructions such as bridges and piers could result in artificially elevated concentrations of suspended sediments if the sampler is immersed in an eddy that is caused by the obstruction. If it is necessary to include an eddy in the cross section to be sampled, consider treating the eddy as a solid obstruction: subtract the eddy width from that of the total cross section, and determine the width of the increments based on the remaining stream width.

RULE OF THUMB

When selecting the number of equal-width increments:

- Cross-sectional width ≥ 5 ft—use a minimum of 10 equal-width increments.
- Cross-sectional width <5 ft—use as many increments as practical, but equally spaced a minimum of 3 in. apart.

Equipment limitations also constrain the number of increments selected; for example:

- When using a D-95 at maximum depth with a 14-L churn splitter, EWI samples can be collected at approximately 14 verticals. If an 8-L churn splitter is used, samples can be collected at approximately 10 verticals.
- When using a D-77 and a 14-L churn splitter, the maximum average depth must not exceed 5 ft when samples are collected at 10 verticals.

Step 3. Select the transit rate.

a. Refer to Appendix A4-A for guidelines for determining the transit rates for collecting isokinetic, depth-integrated samples. Unless the mean velocity is actually determined, use the trial-and-error method to determine the minimum transit rate.

b. Locate the equal-width increment containing the largest discharge (largest product of depth times velocity) by sounding for depth and either measuring or estimating velocity. At the vertical for this increment, use of the minimum transit rate results in the maximum allowable filling of the sampler bottle or bag during one vertical traverse.

c. Determine the minimum transit rate at this vertical for the type of sampler (bottle or bag), size of sampler nozzle, and the desired sample volume.

- Approximate the mean velocity of the vertical in feet per second by timing a floating marker (such as a peanut) as it travels a known distance. (A known length of flagging tape tied to the cable where the sampler is attached often is used to measure the distance.) Divide the distance (in feet) by the time (in seconds) and multiply by 0.86.
- Make sure that the transit rate does not exceed the maximum allowable transit rate to be used at any of the remaining verticals along the cross section. This can be determined by sampling the slowest increment. If the minimum volume of sample (relative to depth of the vertical) is not collected at this vertical, then the EWI method cannot be used at this cross section to collect a discharge-weighted sample (Appendix A4-A).

Guidelines for selecting the transit rate for EWI sampling
• The descending and ascending transit rate must be constant in each direction and must be the same for each vertical along the cross section.
• Do not exceed the maximum allowable transit rate if using EWI. If the transit rate must exceed the maximum allowable rate, use EDI instead of EWI.
• The transit rate selected must be sufficiently rapid to keep from overfilling the sampler. The sampler is overfilled when the water surface in the sampler container is above the bottom edge of the nozzle when the sampler is held in the sampling position.
• The same size sampler nozzle and container must be used at all verticals along the cross section.
• If the total volume collected will exceed the recommended volume for the churn splitter, then a cone splitter must be used.

Step 4. Collect samples.

The sample-collection procedure is the same whether you are wading or using the reel-and-cable suspension method. **Use *CH/DH* techniques, as required (section 4.0.1). Always follow safety procedures (NFM 9)**.

a. Move to the first vertical (midpoint of first EWI near edge of water) and field rinse equipment (section 4.0.2).

b. Record start time and gage height.

c. Lower field-rinsed sampler at the predetermined constant transit rate until slight contact is made with the streambed. Do not pause upon contacting the streambed. Raise the sampler immediately at the same constant transit rate until sampler completes the vertical traverse.

   - Take care not to disturb the streambed by bumping the sampler on it; bed material may enter the nozzle, resulting in erroneous data.
   - Do not overfill the sampler container. Overfilling results in a sample that is not isokinetic and that could be enriched with heavy particulates because of secondary circulation of water through the sampler (from nozzle through air exhaust). This enrichment will result in an artificially increased sediment concentration and will bias particle-size distribution toward heavier and larger particulates.
   - Do not underfill the sampler container (Appendix A4-A). Underfilling will result in a sample that is not isokinetically collected because the maximum transit rate has been exceeded.
   - If the required volume cannot be collected, use the EDI method to obtain discharge-weighted samples.

d. Inspect each subsample as it is collected, looking for overfilling or underfilling of the sampler container and (or) the presence of anomalously large amounts of particulates that might have been captured because of excessive streambed disturbance during sample collection. If you note any of these conditions, discard the sample, making sure there are no residual particulates left in the container, and resample.

e. Move sampling equipment to the next vertical. Maintain the selected transit rate. The volume of the subsample can vary considerably among verticals. Subsamples can be collected at several verticals before emptying the sampler container, as long as the maximum volume of sample in a bottle or bag sampler has not been exceeded. If the container is overfilled, it is necessary to resample.

> TECHNICAL NOTE: The tables in Appendix A4-A apply to the first complete round-trip transit starting with an empty sampler container. These tables cannot be used if the sampler is not emptied between verticals.

f. Continue to the next vertical until no more samples can be collected without overfilling the sampler container. Empty the subsample into a field-rinsed churn or cone splitter and repeat sample collection in the same manner until subsamples have been collected at all the verticals.

- If the total volume of the subsamples to be collected will exceed the operational capacity of the churn, select from the following options: use either a sampler with a smaller bottle or a bag sampler with a smaller nozzle; or use a cone splitter; or use the EDI method, if appropriate.
- To ensure that all particulates are transferred with the sample, swirl the subsample gently to keep particulates suspended and pour the subsample quickly into the churn or cone splitter.
- Sample EWI verticals as many times as necessary to ensure that an adequate sample volume is collected as required for analysis, but sample at each vertical an equal number of times. (The composite cross-sectional sample will remain proportional to flow at the time of sampling.)
- If flow is stable during sampling, then multiple samples can be collected at each vertical during a single traverse along the cross section. If flow is changing, however, study objectives should determine whether to collect multiple samples at each vertical during a single traverse or to collect one sample at each vertical during multiple traverses along the cross section. Document on field forms the method used.

g. Record the following information after all samples have been collected:

- Sampling end time.
- Ending gage height.
- All field observations and any deviations from standard sampling procedures.

Step 5. Process Samples ➜ Refer to NFM 5.

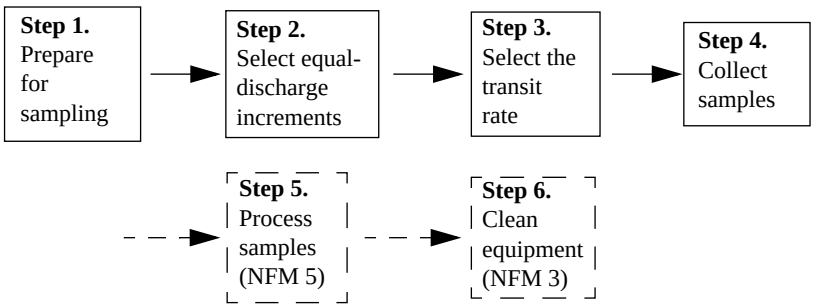
Step 6. Clean Equipment ➜ Refer to NFM 3.

- If the sampler will not be reused during a field trip, rinse sampler components with deionized water before they dry and place them into a plastic bag for transporting to the office laboratory to be cleaned.
- If the sampler will be reused during the field trip, rinse the components with DIW while still wet from sampling and then field-clean while at the sampling site using the prescribed procedures (NFM 3). Reassemble the sampler.
- Collect a field blank, if required, after sampling equipment has been cleaned at the sampling site.
- Place the cleaned sampler into a plastic bag and seal for transport to the next site.

Equal-discharge-increment (EDI) method

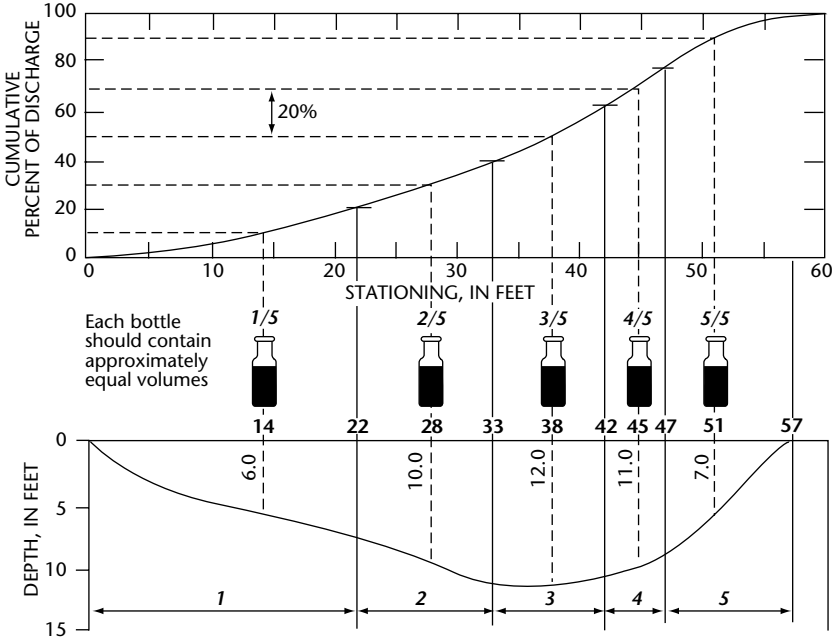
The objective of the EDI method is to collect a discharge-weighted sample that represents the entire flow passing through the cross section by obtaining a series of samples, each representing equal volumes of stream discharge. The EDI method requires that flow in the cross section be divided into increments of equal discharge. Equal-volume, depth-integrated samples are collected at the centroid of each of the equal-discharge increments along the cross section (fig. 4-3). Centroid is defined as that point in the increment at which discharge is equal on both sides of the point.

Guidelines for the EDI sampling method



Be sure that the field effort is adequately staffed and equipped. Check QC requirements before departing—QC samples require additional equipment and supplies.

Example: Sampler D77; nozzle size, 5/16 inches ID; 3 Liter sample bottle; width 57 feet; maximum depth 12 feet; maximum velocity, 5.0 ft/s; width of section containing 20 percent of flow is variable, 5 to 22 feet; 20 percent of flow per section will give 5 sampling verticals; transit rate variable, 0.3 to 1.7 ft/s.



Sampling vertical bottle/ number	Percent discharge	Increment centroid from left edge of water, in feet	Increment depth, in feet	Velocity, in feet per second	Transit rate to give 2.7 liters, in feet per second
1/5	20	14	6	2.5	0.16
2/5	20	28	10	3.0	.33
3/5	20	38	12	3.1	.41
4/5	20	45	11	6.1	.72
5/5	20	51	7	4.8	.37

Figure 4-3. Equal-discharge-increment method for collection of water samples (modified from Bruce Ringen, U.S. Geological Survey, written commun., 1978).

Step 1. Prepare for sampling for inorganic and organic analytes.[8]

a. Upon arrival at the field site, set out safety equipment such as traffic cones and signs. Park vehicle in a location and direction so as to prevent sample contamination from vehicle emissions.

b. Assemble equipment needed and set up a clean work space.

- Organic compounds. Select equipment with fluorocarbon polymer, glass, or metal components if components will directly contact samples to be analyzed for organic compounds. Do not use plastics other than fluorocarbon polymers.
- Inorganic constituents. Select equipment with components made of fluorocarbon polymer or other relatively inert and uncolored plastics or glass if components will directly contact samples to be analyzed for inorganic constituents. Do not use metal or rubber components for trace-element sampling.
- Microbiological analyses. Collect samples for microbiological analyses using equipment and techniques described in NFM 7.

Step 2. Select the number and location of equal-discharge increments.

The number and location of equal-discharge increments should not be determined arbitrarily. Selection of increments for a sampling site is governed by factors described in a, d, and e below.

a. Visually inspect the stream from bank to bank, observing velocity, width, and depth distribution, as well as apparent distribution of sediment and aquatic biota along the cross section. Document location of stagnant water, eddies, backwater, reverse flows, areas of faster than normal flow, and piers or other obstructions along the cross section.

b. Determine stream width from a tagline or from distance markings on bridge railings or on a cableway.

c. At sites with little sampling history—measure, record, and review the cross-sectional variation of field measurements (for example, specific electrical conductance, pH, temperature, and dissolved oxygen).

[8]Preparations for water sampling are described in NFM 1, 2, and 3. Consult NFM 5 for sample processing, NFM 6 for field measurements, NFM 7 for biological indicators, NFM 8 for bottom-material sampling, and NFM 9 for field safety.

d. Measure discharge at the cross section to be sampled or use an existing EDI graph prepared from current or historical discharge measurements (fig. 4-3) (Edwards and Glysson, 1998). An existing EDI graph can be one prepared for the site that shows, for example, cumulative discharge or cumulative percent of discharge versus stationing.

e. Determine volume of discharge that will be represented in each EDI, based on data objectives for the study, variation in field measurements, flow and stream-channel characteristics along the cross section, and volume of sample required for analyses of target analytes.

f. Divide the cross section into equal-discharge increments.

- When determining the number of increments to be sampled, keep in mind that the subsample collected at the centroid of each EDI must represent the mean streamflow measured for that increment. If mean streamflow for the increment is not represented, increase the number of increments by decreasing the volume represented by each discharge increment until the mean streamflow value for the increment is represented.
- As a guide, a minimum of 4 sampling increments is recommended; the number of increments is usually less than 10.

g. Determine the location of the centroid of flow within each increment from the discharge measurement by (1) constructing a curve using cumulative discharge or cumulative percentage of discharge (fig. 4-3) plotted against cross-section stationing, or (2) determining EDI locations directly from the discharge measurement sheet (fig. 4-4; an explanation of this method and definition of midpoint are described in Edwards and Glysson, 1998.) Centroid-of-flow locations also can be determined from an EDI graph, as described below and in the TECHNICAL NOTE that follows the example below.

Station: 11482500 Redwood Creek at Orick, CA

Far-Mid Point	Dist. from initial point	Width	Depth	Observation depth	Revolutions	Time in seconds	Velocity: At point	Velocity: Mean in vertical	Adjusted for hor. angle or	Area	Discharge: Q	Discharge: ΣQ	
4	0	4	0	.6	LE	W	0			0	0	0	
12	8	8	1.00		30	47	1.41			8.0	11.3	11.3	
20	16	8	1.80		30	44	1.51			14.4	21.7	33.0	
26	24	8	2.00		50	44	2.50			16.0	40.0	73.0	62.2
28	32	8	2.00		60	45	2.92			16.0	46.7	119.7	
36	40	8	2.30		50	48	2.29			18.4	42.1	161.8	
44	48	8	2.25		40	44	2.00			18.0	36.0	197.8	186.6
50	56	8	2.25		40	40	2.20			18.0	39.6	237.4	
52	64	8	2.30		40	40	2.20			18.4	40.5	277.9	
60	72	8	2.30		50	45	2.44			18.4	44.9	322.8	311.0
68	80	8	2.20		40	45	1.96			17.6	34.5	357.3	
74	88	8	2.00		40	43	2.05			16.0	32.8	390.1	
76	96	8	1.90		50	47	2.34			15.2	35.6	425.7	
84	104	8	2.00		40	42	2.10			16.0	33.6	459.3	435.4
92	112	8	2.00		40	40	2.20			16.0	35.2	494.5	
100	120	8	1.90		30	43	1.54			15.2	23.4	517.9	
102	128	8	1.80		40	40	2.20			14.4	31.7	549.6	
108	136	8	1.70		50	44	2.50			13.6	34.0	583.6	559.8
116	144	8	1.60		50	44	2.50			12.8	32.0	615.6	
124	152	8	1.00		20	54	.827			8.0	6.6	622.2	
132	160	4	0	.6	RE	W	0			0	0	622.2	
134													
140	160	160								290.4	622.2		
148													
156													
160													

EDI Centroid Location — EDI Cumulative Discharge

Figure 4-4. Discharge-measurement field notes used to determine the equal-discharge-increment centroid locations based on cumulative discharge and far-midpoint stationing (from Edwards and Glysson, 1998, p. 42).

Example:

In this example, each EDI equals 20 percent of discharge.

i. If the stream cross section will be divided into five equal-discharge increments, divide stream discharge by five to determine the discharge increment.

ii. Locate the centroid of the initial EDI where cumulative discharge equals half the discharge increment (10 percent). This is the location of the vertical from which the first subsample is collected.

iii. Locate each of the remaining centroids (four in this example) by adding the discharge increment (20 percent) to the previous centroid discharge (20 + 10 = 30) and determining where that cumulative discharge occurs along the cross section.

iv. The EDI centroids will correspond to locations of 10, 30, 50, 70, and 90 percent of the cumulative discharge along the cross section. In figure 4-3, these percentages of cumulative discharges correspond to locations at 14, 28, 38, 45, and 51 ft from the left edge of the water, whereas in figure 4-4, the centroid locations of the equal-discharge increments are at 26, 50, 74, 102, and 134 ft.

TECHNICAL NOTE: If the stream channel is stable at the cross section to be sampled, graphs of cumulative discharge or percentage cumulative discharge at various stages can be based on historical discharge measurements. Location of EDI centroids can be determined from these EDI graphs so that discharge measurements do not have to be made before each sampling. Linear interpolation based on discharge can be made between curves for different discharges on the EDI graphs. EDI graphs require periodic verification by being compared to recent discharge measurements.

Step 3. Select the transit rate.

a. Determine the sampling depth and the mean stream velocity at the centroid of each equal-discharge increment.

b. Determine the transit rate for each centroid that will yield subsamples with approximately the same volume (within 10 percent) using sampling depth, mean stream velocity, and information in Appendix A4-A. When compositing subsamples, the minimum volume for every equal-discharge increment is the minimum volume for the deepest vertical.

Guidelines for selecting the transit rate for EDI sampling
• Collect samples of equal volumes at each centroid. This is required for EDI if the sample will be composited (fig. 4-3). Generally, transit rates vary from centroid to centroid in order to collect equal volumes. • Keep the transit rate unidirectional, constant, and within the isokinetic transit range of the sampler when collecting isokinetic samples at each centroid. • Do not exceed the maximum transit rate (Appendix 4A-4). The maximum transit rate will be exceeded if the minimum sample volume associated with stream velocity and the selected nozzle and bottle size is not collected. Exceeding the maximum transit rate will affect the concentration of particulates ≥ 0.062 millimeters.

Step 4. Collect samples.

The procedures are the same whether you are wading or using a reel-and-cable suspension method. **Use *CH/DH* techniques, as required (section 4.0.1), and implement safety procedures (NFM 9).**

- ▶ Collect microbiological samples using equipment and techniques as described in NFM 7.
- ▶ Collect subsamples at EDI centroids as many times as necessary to ensure collection of sufficient sample volume for analysis. If the sample is to be composited, care must be taken to obtain approximately the same total volume (± 10 percent) from each EDI centroid so that the composited cross-sectional sample will be proportional to flow at the time of sampling.
- ▶ Stay within the isokinetic transit-rate range of the sampler at each centroid. If flow velocity is less than the isokinetic transit-rate range of the sampler, a discharge-weighted sample still can be obtained by collecting equal volumes at each centroid; however, this sample will not be isokinetic.

a. Move sampling and support equipment to the centroid of the first increment to be sampled. Field rinse the sampling equipment (section 4.0.2) and record sampling start time.

b. Read and record the starting gage height.

c. Lower the sampler at the predetermined transit rate until slight contact is made with the streambed.

   - Do not pause upon contacting the streambed. Raise the sampler immediately at a constant transit rate to complete the vertical traverse. The descending transit rate does not have to equal the ascending transit rate, but each rate must be unidirectional, constant, and within the isokinetic transit range of the sampler.
   - Take care not to disturb the streambed with the sampler. Disturbing the streambed could cause bed material to enter the nozzle, resulting in erroneous data.
   - Ensure that the sampler container has not overfilled. Overfilling will result in enrichment of the sample with heavy particulates due to secondary circulation of water through the sampler (from nozzle through air exhaust). This enrichment will result in an artificially increased sediment concentration and will bias particle-size distribution towards heavier and larger particulates.

d. Inspect each subsample, looking for overfilling and (or) the presence of anomalously large amounts of particulates that might have been captured because of excessive streambed disturbance during sample collection. If you note either or both of these conditions, discard the sample, making sure there are no residual particulates left in the container, and resample.

e. Ensure that the sampler container is not underfilled (that the minimum volume indicated in Appendix A4-A has been collected). Underfilling will result in a subsample that is not isokinetically collected—usually because the maximum transit rate has been exceeded.

f. Depending on study objectives, either process and (or) analyze the subsample collected at the initial centroid as a separate sample, composite this subsample with other subsamples collected along the cross section, or split the subsample for further processing.

   - If the total volume of the subsamples that will be collected will exceed the operational capacity of the churn or cone splitter, decrease the number of increments or use an appropriate sampler with a smaller bottle or with a bag with a smaller nozzle.
   - Ensure that all particulates in the sampler bottle or bag are transferred with the sample by swirling the sample gently to keep particulates suspended, and quickly pouring the sample into the churn or cone splitter.

g. Move equipment to the next vertical.

- Determine the transit rate for this vertical. If the subsamples are composited, the total volume collected at each centroid must be equal.
- Repeat procedures, steps 4 c-f.
- Repeat this process at the remaining verticals along the cross section.

h. Record the following information after all samples have been collected:

- Sampling end time.
- Ending gage height.
- All field observations and any deviations from standard sampling procedures.

Step 5. Process samples ➜ Refer to NFM 5.

Step 6. Clean equipment ➜ Refer to NFM 3.

- If the sampler will not be reused during a field trip, rinse the components with deionized water before they dry and place them into a plastic bag for transport to the office laboratory to be cleaned.
- If the sampler will be reused during the field trip, rinse the components with DIW while still wet from sampling, and then follow the prescribed cleaning procedures while at the sampling site (NFM 3). Reassemble the sampler.
- Collect a field blank, if required, after sampling equipment has been cleaned at the sampling site.
- Place cleaned sampler into a plastic bag and seal for transport to the next site.

Single vertical at centroid-of-flow (VCF) method

The VCF method for collecting water samples is identical to the EDI method except that there is one centroid of flow for the stream cross section and therefore only one vertical is sampled. To use this method, the section must be well mixed vertically and laterally with respect to concentrations of target analytes.

APPENDIX A4-A

TRANSIT RATE AND VOLUME GUIDELINES FOR ISOKINETIC SAMPLING

Prepared by **Wayne E. Webb,**
U.S. Geological Survey, Reston, Va.

The tables in Appendix A4-A apply to the first complete round-trip transit starting with an empty sampler container. **These tables are valid only if the sampler is emptied between verticals.**

Tables showing:

1. Isokinetic transit rates for a 1-liter bottle sampler with a
   a. 3/16-inch nozzle
   b. 1/4-inch nozzle
   c. 5/16-inch nozzle
2. Isokinetic transit rates for a 3-liter bottle sampler with a
   a. 1/4-inch nozzle
   b. 5/16-inch nozzle
3. a. Minimum volumes for isokinetic sampling with a bag sampler
   b. Isokinetic transit rates for a 3-liter bag sampler with a 1/4-inch nozzle
   c. Isokinetic transit rates for a 3-liter bag sampler with a 5/16-inch nozzle

The designations in the **RATE** column of these tables are defined as follows:

full The reeling or transit rate that fills the sampler to its maximum volume.

-10 tip The reeling or transit rate that will result in a volume in the sampler such that if the sampler nozzle is tipped 10 degrees down from the horizontal, no sample will spill from the nozzle.

fastest The reeling or transit rate that is the fastest rate to avoid compression problems in bottle samplers or to not exceed a transit rate that is more than 0.4 times the stream velocity for bag samplers.

The volume designations in these tables are defined as follows:

max vol. The volume that will be in the sampler when the "full" (see definition above) reeling rate or transit rate is used for the specified stream depth and velocity.

-10 vol. The volume that will be in the sampler when the "-10 tip" (see definition above) reeling or transit rate is used for the specified stream depths and velocity.

min vol. The volume that will be in the sampler when the "fastest" (see definition above) reeling or transit rate is used for the specified stream depth and velocity.

APPENDIX A4-A—Table 1a. Isokinetic transit rates for a 1-liter bottle sampler with a 3/16-inch nozzle

[Transit rates in feet per second; Depth is (water depth) - (unsampled zone); max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
1	full	0.02	0.02	0.03	0.03	0.04	0.04	0.05	0.05	0.06	0.07	0.08	0.09	1,050	919
1	-10 tip	0.02	0.03	0.03	0.04	0.05	0.05	0.06	0.07	0.08	0.10	0.11	0.12	798	667
1	fastest	0.12	0.17	0.21	0.25	0.29	0.33	0.37	0.41	0.50	0.58	0.66	0.74	131	--
2	full	0.03	0.04	0.05	0.06	0.07	0.08	0.09	0.10	0.12	0.14	0.17	0.19	1,050	807
2	-10 tip	0.04	0.05	0.07	0.08	0.10	0.11	0.12	0.14	0.16	0.19	0.22	0.25	798	555
2	fastest	0.13	0.18	0.22	0.27	0.31	0.36	0.40	0.45	0.54	0.63	0.72	0.81	243	--
3	full	0.05	0.06	0.08	0.09	0.11	0.12	0.14	0.16	0.19	0.22	0.25	0.28	1,050	711
3	-10 tip	0.06	0.08	0.10	0.12	0.14	0.16	0.18	0.20	0.25	0.29	0.33	0.37	798	459
3	fastest	0.14	0.19	0.24	0.29	0.34	0.38	0.43	0.48	0.58	0.67	0.77	0.87	339	--
4	full	0.06	0.08	0.10	0.12	0.14	0.17	0.19	0.21	0.25	0.29	0.33	0.37	1,050	628
4	-10 tip	0.08	0.11	0.14	0.16	0.19	0.22	0.25	0.27	0.33	0.38	0.44	0.49	798	376
4	fastest	0.15	0.21	0.26	0.31	0.36	0.41	0.46	0.51	0.62	0.72	0.82	0.93	423	--
5	full	0.08	0.10	0.13	0.16	0.18	0.21	0.23	0.26	0.31	0.36	0.41	0.47	1,050	555
5	-10 tip	0.10	0.14	0.17	0.20	0.24	0.27	0.31	0.34	0.41	0.48	0.54	0.61	798	303
5	fastest	0.16	0.22	0.27	0.33	0.38	0.44	0.49	0.55	0.66	0.77	0.88	0.99	496	--
6	full	0.09	0.12	0.16	0.19	0.22	0.25	0.28	0.31	0.37	0.43	0.50	0.56	1,050	490
6	-10 tip	0.12	0.16	0.20	0.25	0.29	0.33	0.37	0.41	0.49	0.57	0.65	0.74	798	238
6	fastest	0.17	0.23	0.29	0.35	0.41	0.47	0.52	0.58	0.70	0.81	0.93	1.05	561	--

APPENDIX A4-A—Table 1a. Isokinetic transit rates for a 1-liter bottle sampler with a 3/16-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
7	full	0.11	0.14	0.18	0.22	0.25	0.29	0.33	0.36	0.43	0.51	0.58	0.65	1,050	432
7	-10 tip	0.14	0.19	0.24	0.29	0.33	0.38	0.43	0.48	0.57	0.67	0.76	0.86	798	180
7	fastest	0.18	0.25	0.31	0.37	0.43	0.49	0.55	0.62	0.74	0.86	0.99	1.11	618	--
8	full	0.12	0.17	0.21	0.25	0.29	0.33	0.37	0.41	0.50	0.58	0.66	0.75	1,050	380
8	-10 tip	0.16	0.22	0.27	0.33	0.38	0.44	0.49	0.54	0.65	0.76	0.87	0.98	798	129
8	fastest	0.19	0.26	0.32	0.39	0.45	0.52	0.58	0.65	0.78	0.91	1.04	1.17	670	--
10	full	0.16	0.21	0.26	0.31	0.36	0.41	0.47	0.52	0.62	0.72	0.83	0.93	1,050	292
10	-10 tip	0.20	0.27	0.34	0.41	0.48	0.54	0.61	0.68	0.82	0.95	1.09	1.23	798	40
10	fastest	0.22	0.29	0.36	0.43	0.50	0.57	0.65	0.72	0.86	1.00	1.15	1.29	759	--
12	full	0.19	0.25	0.31	0.37	0.43	0.50	0.56	0.62	0.75	0.87	0.99	1.12	1,050	218
12	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
12	fastest	0.24	0.31	0.39	0.47	0.55	0.63	0.71	0.78	0.94	1.10	1.25	1.41	832	--
14	full	0.22	0.29	0.36	0.43	0.51	0.58	0.65	0.72	0.87	1.01	1.16	1.30	1,050	156
14	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
14	fastest	0.26	0.34	0.43	0.51	0.60	0.68	0.77	0.85	1.02	1.19	1.36	1.53	894	--
15	full	0.23	0.31	0.39	0.47	0.54	0.62	0.70	0.78	0.93	1.09	1.24	1.40	1,050	129
15	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
15	fastest	0.27	0.35	0.44	0.53	0.62	0.71	0.80	0.89	1.06	1.24	1.42	1.59	922	--

APPENDIX A4-A—Table 1b. Isokinetic transit rates for a 1-liter bottle sampler with a 1/4-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
1	full	0.03	0.04	0.05	0.06	0.06	0.07	0.08	0.09	0.11	0.13	0.15	0.17	1,050	918
1	-10 tip	0.04	0.05	0.06	0.07	0.08	0.10	0.11	0.12	0.15	0.17	0.19	0.22	798	667
1	fastest	0.22	0.29	0.37	0.44	0.51	0.59	0.66	0.73	0.88	1.03	1.17	1.32	132	--
2	full	0.06	0.07	0.09	0.11	0.13	0.15	0.17	0.18	0.22	0.26	0.29	0.33	1,050	806
2	-10 tip	0.07	0.10	0.12	0.15	0.17	0.19	0.22	0.24	0.29	0.34	0.39	0.44	798	555
2	fastest	0.24	0.32	0.40	0.48	0.56	0.63	0.71	0.79	0.95	1.11	1.27	1.43	243	--
3	full	0.08	0.11	0.14	0.17	0.19	0.22	0.25	0.28	0.33	0.39	0.44	0.50	1,050	710
3	-10 tip	0.11	0.15	0.18	0.22	0.25	0.29	0.33	0.36	0.44	0.51	0.58	0.65	798	458
3	fastest	0.26	0.34	0.43	0.51	0.60	0.68	0.77	0.85	1.02	1.19	1.36	1.54	340	--
4	full	0.11	0.15	0.18	0.22	0.26	0.29	0.33	0.37	0.44	0.52	0.59	0.66	1,050	626
4	-10 tip	0.15	0.19	0.24	0.29	0.34	0.39	0.44	0.48	0.58	0.68	0.77	0.87	798	375
4	fastest	0.27	0.36	0.46	0.55	0.64	0.73	0.82	0.91	1.09	1.28	1.46	1.64	423	--
5	full	0.14	0.18	0.23	0.28	0.32	0.37	0.41	0.46	0.55	0.64	0.74	0.83	1,050	553
5	-10 tip	0.18	0.24	0.30	0.36	0.42	0.48	0.54	0.61	0.73	0.85	0.97	1.09	798	301
5	fastest	0.29	0.39	0.49	0.58	0.68	0.78	0.87	0.97	1.17	1.36	1.55	1.75	497	--
6	full	0.17	0.22	0.28	0.33	0.39	0.44	0.50	0.55	0.66	0.77	0.88	0.99	1,050	488
6	-10 tip	0.22	0.29	0.36	0.44	0.51	0.58	0.65	0.73	0.87	1.02	1.16	1.31	798	236
6	fastest	0.31	0.41	0.52	0.62	0.72	0.82	0.93	1.03	1.24	1.44	1.65	1.86	562	--

APPENDIX A4-A—Table 1b. Isokinetic transit rates for a 1-liter bottle sampler with a 1/4-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
7	full	0.19	0.26	0.32	0.39	0.45	0.52	0.58	0.64	0.77	0.90	1.03	1.16	1,050	430
7	-10 tip	0.25	0.34	0.42	0.51	0.59	0.68	0.76	0.85	1.02	1.19	1.36	1.52	798	178
7	fastest	0.33	0.44	0.55	0.65	0.76	0.87	0.98	1.09	1.31	1.53	1.74	1.96	620	--
8	full	0.22	0.29	0.37	0.44	0.52	0.59	0.66	0.74	0.88	1.03	1.18	1.32	1,050	378
8	-10 tip	0.29	0.39	0.48	0.58	0.68	0.77	0.87	0.97	1.16	1.36	1.55	1.74	798	126
8	fastest	0.34	0.46	0.57	0.69	0.80	0.92	1.03	1.15	1.38	1.61	1.84	2.07	672	--
10	full	0.28	0.37	0.46	0.55	0.64	0.74	0.83	0.92	1.10	1.29	1.47	1.66	1,050	288
10	-10 tip	0.36	0.48	0.61	0.73	0.85	0.97	1.09	1.21	1.45	1.69	1.94	2.18	798	37
10	fastest	0.38	0.51	0.63	0.76	0.89	1.01	1.14	1.27	1.52	1.78	2.03	2.28	761	--
12	full	0.33	0.44	0.55	0.66	0.77	0.88	0.99	1.10	1.32	1.55	1.77	1.99	1,050	214
12	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
12	fastest	0.42	0.55	0.69	0.83	0.97	1.11	1.25	1.39	1.66	1.94	2.22	2.50	836	--
14	full	0.39	0.52	0.64	0.77	0.90	1.03	1.16	1.29	1.55	1.80	2.06	2.32	1,050	152
14	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
14	fastest	0.45	0.60	0.75	0.90	1.05	1.20	1.36	1.51	1.81	2.11	2.41	2.71	898	--
15	full	0.41	0.55	0.69	0.83	0.97	1.10	1.24	1.38	1.66	1.93	2.21	2.48	1,050	124
15	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
15	fastest	0.47	0.63	0.78	0.94	1.10	1.25	1.41	1.57	1.88	2.19	2.50	2.82	926	--

APPENDIX A4-A—Table 1c. Isokinetic transit rates for a 1-liter bottle sampler with a 5/16-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
1	full	0.04	0.06	0.07	0.09	0.10	0.12	0.13	0.14	0.17	0.20	0.23	0.26	1,049	918
1	-10 tip	0.06	0.08	0.09	0.11	0.13	0.15	0.17	0.19	0.23	0.26	0.30	0.34	800	668
1	fastest	0.34	0.46	0.57	0.69	0.80	0.92	1.03	1.15	1.38	1.61	1.84	2.07	132	--
2	full	0.09	0.12	0.14	0.17	0.20	0.23	0.26	0.29	0.35	0.40	0.46	0.52	1,049	806
2	-10 tip	0.11	0.15	0.19	0.23	0.26	0.30	0.34	0.38	0.45	0.53	0.60	0.68	800	557
2	fastest	0.37	0.50	0.62	0.74	0.87	0.99	1.12	1.24	1.49	1.74	1.98	2.23	243	--
3	full	0.13	0.17	0.22	0.26	0.30	0.35	0.39	0.43	0.52	0.60	0.69	0.78	1,049	709
3	-10 tip	0.17	0.23	0.28	0.34	0.40	0.45	0.51	0.57	0.68	0.79	0.91	1.02	800	460
3	fastest	0.40	0.53	0.67	0.80	0.93	1.07	1.20	1.33	1.60	1.87	2.13	2.40	340	--
4	full	0.17	0.23	0.29	0.35	0.40	0.46	0.52	0.58	0.69	0.81	0.92	1.04	1,049	626
4	-10 tip	0.23	0.30	0.38	0.45	0.53	0.60	0.68	0.75	0.91	1.06	1.21	1.36	800	376
4	fastest	0.43	0.57	0.71	0.86	1.00	1.14	1.28	1.43	1.71	2.00	2.28	2.57	424	--
5	full	0.22	0.29	0.36	0.43	0.50	0.58	0.65	0.72	0.86	1.01	1.15	1.29	1,049	552
5	-10 tip	0.28	0.38	0.47	0.57	0.66	0.75	0.85	0.94	1.13	1.32	1.51	1.70	800	303
5	fastest	0.46	0.61	0.76	0.91	1.06	1.21	1.37	1.52	1.82	2.13	2.43	2.73	497	--
6	full	0.26	0.35	0.43	0.52	0.60	0.69	0.78	0.86	1.04	1.21	1.38	1.55	1,049	487
6	-10 tip	0.34	0.45	0.57	0.68	0.79	0.91	1.02	1.13	1.36	1.58	1.81	2.04	800	238
6	fastest	0.48	0.64	0.81	0.97	1.13	1.29	1.45	1.61	1.93	2.26	2.58	2.90	562	--

APPENDIX A4-A—Table 1c. Isokinetic transit rates for a 1-liter bottle sampler with a 5/16-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
7	full	0.30	0.40	0.50	0.60	0.71	0.81	0.91	1.01	1.21	1.41	1.61	1.81	1,049	429
7	-10 tip	0.40	0.53	0.66	0.79	0.92	1.06	1.19	1.32	1.58	1.85	2.11	2.38	800	180
7	fastest	0.51	0.68	0.85	1.02	1.19	1.36	1.53	1.70	2.04	2.38	2.73	3.07	620	--
8	full	0.35	0.46	0.58	0.69	0.81	0.92	1.04	1.15	1.38	1.61	1.84	2.07	1,049	377
8	-10 tip	0.45	0.60	0.75	0.91	1.06	1.21	1.36	1.51	1.81	2.11	2.42	2.72	800	128
8	fastest	0.54	0.72	0.90	1.08	1.26	1.44	1.62	1.80	2.16	2.51	2.87	3.23	672	--
10	full	0.43	0.58	0.72	0.86	1.01	1.15	1.29	1.44	1.73	2.01	2.30	2.59	1,049	287
10	-10 tip	0.57	0.75	0.94	1.13	1.32	1.51	1.70	1.89	2.26	2.64	3.02	3.40	800	38
10	fastest	0.59	0.79	0.99	1.19	1.39	1.59	1.78	1.98	2.38	2.77	3.17	3.57	762	--
11	full	0.47	0.63	0.79	0.95	1.11	1.27	1.42	1.58	1.90	2.22	2.53	2.85	1,049	219
11	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
11	fastest	0.60	0.80	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	830	--
12	full	0.52	0.69	0.86	1.04	1.21	1.38	1.55	1.73	2.07	2.42	2.76	3.11	1,049	143
12	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
12	fastest	0.60	0.80	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	906	--
13	full	0.56	0.75	0.94	1.12	1.31	1.50	1.68	1.87	2.24	2.62	2.99	3.37	1,049	68
13	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
13	fastest	0.60	0.80	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	981	--

APPENDIX A4-A—Table 2a. Isokinetic transit rates for a 3-liter bottle sampler with a 1/4-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
2	full	0.02	0.03	0.03	0.04	0.05	0.05	0.06	0.07	0.08	0.10	0.11	0.12	2,832	2,120
2	-10 tip	0.02	0.03	0.04	0.05	0.06	0.06	0.07	0.08	0.09	0.11	0.13	0.14	2,457	1,745
2	fastest	0.08	0.11	0.14	0.16	0.19	0.22	0.24	0.27	0.33	0.38	0.43	0.49	712	--
3	full	0.03	0.04	0.05	0.06	0.07	0.08	0.09	0.10	0.12	0.14	0.16	0.18	2,832	1,840
3	-10 tip	0.04	0.05	0.06	0.07	0.08	0.09	0.11	0.12	0.14	0.17	0.19	0.21	2,457	1,465
3	fastest	0.09	0.12	0.15	0.18	0.20	0.23	0.26	0.29	0.35	0.41	0.47	0.53	992	--
4	full	0.04	0.05	0.07	0.08	0.10	0.11	0.12	0.14	0.16	0.19	0.22	0.25	2,832	1,597
4	-10 tip	0.05	0.06	0.08	0.09	0.11	0.13	0.14	0.16	0.19	0.22	0.25	0.28	2,457	1,222
4	fastest	0.09	0.13	0.16	0.19	0.22	0.25	0.28	0.31	0.38	0.44	0.50	0.56	1,235	--
5	full	0.05	0.07	0.09	0.10	0.12	0.14	0.15	0.17	0.20	0.24	0.27	0.31	2,832	1,383
5	-10 tip	0.06	0.08	0.10	0.12	0.14	0.16	0.18	0.20	0.24	0.28	0.31	0.35	2,457	1,009
5	fastest	0.10	0.13	0.17	0.20	0.23	0.27	0.30	0.33	0.40	0.47	0.53	0.60	1,449	--
6	full	0.06	0.08	0.10	0.12	0.14	0.16	0.18	0.20	0.25	0.29	0.33	0.37	2,832	1,195
6	-10 tip	0.07	0.09	0.12	0.14	0.17	0.19	0.21	0.24	0.28	0.33	0.38	0.42	2,457	820
6	fastest	0.11	0.14	0.18	0.21	0.25	0.28	0.32	0.35	0.42	0.50	0.57	0.64	1,637	--
7	full	0.07	0.10	0.12	0.14	0.17	0.19	0.21	0.24	0.29	0.33	0.38	0.43	2,832	1,028
7	-10 tip	0.08	0.11	0.14	0.17	0.19	0.22	0.25	0.28	0.33	0.39	0.44	0.50	2,457	653
7	fastest	0.11	0.15	0.19	0.22	0.26	0.30	0.34	0.37	0.45	0.52	0.60	0.67	1,804	--

APPENDIX A4-A—Table 2a. Isokinetic transit rates for a 3-liter bottle sampler with a 1/4-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
8	full	0.08	0.11	0.14	0.16	0.19	0.22	0.25	0.27	0.33	0.38	0.44	0.49	2,832	878
8	-10 tip	0.09	0.13	0.16	0.19	0.22	0.25	0.28	0.31	0.38	0.44	0.50	0.57	2,457	503
8	fastest	0.12	0.16	0.20	0.24	0.28	0.32	0.36	0.40	0.47	0.55	0.63	0.71	1,954	--
9	full	0.09	0.12	0.15	0.18	0.21	0.25	0.28	0.31	0.37	0.43	0.49	0.55	2,832	743
9	-10 tip	0.11	0.14	0.18	0.21	0.25	0.28	0.32	0.35	0.42	0.50	0.57	0.64	2,457	368
9	fastest	0.12	0.17	0.21	0.25	0.29	0.33	0.37	0.42	0.50	0.58	0.67	0.75	2,089	--
10	full	0.10	0.14	0.17	0.20	0.24	0.27	0.31	0.34	0.41	0.48	0.55	0.61	2,832	620
10	-10 tip	0.12	0.16	0.20	0.24	0.28	0.31	0.35	0.39	0.47	0.55	0.63	0.71	2,457	246
10	fastest	0.13	0.17	0.22	0.26	0.31	0.35	0.39	0.44	0.52	0.61	0.70	0.79	2,212	--
12	full	0.12	0.16	0.20	0.25	0.29	0.33	0.37	0.41	0.49	0.57	0.65	0.74	2,832	408
12	-10 tip	0.14	0.19	0.24	0.28	0.33	0.38	0.42	0.47	0.57	0.66	0.75	0.85	2,457	33
12	fastest	0.14	0.19	0.24	0.29	0.33	0.38	0.43	0.48	0.57	0.67	0.76	0.86	2,424	--
14	full	0.14	0.19	0.24	0.29	0.33	0.38	0.43	0.48	0.57	0.67	0.76	0.86	2,832	229
14	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
14	fastest	0.16	0.21	0.26	0.31	0.36	0.42	0.47	0.52	0.62	0.73	0.83	0.93	2,603	--
15	full	0.15	0.20	0.26	0.31	0.36	0.41	0.46	0.51	0.61	0.72	0.82	0.92	2,832	149
15	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
15	fastest	0.16	0.22	0.27	0.32	0.38	0.43	0.49	0.54	0.65	0.76	0.86	0.97	2,683	--

APPENDIX A4-A—Table 2b. Isokinetic transit rates for a 3-liter bottle sampler with a 5/16-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
2	full	0.03	0.04	0.05	0.06	0.07	0.09	0.10	0.11	0.13	0.15	0.17	0.19	2,830	2,118
2	-10 tip	0.04	0.05	0.06	0.07	0.09	0.10	0.11	0.12	0.15	0.17	0.20	0.22	2,461	1,749
2	fastest	0.13	0.17	0.21	0.25	0.30	0.34	0.38	0.42	0.51	0.59	0.68	0.76	712	--
3	full	0.05	0.06	0.08	0.10	0.11	0.13	0.14	0.16	0.19	0.22	0.26	0.29	2,830	1,837
3	-10 tip	0.06	0.07	0.09	0.11	0.13	0.15	0.17	0.18	0.22	0.26	0.29	0.33	2,461	1,468
3	fastest	0.14	0.18	0.23	0.27	0.32	0.36	0.41	0.46	0.55	0.64	0.73	0.82	993	--
4	full	0.06	0.09	0.11	0.13	0.15	0.17	0.19	0.21	0.26	0.30	0.34	0.38	2,830	1,593
4	-10 tip	0.07	0.10	0.12	0.15	0.17	0.20	0.22	0.25	0.29	0.34	0.39	0.44	2,461	1,224
4	fastest	0.15	0.20	0.24	0.29	0.34	0.39	0.44	0.49	0.59	0.68	0.78	0.88	1,237	--
5	full	0.08	0.11	0.13	0.16	0.19	0.21	0.24	0.27	0.32	0.37	0.43	0.48	2,830	1,379
5	-10 tip	0.09	0.12	0.15	0.18	0.21	0.25	0.28	0.31	0.37	0.43	0.49	0.55	2,461	1,010
5	fastest	0.16	0.21	0.26	0.31	0.36	0.42	0.47	0.52	0.62	0.73	0.83	0.94	1,451	--
6	full	0.10	0.13	0.16	0.19	0.22	0.26	0.29	0.32	0.38	0.45	0.51	0.58	2,830	1,190
6	-10 tip	0.11	0.15	0.18	0.22	0.26	0.29	0.33	0.37	0.44	0.52	0.59	0.66	2,461	820
6	fastest	0.17	0.22	0.28	0.33	0.39	0.44	0.50	0.55	0.66	0.77	0.88	0.99	1,641	--
7	full	0.11	0.15	0.19	0.22	0.26	0.30	0.34	0.37	0.45	0.52	0.60	0.67	2,830	1,021
7	-10 tip	0.13	0.17	0.21	0.26	0.30	0.34	0.39	0.43	0.52	0.60	0.69	0.77	2,461	652
7	fastest	0.18	0.23	0.29	0.35	0.41	0.47	0.53	0.58	0.70	0.82	0.93	1.05	1,809	--

APPENDIX A4-A—Table 2b. Isokinetic transit rates for a 3-liter bottle sampler with a 5/16-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		1.50	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00		
8	full	0.13	0.17	0.21	0.26	0.30	0.34	0.38	0.43	0.51	0.60	0.68	0.77	2,830	870
8	-10 tip	0.15	0.20	0.25	0.29	0.34	0.39	0.44	0.49	0.59	0.69	0.79	0.88	2,461	501
8	fastest	0.18	0.25	0.31	0.37	0.43	0.49	0.55	0.62	0.74	0.86	0.99	1.11	1,960	--
9	full	0.14	0.19	0.24	0.29	0.34	0.38	0.43	0.48	0.58	0.67	0.77	0.86	2,830	734
9	-10 tip	0.17	0.22	0.28	0.33	0.39	0.44	0.50	0.55	0.66	0.77	0.88	0.99	2,461	365
9	fastest	0.19	0.26	0.32	0.39	0.45	0.52	0.58	0.65	0.78	0.91	1.04	1.17	2,096	--
10	full	0.16	0.21	0.27	0.32	0.37	0.43	0.48	0.53	0.64	0.75	0.85	0.96	2,830	610
10	-10 tip	0.18	0.25	0.31	0.37	0.43	0.49	0.55	0.61	0.74	0.86	0.98	1.10	2,461	241
10	fastest	0.20	0.27	0.34	0.41	0.48	0.54	0.61	0.68	0.82	0.95	1.09	1.22	2,220	--
12	full	0.19	0.26	0.32	0.38	0.45	0.51	0.58	0.64	0.77	0.90	1.02	1.15	2,830	396
12	-10 tip	0.22	0.29	0.37	0.44	0.52	0.59	0.66	0.74	0.88	1.03	1.18	1.32	2,461	26
12	fastest	0.22	0.30	0.37	0.45	0.52	0.60	0.67	0.74	0.89	1.04	1.19	1.34	2,435	--
14	full	0.22	0.30	0.37	0.45	0.52	0.60	0.67	0.75	0.90	1.05	1.19	1.34	2,830	215
14	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
14	fastest	0.24	0.32	0.40	0.48	0.57	0.65	0.73	0.81	0.97	1.13	1.29	1.45	2,615	--
15	full	0.24	0.32	0.40	0.48	0.56	0.64	0.72	0.80	0.96	1.12	1.28	1.44	2,830	135
15	-10 tip	--	--	--	--	--	--	--	--	--	--	--	--	--	--
15	fastest	0.25	0.34	0.42	0.50	0.59	0.67	0.76	0.84	1.01	1.18	1.34	1.51	2,695	--

APPENDIX A4-A—Table 3a. Minimum volumes for isokinetic sampling with a bag sampler

[The volumes listed below are the minimum volumes that must be collected in a bag sampler to have not exceeded 0.4 times the mean stream velocity. Generally, bag samplers must be operated in water warmer than 7 degrees Celsius and where the velocity is greater than 3 feet per second.]

Water depth minus unsampled zone, in feet	Minimum volume (in milliliters) for the nozzle diameter (inch) shown[1]		
	3/16 inch	1/4 inch	5/16 inch
1	27	48	75
2	54	96	151
3	81	145	226
4	109	193	301
5	136	241	377
6	163	289	452
7	190	338	528
8	217	386	603
9	244	434	678
10	271	483	754
11	298	531	829
12	326	579	904
13	353	627	980
14	380	675	1,055
15	407	724	1,131
20	543	965	1,507
25	678	1,206	1,884
30	814	1,447	2,262
35	950	1,688	2,638
40	1,085	1,930	3,015
45	1,221	2,171	3,392
50	1,357	2,412	3,769
55	1,492	2,653	4,146
60	1,629	2,894	4,524
65	1,764	3,136	4,899
70	1,899	3,377	5,276
75	2,035	3,618	5,653

APPENDIX A4-A—Table 3a. Minimum volumes for isokinetic sampling with a bag sampler— *Continued*

Water depth minus unsampled zone, in feet	Minimum volume (in milliliters) for the nozzle diameter (inch) shown [1]		
	3/16 inch	1/4 inch	5/16 inch
80	2,171	3,859	6,030
85	2,306	4,100	6,407
90	2,442	4,342	6,784
95	2,578	4,583	7,161
100	2,713	4,824	7,537
120	3,257	5,789	9,045
140	3,799	6,754	10,552
160	4,342	7,718	12,060
180	4,884	8,683	13,567
200	5,427	9,650	15,075

[1] Minimum volume = area of nozzle x time in water x mean stream velocity in vertical; minimum volume in milliliters = 15 x 3.14 x 2.54 cubed x nozzle diameter, in inches squared x depth, in feet.

APPENDIX A4-A—Table 3b. Isokinetic transit rates for a 3-liter bag sampler with a 1/4-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00	10.00	12.00		
6	full	0.11	0.13	0.16	0.18	0.20	0.22	0.27	0.31	0.36	0.40	0.44	0.53	2,607	2,318
6	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	290	--
8	full	0.15	0.18	0.21	0.24	0.27	0.30	0.36	0.41	0.47	0.53	0.59	0.71	2,607	2,221
8	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	386	--
10	full	0.19	0.22	0.26	0.30	0.33	0.37	0.44	0.52	0.59	0.67	0.74	0.89	2,607	2,125
10	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	483	--
12	full	0.22	0.27	0.31	0.36	0.40	0.44	0.53	0.62	0.71	0.80	0.89	1.07	2,607	2,028
12	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	579	--
14	full	0.26	0.31	0.36	0.41	0.47	0.52	0.62	0.73	0.83	0.93	1.04	1.24	2,607	1,931
14	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	676	--
16	full	0.30	0.36	0.41	0.47	0.53	0.59	0.71	0.83	0.95	1.07	1.19	1.42	2,607	1,835
16	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	773	--
18	full	0.33	0.40	0.47	0.53	0.60	0.67	0.80	0.93	1.07	1.20	1.33	1.60	2,607	1,738
18	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	869	--
20	full	0.37	0.44	0.52	0.59	0.67	0.74	0.89	1.04	1.19	1.33	1.48	1.78	2,607	1,642
20	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	966	--
22	full	0.41	0.49	0.57	0.65	0.73	0.81	0.98	1.14	1.30	1.47	1.63	1.96	2,607	1,545
22	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,062	--

APPENDIX A4-A—Table 3b. Isokinetic transit rates for a 3-liter bag sampler with a 1/4-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00	10.00	12.00		
24	full	0.44	0.53	0.62	0.71	0.80	0.89	1.07	1.24	1.42	1.60	1.78	2.13	2,607	1,449
24	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,159	--
26	full	0.48	0.58	0.67	0.77	0.87	0.96	1.16	1.35	1.54	1.73	1.93	2.31	2,607	1,352
26	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,255	--
28	full	0.52	0.62	0.73	0.83	0.93	1.04	1.24	1.45	1.66	1.87	2.07	2.49	2,607	1,255
28	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,352	--
30	full	0.56	0.67	0.78	0.89	1.00	1.11	1.33	1.56	1.78	2.00	2.22	2.67	2,607	1,159
30	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,449	--
35	full	0.65	0.78	0.91	1.04	1.17	1.30	1.56	1.81	2.07	2.33	2.59	3.11	2,607	917
35	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,690	--
40	full	0.74	0.89	1.04	1.19	1.33	1.48	1.78	2.07	2.37	2.67	2.96	3.56	2,607	676
40	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,931	--
45	full	0.83	1.00	1.17	1.33	1.50	1.67	2.00	2.33	2.67	3.00	3.33	4.00	2,607	435
45	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,173	--
50	full	0.93	1.11	1.30	1.48	1.67	1.85	2.22	2.59	2.96	3.33	3.70	4.44	2,607	193
50	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,414	--
53	full	0.98	1.18	1.37	1.57	1.77	1.96	2.36	2.75	3.14	3.53	3.93	4.71	2,607	48
53	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,559	--

APPENDIX A4-A—Table 3c. Isokinetic transit rates for a 3-liter bag sampler with a 5/16-inch nozzle

[Transit rates in feet per second; Depth is water depth - unsampled zone; max, maximum; vol, volume; min, minimum; mL, milliliter; --, not applicable]

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00	10.00	12.00		
2	full	0.06	0.07	0.08	0.09	0.10	0.12	0.14	0.16	0.19	0.21	0.23	0.28	2,604	2,453
2	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	151	--
4	full	0.12	0.14	0.16	0.19	0.21	0.23	0.28	0.32	0.37	0.42	0.46	0.56	2,604	2,302
4	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	302	--
6	full	0.17	0.21	0.24	0.28	0.31	0.35	0.42	0.49	0.56	0.63	0.70	0.83	2,604	2,151
6	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	453	--
8	full	0.23	0.28	0.32	0.37	0.42	0.46	0.56	0.65	0.74	0.83	0.93	1.11	2,604	2,000
8	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	604	--
10	full	0.29	0.35	0.41	0.46	0.52	0.58	0.70	0.81	0.93	1.04	1.16	1.39	2,604	1,849
10	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	755	--
12	full	0.35	0.42	0.49	0.56	0.63	0.70	0.83	0.97	1.11	1.25	1.39	1.67	2,604	1,698
12	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	906	--
14	full	0.41	0.49	0.57	0.65	0.73	0.81	0.97	1.14	1.30	1.46	1.62	1.95	2,604	1,547
14	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,057	--
16	full	0.46	0.56	0.65	0.74	0.83	0.93	1.11	1.30	1.48	1.67	1.86	2.23	2,604	1,396
16	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,208	--
18	full	0.52	0.63	0.73	0.83	0.94	1.04	1.25	1.46	1.67	1.88	2.09	2.50	2,604	1,245
18	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,359	--

APPENDIX A4-A—Table 3c. Isokinetic transit rates for a 3-liter bag sampler with a 5/16-inch nozzle—*Continued*

Depth (in feet)	Rate	Mean stream velocity in vertical (feet per second)												Max. vol. -10 vol. min. vol.	Volume-min. vol. (mL)
		2.50	3.00	3.50	4.00	4.50	5.00	6.00	7.00	8.00	9.00	10.00	12.00		
20	full	0.58	0.70	0.81	0.93	1.04	1.16	1.39	1.62	1.86	2.09	2.32	2.78	2,604	1,094
20	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,509	--
22	full	0.64	0.77	0.89	1.02	1.15	1.28	1.53	1.79	2.04	2.30	2.55	3.06	2,604	943
22	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,660	--
24	full	0.70	0.83	0.97	1.11	1.25	1.39	1.67	1.95	2.23	2.50	2.78	3.34	2,604	792
24	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,811	--
26	full	0.75	0.90	1.06	1.21	1.36	1.51	1.81	2.11	2.41	2.71	3.01	3.62	2,604	642
26	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	1,962	--
28	full	0.81	0.97	1.14	1.30	1.46	1.62	1.95	2.27	2.60	2.92	3.25	3.90	2,604	491
28	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,113	--
30	full	0.87	1.04	1.22	1.39	1.57	1.74	2.09	2.43	2.78	3.13	3.48	4.17	2,604	340
30	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,264	--
32	full	0.93	1.11	1.30	1.48	1.67	1.86	2.23	2.60	2.97	3.34	3.71	4.45	2,604	189
32	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,415	--
34	full	0.99	1.18	1.38	1.58	1.77	1.97	2.37	2.76	3.15	3.55	3.94	4.73	2,604	38
34	fastest	1.00	1.20	1.40	1.60	1.80	2.00	2.40	2.80	3.20	3.60	4.00	4.80	2,566	--
35	full	--	--	--	--	--	--	--	--	--	--	--	--	--	--
35	fastest	--	--	--	--	--	--	--	--	--	--	--	--	--	--

PART 2D

Long-Term Ambient Trend Monitoring -- Automatic Sampling

This section contains the guidance for the District Long-Term Ambient Trend Monitoring using the automatic sampling option. These procedures and protocols were modified from the Atlanta Region NPDES MS4 Phase I Standard Operating Procedures for Stormwater Monitoring (2000).

2D.1 Overview

Where possible, it is recommended that wet weather and dry weather monitoring samples be collected at long-term ambient trend sites using automated composite samplers. The main advantage of automatic sampling is that it is more convenient than manual composite grab techniques and can substantially reduce the amount of labor-hours required for monitoring personnel, particularly for wet weather events.

The methods discussed here refer to flow-weighted techniques. A discussion of time-weighted vs. flow-weighted composite sampling are provided in Appendix 2C-2 along with descriptions of the different ways to collect flow-weighted samples.

The equipment manufacturer's manual should always be consulted for specific procedures for equipment installation, calibration, operation and maintenance.

2D.2 Setting up the Sampling Station

This section describes site preparation and pre-event activities associated with collection of both dry and wet weather samples for long-term ambient trend monitoring.

2D.2.1 Equipment Requirements

A typical automated sampler site should include the following equipment:

- Automate composite sampler with data recorder;
- Marine battery (or other power source);
- Flow monitoring device;
- Rain gage; and
- Shelter housing (optional, but recommended for security reasons)

If electricity is available at the site, an optional refrigerated sampler can be installed to eliminate the need to ice samplers prior to rain events. Also, telemetry equipment can be installed to allow remote manual activation and access to the data. Precipitation data (including duration, volume, and antecedent dry period) must be provided for all monitored storm events. For sites which can not be covered by existing rain gages, a rain

gage should be installed which is clear of surrounding rooftops and tree canopy. Monitoring sites in proximity to one another may share rain gage information.

2D.2.2 Establishing the Rainfall-Runoff Relationship

The amount of runoff generated from a storm of a given rainfall volume is dependent upon the characteristics of the watershed (such as soils, drainage patterns, land-use and channel properties), antecedent rainfall conditions, and the intensity and duration of the precipitation event. The best source of estimating runoff is to have existing flow and rainfall data from the drainage area being studied. However, when new monitoring sites are being developed it is typically not possible to rely on existing data.

Without historical flow data, initial runoff volume estimates can be made through the use of a rainfall-runoff relationship to develop a synthetic hydrograph. A commonly used rainfall-runoff relationship is the runoff coefficient method, described in Appendix 2C-6. The advantage of this method is that it is a very simple method of calculating runoff volumes and can be easily applied to estimate runoff volumes for a range of watershed sizes. However, the result is only used as a starting point for calculating anticipated runoff volumes. Other methods of estimating the runoff produced include the SCS hydrologic method and the USGS regression equations. Further information on these methods can be found in the Georgia Stormwater Management Manual (2001).

2D.2.3 Flow Monitoring

Flow monitoring is essential because stormwater pollutant loads cannot be estimated without accurate flow measurements. Once a monitoring station has been established and calibrated, flow measurements should be based upon actual flow data from control devices or depth and velocity methods.

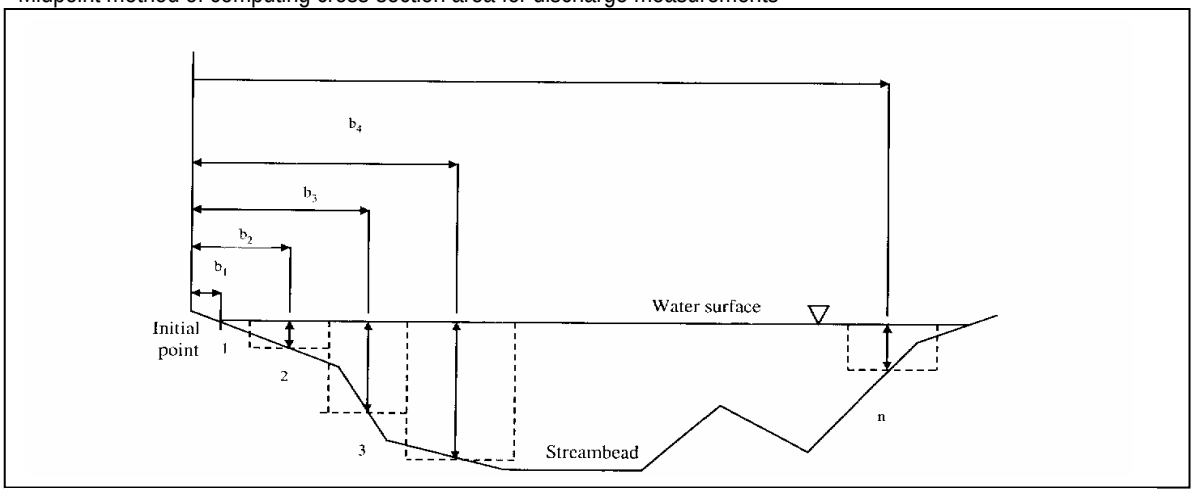
Flow monitoring equipment should be capable of producing data to generate storm hydrographs (e.g., plots of flow rate versus time) for monitored storm events. The storm hydrographs should be used to derive the required storm summary information including: storm runoff volume, peak flow, and duration. The time interval between samples will vary over the storm hydrograph depending on the flow rate. This sampling methodology is used to achieve a representative flow-weighted composite sample. An appropriate flow monitoring system should be installed so that accurate flow measurements can be obtained over the range of expected storm flows.

2D.2.3.1 Developing Stage-Discharge Relationships

At in-stream monitoring locations, the use of primary control devices such as flumes and weirs is rather limited because of expense and potentially harmful environmental impacts of constructing large devices capable of providing measurements for a range of storm flows. Therefore, open channel measurements at in-stream locations should be conducted by developing a stage-discharge relationship across the stream cross-section. This method applies the principal of continuity where discharge is calculated as the sum of the products of the velocity and the cross-sectional area of the stream. Where available, it is recommended that monitoring sites be located near existing USGS stations so that new stage-discharge relationships do not have to be developed at each in-stream station.

To apply the continuity equation, stream depth and flow velocity measurements must be taken at various points across the stream. Depth and velocity measurements should be taken for each change in depth or velocity across the stream. A diagram of this process is shown in Figure 2D-1.

FIGURE 2D-1
Midpoint method of computing cross-section area for discharge measurements



Source: USGS, 1982 *Measurement and Computation of Streamflow: Volume 1. Measurement of Stage and Discharge.*

It is recommended that as many data points as possible be taken for each cross-section of interest. Typically, 25 to 30 data points are collected (USGS, 1982). However, in small streams, it not always possible to obtain this amount of data. As a practical guide, at least five measurements should be made for small streams (typically less than 10 cfs, and less than 4 feet wide). In general, the more data the better. In any case, the user should understand that the calculations assume that the depth and velocity at the point of measurement is the mean for that segment of the stream.

A staff gage should be set up at or near the instream monitoring point of interest to correlate measurements to a reference depth. Cross-sectional depth and velocity measurements should be collected at several points near the monitoring point of interest. The continuity equation [$Q=\Sigma(AV)$] should then be applied to the collected data to calculate the flow. A sample collection form for developing open channel stage-discharge relationships is provided in Appendix 2A-3.

The discharge measurement procedure should be applied over a range of wet weather flows so that the discharge can be determined for various stages or depths of flow. Flow measurements taken during this period serve to develop the stage-discharge relationship. The discharge rating may be a simple linear relationship or more complex relation such as an exponential, logarithmic, or power equation. Then, the stage-discharge measurements should be plotted over the range of flows. Spreadsheet analysis using regression techniques and trendline functions can be a valuable tool in developing an equation or series of equations to fit the data. In many cases, it is possible that multiple equations will be needed to develop an equation with a good fit.

Initially, discharge measurements should be made with the frequency necessary to define the discharge relation. Measurements should also be made at periodic intervals (at least every 3 months) and after major storm events to verify the rating. If needed, the rating should be revised to reflect the changes to the stream.

2D.2.3.2 Operation of the Current Metering Equipment

The flow monitoring procedure described above requires the use of a current meter and a depth measurement device. Wading rods are typically used to determine stream depth. The calculation of the current of the stream is determined by placing a meter at a point in the stream and counting the number of revolutions within a given time period. Typically used current meters include the Price type AA for general use and the Price pygmy meter for shallow flow applications. For these types of devices, the current meter attaches to the wading rod. Both of these types of meters operate by sending an electrical impulse to headphones worn by the user. The electrical impulse produces an audible click in the headphones from which the user can count the number of revolutions over a given time period. The number of revolutions over a given time period is then converted to a velocity. Rating tables have been developed by the USGS for these current meters.

Current measurements should be taken perpendicular to the cross-section of the stream being surveyed. Special care should be taken of the current meter to ensure that it is kept in good condition. Additional discussion of the Price type AA and pygmy meters can be found in USGS, 1982.

The procedure described here applies to top settling rods. These are the preferred type of wading rod because of the convenience in setting the current meter at the proper depth (USGS, 1982). For streams with depths of less than 2.5 feet, the average current is taken to be the current at 0.6 times the stream depth. For streams of greater depth, the velocity at 0.2 and 0.8 times the depth are taken and the average of the two values is taken as the mean. Before using either the Price type AA or pygmy meters, a spin test should be performed to ensure that the rotor freely rotates. Consult the manufacturer's specifications for the procedure for conducting a spin test.

2D.2.4 Conducting Flow Measurements

After using area-velocity measurements to develop a stage-discharge relationship, a device to measure water level should be placed within the stream. The stage-discharge relationship should be programmed into the data recorder or automated sampling equipment so that the depth of the water can be converted to flow, and logged.

Pressure transducers are recommended as the measurement device because of their reliability and ease of use. A pressure transducer senses changes in pressure exerted by a column of water above a "strain gage." The pressure transducer measures the changes in pressure, and converts that change into an electronic signal. This electronic signal is then sent to the data logger and translated into a head reading based on the range of the transducer. Pressure transducers must be handled with care during installation and modification. While transporting the equipment, transducers should be protected with protective covers.

Samplers can be triggered by an increase in flow and/or rainfall. For instance, a sampler could be programmed to activate when a storm produces an increase over base flow of at least 1.0 inch when there is 0.10 inch of rain within a 30 minute period.

2D.3 Sampling Preparation

2D.3.1 Wet Weather Event Considerations

Obtaining the most reliable and current information on the intensity and duration of forecasted precipitation events is critical to sampling wet weather events. Once it has been determined that a storm event is expected that is predicted to meet the 0.3 inch rainfall criteria and antecedent dry period, the field team should begin final preparation for the sampling event. The analytical laboratory should be contacted to alert them to the potential of an upcoming sampling event.

The day prior to commencement of a wet-weather sampling event, the field team shall conduct the following activities on-site:

- Check equipment (operation and placement) to make sure it is free of debris and has not been tampered with;
- Inspect the sample tubing and pump tubing for cracks;
- Download data from the data logger, if necessary, and check the rainfall data to verify that antecedent dry weather conditions have been met;
- Replace the battery, if necessary;
- Calibrate the sampler according to the manufacturers instructions;
- Install a new, pre-cleaned sample collection container (if necessary) and pack fresh ice around it;
- Reprogram the sampler, if necessary, to adjust the incremental sample volume to increase or decrease the frequency of sample collection based on total rainfall predictions (the sampler should be programmed for an average rainfall of 0.7 inches); and
- Activate the sampler so that it is ready to begin sampling when triggered.

2D.3.2 Sampling Equipment Checklist

Before visiting a sampling station, the field team should ensure that all of the necessary equipment is present and in order. Table 2D-1 shows the required equipment needed for long-term ambient trend monitoring (*this checklist assumes that sample filtration and preservation occurs at the laboratory rather than in the field – see the next section for more details*).

TABLE 2D-1
List of Equipment and Supplies for Long-Term Ambient Trend Monitoring

Field Equipment	Function
Sample bottles with labels	For collection of base flow grab samples only
Composite jar	To replace in automatic sampler for collection of wet weather composite samples only
Reagent-grade water (1 L wide mouth HDPE bottle and gallon container)	Calibration of automatic sampler and collection of equipment blank
1 L graduated cylinder and bucket	Calibration of automatic sampler
Clear tape and applicator	To apply over label
Coolers	For transport of grab samples
Cloth or mesh grocery bag	For transport of composite jar
Ice/ ice packs	To keep samples preserved after collection and during transport from the site
Clipboard or notebook with data collection forms and COC forms / Pens	To document field data and activities
List of outfalls, directions, protocols, and H&S plan.	For reference in the field
Field logbook	To record notes
Sharpie (extra fine)	Label sample bottles
Cell phone	Communication in the field.
First Aid Kit	Health and Safety Plan
Disposable gloves, safety shoes, and safety glasses	Health and Safety Plan

2D.4 Sample Collection and Handling

2D.4.1 Flow-Weighted Composite Sample Collection

The field crew should start to mobilize to collect flow-weighted composite samples as soon as it is practical after the start of a wet weather event, and should collect the sample no later than 24 hours after the storm event begins. Samplers may continue to take samples after the rain has stopped if the storm hydrograph has not returned to baseline conditions. The following activities should be conducted by the field crew:

- Check the automated sampler to ensure that it has stopped taking samples;
- If sampling has ceased, collect the composite jar from the automated sampler;
- Observe the sample and complete the field data collection form;
- Place the composite jar in a cloth or mesh bag and surround it with ice for transport back to a staging area or laboratory;
- Check the automated sampler's electronic log to ensure that all samples were collected without incident. Record any anomalies in the field logbook;
- Check the flow meter and sample intake to clear any built-up materials;
- Replace the composite jar with a new or pre-cleaned composite jar and activate the sampling program (alternatively, this can be done as a pre-event activity); and
- Secure the sampling equipment.

2D.4.2 Dry Weather (Baseflow) Sample Collection

Sampling for dry weather baseflow measurements can be performed by manually operating the composite sampler to take a discrete sample or by manual grab samples (inserting a sample container under or down current of a discharge or stream). Use of the automated composite sampler to collect the grab sample may be advantageous, especially for less accessible instream sites since the sample intake line should already be in place.

To ensure that the grab samples are representative, the following procedures should be followed.

- For grabs using the automatic sampler, open the lid of the sampler and hold the sample container under the center of the lid. Activate the sampler pump to manually collect a sample.
- For manual grabs, follow the procedures for grab sampling in Section 2B.

2D.5 Equipment and Site Maintenance

Routine maintenance activities in the field should be conducted once a week and should include the following.

- Conduct a general inspection of the site and equipment.
- Cleaning the debris around the sample tubing. If the automated sampler is used for grab sample collection, then 2.5 times the length of the tube should be cut off and discarded.
- Change battery (if an extra power source is required). Battery storage, recharging and installation guidance is found in Appendix 2D-6.

Maintenance activities that can be performed off the field include the following.

- Inspect and calibrate water quality monitoring equipment (i.e., conductivity meter, pH meter, thermometer).
- Recharge 12-volt marine batteries on a rotating schedule (if these are used).

If the equipment has malfunctioned during a storm event, it should be inspected to identify the problem(s) and the appropriate corrective action taken to ensure the problem(s) has been fixed. The specific manufacturer's instructions should be referred to for troubleshooting.

Additional information and guidance on the operations and maintenance of automatic sampling equipment is provided in Appendix 2D-5.

References

Atlanta Regional Commission (ARC). 2000. Atlanta Region NPDES MS4 Phase I Standard Operating Procedures for Stormwater Monitoring.

Brown, W. and Schueler, T. 1997. National Pollutant Removal Performance Database for Stormwater BMPs: A National Examination of Pollutant Removal Capability, Center for Watershed Protection, Silver Spring, MD.

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U.S. Geological Survey (USGS). 1982. Measurement and Computation of Streamflow: Volume 1. Measurement of Stage and Discharge. Geological Survey Water-Supply Paper 2175.

U.S. Environmental Protection Agency (EPA). 1992. NPDES Stormwater Sampling Guidance Document. EPA-833-92-001, U.S. Environmental Protection Agency, July 1992.

U.S. Environmental Protection Agency (EPA). 1983. Methods for Chemical Analysis of Water and Wastes. EPA-600-479-020, U.S. Environmental Protection Agency, March 1983.

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APPENDIX 2D-1
Automatic Sampler
Data Collection Forms

Automatic Sampler Equipment and Site Maintenance Checklist			
Name of City or County:			
Permanent Station Name/Location:			
Site Maintenance			
Date and time of sampling: @ Location?			
Maintenance performed by:			
Maintenance conducted weekly? Yes / No Clean debris around the sampling tube? Auto-sampler used for grab sample collection? Yes / No If yes, cut off 2x (or 2.5 x) of the tubing			
General inspection of the site.			
Extra power source is required? Yes / No			
If yes, change the battery			
Equipment Maintenance			
	Equipment	Inspection	Calibration
Conductivity meter			
pH meter			
Thermometer			
Extra power source is required? Yes / No If yes, change the battery			

APPENDIX 2D-2
Field Measurement of Stream Discharge in Open Channels

Background

This protocol describes the method for rating an open channel using a Price Type AA or Pygmy meter. This procedure will be applied to develop rating curves for both primary control devices and natural streams.

Equipment

- Current meter (Type AA or Pygmy velocity meter)
- Measuring tape
- Stopwatch
- Headset
- Notepad and pencil
- Screwdriver

Pre-Procedure Requirements

Before conducting field measurements of stream discharge, the equipment should be tested and inspected to ensure that all parts are in good working order. Proper cleaning of the meter should be performed after every use.

Before beginning the field measurements, a spin test should be performed to ensure that the rotor operates freely. For the pygmy meter, the rotor should rotate freely for between 30 and 90 seconds when spun by the user. If the rotor does not spin freely for this period of time, the rotor should be re-oiled and the test performed again.

Procedure

The current-meter measurement method computes flow by summing the products of individual subsection areas and average velocity across the subsection. A sample data collection and flow computation form is provided to assist the user with this methodology.

The following steps will be followed in computing open channel flow:

1) Choose an initial point on the stream bank higher than the limits of expected high flows. The cross-section should have as even velocity and depth as possible.

2) Stretch the tagline across the stream (perpendicular to flow) and anchor.

3) The first point data point is the intersection of the water and ground surface on the stream bank. Record the values at this point as depth = 0 and velocity = 0.

4) For stream depths less than about 2.5 feet, set the meter depth at 0.6 to record the velocity at 6/10 the depth of the water. For streams depths greater than 2.5 feet, set the meter at 0.2 to record the velocity at 2/10 the depth of the water and repeat the measurement at 0.8 to record the velocity at 8/10 the depth of the water.

5) At 25-30 points across the open channel (25-30 is an ideal number, as a minimum 5 sections will be chosen for small streams), record the horizontal distance from initial point, the vertical depth from the water surface to the bottom of the stream, and the velocity of the stream. To compute the velocity, plug the headset into the meter and count the number of beeps over a given time interval (a 30-60 second time interval shall be used). At each vertical, the velocity sampled is assumed to represent the mean velocity for the subsection.

6) Compute the total flow through the open channel by summing the products of the individual area and velocities for each subsection. The field data sheet provided guides the user through this process. This step can also be done on a spreadsheet program in the office.

7) Repeat this procedure for various stream stages.

8) Plot the data points (gauge height vs. flow) and compare with previous rating curves (if applicable). Once again, this step can be aided with a spreadsheet program.

References

U.S. Geological Survey (USGS). 1982. *Measurement and Computation of Streamflow: Volume 1. Measurement of Stage and Discharge*. Geological Survey Water-Supply Paper 2175.

Flow Rating Measurements

FLOW MEASUREMENT FORM **PAGE 1 OF 1**

Information

Location:

Date:

Field measurements by:

Stage at start of rating: Start time:

Stage at end of rating: End time:

Average stage

Meter type: Serial #:

Spin Test:

Other special notes:

Calculations

Distance	Depth	Rev	Time		Distance	Depth	Rev	Time		Qi

Computed Total Qtotal=

Calculatation made by:

Date:

Other notes:

APPENDIX 2D-3
Flow Measurement in Closed Conduits

Background

This procedure describes the three methods of flow monitoring in closed conduits using primary control devices. The methods discussed here are the Palmer-Bowlus Flume, the Type-H flume and the V-notch weir.

Equipment

- Purchased or fabricated flume or weir
- Secondary level measurement device
- Harness or mounting device for secondary measurement
- WetStop Grout or similar water tight grout

Description – Palmer Bowlus Flume

A Palmer-Bowlus flume is a restriction that is designed to produce critical flow in the throat of the flume. By producing critical flow through the throat of the flume, the level of the sub-critical liquid upstream of the flume can be measured and converted to a flow value (based upon the dimensions of the flume). Critical flow occurs as flow transitions from super-critical (upstream of the throat) to sub-critical flow (downstream of the throat). The depth of flow above the throat is the index of the discharge rate. As flow passes along the throat, critical depth is obtained. At the outlet of the flume, the water passes into super critical flow and a small hydraulic jump occurs.

Certain site characteristics should be in place in order for a Palmer-Bowlus flume to provide accurate flow measurement. Installation considerations and recommendations are discussed in the Isco *Open Channel Flow Measurement Handbook*. In general, minimum and maximum slopes on the approach are necessary; bends, turbulence and other obstructions should be avoided upstream of the device (smooth and laminar flow); and the flume should not be subject to backwater conditions.

Description – Type-h Flumes

A Type-H flume is an open channel device that discharges through a sharp edged opening. A Type-H flume is designed to incorporate the accuracy of a weir with the self-cleaning advantages of a flume. Type-H flumes differ from weirs in that flow is contracted gently from the sides only, like a flume. Type-H flumes provide good accuracy for determining small flows.

The water surface level is measured upstream of the discharge point at a distance equal to the height of the flume (D). The flow of the water as it approaches the Type-H flume must be sub-critical. As with the Palmer-Bowlus flume, flow during the approach should be smooth and laminar. Water must spill from end unimpeded, such that the flume is not operating under submerged condition. The preferred approach channel is rectangular, having the same depth and width as the depth the flume.

Description – Weirs

For measurements over a long period of time or for controlling discharges from a storm water detention pond, a more permanent structural device (such as a weir), is required. A weir is an obstruction or dam of a specific geometry that allows flow to pass over it. Weirs are normally classified according to the shape of the notch. Commonly used geometric shapes for weirs include rectangular, trapezoidal and V-notch shape. Each type of weir has an associated equation for determining the flow rate through the weir, based on the depth of the upstream water surface elevation. The edge or surface over which the liquid passes is called the crest of the weir. The sheet of water leaving the weir crest is called the nappe.

The V-notch type of weir is particularly accurate for low flow measurement. V-notch weirs, unlike rectangular or trapezoidal weirs, come to a point at the bottom and thus have no actual crest length.

The discharge equation for a V-notch weir is of the form:

$$\mathbf{Q=KH^{2.5}}$$

where:

Q = Flow Rate

H=Head on the water

K = a constant, depending upon angle of notch and units

In general, it is recommended that the minimum head on a V-notch weir should be at least 0.2 foot to prevent the nappe from clinging to the crest.

Description – Secondary Measuring Devices

A secondary measuring device must accompany a primary measuring device in order to complete a system of flow measurement. The primary objective of a secondary measuring device is to measure the level of the water surface in the primary measuring device. Many secondary measuring devices convert this value to a corresponding flow rate automatically.

A data recorder or the automated sampler receives the level value from the secondary measuring device. The data recorder or sampler converts the level value into a flow rate depending upon the geometric characteristics of the closed conduit.

Pressure transducers are recommended for level measurement. A pressure transducer is a device that senses changes in pressure exerted by a column of water above a "strain gauge". The pressure transducer measures the changes in pressure, and converts that change into an electronic signal. This electronic signal is then sent to the data logger and translated into a head reading based on the range of the transducer.

Pressure transducers must be handled with care during installation and modification. While transporting the equipment, transducers should be protected with protective covers.

Description – Staff Gauges

While automatic devices (such as pressure transducers) allow for continuous level measurement, a manual method of checking level measurement is recommended to accompany automatic devices. A staff gauge is commonly used to perform this function. A staff gauge is a fixed scale on which the level of liquid in the primary device can be read. A portion of the staff gauge is often set so that it is immersed in water (if base flow is present). Staff gauges should be installed at each of the monitoring locations to verify the level measurements collected. Staff gauge readings should be recorded by field team members during sampling events.

Procedure

Installation of Primary Control Devices

The flume or weir should be installed on a section of pipe with smooth, uniform flow and relatively constant slope. Ideally, there should be no bends, drop manholes or flow junctions within 25 pipe diameters upstream of the flume location.

After a suitable location for the flume has been selected, the section should be placed within the closed conduit. The approach section of the pipe (end farthest from the flume) will be placed upstream.

The pipe will be grouted in place using WetStop grout (or similar). The pipe will be grouted to the conduit such that the connections are as smooth as possible.

Maintenance of Primary Control Devices

Debris and other obstructions to flow may accumulate upstream of the flume or weir. The flume or weir should be inspected during site visits to ensure that clogging does not become a problem.

The seal between the flume and the pipe should be investigated for evidence of any leaks. If necessary, additional grout will be applied to the seal and the connection will be reevaluated.

References

ISCO. 1996. Open Channel Measurement Handbook. ISCO, Inc.

APPENDIX 2D-4
Estimation of Runoff Volumes for Storm Events

Background

This procedure presents a method of estimating runoff volumes for storm events. This method can be used as a preliminary method of predicting runoff volume for a given storm event during the establishment of a new monitoring location.

Procedure

The runoff coefficient method calculates total discharge volume using an equation that relates the amount of rainfall to the volume of discharge runoff from the contributing drainage area. The total runoff volume (V) is calculated as the product of the rainfall depth (R), the drainage area (A), and a runoff coefficient (C). The equation is written as:

$$V_t = R_t \times [(A_{paved} \times C_{runoff}) + (A_{unpaved} \times C_{runoff})]$$

where:

Vt = the total runoff volume in cubic feet
Rt = the total rainfall measured in feet
A paved = the area (sq ft.) within the drainage basin that is paved or roofed
C runoff = a specific runoff coefficient (no units) for the drainage area ground cover

The runoff coefficient represents the fraction of the total rainfall that is transmitted as runoff from the drainage area flowing into the point of interest. Typically used runoff coefficients are listed in the Table below.

RUNOFF COEFFICIENTS C, FOR RECURRENCE INTERVALS UP TO 10 YEARS

Business	Runoff Coefficients	Pavement	Runoff Coefficients
Downtown	0.70 to 0.95	Asphalt or concrete	0.70 to 0.95
Neighborhood	0.50 to 0.70	Brick	0.70 to 0.85
Residential		Roofs	0.70 to 0.95
Single Family	0.30 to 0.50	Lawns, sandy soil	
Multifamily, detached	0.40 to 0.60	Flat, 2%	0.05 to 0.10
Multifamily, attached	0.60 to 0.75	Average (2-7%)	0.10 to 0.15
Residential, suburban	0.25 to 0.40	Steep (7% or more)	0.15 to 0.20
Apartment	0.50 to 0.70		
		Lawns, heavy soil	
Industrial		Flat, 2%	0.13 to 0.17
Light	0.50 to 0.80	Average (2-7%)	0.18 to 0.22
Heavy	0.60 to 0.90	Steep (7% or more)	0.25 to 0.35
Parks, cemeteries	0.10 to 0.25		
Railroad yard	0.20 to 0.35		
Unimproved	0.10 to 0.30		

Source: From *Design and Construction of Sanitary and Storm Sewers. ASCE Manual of Practice No. 37,* 1970.

This procedure is designed to provide a straightforward method of calculating the anticipated runoff volume produced by a storm event. Further information on the open channel discharge calculation procedure, including the limitations of this method and factors influencing the accuracy of this method, can be found in USGS, 1982.

References

ASCE. 1970. *Design and Construction of Sanitary and Storm Sewers. ASCE Manual of Practice No. 37.*

U.S. Geological Survey (USGS). 1982. *Measurement and Computation of Streamflow: Volume 1. Measurement of Stage and Discharge*. Geological Survey Water-Supply Paper 2175.

APPENDIX 2D-5
Operation & Maintenance of Automated Composite Samplers for Flow-Weighted Sampling

Background

This protocol describes the methods for operating and maintaining the composite samplers used for storm water sample collection. The purpose of the protocol is to ensure that the samplers are configured, and operated appropriately and consistently at each sampling location. This protocol assumes that the sampler has already been installed and connected to a power source at the sampling location, the sample tubing has been attached, and the end of the sample tubing has been located in a free-flowing, well-mixed portion of the channel to be sampled.

If information is required concerning the sampler and it does not appear in this protocol, the Manufacturer's manual should be consulted.

Equipment

- Automated composite sampler
- Glass or plastic sample bottle
- Graduated cylinder (for calibration),
- Spare sample tubing (if needed for replacement),
- Spare pump tubing (if needed for replacement)

Pre-Procedure Requirements

A sampler clock is used by most automated samplers to log the time/date of collected samples and sample faults. For this reason it is important that the sampler clock is synchronized with clock of the associated data recorder. Consult the manufacturers operating manual for specific information regarding clock set-up.

Procedure

This procedure describes what parameters typically need to be configured during set-up of automated composite samplers for the collection of flow-weighted sampling. The sampler parameters describe sampler characteristics, such as number of bottles and sample tubing length. Typical Composite Sampler Parameters that need to be configured include:

- Number of Bottles #
- Bottle Unit of Measure (e.g., gallons)
- Bottle Volume
- Tubing Internal Diameter (i.d.) Unit of Measure
- Tubing Length Unit of Measure

- Tubing Length [e.g., tubing length to nearest foot]
- Timed or Flow Mode?
- Interval
- Mode (Composite/Continuous)
- Sample Volume
- Volume Calibration
- Intake Rinses
- Intake Faults

Calibration of Sample Volume

Calibration of sample volume should be performed regularly to ensure that the sampler is pumping the correct volume of storm water into the collection bottle when it takes a sample.

Operation and Sample Collection

Prior to a potential sample event, the sample program should be checked, the sample volume calibrated, and a clean sample collection bottle placed inside the sampler. If refrigeration is not available, ice should be packed around the bottle to facilitate sample preservation. The sampler should be then be secured to prevent tampering (either inside a locked shed or using padlocks on the unit itself).

Sampler Maintenance

During each field visit, the sampler should be inspected and maintained as necessary by the field crew(s). Required maintenance activities include cleaning sample tubing, clearing debris from inside and around the strainer, and replacing sample/pump tubing if necessary.

Cleaning the Sample Tubing

When required, the sample tubing should be cleaned by pumping water with mild detergent or other cleaning solution through the tubing, using the Manual Pump Mode. The tubing should then be rinsed with water several times before being set to sample. Note that this may not be practical at all of the sample locations.

Clearing the Strainer

If routine inspection of the strainer reveals that silt or other debris has accumulated around it, the strainer should be rinsed and cleared manually by the field team(s) then put back in place.

Replacement of Sample Tubing

If the sample tubing at a site becomes dirty or gets punctured, it should be replaced. This should be done by cutting a new length of sample tubing and fitting it on to the sampler.

Replacement of Pump Tubing

If the pump tubing on a sampler wears out, becomes dirty or gets punctured, it should be replaced. This should be done by cutting a new length of silicon pump tubing and fitting it on to the sampler.

To fit the new tubing, the front cover of the pump housing should need to be removed. This is done by removing the screws from the front cover then lifting the cover off to reveal the pump tubing. The tubing can then be replaced. Refer to the sampler operating manual for the correct type and length of tubing to be installed.

Desiccant Replacement

Most automated samplers have provisions for humidity control. A humidity indicator is typically located on the front side of the controller and humidity inside the controller enclosure is shown by the color of the indicator. If the indicator turns pink or white, the electronics housing should be inspected for seal failure and the desiccant module should be replaced.

APPENDIX 2D-6
Battery Storage, Recharging, and Installation

Background

If electricity or solar power is not available at the site, use the following procedure.

Equipment

- 6/2 Amp Battery Charger
- 12 volt deep cycle marine batteries

Safety

For safety, batteries should be stored on wood or plastic and off of the floor. Completely charged batteries should be stored on the top wooden shelf in the designated battery area. Partially charged batteries should be stored on the bottom wooden shelf in the designated battery area.

Procedure

Batteries should be recharged in a fume hood or a well ventilated area. Wear face and eye protection when connecting or disconnecting leads and turning power on or off.

Set the battery charger to 6 amps. With the battery charger unplugged, connect the red clip to the positive terminal. Connect the black clip to the negative terminal. Plug in the power cord to a grounded outlet. Close the fume hood door. To disconnect the leads, unplug the power cord, disconnect the black lead and then the red lead.

The battery should be recharged for approximately 10-12 hours or overnight (for a partially charged battery). The battery may need to be charged longer than that if the unit has a very small charge left. The unit should be turned off and the leads disconnected the following morning.

Installation

Install batteries by connecting the read lead to the positive terminal and then connecting the black lead to the negative terminal.

Replace batteries by disconnecting the black lead and then the red lead.

PART 3

Dry Weather Illicit Discharge Screening

Illicit discharges are unpermitted non-stormwater flows to the stormwater drainage system that contain pollutants or pathogens. Illicit discharges can be direct discharges or dumping to the stormwater system, or can occur through upstream activities that eventually flow to storm drain or drainage channel. Illegal connections are physical connections such as pipes that allow illicit discharges to the stormwater system on an ongoing basis.

Screening of stormwater outfalls during dry weather is an important tool for investigating potential non-stormwater entries to the storm drainage system. Subsequent identification and elimination of illicit discharges and illegal connections can result in substantial improvements to local water quality.

3.1 Monitoring Overview

Dry weather screening is performed on prioritized stormwater outfalls which are selected based on the potential for illicit discharges.

Screening of stormwater outfalls for illicit discharges is performed during periods of dry weather, which is defined as rainfall of less than 0.1 inch per day for at least 72 hours. This criterion avoids the screening of flows that may have resulted from wet weather (stormwater) events.

Each outfall is to be inspected for flow. When a dry weather flow is observed at an outfall, the following are to be performed on the flow:

1. **Field observations and measurements –** Site descriptions and qualitative observations of physical conditions of the outfall and flow, as well as measurement of several in-situ water quality parameters.
2. **Water Quality Sampling –** Collection of water quality samples for field analysis or laboratory analysis when indicated by the field observations and measurements.

In dry weather outfall screening, the field team is looking for indicators that point to or confirm an illicit discharge or illegal connection. Appendix 3-2 provides guidance on potential sources of pollution based upon the findings of the screening.

The discovery of an illicit discharge will warrant a more detailed pollutant source identification investigation.

Note: Local governments may develop and utilize alternative illicit discharge detection methods and procedures with the approval of Georgia EPD.

3.2 Outfall Screening Locations

Local governments should select screening locations based on the potential for illicit discharges. The following guidelines should be used to prioritize stormwater outfalls within a jurisdiction for dry weather screening of potential illicit connections:

- Utilize an up-to-date inventory of the city or county separate storm sewer system outfalls;
- Review records of previously screened outfalls to identify any subset of outfalls that have previously, and consistently, had illicit dry weather flows;
- Identify any new outfalls, or outfalls not previously screened, or outfalls identified by citizen complaints;
- Identify outfalls that drain into 303(d) listed waters, or have significant industrial land use, or discharge to streams with water quality concerns without obvious point sources;
- Rank previously screened outfalls by quarter since last screening; and
- Prioritize the set of outfalls for quarterly screening by adding the number of problem outfalls to the number of previously unscreened outfalls.

In order to provide a comprehensive screening of outfalls within a community, sites should be rotated on an annual basis.

3.3 Outfall Screening Preparation

3.3.1 Preliminary Mapping and Land Use Evaluation

Before outfall screening can be performed, preliminary mapping and land use evaluation should be completed following the prioritization and identification of target outfalls or drainage areas. Required information includes:

- Outfall locations;
- Outfall drainage areas;
- Commercial and industrial activities in each drainage area; and
- Locations of septic tanks in each drainage area.

Field maps are prepared to guide the screening team. These maps, at a minimum, should have labeled streets and hydrologic features so field teams can orient themselves.

3.3.2 Field Sampling and Analysis Equipment

Table 3-1 lists the recommended equipment for dry weather outfall screening. Before undertaking field work, the field team should ensure that all of the necessary equipment is present and in order. Both the pH meter and the conductivity meter should be calibrated. In addition, field test kits should be inspected to ensure that they have sufficient reagents and test strips/discs.

TABLE 3-1
List of Equipment and Supplies for Dry Weather Outfall Screening

Field Equipment	Function
Field maps (with outfall locations, drainage areas, and street information)	Locating outfalls for screening
Field measurement equipment (temperature, pH, conductivity meters)	Measuring field temperature, pH and specific conductivity of dry weather flows
Field test kits	Measuring fluoride, surfactants and fecal coliform
Sample bottles with labels	For collection of grab samples
Sealed, sterile sample bottles with labels	For collection of bacteria grab samples
Grab water sampler (dipper on long pole)	For outfalls/flows that are difficult to reach
Waders and walking stick	For reaching outfalls near a stream or waterbody
Hand-operated vacuum pump sampler	For shallow dry weather flows
Clear tape and applicator	To apply over label
Coolers	For transport of grab samples
Ice / ice packs	To keep samples preserved after collection and during transport from the site
Clipboard or notebook with data collection forms and COC forms / Pens	To document field data and activities
List of outfalls, directions, protocols, and H&S plan.	For reference in the field
Field logbook	To record notes
Permanent marker (extra fine)	Label sample bottles
Cell phone	Communication in the field
Handheld GPS receiver	Determining outfall locations
Digital camera	To document dry weather flow and/or conditions
Flashlight	Recording visual conditions
First Aid Kit	Health and Safety Plan
Disposable gloves, safety shoes, and safety glasses	Health and Safety Plan

3.3.3 Weather Considerations

Prior to any screening field work, check local rain gages to ensure that the conditions are appropriate for dry weather outfall screening. Dry weather is defined as rainfall of less than 0.1 inch per day for at least 72 hours.

3.4 Outfall Screening Procedures

An example Dry Weather Outfall Screening Form is found in Appendix 3-1 which can be used to record the observations and analytical results of the dry weather screening procedures. A local government may design their own form, however it should contain all of the relevant information for the dry weather screening protocol.

3.4.1 Field Observations and Measurements

Outfall screening is initiated by driving or walking to the outfall location. When an outfall is reached, it should be physically marked or labeled, and the coordinates logged using the GPS receiver (if available).

Basic descriptive information is recorded at the top part of the Dry Weather Outfall Screening Form:

- Outfall location
- Outfall ID number
- Outfall type, material and size
- Receiving stream and/or watershed name
- Date and time of screening
- Weather observations
- Staff person(s) undertaking the screening

Digital photographs are taken of the outfall and photo numbers recorded on the screening form.

Physical observations of the site are recorded on the screening form under *Field Observations and Measurements*. If no flow is observed during the outfall screening, the "Flow from outfall?" field should be checked "No" and the screening is complete. This result will be counted towards the total number of outfalls screened.

If flow is observed, then "Yes" should be checked and the following physical indicators recorded. Each of these observations associated with flowing outfalls may predict the presence of an illicit discharge or illegal connection:

- **Odor –** Description of any odors that emanate from the outfall and an associated severity score. Since noses have different sensitivities, the entire field team should reach consensus about whether an odor is present and how severe it is. A severity score of one means that it is faint or the team cannot agree on its presence or origin. A score of two indicates a moderate odor within the pipe. A score of three is assigned if the odor is so strong that the field team smells it a considerable distance away from the outfall.

- **Color –** The visual assessment of the discharge color. The intensity of color is ranked from one (slightly tinted) to three (clearly visible in the flow). The best way to measure color is to collect the discharge in a clear sample bottle and hold it up to the light. Field teams should also look for downstream plumes of color that appear to be associated with the outfall.

- **Turbidity –** The visual estimate of the turbidity of the discharge, which is a measure of the cloudiness or opaqueness of the water. Turbidity is ranked from one (slight cloudiness) to three (opaque). Like the color observation, turbidity is best observed using a clear sample bottle. The field team should also look for turbidity in the plunge pool below the outfall, and note any downstream turbidity plumes that appear to be associated with the outfall.

- **Floatables –** The presence of any floatable materials in the discharge or the plunge pool below. Sewage, oil sheen or film, and suds are all examples of floatable indicators. [Note that for dry weather screening, trash and debris are not considered indicators of an illicit discharge or illegal connection.]

Upon completing the physical observations, measure temperature, pH, and specific conductivity of the dry weather flow (either in-situ or using a sample bottle), and record the readings on the screening form.

3.4.2 Water Quality Sampling

Water quality sampling of a dry weather flow is performed to look for chemical indicators which may detect, characterize or confirm the presence of an illicit discharge or illegal connection.

Water quality sampling is required for a dry weather flow that meets any of the following criteria:

- Visible sewage or sewage odor
- Physical indicator of potential illicit discharge (color, odor, turbidity or floatables)
- pH lower than 6.5 or higher than 7.5
- Specific conductivity greater than 300 μmho/cm

Sampling may be undertaken either using field test kit equipment or by collecting grab samples for laboratory analysis. Water samples should be tested for following parameters:

- Fluoride
- Surfactants (detergents)
- Fecal coliform

Note: A local government may utilize alternate or additional chemical indicators with the approval of Georgia EPD.

3.4.2.1 Field Sampling and Analysis

Field test kits with appropriate reagents, test strips/discs, and sampling equipment should be used. The test kits must have the ability to detect fluoride within the range 0 to 2.00 mg/L and surfactants within the range 0 to 3.0 mg/L.

Follow the kit manufacturer's procedures for obtaining a test sample and completing the field analysis. Record the field analysis results on the screening form.

3.4.2.2 Grab Samples

Grab samples and subsequent laboratory analysis may be performed in lieu of field sampling for one or more of the water quality parameters. Grab samples should be analyzed using EPA-approved laboratory analysis methods.

3.4.2.3 Grab Sample Collection

A manual grab sample for a dry weather flow is accomplished by inserting the sample container (either plastic or glass depending on the parameter) under or down current of a discharge with the container opening facing upstream. In many cases, the sample container itself can be used to collect the sample. Less accessible outfalls will require the use of poles and buckets to collect the grab sample. A pre-measured cut-off milk jug can be used to

capture shallow flows from the outfall. To ensure that the manual grab samples are representative, the following procedures should be followed:

- Do not open sample bottle until sample is to be actually collected.
- Use gloves at all times when handling sampling bottles.
- Take the grab from the horizontal and vertical center of the outfall.
- Make sure not to disturb any sediments or benthic growth in the outfall.
- Transfer samples into proper container (e.g., from bucket to sample container). Fecal coliform grab samples must be collected directly into the sterile sample container.

All of the equipment and containers that come into contact with the sample should be cleaned in order to avoid contamination, and be non-reactive to prevent leaching of pollutants.

3.4.3.3 Grab Sample Handling

The grab sample bottle type, preservation requirements, and holding time requirement for those parameters being tested are listed in Table 3-2. Proper preservation and maintenance of the holding times for each parameter is essential for the integrity of the sampling results. Note that fecal coliform samples have a **short holding time of six hours** and must be returned to the lab for analysis within this time or the results may be unrepresentative of the flow.

TABLE 3-2
Modified Handling Requirements for Samples

Parameter	Container Type[1]	Sample Volume (g)	Sample Preservation	Maximum Holding Time
Fluoride	P,G	500ml	Cool, 4ºC	28 days
Surfactants (detergents)	P	500ml	Cool, 4ºC	48 hours
Fecal Coliform[2]	PP,G	100 ml	Cool, 4ºC	6 hours

[1] Polyethylene (P), Polypropylene (PP), Glass (G) – EPA-approved sample containers (40 CFR 136)
[2] In chlorinated waters, dechlorinate the sample with sodium thiosulfate by adding 1 ml of 10% $Na_2S_20_3$ to the 100 ml sample

3.4.3.4 Grab Sample Identification and Labeling

A sample numbering system should be used to ensure that each sample is uniquely identified in the field and tracked on field data collection forms. The sample numbering should be as follows:

###-MMDDYY-HH:MM

where:

- ### = A unique number for each sample location
- MMDDYY = Month, day, year
- HH:MM = Time in military units

All of the samples collected at the site should be placed in the appropriate sample containers for preservation and shipment to the designated laboratory. Each sample should be identified with a separate identification label. A waterproof, gummed label should be attached to each sampling container. Information to be recorded on the label should include:

- Site name;
- Sample number;
- Analysis to be performed;
- Date and time of collection;
- Preservation used and any other field preparation of the sample; and
- Initials of field crew collecting the sample.

3.4.3.5 Grab Sample Documentation

A chain-of-custody (COC) form should accompany all samples. A sample COC form is found in Appendix 2A-1 of this document. The COC form shall include all of the information provided on the sample label discussed in the preceding section.

The purpose of the COC form is to provide a mechanism for tracking each sample submitted for laboratory analysis. The information on the COC form must be identical to the information of the sample label. A COC form should be prepared by the sample collector for each set of samples submitted for laboratory analysis. The form should be placed in a re-sealable plastic bag (to keep the form dry) and sealed inside each sample cooler. When transferring possession of the samples, the individual relinquishing and receiving samples should sign, date, and note the time on the COC form. This record documents the transfer of custody from the sampler to another person, to/from a secure storage area, and to the laboratory. Copies of the COC forms should be kept for future reference.

3.4.3.6 Analytical Laboratory Coordination and Sample Delivery

The samples should be packed in coolers with ice (or ice packs) to ensure they maintain the required temperature of less than or equal to 4°C during transport to the designated laboratory. Contact the laboratory prior to sampling to assure that the samples will be analyzed within their holding time. Samples may be placed in individual one-gallon re-sealable bags as a precaution to avoid spilling the sample. All glass bottles should be individually bagged and bubble-wrapped to prevent breakage on the way to the lab. Samples may be placed in a large trash bag inside a cooler (to ensure against the sample leaking) with ice completely covering the samples.

3.5 Quality Assurance/Quality Control

This section describes the elements of the field quality assurance/quality control (QA/QC) program. The overall QA/QC objective for the monitoring program is to ensure that the data collected are of good quality.

3.5.1 Field QA/QC

Field quality control procedures include calibration procedures, field blanks and field duplicates. The field equipment should be calibrated appropriately prior to leaving for the

sampling site to ensure proper performance of the equipment. This includes the pH meter, conductivity meter, and the thermometer. The pH meter should be calibrated using two buffers that bracket the expected pH range (typically 4 and 7). The conductivity meter is calibrated by rotating the probe below the surface in a standard Potassium Chloride solution in a circular motion. The readings must be within 10 percent to be acceptable. The thermometers used should be accurate to ± 5°C.

Quality control blanks should be used in the field to determine potential sample contamination during sample collection, handling, shipment, storage, or laboratory handling and analysis. Reagent grade water should be used for the quality control blanks. A minimum of one field blank for surfactants (detergents) and fecal coliform is required each day with scheduled field screening. For fluoride, a field blank should be used with approximately 10 percent of samples (or as required by the lab).

Field duplicates should be collected on approximately 10 percent of the samples to assess the representativeness of sampling procedures in addition to the normal uncertainty associated with the analysis.

3.5.2 Laboratory QA/QC

The laboratories should follow Georgia EPD- approved methods and routinely perform quality control checks during laboratory analysis, including calibration standards, blanks, laboratory control samples, laboratory control duplicate samples, matrix spikes, and matrix spike duplicates. Spikes and duplicates should be performed on a minimum of 10 percent of the samples and should meet data quality objectives established by the client.

References

Center for Watershed Protection. 2004. Illicit Discharge Detection and Elimination – A Guidance Manual for Program Development and Technical Assessments.

U.S. Environmental Protection Agency (EPA). 1993. Investigation of Inappropriate Pollutant Entries into Storm Drainage Systems – A User's Guide. EPA/600/R-92/238, U.S. Environmental Protection Agency, January 1993.

U.S. Environmental Protection Agency (EPA). 1992. NPDES Stormwater Sampling Guidance Document. EPA-833-92-001, U.S. Environmental Protection Agency, July 1992.

Appendix 3-1
Dry Weather Outfall Screening
Data Collection Forms

Dry Weather Outfall Screening Form	
Name of City or County:	Data Sheet Number:
Date of screening (MM/DD/YY):	Time of screening:
Weather conditions:	
Sampling performed by:	

Outfall Description	
Outfall Location:	Outfall I.D. Number:
Outfall Type/Material: ❒ Closed Pipe (circle): RCP CMP PVC HDPE Other: _____ ❒ Open Channel (circle): Concrete Earthen Grassy Other: _____	Outfall Diameter/Dimensions:
Receiving stream and watershed name:	
Land use/industries in drainage area:	
GPS Coordinates:	Photo numbers:

Field Observations and Measurements
Flow from outfall? ❒ Yes ❒ No Flow Description: ❒ Trickle ❒ Moderate ❒ Substantial
Odor: ❒ None ❒ Sewage ❒ Sulfide (rotten eggs) ❒ Petroleum/gas ❒ Rancid/sour ❒ Other _____ Relative severity: ❒ 0-None ❒ 1-Faint ❒ 2-Easily Detected ❒ 3-Noticable from a distance
Color: ❒ Clear ❒ White ❒ Gray ❒ Orange/Rust ❒ Red ❒ Yellow ❒ Green ❒ Brown/Black ❒ Other Relative severity: ❒ 0-None ❒ 1-Faint ❒ 2-Clearly visible in bottle ❒ 3-Clearly visible in flow _____
Turbidity: ❒ None ❒ Cloudy ❒ Opaque ❒ Silty ❒ Muddy ❒ Other _____ Relative severity: ❒ 0-None ❒ 1-Slight cloudiness ❒ 2-Cloudy ❒ 3-Opaque
Floatables: ❒ None ❒ Sewage ❒ Petroleum (oil sheen) ❒ Suds ❒ Other _____ Relative severity: ❒ 0-None ❒ 1-Few/slight ❒ 2-Some ❒ 3-Heavy
Flow Temperature (ºC):
Flow pH: pH meter calibrated? ❒ Yes ❒ No
Flow Conductivity (µmho/cm): Conductivity meter calibrated? ❒ Yes ❒ No

Water Quality Sampling			
Field Test Kit Manufacturer:		Model:	
Fluoride (mg/L):		Fecal Coliform (MPN/100ml):	
Surfactants (mg/L):		Analysis Comments:	
Grab sample for lab? (fluoride/surfactants) ❒ Yes ❒ No		Bacteria Grab sample for lab? (fecal coliform) ❒ Yes ❒ No	
Grab Sample ID:		Bacteria Grab Sample ID:	

Outfall Potential for Illicit Discharge:

❒ Unlikely - or- No Flow ❒ Possible (presence of two or more indicators)

❒ Suspect (one or more indicators with severity of 2 or 3) ❒ Obvious - or- Confirmed

NOTE: Water quality sampling (using a field test kit and/or grab samples) is required for a dry weather flow that meets any of the following criteria: Visible sewage or sewage odor; physical indicator of potential illicit discharge (color, odor, turbidity or floatables); pH lower than 6.5 or higher than 7.5; or specific conductivity greater than 300 µmho/cm.

TABLE 3-3
Dry Weather Outfall Screening – Sample Data Tracking Form

Date	Outfall I.D. Number	Flow? (Y/N)	Odor (describe)	Color (describe)	Turbidity (describe)	Floatables (describe)	Temp (°C)	pH	Conductivity (µmho/cm)	Fluoride (mg/L)	Surfactants (mg/L)	Fecal Coliform (MPN/100ml)	Follow-up Actions

APPENDIX 3-2
Evaluating Dry Weather Screening Results

Background

Dry weather screening of stormwater outfalls is an important tool used to evaluate non-stormwater flows in the storm drainage system. Effectively evaluating and interpreting dry weather screening results and data is the first step in identifying and tracing a potential illicit discharge or illegal connection.

Field Observations

Field observations of a dry weather flow include odor, color, turbidity and floatables. These parameters are qualitative indicators detected by visual inspection and smell, and require no measurement equipment. They are important in evaluating a dry weather flow for a potential illicit discharge, and may confirm the most severe or obvious discharges.

Table 3-4 lists the field observation parameters, along with potential sources for a number of observed conditions.

Field Measurements and Water Quality Sampling Results

Field measurements and water quality sampling provide additional information which may detect, characterize or confirm an illicit discharge or illegal connection. Temperature, pH and conductivity measurements are completed in-situ using probes or other equipment that is calibrated prior to field work. Water quality sampling for the presence of fluoride, surfactants and fecal coliform is performed either in-field using test kit equipment or by collecting grab samples for laboratory analysis.

Table 3-5 lists the various parameters included in the dry weather screening protocol along with benchmarks and guidance on evaluating results. Figure 3.1 provides a flow chart which can be used to identify illicit discharges based upon findings.

Ranking the Potential for an Illicit Discharge

Based upon the screening results, all outfalls should be ranked for their potential for an illicit discharge:

- Those outfalls without flow or that appear to be from an uncontaminated source would be ranked "Unlikely or No Flow."
- Any flow that shows two or more suspect field observation or chemical indicator that falls outside of the range of normal stormwater or groundwater should be marked as "Possible" for an illicit discharge.
- The presence of one or more field observations with a rank of two or three, or chemical indicators far outside of the range of normal stormwater or groundwater should be ranked "Suspect."
- Any flow that is clearly an illicit discharge should be listing as "Obvious or Confirmed."

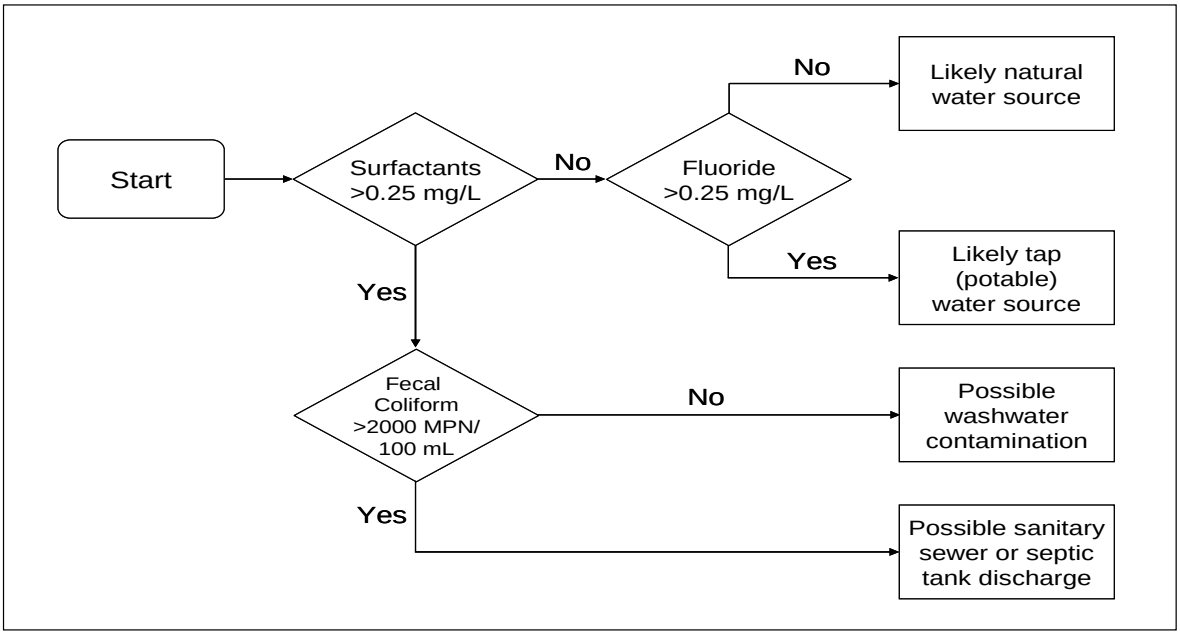
TABLE 3-4
Physical Observations and Potential Sources

Parameter	Observations	Potential Source(s)
Odor	Sewage	Sanitary sewer; septic tank discharges
	Sulfur (rotten eggs)	Industrial discharge (sulfides and/or organics); sanitary sewer; septic tank discharges
	Oil / gasoline	Facilities associated with vehicle maintenance and operation; petroleum product manufacturing or storage; industrial discharge
	Rancid / sour	Food preparation facilities (restaurants, hotels, etc.)
Color	Orange / rust	Construction site or unstabilized soil (eroded soil and clay)
	White / milky	Sanitary sewer; septic tank discharges; residential or commercial washwater; concrete or stone operations; fertilizer
	Grey	Residential or commercial washwater; dairies
	Red	Meat packers
	Yellow	Industrial discharge
	Green	Industrial discharge; Facilities associated with vehicle maintenance and operation (antifreeze)
	Brown / black	Industrial discharge
Turbidity	Cloudy	Sanitary sewer; septic tank discharges; residential or commercial washwater; concrete or stone operations; fertilizer; industrial discharge
	Opaque	Food preparation facilities (restaurants, hotels, etc.); industrial discharge
	Silty / Muddy	Construction site or unstabilized soil (eroded soil and clay)
Floatables	Sewage	Sanitary sewer; septic tank discharges
	Petroleum (oil sheen)	Facilities associated with vehicle maintenance and operation; petroleum product manufacturing or storage; industrial discharge
	Suds	Sanitary sewer; septic tank discharges; residential or commercial washwater

TABLE 3-5
Interpretation of Field Measurements and Water Quality Sampling Parameters

Parameter	Benchmarks	Evaluation
Temperature	Temperature should be near or below ambient conditions for groundwater or stormwater runoff.	Higher than ambient temperature may indicate stream condensate or industrial process water.
pH	The normal pH range for stormwater runoff is between 6 and 8, with 7 being neutral.	pH is a relatively good indicator of liquid wastes from industries, which can have very high or low pH values (ranging from 3 to 12). The pH of residential and commercial washwater tends to be in the range of 8 or 9.
Conductivity	Stormwater should have a low conductivity (under 300 μmho/cm).	Conductivity greater than 300 μmho/cm indicates a high dissolved solids content in the flow which may be from an illicit discharge or illegal connection
Fluoride	There should no traces of fluoride in the stormwater.	Presence of fluoride indicates the presence of potable (treated) water. Fluoride can often be used to separate treated potable water from untreated water sources, such as stormwater, groundwater or non-potable industrial waters.
Surfactants (detergents)	There should be no traces of surfactants (detergents) in the stormwater.	This parameter is associated with cleaning/washing operations and may indicate residential or commercial wastewater.
Fecal Coliform	Fecal coliform is an indicator of fecal bacteria from warm-blooded animals.	Its presence in high numbers often indicates contamination with sanitary waste, although high levels of pet waste may also produce similar results.

FIGURE 3-1 Flowchart to Identify Illicit Discharges using Outfall Screening Sampling Results



Following Up on Potential Illicit Discharges

All outfalls ranked as possible, suspect or obvious illicit discharges require follow-up actions and activities to determine the specific source(s) of contamination. There are a variety of methods for illicit discharge source identification, including:

- **Mapping Analysis –** Evaluation of the drainage area, land uses and properties above the outfall including the route of the storm drainage system and locations of storm drains. This enables local staff to predict the likely locations of illicit discharges and illegal connections. Geographic Information Systems (GIS) are a useful tool for identifying illicit discharges through mapping analysis.
- **Drainage Area Investigation –** A windshield survey or more detailed property inspections in the drainage area that has the illicit discharge. These inspections are often performed following a mapping analysis.
- **Piping Schematic Review –** Examination of building plans and plumbing details for potential sites where improper connections to the storm drainage system may have occurred.
- **Smoke Testing –** Testing of pipes to locate connections by injecting a non-toxic vapor (smoke) into the system and following its path of travel.
- **Dye Testing –** Addition of colored dye to the drain water in suspect piping and subsequent surveillance to determine if dyed water appears in the storm drain system, thus indicating an illegal connection.
- **Septic System Investigation –** Low density residential watersheds may require special investigation methods when failing septic systems are suspected. Homeowner surveys, surface investigations and infrared photography have all been used successfully to identify problem septic system facilities.

The appropriate method for any given outfall or area will be heavily dependent on the watershed and land use conditions, drainage system characteristics, available resources and the nature of the discharge and screening results.

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PART 4

Monitoring for Assessing TMDL Implementation and Delisting of Impaired Waters

This section contains guidance from Georgia EPD on the methodology and data requirements for listing and delisting waterbodies on the State's 305(b)/303(d) list of impaired waters.

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Guidance On Submitting Water Quality Data For Use By The Georgia Environmental Protection Division In 305(b)/303(d) Listing Assessments

Georgia Department of Natural Resources
Environmental Protection Division
October 2002

Guidance On Submitting Water Quality Data For Use By The Georgia Environmental Protection Division In Listing Impaired Waters
October 2002

Requirements for the submission and acceptance of water quality data for use in listing impaired waters by the Environmental Protection Division (EPD) are set forth in the *Rules And Regulations For Water Quality Control, Chapter 391-3-6-.03-(13)*, hereinafter referred to as the "Rule," (refer to Appendix A). The purpose of this document is to outline the general considerations and, where necessary, the specific requirements and procedures to ensure that submitted data is useful in the listing process.

The most important component in ensuring data acceptance is the preparation of a Sampling and Quality Assurance Plan (SQAP). The Rule requires that the Division concur with the Plan prior to monitoring. Division concurrence with a SQAP will be provided in writing within three weeks of receipt if the Plan is determined to be acceptable. Specific guidance on SQAP preparation may be found in the following section. For on-going, long term monitoring projects, the SQAP does not have to be resubmitted if there are no changes to monitored parameters, certification/accreditation provisions or quality assurance measures. Sampling locations may be added to a project by amending the SQAP, although the reduced data set from new stations may be insufficient for use in making listing decisions. Such amendments must be submitted in writing with the associated map revisions; approval of Plan amendments by the Division will also be made in writing.

EPD has guidelines for the number of measurements necessary for data to be used in listing decisions for all parameters. Special monitoring requirements apply for some parameters based on seasonal or flow variations. These are summarized in Appendix B. The guidelines for sampling frequency and number of observations should be considered carefully in preparation of the SQAP and the overall design of the study.

The data generated during an approved study must be presented in a final report to the Division. These reports are due on or before June 1st of odd numbered years if the data is to be considered for the subsequent year's 305(b)/303(d) listing assessments. Elements of a final report should follow the outline of the SQAP. At a minimum, the final report should include the following:

1. Narrative on water quality conditions documented.
2. Rainfall data for the study period if available.
3. Pollutant sources identified (if any).
4. Parameter values that did not meet Quality Assurance standards.
5. Presentation of all data obtained in tabular form along with sampling dates, times, station numbers, location descriptions and map.
6. Geometric means calculated for bacteria data.
7. If metals data are not dissolved values, the total recoverable metals results should include corresponding total suspended solids and hardness data for calculation of dissolved metals values.

Required Elements Of A "Sampling & Quality Assurance Plan"

PART ONE: Introduction & Study Objectives

1. The organization conducting or coordinating the project should be named and an official liaison with EPD identified. Contact information including telephone and facsimile numbers, mailing address and e-mail parameters (if applicable) should be provided. A listing of individuals or organizations participating in the project should be included but is not required.

2. Background information detailing the need for the study must be discussed. This should include water quality concerns in the study area, the current listing status and designated use categories of the streams to be monitored and the results of any "screening" data (if available).

3. The objective(s) of the project must be identified. In addition to generating water quality data to be used by EPD in listing decisions, this could include documentation of water quality conditions, identification of pollutant sources (point or non-point), long-term stream monitoring or stakeholder involvement in watershed protection.

PART TWO: Sampling Plan

1. The location of the study area must be delineated. This must include county/city jurisdictions and the river basin or watershed and streams to be sampled, by name. Sampling stations must be identified by general narrative and specific location (GPS coordinates, for example). The inclusion of sampling station maps is also required; the preferred scale is that used on United States Geological Survey (USGS) 1:24,000 topographic quadrangle maps.

2. The sampling parameters to be monitored during the study must be identified. If different parameter suites are to be analyzed at different stations or station groups, these must be defined. Parameters should also be differentiated between those that are to be measured in the field, those that will be analyzed in a laboratory by project personnel and those that will be tested by an outside "contract" or commercial laboratory.

3. Sampling schedules must be discussed in a qualitative manner. (These will be largely determined by the parameters analyzed and Assessment Data Sampling Requirements; refer to Appendix B). Special scheduling considerations for individual parameters should be addressed ("wet season" and "dry season" sampling for metals, for example).

4. Personnel and required material resources available should be discussed realistically so as to indicate they are sufficient to perform the planned sampling schedule. In addition to project field personnel, other necessary resources such as vehicles, sample bottles and preservatives, field instruments and standards should be provided for in the plan.

PART THREE: Quality Assurance Plan

1. Statements that the requirements of the *Water Protection Branch Quality Assurance Manual* (June, 1999) and *Title 40* of the *Code of Federal Regulations, Part 136* will be adhered to must be included in all plans. (These requirements are specified in the Rule).

2. The project provisions for field quality assurance must be comprehensively addressed. This shall include the following topics:

a. Sample collection technique (by parameter) and sample representativeness

b. Considerations for proper sample containers, required preservatives, refrigeration/ storage and adherence to holding time limitations

c. Field instrument calibration, quality assurance measures on meter and probe response; analytical duplicates, standards and record keeping

d. Sampling personnel training in all applicable procedures

3. Laboratory Analyst Certification / Laboratory Accreditation: The Rule requires that laboratory analyses be performed by a Certified Laboratory Analyst or an Accredited Laboratory.

a. If analyses will be performed by a Certified Laboratory Analyst, the analyst must be identified and their certification number and expiration date must be provided.

b. If analyses will be conducted by an Accredited Laboratory, the accrediting organization and the accreditation expiration must be cited. The laboratory must be accredited for the media and specific analyses it is expected to perform. The plan must include a statement that the accredited laboratory will perform all Quality Assurance/Quality Control measures required by the accrediting organization on samples analyzed for the study.

c. For all laboratories, the Quality Assurance/Quality Control measures required by specific methods referenced in 40 CFR Part 136 must be implemented, and a statement to that effect must be included in the plan. The plan shall also state that adequate records on analytical procedures ("bench sheets") and the Quality Assurance/Quality Control measures shall be maintained to document their proper implementation and performance, and that the records shall remain on file and available for review for a minimum of three years.

Appendix A

Rule 391-3-6-.03 -- Water Use Classifications and Water Quality Standards.

(13) **Acceptance of Data.** Sampling methods for water quality samples collected and reported by any person to the Division for its use in listing or delisting impaired waters pursuant to the State's responsibilities under Sections 303(d) and 305(b) of the Federal Act shall conform to the guidance in the *Water Protection Branch Quality Assurance Manual* (June, 1999), Georgia Department of Natural Resources, Environmental Protection Division, Water Protection Branch, Atlanta, GA 30354. Analytical standards for these samples must comply with the requirements of *Title 40, Code of Federal Regulations*, Part 136. Sample analyses shall be performed by an analyst certified in compliance with the *Georgia State Board of Examiners for Certification of Water and Wastewater Treatment Plant Operators and Laboratory Analysts Act*, as amended, or by a laboratory facility accredited in compliance with the *Georgia Rules for Commercial Environmental Laboratory Accreditation* (O.C.G.A. 12-2-9). A site-specific sampling and quality assurance plan is required if the data is to be considered and Division concurrence must be obtained prior to monitoring. Laboratories operated by Federal and State government agencies and laboratories at academic institutions with active or current contracts with the Division are exempt from these provisions.

APPENDIX B

Minimum Number of Samples for Assessment of Data for 303(d) Listing Purposes		
Criterion	**Type of Sample**	**No. of Samples**
Dissolved Oxygen	Instantaneous Field Reading	20 measurements within a 12 month period (1-2 measurements per month)
pH	Instantaneous Field Reading	20 measurements within a 12 month period (1-2 measurements per month)
Temperature	Instantaneous Field Reading of Water Temperature	20 measurements within a 12 month period (1-2 measurements per month)
Bacteria	Grab	16 Samples (4 samples collected within a 30 day period over 4 calendar quarters to calculate 4 geometric means). **Note: The 30 day sampling period should not overlap the months of April/May and October/November due to changes in the in-stream water quality standards for bacteria.**
Metals (including Mercury)	Grab using Clean Sampling Techniques. **Note: Samples may be analyzed for dissolved or total recoverable metals. If dissolved is used, total recoverable must also be analyzed and reported. Total Suspended Solids and hardness must also be analyzed and reported for every sample.**	2 Samples (collected during one winter season and one summer season)
Organic Chemicals (including Pesticides)	Grab	2 Samples (collected during one winter season and one summer season)
Flow/Precipitation	If stream gage is in the vicinity of the sampling location, a gage height must be reported. Flow conditions at the time of sampling and recent precipitation measurements from the weather service should be reported.	Noted for each sampling event.

PART 5A

Biological/Habitat Assessment -- Benthic Macroinvertebrates

This section contains guidance on biological monitoring and analysis for benthic macroinvertebrates for the biological/habitat assessment component of the District's Water Quality Monitoring Plan. These procedures and protocols were taken directly from Georgia EPD's Standard Operating Procedures for Macroinvertebrate Biological Assessment of Wadeable Streams in Georgia, 2007.

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Macroinvertebrate Biological Assessment of Wadeable Streams in Georgia

STANDARD OPERATING PROCEDURES

March 2007

Georgia Department of Natural Resources

Environmental Protection Division

Watershed Protection Branch

Version 1.0

TABLE OF CONTENTS

Chapter 3 Macroinvertebrate Biological Assessment Sorting and Subsampling Procedures

Chapter 4 Macroinvertebrate Biological Assessment Identification Procedures

Chapter 5 Macroinvertebrate Biological Assessment Data Analysis Procedures

List of Tables and Figures

Tables

Figures

Chapter 1

Georgia Department of Natural Resources

Environmental Protection Division

Introduction and Background Information

1.1 Introduction

The Standard Operating Procedures (SOP) Manual is designed to describe and document the Georgia Department of Natural Resources (GADNR), Environmental Protection Division's (EPD) operations associated with aquatic macroinvertebrate biological monitoring in **wadeable streams**. The primary purpose of this document is to establish and maintain uniform methodology and quality control guidance for biological data collected within the state. Compliance with these procedures is essential to produce reliable biological data. This document is intended for use as a training resource for all Ambient Monitoring Unit sampling employees as well as a technical manual to be used as a reference for study plans and reports. Deviations from this SOP manual should receive prior approval from EPD staff and be explained and documented.

Resident biota in a water body functions as continual monitors of environmental quality. They are capable of detecting the effects of episodic and cumulative pollution, as well as the effects of altering available habitat. Any stress (biological, chemical, or physical) imposed on an aquatic ecosystem manifests its impact on the biological organisms present in that ecosystem such that the organisms may not sufficiently recover to reestablish their pre-stress community structure (Loeb and Spacie, 1994). Aquatic macroinvertebrates are good indicators of contamination because they are resident monitors of pollution and are less able to migrate from the impairment. Since macroinvertebrates must persist in the contaminant field, they indicate past conditions as well as current conditions. Therefore, the structure and function of resident biota is a direct measurement of the condition of the aquatic ecosystem.

Other advantages of using benthic macroinvertebrates include: (1) large number of species offering a spectrum of responses to perturbations; (2) well developed qualitative sampling and analytical methods; (3) simple inexpensive equipment requirements; (4) the responses of many common species to different types of pollution have been established; (5) macroinvertebrates are well suited to experimental studies of perturbation; and, (6) the taxonomy of many groups is well known and identification keys are available (Hauer and Lamberti 1996).

When simultaneously monitoring the chemical, physical, and biological integrity of a water body, the ecological integrity or health of the aquatic ecosystem can be determined (USEPA, 1990). Both biological and chemical methods play critical roles in a successful pollution control program. They should be considered complementary rather than mutually exclusive approaches that will enhance overall program effectiveness when used appropriately (Plafkin et al., 1989). In turn, the ecological condition of an aquatic ecosystem provides an evaluation for determining whether or not the water body is, or is not, impaired. In turn, with these tools, aquatic biologists are able to evaluate the ecological condition for determining whether or not the waterbody is impaired.

1.2 Laws and Regulations

The condition of surface waters in the Unites States is covered by a number of regulations regarding monitoring and control of identified pollutants, non-point sources of pollutants, the maximum load of both point and non-point pollutants, and the development of new and better monitoring strategies.

Section 101(a) of the Clean Water Act (CWA) states the restoration and maintenance of the chemical, physical, and biological integrity of the nation's waters as its primary objectives. Section 305 (b) of the CWA requires states to regularly report the condition of their waters (EPA 1997). This is accomplished by conducting ambient water monitoring to determine changes in water quality over time, designating the sources of water quality problems, and determining if pollution control programs are working.

A point source is defined as any discernible, confined, and discrete conveyance, including but not limited to any pipe, ditch, channel, tunnel, conduit, well, discrete fissure, container, rolling stock, concentrated animal feeding operation, or vessel or other floating craft, from which pollutants are or may be discharged (EPA 1992a). According to section 502(14) of the CWA, nonpoint sources are defined as sources of water pollution that do not meet the legal definition of a "point source". Section 319 of the 1987 CWA indicates that states are; "(1) required to conduct statewide assessments of their waters to identify those that were either impaired (did not fully support state water quality standards) or threatened (presently meet water quality standards but are likely not to continue to meet water quality standards fully) because of Nonpoint Source Programs (NPS's); (2) required to develop NPS management programs to address the impaired or threatened waters identified in their nonpoint assessments; and, (3) entitled to receive annual grants from the EPA to assist them in implementing their NPS management programs once the EPA has approved the assessments and programs" (EPA 1997).

Total maximum daily loads (TMDLs) allocate allowable loads among different pollutant sources (both point- and nonpoint-) so that appropriate control actions can be taken, water quality standards may be achieved, and human health and aquatic resources can be protected (EPA 1994a). TMDLs are a significant issue throughout the nation and it is important to understand the relationship between nonpoint sources and biological assessments and criteria. The total maximum daily load (TMDL) program is designed to identify those waters that do not meet non-point source water quality standards, required by section 303 (d) of CWA (EPA 1997). States are required to develop TMDL's for each chemical parameter and a priority ranking for those waters not meeting water quality standards. The TMDL program helps to identify and establish controls to reduce nonpoint source pollution.

1.3 Site Selection

When selecting sample sites one should take into consideration the specific monitoring issue (*e.g.* short-term impacts or long-term trends), possible point and non-point influences in the watershed of the stream to be sampled, structural influences (bridges,

dams) that may fall into the selected reach, accessibility, and safety. Before any sampling is conducted, an initial reconnaissance inspection should be accomplished to determine if the selected sample locations are feasible. The field investigator should fill out a Waterbody Reconnaissance Report (see p.2A-2) during this time.

Sampling locations immediately above or below the confluence of two streams, or a stream and a point/non-point source discharge, should be avoided as mixing does not occur immediately. Unless the stream is extremely small or turbulent, an in-flow will usually flow along the stream bank with little lateral mixing for some distance. This could result in two very different biological populations and an inaccurate assessment of stream conditions. This can be avoided by sampling the area where mixing has occurred. If the study is to determine the distance of effects caused by a pollution source then a series of sampling stations at points of increasing distance from the impact source(s) can be set up. These stations will provide a basis for delineating impact and recovery zones.

Sampling near the mouths of tributaries entering large waterbodies should be avoided because these areas will have habitat more typical of the large waterbody (Karr et al., 1986). Streams in different regions of the state can have structures that deviate greatly from the morphology of a "typical" stream. What follows is an attempt to help you determine how to sample those streams. **Braided streams** - all of the braids in the 100-meter reach should be sampled; the jabs should be divided among the braids as evenly as possible. **Swampy** - some of the streams in South Georgia can spread out and be swampy in appearance, these should only be sampled if the main channel is discernible and the wet areas outside of the channel are at most only a few inches deep. The jabs in these streams should be collected within the channel and not the shallow backwater areas. **Tidal** – these should be sampled only at low tide; this is the only time true discharge can be calculated. This will also allow you to collect the freshwater organisms that may have been hiding from possible salt influence at high tide. It should be noted here that salt influence could be easily determined during reconnaissance by taking a conductivity reading at high tide. This reading is important because until the Wildlife Resources Department (WRD) or the Coastal Resources Division (CRD) develops metrics for estuarine fish, salt influenced tidal sites do not need to be sampled for fish. Valid conductivity readings must be taken and presented to allow for fish sample exclusion for watershed assessments (WSA's) and watershed protection plans (WPP's). **Blackwater** – Some of the subecoregions are dominated by, or completely made up of, blackwater streams. These are sampled the same way as other streams in the area; the only distinction that determines a difference in sampling protocols is high gradient or low gradient.

Locally modified sites, such as small impoundments (man and beaver made) and bridge areas, should be avoided unless these data are needed to assess their effects. Bridges and impoundments may alter the water flow or cause sediment deposition, which may result in major changes within the macroinvertebrate population diversities. Due to the fact that material is often thrown from bridges, it is advisable to sample the waterbody at least 100 meters upstream of the bridge to reduce the chance of influence. Being upstream at least 100 meters minimizes the effect on stream velocity, depth, and overall habitat quality.

The sampling reach should be representative of the characteristics of the stream. No major tributaries should be discharging within the sampling reach. When selecting the 100 meter reach, every effort should be made to avoid sampling in areas where beaver dams are present. The presence of beaver dams may result in inaccurate data.

1.3.1 For Monitoring Plans and Watershed Assessments

County and topographic maps should be used to locate all possible access points within the scope of the study. It is also strongly encouraged that a pre-submittal meeting be scheduled with EPD staff to discuss the components of the Monitoring Plan. This will cut down on the amount of time spent in transit for revisions.

1.4 When to Sample

Reproductive periods and different life stages of aquatic insects are related to the abundance of particular food supplies (Cummins and Klug, 1979). Peak emergence and reproduction typically occur in the spring and fall, although onset and duration vary somewhat across the United States. During peak reproduction, approximately 80 percent of the macroinvertebrates will be too small to be captured in sufficient numbers to accurately characterize the community. Additionally, food source requirements for early instars are different from those of later instars. Therefore, the biologically optimal sampling season would occur when the habitat is utilized most heavily by later instars and the food resource has stabilized to support a balanced indigenous community (Plafkin et al., 1989). Biologically optimal periods start in the fall and continue through winter. **Georgia's Index Period for sampling is mid-September through February**.

If the study objective is to document stream conditions due to improvement of wastewater treatment, removal of a treatment system, etc., then the biological evaluation could be limited to one sampling event during periods of greatest environmental stress such as low flow, high temperature periods for point source discharges or high flow, runoff periods for nonpoint source discharges. Assessment of worst-case conditions may be needed under certain permitting regulations, or as a follow-up to sampling during biologically optimal periods, where impairment is detected (Plafkin et al., 1989).

In evaluating the success of a Best Management Practice (BMP), it is necessary to compare pre-improvement biological conditions to post-improvement biological conditions. Thus, sampling strategies should be designed to allow such comparison.

Reasons for sampling during the specific index period:
1. Watershed assessments to evaluate general ecosystem health.
2. Problem identification of specific sites.
3. Trend monitoring efforts.
4. Measuring BMP effectiveness.

1.4.1 Severe Weather Events

Macroinvertebrate communities can be heavily influenced by severe weather events such as rain and drought. Sampling immediately after a major rain event (>1 inch) should be avoided because of the increased velocities and scour, both of which can cause drift of the macroinvertebrates. Collection of samples from affected streams should be postponed at least two weeks after a major rain event to allow the macroinvertebrate communities to stabilize. Drought can have just as profound of an effect on macroinvertebrate communities; caused by both reduction of dissolved oxygen from high temperatures and of available living space from decreased flow. Streams that have extremely low flow due to severe drought should not be sampled. Additionally, streams that exhibit increased water levels due to drought relief caused by increased rains should not be sampled until two to three weeks of sustained higher flows have passed. This will allow proper colonization and stabilization of the recently wetted habitats.

Sampling of fish and macroinvertebrate communities in a reach should not be done concurrently. The process of sampling one of the communities will invariably disturb the other. If fish are sampled first then two weeks should be allowed for stabilization of the macroinvertebrate communities. If macroinvertebrates are sampled first there would be substantially less wait time for the fish communities to stabilize due to the higher mobility of fish.

Catastrophic events such as fish kills or spills can occur at any time. Therefore, the evaluation of these sites might not occur during specific index periods.

Some pages are intentionally left blank to allow for copying of field and laboratory sheets.

This document is to be considered a living document and may be updated as new data and procedures become available. Updates will be posted on the website listed below.

If you have any questions or comments about this manual, please contact the Ambient Monitoring Unit at:

Georgia Environmental Protection Division
Watershed Protection Branch
Watershed Planning and Monitoring Program
Ambient Monitoring Unit
4220 International Parkway Suite 101
Atlanta, Georgia 30354
Phone: 404-675-6236
Fax: 404-675-6244
http://www.gaepd.org/

1.5 References

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Chapter 2

Georgia Department of Natural Resources

Environmental Protection Division

Macroinvertebrate Biological Assessment Field Procedures

Sampling Procedure:

GA DNR Macroinvertebrate Bioassessment

Sampling Process Organizational Chart

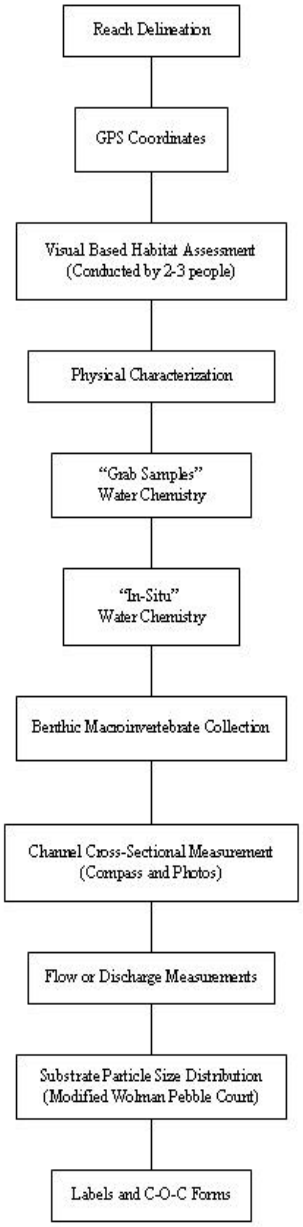

Figure 2-1: GA DNR Sampling Procedure Organizational Chart

These procedures are designed and intended for the assessment of **wadeable** streams only. The monitoring protocols used in Georgia consist of three basic components: **physical characterization**, **water quality**, and a **biosurvey of the aquatic benthic macroinvertebrates**. The physical characterization evaluates habitat quality using key structural parameters (habitat assessment, pebble count, cross section, and flow) and can be used in conjunction with water quality data to determine potential sources and/or probable causes of impairment. If a degraded community is discovered from the results of the biosurvey, physical information will aid interpretation of effects relative to the biotic potential of a site. Conducting a pebble count will establish a representation of a stream's substrate composition at the time of sampling and can be used during future studies to detect changes related to sediment loading. If impairment is detected, the investigator provides an estimation of the probable cause and source(s) on the Physical Characterization and Water Quality Field Data Sheet (see p. 2A-12). For the order of operations on field procedures, please refer to flow chart on p. 2-1 (Figure 2-1).

2.1 Physical Characterization

2.1.1 Reach Delineation

The sampling location on a stream should be a minimum of 100 meters upstream from a road or bridge crossing. Being upstream at least 100 meters minimizes the effect on stream velocity, depth, and overall habitat quality. The sampling reach should be representative of the characteristics of the stream. No major tributaries should be discharging within the sampling reach. When selecting the 100 meter reach, every effort should be made to avoid sampling in areas where beaver dams are present. The presence of beaver dams may result in inaccurate data.

Once a location has been selected, the first step of the GA DNR Macroinvertebrate Bioassessment Protocol is to establish a reach of 100 meters. This can be done either during site reconnaissance or before any sampling has been conducted.

Equipment/Materials:

- Meter tape (either 100 m or 50 m)
- Flagging tape or Flags
- Flagging stakes

Procedure:

1.) The lower portion of the selected reach (the downstream end) is the zero mark. Flag off this location with brightly colored flags or flagging tape (Flagging tape can be tied around trees, bushes, or other overhanging material. Just make sure it is visible while sampling in the stream.). If the Waterbody Reconnaissance Report (see p. 2A-2) has not been completed during a previous visit, do so now.

2.) Either tie off the meter tape or have another assessor hold the meter tape as you walk along banks to measure the reach. Make sure not to walk in sampling reach prior to sampling. Closely follow the channel (In **rare** cases vegetation may prevent field assessors from walking along the bank, in these cases use great caution not to disturb the sampling reach as you walk along the waters edge.). If the reach is being delineated at least three weeks before the sample date then the person marking off the reach can walk up the stream channel. In cases of high sinuosity, the reach should follow the sinuous channel.

3.) Mark the 50m mark either with bright colored flags or flagging tape. Flagging tape can be tied around trees, bushes, or other overhanging material. Just make sure it is visible while sampling in the stream.

4.) Continue to walk along the bank to measure the reach. Depending on the length of your meter tape, you may need to tie off again at the fifty or have another field assessor hold the tape at the 50-meter mark.

5.) Mark the 100m mark with either brightly colored flags or flagging tape (This will be your upper portion of the reach, the upstream end.). Flagging tape can be tied around trees, bushes, or other overhanging material.

6.) Walk back with the meter tape in hand to the zero meter mark. Making sure not to walk in the sampling reach.

2.1.2 GPS Coordinates

Equipment/Materials:

- Pebble Count Field Sheet (p. 2A-26)
- GPS unit
- Pencils
- Clipboard

After the reach has been delineated, the GPS coordinates at the start of the reach should be recorded. The latitude and longitude should be recorded in both decimal degrees and degrees, minutes, seconds. This eliminates possible difficulties with coordinate conversions. Make sure to legibly record the coordinates on all associated field sheets After sampling the site, make sure all field sheets are completed with the correct latitude and longitude, making sure not to transpose numbers. Try to take the coordinate readings at the zero meter mark on the bank (near water's edge). If you receive a message of poor satellite reception, move further up the bank to take readings. If the unit still indicates poor satellite reception, move to the top of the bank or a nearby clearing in close proximity to the zero mark. On the Pebble Count Field Sheet (see p. 2A-26) record the coordinate location description. For example, in most cases the reading will be taken at the zero meter mark. Note from which bank the coordinates were taken (left bank or right bank). Record the direction and distance of the location in the coordinate location

description (when not at the zero meter mark). For some units, you may be able to record the Position Dilution of Precision (PDOP) and differential correction. Most readings will have an error of at least ± 10-15 feet. Record any error given by the unit.

2.1.3 Visual-Based Physical Habitat Assessment

The habitat assessment process involves rating the ten parameters as Optimal, Suboptimal, Marginal, or Poor based on the criteria included on the Habitat Assessment Field Data Sheets (see p. 2A-4). The optimal category describes criteria for each parameter that meets natural expectations. The suboptimal category includes criteria that is less than desirable, but satisfies expectations in most areas. The marginal category includes judgement criteria describing moderate levels of degradation with severity at frequent intervals in the area. The final category, poor, encompasses criteria for streams having been substantially altered with severe degradation characteristics.

Equipment/Materials:

- Habitat Assessment Field Data Sheet (High Gradient or Low Gradient) (p. 2A-4)
- Habitat Assessment Average (High Gradient or Low Gradient) (p. 2A-8)
- Supplemental Information (High Gradient or Low Gradient) (Appendix 2B)
- Pencils
- Clipboards

(The following procedure was developed by Columbus State University (2000) and part of this method was taken from Barbour *et al.* (1999).)

The habitat assessment is conducted after the reach has been delineated and the GPS coordinates taken. For Quality Assurance/Quality Control (QA/QC) purposes, each biologist individually performs the assessment and the results are recorded on the Habitat Assessment form (see p. 2A-4). The habitat assessment should be conducted by two to three investigators (preferably three). This will help reduce the subjectivity of the assessment. The appropriate Habitat Assessment field sheet should be filled out separately by each investigator. The investigator completing the physical characterization / water quality field sheet can complete the habitat assessment at the same time. The habitat assessment is conducted by walking the entire 100 meter reach prior to sampling. (Walk along the banks when conducting the assessment so as not to disturb the habitat.) If the total habitat scores deviate 30 or more points from one another, then the biologists should take the time to discuss their individual parameter scores while at the sampling location. If three or more people are conducting the assessment and one of the total habitat scores deviates 30 or more points from the other assessors' scores, then the team leader has the option to discard the outlier before calculating the average total habitat score. (Retain all original data sheets for each site.)

All personnel conducting the habitat assessments will be trained in a consistent manner to ensure both standardization and proper performance of the evaluations. Field validation,

conducted at least once per year, will involve comparison of independent habitat evaluations by all investigators of a particular field site.

Streams are assessed for habitat based on high (riffle/run) or low (glide/pool) gradients. The choice of using the high or low gradient field data sheet will depend on: (1) Location of sample site within the state; (2) The presence or absence of riffles; and, (3) Best professional judgment. During drought conditions or low flow conditions, high or low gradient is based on whether there should or should not be riffles. One way to determine this is, in high gradient streams, there may be areas where small gravel and cobble is in the stream, indicating it was once a riffle at higher flow. High gradient (riffle/run prevalent) streams are those in moderate to high gradient landscapes that sustain water velocities of approximately 1.0 ft/sec or greater. These streams have substrates primarily composed of coarse sediment particles (*i.e.* gravel or larger) or frequent coarse particulate aggregations along the stream reaches. Low gradient (glide/pool prevalent) streams are those in low to moderate gradient landscapes that have water velocities rarely greater than 1.0 ft/sec, except during storm events. These streams have substrates of fine sediment or infrequent aggregations of coarser (*i.e.* gravel or larger) sediment particles along the stream reaches (Barbour and Stribling 1995). Generally the streams north of the fall line are high gradient and those below the fall line are low gradient. However, there are always exceptions to the rule. Areas near and around the fall line are examples of where a low gradient stream may occur above the fall line or a high gradient stream may occur below the fall line. Thus, it is important to determine whether the stream has riffles, or should have riffles under normal flow conditions.

The habitat parameters evaluated are related to overall aquatic life use and are a potential source of limitation for the aquatic biota (Plafkin et al., 1989). The physical parameters of the habitat assessment are broken into primary, secondary, and tertiary categories. Primary parameters describe those instream physical characteristics that directly affect the biological community. Primary conditions include epifaunal substrate/available cover, embeddedness, velocity/depth regime, and pool substrate/variability. Secondary parameters include channel alteration, sediment deposition, channel flow status, frequency of riffles, and channel sinuosity. These parameters relate to channel morphology, which controls the behavior of stream flow and the sediment deposits, which the stream collects. The tertiary parameters in the habitat assessment matrix pertain to the riparian vegetation and stream bank structure. These include bank stability, bank vegetative protection, and the riparian vegetative zone. The stability of a streambank indirectly affects the type of habitat available within a stream. Vegetated banks reduce the amount of sediment that washes from the streambank by absorbing energy from the raindrops, binding soil particles, and reducing the velocity of runoff water. Less sediment to cover rocks and logs results in more habitats available for colonization by invertebrates. All parameters are usually evaluated over the designated area (100 meters) of stream (Plafkin et al. 1989; Ball 1982; Platts et al. 1983).

Make sure to completely fill out the Habitat Assessment form (see both high and low gradient forms (see p. 2A-4 to 2A-7) and the average Habitat Assessment form (see p. 2A-8 to 2A-10)) and use the correct form based on stream type. Remember to

determine left bank and right bank by looking downstream. Supplemental information has been provided (see p. 2B-1 to 2B-20), which will help with the breakdown of the point scale for individual parameters. New investigators should use this information in conjunction with the Habitat Assessment form if questions arise at a site.

2.1.4 Physical Characteristics/Water Quality Field Data Sheet

The Physical Characteristics and Water Quality Field Data Sheet (see p. 2A-12) is completed after the reach has been delinated and the GPS coordinates recorded. The Physcial Characteristics and Water Quality Field Data Sheet can be completed by one person at the same time the habitat assesment is being conducted. Modifications to the sheet may be necessary once sampling is complete. For example, when accessing the stream from the bank, the invesigator may not have noticed an oil slick. Thus, this would need to be recorded. All personnel present during sampling should be consulted for additional observations that may have been overlooked by the principal investigator. Make sure to fill out the Physical Characteristics and Water Quality Field Data Sheet completely before leaving the sampling location.

Equipment/Materials :

- Physical Characteristics/Water Quality Field Data Sheet (p. 2A-12)
- Pencils
- Clipboard

Procedures:

(The following procedure was developed by Columbus State University (2000) and Barbour *et al.* (1999).)

1. **Site Location/Map:** The first step for assessing the physical parameters is to draw a map of the stream reach. Include items such as large trees on each bank, large snags, undercut banks, other available habitats and features that are deemed important by the investigator. The hand-drawn map is useful to illustrate major landmarks, channel morphology, vegetative zones, buildings, and other features that might be used to aid in data assessment.

2. The next step is to fill in the rest of the field sheet completely for all parameters. The following are general comments for each of the field parameters that should be measured/observed and documented on the Physical Characteristics and Water Quality Field Data Sheet:

a. Stream Characterization:

i. **Subsystem Classification:** The stream subsystem is marked to indicate whether the stream is perennial or intermittent in nature and if the stream is tidally influenced.

ii. **Stream Type:** Coldwater and warmwater streams are different. Many states have established temperature criteria that differentiate these two stream types. Clearwater and Blackwater sites may require separate biocriteria in Georgia. Thus, it is important to determine and mark which stream type is present.

iii. **Stream Origin:** Note the origination of the stream under study, if it is known. Examples are glacial, montane, swamp, bogs, and spring fed. As the size of the stream or river increases, a mixture of origins of tributaries is likely.

b. Weather Conditions: Note the present weather conditions on the day of the survey and those immediately preceding the day of the survey. This information is important to interpret the effects of a storm event on the sampling effort.

c. Riparian Zone/Instream Features:

i. **Watershed Features:**

1. **Predominant Surrounding Land Use**: Observe the predominant land use type in the vicinity. Document the predominant land-use type in the catchment. Note any other land uses that may not be predominant but may potentially affect the water quality. Land use maps can be used to determine the percentage of each type of land use. This information may be recorded in the database (can use GIS to determine this information). On the field sheet, if livestock are present, note the type of livestock, accessibility to the stream, and also note any crops planted (if determinable).

2. **Local Watershed Nonpoint Source Pollution:** This refers to the problems and potential problems existing in the watershed. Nonpoint source pollution is defined as diffuse agricultural and urban runoff. Indicate if there is no evidence, some evidence, or obvious sources of nonpoint pollution on the field sheet.

a. Indicate in the notes section on the field data sheet any instream impact that is visible. For example: Agricultural runoff, Permitted or Illegal discharge, Runoff from road crossing, Fjord in stream, Storm water, Landfill, Stream alteration, Construction, Feedlots, Constructed wetlands, Septic Systems, Dams & Impoundments, Mine seepage, and Logging. Indicate if the possible impairment results from recent or previous activity.

3. **Canopy Cover:** Estimate the percentage to which the stream is shaded by overhanging branches. Look straight up, and then scan back and forth to determine how much of the sky is blocked by trees. An exposed stream may experience increased water temperature that may be limiting to some organisms and may be favorable to nuisance algal blooms, which along with the elevated temperature can cause a decreased dissolved oxygen level. A fully shaded stream can inhibit the growth and reproduction of herbaceous aquatic and riparian plants, inhibit primary production, and increase available habitat. (A densiometer may be used in place of visual estimation.)

4. **Local Watershed Erosion:** The existence of, or potential for, detachment of soil within the local watershed (the portion of the watershed or catchment that directly affects the stream reach) and its movement into the stream should be noted. Erosion can be rated through visual observation of watershed and stream characteristics (turbidity noted in water quality section). Indicate whether there is no erosion, moderate erosion, or heavy erosion.

ii. **Instream Features:** Instream features are measured or evaluated in the sampling reach and catchment as appropriate.

1. ***High Water Mark:*** Estimate the vertical distance from the bankfull margin of the stream bank to the peak overflow level, as indicated by debris hanging in riparian or floodplain vegetation, and deposition of silt or soil. In instances where bank overflow is rare, a high water mark may not be evident.

2. ***Estimated stream width:*** In order to determine stream width, estimate the distance (or measure) from bank to bank at different locations of the stream. Estimate an

average stream width, which is representative of the reach, and record measurement in meters.

3. ***Estimated Stream Depth:*** Estimate the vertical distance from the top of the water surface to the stream bottom at a representative depth in runs, pools, and riffles (when present).

4. ***Portion of reach Represented by Stream Morphological Types:*** The proportion represented by riffles, runs, and pools should be noted to describe the morphological heterogeneity of the reach.

5. ***Mean Velocity:*** Record the average of stream velocity in a representative run area. If equipment for measuring velocity is not available, use the float method. Stream velocity can have a direct influence on the health, variety, and abundance of aquatic communities. The velocity of water flowing through the system influences the amount of dissolved oxygen in the water and the movement of materials and food particles through the system. High water velocities may result in scour and drift, reducing both the abundance and types of macroinvertebrates present. Low water velocities may result in increased sediment embeddedness caused by silt deposition. The presence of dams, channelization, terrain, runoff, and other factors can affect velocity. (For float method refer to Velocity/Discharge Procedures p. 2-14)

6. ***Reach Length:*** For the GA DNR Macroinvertebrate Bioassessment Program the reach length will be 100 meters.

7. ***Channelized***: Indicate whether or not the area around the sampling station is channelized. Natural, sinuous channelization caused by normal hydrological forces is not considered here. Look for spoil banks, straightening of stream, channel cross section appears box-cut, presence of riprap or other artificial stabilization, and diversions.

8. ***Dredging:*** Indicate if the stream channel has been dredged.

9. ***Dam Present:*** Indicate the presence of a dam upstream or downstream of the sampling area that may alter the flow regime, drift, or movement of biota.

10. ***Beaver Activity Based on Observations:*** Observe the watershed and indicate whether there is active beaver activity, previous activity or no activity. However, **when selecting the 100 meter reach, every effort should be made to sample in areas without beaver dams**. Beaver dams may result in inaccurate data being collected.

11. ***Livestock Damage Based on Observations:*** Observe the watershed and indicate if there has been livestock damage to the stability of the bank. Estimate the amount of observed damage to banks from livestock entering/exiting the channel within the stream reach. Indicate whether there has been no damage; the bank is stable, moderate damage, high damage, or severe damage due to livestock only.

d. Riparian Vegetation: An acceptable riparian zone includes a buffer strip which extends a minimum of 18 m (Barton et al. 1985) from the stream on either side. The acceptable width of the riparian zone may also be variable depending on the size of the stream. Streams over 4 meters in width may require larger riparian zones. Document the vegetation within the riparian zone for dominate type and species, if known. Also indicate whether brush, forest, grass, or exposed represent the riparian zone best. (Some of the common trees found in Georgia include river birch, laurel oak, southern red oak, post oak, willow, red maple, sycamore, locust, hickory, red cedar, cypress, sweet gum, and slash, loblolly, and longleaf pine.)

e. Aquatic Vegetation: Document the general type and relative dominance of aquatic plants. List the species, if known. An estimation of the extent of aquatic vegetation is made. Aquatic vegetation provides refugia and food for aquatic fauna. It is also an ecological assemblage that responds to perturbation.

f. Sediment/Substrate:

i. ***Sediment Odors:*** Disturb the sediment in a pool or other depositional area and note any odors described (or include any other odors not listed) which are associated with sediment odors observed in the sampling area.

ii. ***Sediment Oils:*** Note the term which best describes the relative amount of any sediment oils observed in the sampling areas.

iii. ***Sediment Deposits:*** Note the deposits described (or include any other deposits not listed) which are present in the sampling area. Also indicate whether the undersides of rocks not deeply

embedded are black (which generally indicates low dissolved oxygen or anaerobic conditions).

iv. ***Organic Substrate Components:*** Indicate relative abundance of the three substrate types listed. **This may not add up to 100%**.

v. ***Inorganic Substrate Components:*** Visually estimate the relative proportion of each of the seven substrate/particle types listed that are present over the sampling reach. **This should add up to 100%**.

g. Water Quality:

i. ***Water Odors:*** Note those odors described (or include any other odors not listed) that are associated with the water in the sampling reach.

ii. ***Water Color:*** Circle the color described (or include any other color not listed) that is associated with the water in the surrounding area. Planktonic algae, suspended solids, dyes, chemical discharges, etc., may cause color variations.

iii. ***Water Surface Oils:*** Note whether any oils are present on the water surface. Evaluate the water surface for oils before disturbing the sediment. If possible, indicate whether oils are a result of bacterial growth, petroleum, etc.

iv. ***Water Clarity (Turbidity):*** Note the term, which, based upon visual observation, best describes the amount of material suspended in the water column. Turbidity is defined as a cloudy condition in water, due to the suspension of silt or finely divided organic matter, that affects light penetration and the productivity of algae and aquatic plants. The settling of solids alters the nature of the substrate.

v. ***Water Chemistry:*** For *in-situ* and grab methods chemical data see water chemistry procedures (p. 2-18).

2.1.5 Stream Cross-Sectional Measurement Procedure

The cross-section is conducted after the water samples have been collected. With a three to four member team, this can be sampled at the same time the benthic macroinvertebrates are being collected, thus being very careful not to disturb the habitat. If using a two-member team, this would be conducted post macroinvertebrate sampling. The section of the stream to be measured should be as uniform as is available within the reach. The location should be as unobstructed as possible and be as proximal to the 50-meter mark of the sampled reach as is feasible within the reach.

Note: If discharge measurements are to be conducted, take this into account when selecting the cross section location as both can be performed at the same location.

Equipment/Materials:

- Stakes (rebar)
- 4 lb. sledge hammer (or regular hammer)
- 100 meter tape
- Meter stick or Surveying Rod (m)
- Line level
- Compass
- Camera
- Pencils
- Channel Cross-Section Field Sheet (p. 2A-22)
- Clips to hold tape measure
- Clipboard

Procedures:

(Columbus State University developed this protocol (2000) and used the USDA RM-245 method.)

1. Determine if the 50 m mark in the reach is a representative area for the cross sectional measurements. If not, determine a representative area that is near the 50 meter mark. Make sure to record the location on the Channel Cross-Section Field sheet (p. 2A-22).

2. Establish monuments for measurement points (using stakes). First, determine the lowest bank in the reach. On that side of the stream, drive the re-bar stake; this will be point A (Figure 2-2). Record the height of the stake (point A) above the surface of the ground; record this under "pin height" on Channel Cross-Section Field sheet (p. 2A-22). Gently brush away any leaves or other organic matter around the stake (monument) before taking this measurement. [The stake on the lower bank can also be used as a permanent monument if multiple samples will be taken at this location. Drive the re-bar stake in the top edge of the stream bank leaving approximately 3-6 centimeters exposed. Spray paint exposed end of stake with a bright color (*i.e.* hunter orange) and place survey cap on the top of monument. This will allow the cross-section to be established easier and will allow comparability of the data over time.]

3. Secure one end of your 100 meter tape to the rebar and move it across the stream to the opposite bank, the highest bank, (this will be point B), secure to another stake (This can also be done with a nylon line or camline). This line (tag line) is the fixed point in the landscape to which you will relate your stream cross-section.

Clip the line level to the meter tape to make sure the line is level. **It is critical that the tag line be as level as possible!**

4. Establish your tag line so that it is perpendicular to the streamflow.

5. Establish at least one benchmark, but two or three is preferable. The benchmark is something prominent and distinctive in the area of each cross sectional site to use to find the site at a later date (*i.e.* manhole cover, boulder, tree, building). Note the **direction and distance** to Point A on the Channel Cross-Section Field sheet.

6. Record the distance from Point A – (the lower bank rebar) to Point B – (the higher bank rebar). Also record the width of the active stream channel.

7. Use the compass to measure direction from Point A to Point B. Record on field sheet. Be as accurate as possible. (Record left to right or right to left bank; look downstream to establish left or right bank.)

8. Once the tag line has been established and located, divide the active channel width (Point A to Point B) by 10 to determine the minimum number of intervals for measurements (should be rounded to nearest centimeter). Enough measurements should be made so that the topographic heterogeneity of the channel is captured. To capture the topographic heterogeneity of the channel, the intervals may need to be divided up further. At least ten measurements **must** be recorded. Use a meter stick or surveying rod held against tag line for vertical distance measurements. Measure from top of tag line to water surface for elevation and then from water surface to substrate for depth. Record all measurements on field sheet. Record the horizontal distance from the monument (using the meter tape).

9. Record (in remarks section) the locations of various depositional features or other channel characteristics as measurements are taken across the stream. For example LB=Left Bank, WDJ=Woody Debris Jam, etc. (See field sheet for more examples, p. 2A-23.) Make sure to fill in a remark for each measurement. Thus, in some cases, water may be recorded.

10. After all measurements have been recorded, pictures should be taken. If possible, take the picture with the tag line visible so it can be used as a reference in the picture. Take photos of the cross section setup from the following vantage points: facing upstream, facing downstream, facing the left bank, and facing the right bank. Photos should also be taken of the benchmarks to allow for ease of locating the cross section point on return possible visits. More photos can be taken if the investigator determines it may be of use. (For example: eroded banks, log jams, sedimentation, etc.) Record the picture numbers on the Channel Cross-Section field sheet in the appropriate location. (Left bank and right bank is determined by looking downstream.)

Return Trips:

11. During subsequent site visits and upon arrival at a previous cross-sectional site, locate benchmark(s) and consult field sheets for location of site from benchmark(s).

12. Perform horizontal and vertical measurements as given in Figure 2-2.

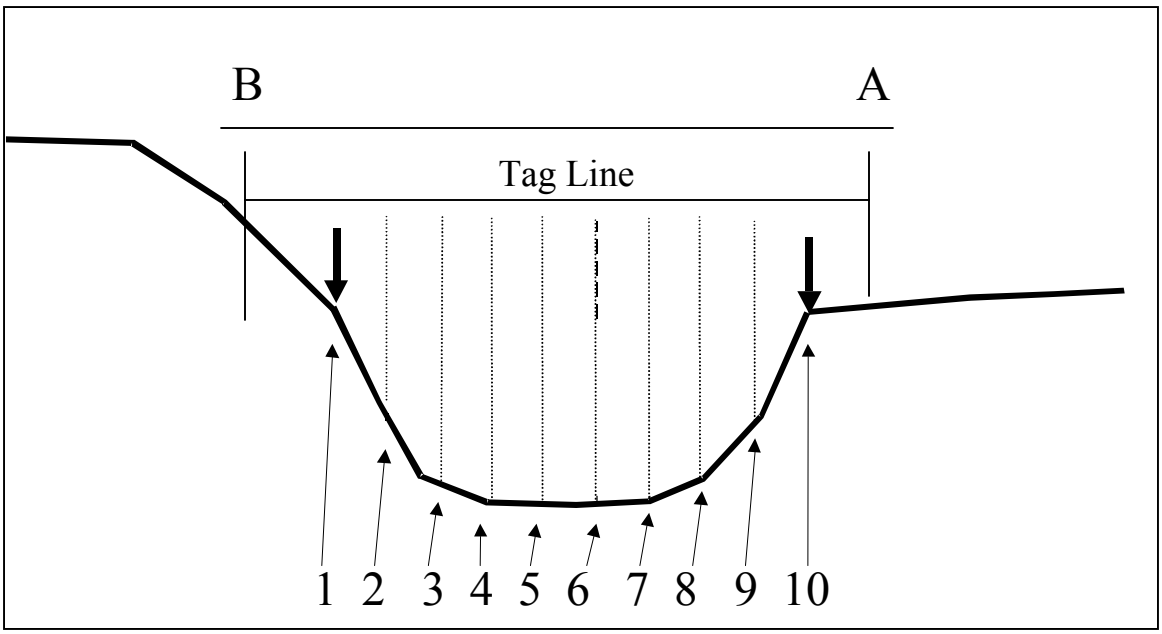

Figure 2-2: Stream Channel Cross Section

2.1.6 Velocity/Discharge Measurements

This measurement can be taken with a flow meter or by using the float method. The flow meter used should conform to USGS/ISO methods for wading discharge measurements. Follow the manufacturers instruction manual for flow meter use. (Use of a flow meter for this measurement is preferable, and is required, if loading analysis is considered a possibility)

Equipment/Materials:

- Flow meter
- Wading Rod

OR

- Tennis ball with holes or orange
- Stopwatch

AND

- Pencils

- Velocity/Discharge Field Sheet (p. 2A-24)
- Meter stick
- 100 meter tape
- Clipboard

Procedures:

2.1.6a Flow Meter

Discharge readings should be conducted at the same location and time as the cross section and using the same tape measure to determine the location of each measurement station. The section of the stream to be measured should have as uniform of a profile as possible with minimal solid obstructions (*i.e.* rocks, woody debris, vegetation, etc.), as these objects will interfere with the velocity readings. The location should be as representative as possible to the rest of the stream and be as close to the 50-meter mark of the sampled reach as possible. The cross section should be divided into **at least** 12 sections. This would result in at least 14 measurements (including the starting-edge and ending-edge measurements); the 12 in-stream measurements should be made at the section division points. For example: If the stream has a width of 8'(96"), with a minimum of 12 measurable sections, each section would be 8" wide (96/12). The first **in-stream** measurement should be at the second edge of the first section (the first edge being the starting-edge measurement); this would put it at 8" from the starting-edge measurement. Every subsequent measurement (8" apart) would be at the edge of the remaining segments. Velocity/discharge readings should be conducted after the chemical data samples have been collected. With a three to four member team, this can be conducted at the same time the benthic macroinvertebrates are being collected, being careful to disturb the habitat as little as possible.

2.1.6b Float Method

To perform the float method, use an orange, a tennis ball with holes in it, or other material that sinks at least halfway into the water. Measure and mark a two meter section of the stream. Two observers will provide the best results. One places the float into the channel upstream of the two points and calls out when it crosses the upstream point. The downstream observer starts the timer. When the float passes the lower point, the timer is stopped and the time is recorded. Repeat the procedure six times at different distances from the bank, within the marked two meter stretch, to get a rough average of velocities. Record the average of the six velocity measurements in m/sec for the mean surface velocity measurement on the Physical Characterization and Water Quality field data sheet (p. 2A-12). **Remember:** this measurement can be done at the same location as the cross section.

If the float method is used, discharge (**Q**) can be calculated using a simple formula. The average velocity of the measured section must first be multiplied by 0.85 to get an estimate of the water velocity (V) at 0.6 depth. This number is then multiplied by the total area (A) of the cross section (this can be determined by adding together the areas of

the incremental cross section measurements).

The formula for discharge using the float method is: **Q=AV**

2.1.6c Quality Control/Quality Assurance (QC/QA)

Proper maintenance and diagnostics, according to manufacturer specifications, should be conducted and documented on a regular basis.

2.1.7 Modified Wolman Pebble Count

The pebble count is conducted after the macroinvertebrates are sampled, so as not to disturb the habitat. For field teams of 3 to 4 individuals, the pebble count can be conducted after the cross-section and discharge measurements while the other team members are sampling the macroinvertebrates. The individual(s) will stay behind the macroinvertebrate collection individuals.

Equipment/Materials:

- Clipboard
- Pencils
- Small Calipers (need for all areas)
- Large Calipers (need for North GA Streams)
- Pebble Count Field Sheet (p. 2A-26)
- Meter stick (or other measurement device for measuring large cobble)
- Calculator
- Sand card

Procedures:

This method can be used in conjunction with a Rosgen Level II assessment procedure in the conduct of a watershed-wide assessment, or as a stand-alone method of characterizing particle size distribution in running waters. For GA DNR purposes the pebble count will be a stand-alone method of characterizing particle size distribution in **wadeable** streams.

(This procedure was developed by Columbus State University (2000) and was taken from Harrelson *et al.* (1994) and Rosegn (1996) methods.)

1. The stream or stream reach of interest is located in the field by the crew. After a thorough visual assessment of channel characteristics is made, a representative section, based upon the best professional judgment of the crew leader, is selected for analysis as the assessment reach (AR). This section should most closely characterize the channel condition and form within the reach of interest. Unusual conditions, atypical channel forms (*i.e.* short, transitional areas between longer homogeneous channel reaches) or other disturbances should be avoided unless assessing atypical areas is the purpose of the work.

2. An estimate of the distribution of channel features (*i.e.* riffles, pools, runs, steps,…etc.) within the AR or reach of interest (ROI) is made.

3. A total of 20 zig-zag transects are proportionally distributed through the AR or ROI. Within each zig-zag transect (see Figure 2-4), a total of 5 particles are measured, spanning the width of the wetted channel and point bars. Particles are measured with calipers, or with a sand card, along the intermediate axis as illustrated in Figure 2-3. A hash mark is made on the Pebble Count field sheet in the appropriate size class. A total of 100 particles are counted throughout the AR or ROI. The angle of the zig-zag depends upon the meander pattern of the stream reach being sampled. Stream reaches that exhibit little or no sinuosity do not require as sharp of an angle as highly sinuous reaches.

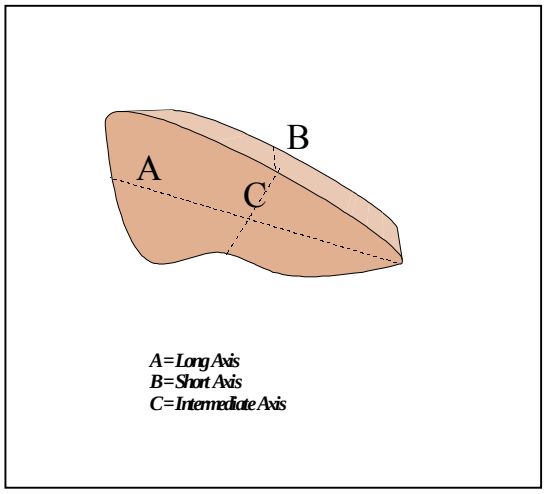

Figure 2-3 Particle Schematic

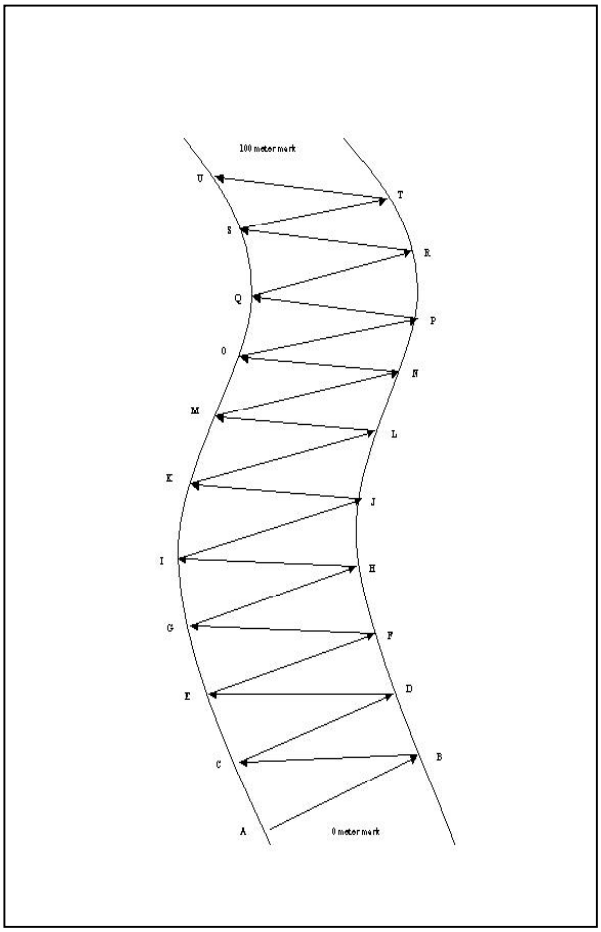

Figure 2-4 Pebble Count Transects

4. Particles are selected at equally spaced intervals across the transects. Thus, if the transect is 10 feet wide, then a particle is selected for analysis at two feet intervals. This can be estimated by eye or a tape can be drawn across the channel to guide particle selection.

   In order to avoid bias when selecting a particle, the operator does not look down. Looking straight ahead, reach down with the index finger extended. Pick up the first particle encountered by that leading finger and measure as described in step 4.

   Measure embedded particles, or those too large to be moved, in place. During these situations, measure the smaller of the two exposed axes.

   Make sure to place a hash mark on the pebble sheet for each measurement. If a second individual is recording the data, the pebble count should be repeated to the person that is measuring for accuracy.

5. The total number of particles should be checked on the pebble count field sheet, before leaving the field. Add up the total number of each particle. A total of 100 particles must be counted for the entire reach.

6. Make sure the Pebble Count field sheet is completely filled out. Calculate the % cumulative numbers for each of the major particle categories: Silt/Clay, Sand, Gravel, Cobble, Boulder, and Bedrock. (For example, you counted 100 particles and 20 of the particles were in the gravel category (20/100 X 100 = 20%), thus 20 percent of the particles are gravel.)

2.2 Water Quality Chemistry Protocol

Water samples can be collected either after the habitat assessment and physical characterization portions of the assessment are finished or while the reach is being delineated if enough people are on hand. (If the reach is being delineated at the time of sampling it must be done along the bank in a manner that will least disturb the sample area.) The important point is: **Water chemistry samples should be collected before anyone in the group has disturbed the stream in the intended reach**. The samples should be taken in mid-channel at the zero mark, being careful not to disturb the downstream side. Chemical sampling is collected prior to making any other measurements. The grab sample should be collected in an area with cross-sectional homogeneity where the water is well mixed. Since turbulence and water velocity principally govern mixing, the selection of a site immediately downstream of a riffle area (in high gradient streams) will ensure good vertical mixing. Horizontal mixing occurs in constrictions in the channel. Collect the grab sample first. Prior to collecting the grab sample, put the guard on the water quality multiprobe unit and let it stabilize to take a reading of the air temperature. After you have collected all your grab samples record the air temperature and place the water quality multiprobe unit into the stream and wait for it to stabilize before taking readings. If the multiprobe is equipped with a circulator ensure

that it is functioning and on at this time. While the water quality multiprobe unit is stabilizing, the *in-situ* turbidity can be measured. (See p. 2-19 to 2-23 for both *grab* and in-situ sampling methods.)

The parameters that will be measured are total kjeldahl nitrogen (TKN), ammonia (NH_3), nitrate-nitrite (NO_2-NO_3), total phosphorus, ortho-phosphate, total organic carbon (TOC), total alkalinity, hardness, suspended solids, Clean Metals (ICP/MS), water temperature (ºC), depth (m), specific conductance (µmhos/cm), salinity, dissolved oxygen (mg/L), dissolved oxygen (%), pH, and turbidity (NTU). The following sampling methods are examples that can be used. However, any EPA/EPD method can be used. Other chemical parameters are required for Watershed Assessments (WSA) and Watershed Protection Plans (WPP). Refer to the WSA and WPP guidelines at www.gaepd.org for more information. Refer to **Table 2-1** for bottle size, preservative, and holding time for parameters. The grab method procedure and clean hand technique (to be used for collecting Clean metals) are given in the following pages.

Table 2-1: DNR Macroinvertebrate Bioassessment Chemical Parameters

Parameters	*Bottle	Preservative	Holding Time
Total Suspended Solids	Half-Gallon	No preservative	7 days
Alkalinity			14 days
Clean metals (ICP/MS)	500 mL plastic	HNO3, <2 pH	6 months
Total Kjeldahl Nitrogen (TKN)	250 mL plastic	H2SO4, pH <2	28 days
Ammonia (NH3)			
Nitrate-Nitrite (NO2-NO3)			
Total Organic Carbon (TOC)			
Ortho-phosphate	250 mL plastic	H2SO4, pH <2	28 days
Total Phosphorus			

***Bottle size should be determined by the lab conducting the tests**

2.2.1 Water Quality Chemistry Collection Protocol – "Grab Method"

This procedure is to be used for collection of water samples for nutrient analyses from small shallow streams of 20 ft width or less. This is a two-person procedure and as such should not be modified to accommodate collection by a single sampler. Depth and velocity of water should be carefully considered in determining whether the sample can be safely taken by wading to mid-channel. Procedure as dictated below may be modified somewhat as circumstances dictate. However, sample hands person must not contaminate their hands by contact with other surfaces and must be the only person to touch the sample bottle.

Equipment/Materials:

- Appropriate size, clean polyethylene sample bottle filled with appropriate, pre-measured preservative solutions for parameters to be measured (nitric acid and sulfuric acid) **Gloves and safety goggles must be worn when adding preservative to samples.**
- Gloves (clean, non-talc polyethylene, latex, vinyl, or PVC; various lengths and shoulder-length)
- Storage bags (clean, zip-type, non-vented, colorless polyethylene [various sizes])
- Cooler (clean, nonmetallic, with white interior for shipping sample)
- Ice or chemical refrigerant packs (to keep samples chilled in the cooler during transport and/or shipping)
- Printed labels
- Pencils
- Clipboards
- *In-situ* and Grab Sample Water Chemistry Field Sheet (p. 2A-14)
- Water Chemistry Analysis Chain of Custody (p. 2A-28)

Procedures:

This procedure was taken from EPA method #1669 (EPA July 1996 & EPA June 1999).

1. All sample bottles should be double bagged in plastic zip lock bags and stored in the cooler chest. The chemical samples are taken at the downstream end of the reach to be sampled (0 meter mark), prior to making any other measurements (biological sampling, morphological or habitat measurements). To minimize contamination from trace metals in the atmosphere, and other potential sources of contamination, water samples should be collected from sites that are as far as possible (e.g., at least several hundred feet) from any metal supports, bridges, wires, poles, or heavily traveled roads. Ideally this would be at the start of the reach, which should be at least 100 meters upstream of any bridge or road crossing.

2. The team should approach the site from down current and downwind to prevent contamination of the sample. One team member will be designated “sample hands” and will handle the sample container, and not touch anything else. The other team member will be designated “bag hands” and will touch everything else (but not the sample container) to prevent contamination of the sample.

3. At the site, all sampling personnel must put on gloves before sampling, with "sample hands" donning shoulder-length gloves. Make sure to rinse the gloves in the stream before collecting the sample. "Bag hands" must unzip the bags. Next, "sample hands" removes the bottle. "Bag hands" then rolls top of bags and holds. “Sample hands” moves to mid channel of stream and facing upstream submerges the bottle with the cap on. Samples…or the sample should be taken well below the surface to eliminate chance of collecting surface film. “Sample hands” removes top from bottle under water allowing it to fill then, while the bottle is still inverted so

that the mouth of the bottle is underwater, "sample hands" replaces the cap of the bottle. Once the bottle lid has been replaced, "bag hands" unrolls the plastic bags, and "sample hands" opens the inside bag, places the bottle inside it. "Bag hands" zips the plastic bags. This procedure is then repeated for each of the samples. Metal samples are collected with new clean gloves each time a sample is taken.

4. Documentation: The sample label is completed after each sample is collected. This includes any comments, and anything unusual observed, concerning both the sample and the sampling process. The Chain of Custody Record will also be completed, annotating the time of collection, and identification number of each sample. Record the sample identification number (same as site number) and number of bottles collected on the *In-situ* and Grab Sample Water Chemistry Field Sheet.

5. After all bottles have appropriate labels affixed (and preservative added if needed), and are re-bagged, samples are placed back in the cooler chest on ice. **Gloves and safety goggles must be worn when adding preservative to samples.**

2.2.2 Water Chemistry Collection Protocol – "Clean Hands" Method for Metals

This procedure is to be used for collection of water samples for low level metals analyses from small shallow streams of 20 ft width or less. This is a two-person procedure and as such should not be modified to accommodate collection by a single sampler. Depth and velocity of water should be carefully considered in determining whether the sample can be safely taken by wading to mid-channel. Procedure as dictated below may be modified somewhat as circumstances dictate. However, clean hands person must not contaminate their gloves by contact with other surfaces and must be the only person to touch the sample bottle.

Equipment/Materials:

- Clean Metals Sample Bottle Kit (certified metals free bottle double bagged with two pair of gloves) -- Storage bags should be: clean, zip-type, non-vented, colorless polyethylene [various sizes] -- Gloves should be: clean, non-talc polyethylene, latex, vinyl, or PVC
- Clean Metals Field Blank
- Cooler (clean, nonmetallic, with white interior for shipping sample
- Ice or chemical refrigerant packs (to keep samples chilled in the cooler during transport and/or shipping)
- Printed labels
- Pencils
- Clipboard
- *In-situ* and Grab Sample Water Chemistry Field Sheet (p. 2A-14)
- Water Chemistry Analysis Chain of Custody (p. 2A-28)

Procedures:

(This procedure was taken from EPA method #1669 (EPA July 1996 & EPA June 1999).

1) Samplers decide who will be the "clean hands" and who will be the "dirty hands".
2) "Clean hands" person opens the outer bag of the metals sampling kit.
3) "Clean hands" reaches in and retrieves one pair of gloves from the outer bag. "Clean hands" should only touch cuff portion of clean hands gloves.
4) "Clean hands" person rolls top of bags to seal them and assist "dirty hands" in putting on first pair of gloves.
5) "Clean Hands" reopens bag and dirty hands (with gloves on) retrieves second pair of gloves from outer bag. "Dirty hands" should only touch cuff portion of clean hands gloves.
6) "Clean hands" rolls top of bag and holds under arm or transfers to "dirty hands" who holds under arm.
7) "Dirty hands" assists clean hands in putting on second pair of gloves. Contact with outside surface of clean hands gloves should be limited to the clean sample bottle.
8) "Dirty hands" then unrolls bottle bag.
9) "Clean Hands" unseals the inner bottle bag and retrieves metals bottle from inner bag with minimal contact with bags.
10) "Clean hands" moves to mid channel of stream and facing upstream submerges metals bottle with cap on. Sample should be taken well below the surface to eliminate chance of collecting surface film.
11) "Clean hands" removes top from bottle allowing it to fill then recaps bottle before bringing it back to surface.
12) "Dirty hands" unrolls bottle bags and "clean hands" places filled sample bottle in inner bag and seals it.
13) "Dirty hands" compresses bags to expel air and closes outer bag.
14) Sample label is prepared prior to sampling and taken to sample location. Sample label is placed in the outer bag. Sample is put on ice.

Notes:

a) Sample contact with air must be kept to a minimum. It is important that bags be sealed or rolled immediately after opening to keep air out. In addition, the sample bottle should not be opened at any time except when submerged in the stream.
b) Clean Hands (wearing gloves) should not touch any surface other than the outside of the sample bottle or the inner bag.
c) A clean metals field blank should be collected at each sampling location.

2.2.2a Quality Control/Quality Assurance (QC/QA)

This procedure was taken from EPA method #1669 (EPA July 1996 & EPA June 1999).

A clean metal field blank will be collected at the first sample site for each trip, if more than ten samples are taken per trip then the first blank will be collected at the first site and then another blank collected at the eleventh site sampled. Field blanks are collected to demonstrate that sample contamination has not occurred during field sampling and sample processing. Field blanks are conducted at the stream bank. The clean hand/dirty hand method is used. A prepared blank will be opened to air for about 15 seconds, closed up, and then placed back into the zip bags. The field blank is then taken back to the laboratory and analyzed with the other samples. To minimize contamination from trace metals in the atmosphere, water samples should be collected from sites that are as far as possible (e.g., at least several hundred feet) from any metal supports, bridges, wires, poles, or heavily traveled roads. Ideally this would be at the start of the reach, which should be at least 100 meters upstream of any bridge or road crossing.

2.2.3 *In-situ* measurements (Multi-probe)

The *in-situ* water quality multiprobe unit can be any brand that conforms to the EPD/EPA methods. The parameters to measure are: water temperature (ºC), depth (m), specific conductance (uhoms/cm), salinity, dissolved oxygen (mg/L), dissolved oxygen (%), and pH. Prior to grab sampling, the probe, with guard, should be laid aside to measure air temperature. You may also use another approved method to collect the air temperature. Record the air temperature on the *In-situ* and Grab Sample Water Chemistry Field Sheet (see p. 2A-14).

Equipment/Materials:

- Water Quality Multiprobe
- Data logger
- Weighted sensor guard
- Cable
- Storage cup
- Calibration cup
- Maintenance kit (spare DO membranes, DO electrolyte, pH reference, and o-ring grease)
- Data logger battery charger
- DI water (deionized)
- pH buffer (4.0, 7.0, or 10.0 depending on sampling sites; only need a two point calibration)
- Kimwipes
- Conductivity solution
- Calibration Sheet (see p. 2A-16)
- Pencils
- *In-situ* and Grab Sample Water Chemistry Field Sheet (see p. 2A-14)

The readings should be taken in mid-channel at the zero mark, being careful to not disturb the downstream side. The unit is then placed in the middle of the channel and left to stabilize. Either walk out of the stream and let the unit stabilize or stand out of the way and be very still. Record this data on the *In-situ* and Grab Sample Water Chemistry Field Sheet. Once the data has been recorded place storage cup on unit and put away to prevent damage to instrument.

2.2.3a Quality Control/Quality Assurance (QC/QA)

The water quality multiprobe units are to be calibrated prior to sampling each day, either the evening before or the morning of sampling. The units should than be calibrated once you have returned to the lab. These calibrations will be your starting and ending value for each sampling day. This is to prevent data from being collected using an erroneous calibration. An erroneous calibration can possibly result in invalidation of collected data. Follow manufacturer guidelines for calibration. For quality control/quality assurance, follow manufacturer maintenance and cleaning procedures periodically.

2.2.4 *In-situ* Turbidity measurements

Turbidity can be measured with any turbidity meter that follows the EPA/EPD guidelines. GA DNR currently is using an *in-situ* method. The turbidity measurement can be taken while the water quality multiprobe unit is stabilizing; however the water sample would need to be taken prior to stabilization of the water quality multiprobe unit. Otherwise the unit would be disturbed and the reading incorrect. The water sample should be collected in a one liter bottle near the zero meter mark. Collect the sample in a representative area in the reach. Mix sample gently, but thoroughly to ensure a representative sample before taking the measurement. Make sure the sample is not allowed to settle. In streams that are not uniform, collect several locations at varying depths and combine the samples into a single, well-mixed composite sample before measurement.

2.2.4a Quality Control/Quality Assurance (QC/QA)

For quality control/quality assurance follow all safety, calibration, measurement techniques, and cleaning techniques in the instruction manual. When measuring very low turbidity, follow procedure for orienting sample cells and dilution water for testing the turbidimeter (see manufacturer instruction manual for details).

2.3 Georgia Macroinvertebrate Collection Protocol (GMCP) for Wadeable Streams

2.3.1 Aquatic Dip Net – 20 Jab Method

After the water chemistry data has been collected, the next step is to collect the benthic macroinvertebrates using a D-frame net. Samples are collected throughout the 100-meter reach. Make sure the sampled habitats are distributed throughout the reach.

Macroinvertebrate collection should be conducted with two team members. One individual will collect the samples using a D-frame net. The other member will keep up with the number of jabs, compiling the material in the sieve bucket, checking large debris for organisms, and elutriating the sieve bucket to reduce the amount of silt in the sample. After sampling is complete, the debris will be placed into plastic sample containers and the Benthic Macroinvertebrate Field Data sheet (see p.2A-20) and the internal and external sample labels must be filled out completely.

Equipment/Materials:

- Standard aquatic dip net, D-frame dip net, 500 µm openings, 0.3 meter width (~1 foot)
- Sieve bucket, with 500 µ opening mesh (with Table 2-2 affixed to the bucket)
- 95 percent ethanol
- Sample containers
- Labels
- Forceps
- Pencils
- Clipboard
- Benthic Macroinvertebrate Field Data Sheet (p. 2A-20)
- Grease pencils

Procedures:

Habitat: riffles, woody debris/snags, undercut banks/rootwads, leafpacks, soft sediment/sandy substrate, and submerged macrophytes (when present)

Area:	20 jabs, each 1-m in length
Mesh size:	500-µm
Index Period:	Fall/Winter (mid September – February)

1. The sample reach should consist of a 100-meter instream segment having **no major tributaries** discharging into the assessment reach. Sampling should be conducted **at least 100-meters upstream of any road or bridge crossing** to minimize the effects on stream velocity, depth, and overall habitat. If the objective is to assess overall watershed conditions, which include bridge/road stressors, sampling should be targeted to a reach at least 100 meters downstream of bridge crossings.

2. Sampling is conducted from downstream to upstream by jabbing the D-frame net into productive and stable habitats at 20 different locations. A single jab consists of forcefully thrusting the net into a productive habitat for a linear distance of 1-meter. A kick consists of "kicking" or disturbing the substrate upstream of a stationary D-frame net for a linear distance of 1-meter.

3. Different types of habitats should be sampled according to the following guidelines. Unique habitat types (*i.e.*, those consisting of less than 5 percent of

stable habitat within the sampling reach) should not be sampled. The following are specific sampling techniques for different productive and stable habitats:

a.) Riffles – Riffle areas are made up of cobble/gravel rock substrates and/or stable woody debris. A riffle area is characterized by three components: (1) a change in elevation, (2) oxygenation of the water, and (3) the riffle area is audible. Six riffle kicks should be collected in areas of different water velocities, *i.e.* three in fast, three in slow. Usually not a common feature of most coastal or low gradient streams. Sample by holding the bottom rim of the dip net against the substrate downstream of the riffle and perpendicular to the flow while disturbing the substrate just upstream of the net with feet and hands to dislodge organisms. If the net moves, it may allow organisms to drift under the net, if this happens the kick should be repeated in a different riffle. Gently rub (with your hands) any loose debris off rocks and sticks so that all organisms are captured. When all rocks have been "washed off", kick the streambed in the area just upstream of the net, this will dislodge any burrowing organisms, or those clinging to the substrate.

b.) Woody debris/Snags – Snags and other woody debris that have been partially or fully submerged and show evidence of decomposition provide excellent colonization habitat. Submerged, woody debris is sampled by jabbing in medium-sized snag material (sticks and branches). Collect samples from different logs and/or stick clumps. The 1-meter section of this habitat is estimated. The snag habitat may be kicked first to help dislodge organisms, but do so only after placing net in water downstream of the snag. Material too large to go into the sample should be rubbed clean into the net in a similar manner as the rocks in a riffle. Accumulated woody material in pool areas can also be considered as snag habitat. Large materials (*e.g.*, logs) are usually avoided since they are not generally productive.

c.) Banks/Rootwads - Collect jabs in different locations exhibiting a variety of bank types, incorporating both left and right banks and areas with different flow regimes. When banks have roots, plants, and snags associated with them, they are sampled in a fashion similar to snags. When the banks consist of unvegetated or soft soil, they are sampled by bumping the net along the substrate rather than dragging the net through soft substrates; this will reduce the amount of detritus (defined as sticks, leaves, and/or pieces of bark) through which you would have to pick. After placing the net downstream, the bank habitat can also be kicked first in order to help dislodge organisms.

d.) Leaf packs - Accumulations of deciduous leaf material on snags, root wads, large substrate particles; *one large handful* of well-conditioned (*i.e.*, partially broken down) leaves should be gathered for each of the three samples; new leaf fall that has not become conditioned should be avoided, if possible.

e.) Soft sediment/sandy substrate – This may be a common stream habitat in watersheds experiencing substantial land use changes, bank instability, and accelerated channel erosion. Using your foot, thoroughly disturb the substrate in an area of approximately 0.3 square meters, and sweep the D-frame net through the suspended materials. For this type of habitat in a strong current, place the net on the bottom (with opening upstream), and disturb the same area upstream of the net, allowing the current to move the materials into the net.

f.) Submerged macrophytes – These are aquatic plants that are rooted on the bottom of the stream. They are sampled in deep water by drawing the net through the vegetation from the bottom to the surface of the water. In shallow water, they are sampled by bumping the net along the bottom in the rooted area. This habitat is generally not present. If present, add a maximum of three jabs to total number (*i.e.*, total number of jabs could potentially be 23).

4. Although sampling techniques remain the same for both low gradient and high gradient streams, the number and kind of samples vary. Table 2-2 on p.2-28 describes the differences. The following explains how to reallocate jabs from missing habitats.

    a) When habitats are not present, jabs will be reallocated using the priority list given in Table 2-2. This allows an equal level of effort, 20 (+3) jabs or kicks, to be obtained at biomonitoring sites. Macrophyte jabs are not included as part of the reallocation of habitat and are not reallocated if the habitat is missing. **Example:** if the riffle habitats are completely absent from the sample reach, then the six jabs would be equally dispersed among the remaining available habitats shown in Table 2-2, starting with woody debris/snags. Each habitat down the list in Table 2-2 would receive an extra jab until the six jabs were reallocated. When all remaining habitats have been increased by one jab, go back to the top of the list and begin distributing the remaining jabs the same way, adding one more to woody debris/snags, one more to undercut bank/root mats, and so on until all jabs have been reallocated.

5. The collected sample is washed by running clean stream water through the net 2-3 times; transfer the sample to the sieve bucket. Samples should be cleaned and transferred to the sieve bucket at least every three to four jabs, more if necessary. Do not let the net become so clogged with debris that it results in the diversion of water around the net rather than through the net. If clogging occurs, discard the sample in the net and recollect that portion of the sample in a different location.

6. As the sample is added to the sieve bucket (500*um* open mesh), it should be further washed to remove fines. All jabs will be composited in the sieve bucket, resulting

in a single, homogenous sample. Mix the sample by hand while sieving, removing large debris from the sample after rinsing and inspecting for organisms. Place any detected organisms back into the sieve bucket. Do not attempt to inspect small debris. Using the grease pencil, mark the habitats sampled on the habitat table (Table 2-2) that is affixed to the sieve bucket, so as to keep up with which habitats have been sampled.

Table 2-2: Prioritized List of Habitat Types for Sampling and Sample Reallocation

HIGH GRADIENT STREAMS		
Priority	**Habitat Type**	**Number of Samples**
1	Fast Riffle	3
2	Slow Riffle	3
3	Woody debris/Snags	5
4	Undercut Banks/Rootwads	3
5	Leaf Packs	3
6	Soft Sediment/Sand	3
	Macrophytes (if any)	3
LOW GRADIENT STREAMS		
Priority	**Habitat Type**	**Number of Samples**
1	Woody debris/Snags	8
2	Undercut Banks/Rootwads	6
3	Leaf Packs	3
4	Soft Sediment/Sand	3
	Macrophytes (if any)	3

7. Transfer the sample from the sieve bucket to the sample container(s) and preserve in 95 percent ethanol. The stream water present in the sample will cut the ethanol to 70-80%. Forceps may be needed to remove organisms from the sieve screen and dip net.

8. The sample containers are labeled with the following information externally and internally: log number/identification number; site designation; stream name; preservative type; reallocation; designation as a quality control site; date of collection; number of bottles; and, collectors' initials. Chain-Of-Custody (C-O-C) forms should include the following information: location; date and time; preservative; site designation; sampling gear; and sampler's name. Proper C-O-C procedures are necessary for tracking sample possession from the field to the lab. Sample containers can either be labeled in the field or at the truck. Containers must be labeled before leaving a sampling location. (See Labels and Chain-of-Custody section for an example, p. 2-29.)

9. Field notes should be taken on the overall habitat condition (in addition to habitat assessment), weather, observations on condition of the macroinvertebrate and fish communities, and other wildlife observed. Notes on the stable habitats sampled should be recorded on the habitat assessment sheets (*i.e.,* the proportion of snags,

macrophytes…etc. sampled; type of substrate; condition of habitats). This information should be recorded on the Benthic Macroinvertebrate field data sheet (p. 2A-20) and stored in the laboratory.

2.4 Labels and Chain-of Custody

After sampling is complete, make sure to pack up all equipment and sample bottles. Once you have returned to your vehicle you will need to fill macroinvertebrate bottles with ethanol, affix labels, and fill out Chain-of-Custody (C-O-C) forms. If your water sample bottles have not been preserved ahead of time, you will need to preserve the samples with the proper acid. The following are examples of labels, C-O-C sheets, and naming methods to be used. Other sheets may be used as long as they contain all pertinent information.

Equipment/Materials:

- Printed Labels
- *In-situ* and Grab Sample Water Chemistry Field Sheet (p. 2A-14)
- Water Chemistry Analysis Chain of Custody (p. 2A-28)
- Macroinvertebrate Chain of Custody (p. 2A-30)
- Water Sample Labels
- Macroinvertebrate Sample Labels
- Pencils
- Clipboard
- 95 percent ethanol

Procedures:

1.) Preserve the macroinvertebrates in 95 percent ethanol, which will be cut to approximately 70%, since you have already added 1/4 - 1/5 volume of stream water.

2.) Once you have preserved your samples, the sample containers are labeled with the following information **externally and internally**: log number/identification number; site designation; stream name; preservative type; reallocation; if designated as a quality control site; date of collection; number of bottles; and, collectors' name.

Example Label:

PROJECT NAME

HERE

Stream Macroinvertebrate Sample

Log # / ID #_____ Site # _____

Stream _____ Preservative *Ethanol*

QC Yes_____No_____ Reallocation Yes____ No____

Collected by _____

Date _____ Time _____ Bottle_____ of_____

Macroinvertebrate samples are assigned a unique inventory number used to identify the sample bottles and are recorded on bench sheets, logbooks, chain-of-custody, QA/QC records, and any other time the sample is documented or discussed.

YYYYMMDDNN

Where:

YYYY is the year
MM is the month
DD is the day of the month
NN is the bottle number for the day (starts with 01 for the first bottle of each day)

For Example:

If you collected 2 bottles from the first stream, 3 bottles from the second stream, and 1 bottle from the third stream sampled on October 01, 2005. The first site's bottle numbers would be 2005100101 and 2005100102; the second site's bottle numbers would be 2005100103, 2005100104, 2005100105; and the third site's bottle number would be 2005100106. By using this system, log numbers (identification numbers) will not be repeated.

3.) The water samples can be preserved either before sampling or after. If you choose to preserve after sampling, then you will need to preserve as soon as you return to your vehicle. Preserve with either sulfuric or nitric acid depending upon which parameters you will be analyzing, refer to EPA/EPD methods for appropriate preservatives. The sample containers are labeled with the following information

externally: log number/site #; stream name; preservative type; parameter that will be tested; if designated as a quality control site; date of collection; number of total bottles for sample location; and, collectors' name.

Example Label:

PROJECT NAME

HERE

Laboratory Name: Water Chemistry Sample

CLEAN METALS: Preservative: HNO_3, <2 pH

Stream Name _____

Site # / Log # _____ QC: Yes___ No____

Collected by _____

Date _____ Time _____ Bottle ____ of____

4.) Chain-Of-Custody (C-O-C) forms should include the following information: location; date; time; preservative; site designation; sampling gear used; and, sampler's name. Proper C-O-C procedures are necessary for tracking sample possession from the field to the lab. Sample containers can either be labeled in the field or at the truck. Containers must be labeled before leaving a sampling location. The chain-of-custody forms need to be filled out before leaving the sampling location.

a. For the water chemistry samples, you will have two forms for Chain of Custody. The first form is the *In-situ* and Grab Sample Water Chemistry Field Sheet. On this form the sampler will sign over the samples to the team leader include date and time, the date and time delivered or sent to the laboratory should be recorded. The second form (see p. 2A-28) is the Water Chemistry Analysis Chain of Custody. This form includes the parameters being analyzed, date, time, site number, stream name, number of bottles, and is signed by the team leader and the laboratory. (The chain of custody form can be designed based on your laboratory requirements.)

b. The macroinvertebrate chain of custody (see p. 2A-30) gives the following information: date; time; sample identification; stream name; site number; contact information; preservative; and collection method. This form is to be signed, initialed, dated for each method conducted: collected, sample recharged with fresh alcohol to prevent decay, subsampled, quality control for sort residue, identification, and quality control for identification.

2.5 References

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Columbus State University. 2000. Quality Assurance Project Plan (QAPP): Ecoregions Reference Site Project for Wadeable Streams in Georgia. Prepared for: Georgia Environmental Protection Division (GAEPD) and U.S. Environmental Protection Agency (USEPA).

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream Channel Reference Sites: An Illustrated Guide to Field Technique. Gen. Tech. Rep. RM-245. Fort Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 61 pp.

Rosgen, D.L. 1996. Applied River Morphology. Wildland Hydrology, Pagosa Springs, Colorado. pp. 5-1 through 5-34.

Sampling Ambient Water for Trace Metals at EPA Water Quality Criteria Levels, July 1996, U.S. Environmental Protection Agency. EPA 821/R-96-008.

United States Environmental Protection Agency. Office of Water: Washington, D.C. June 1999. *Methods and Guidance for Analysis of Water*. EPA 821-C-99-004.

USDA, General Technical Report, RM-245, *"Stream Channel Reference Sites: An Illustrated Guide to Field Technique".*

Appendix 2A
Forms

WATERBODY RECONNAISSANCE REPORT

STREAM NAME:	**SITE# (or ID#):**	**DATE:**
PROJECT:	**REASON FOR SURVEY:**	**TIME:**
COUNTY:	**NEAREST CITY:**	
LATITUDE (D,M,S):	**LONGITUDE (D,M,S):**	
LATITUDE (DD):	**LONGITUDE (DD):**	
INVESTIGATORS:	**FORM COMPLETED BY:**	
POINT OF ASSESSMENT:		
ASSESSMENT TYPE: LOOKING OVER BRIDGE STANDING BY CREEK IN CREEK OTHER:_____		

STREAM SAMPLING (TYPE CONDUCTED)

METHOD: GA RAPID BIOASSESSMENT PROTOCOL

STREAM CONDITION: REFERENCE CONDITION IMPAIRED CONDITION NOT YET DETERMINED
POTENTIAL REFERENCE CONDITION POTENTIAL IMPAIRED CONDITION

STREAM ANALYSIS: CONTINUAL MONITORING METRIC REFINEMENT OTHER:_____

ACCESSIBILITY OF SITE:

ENTER STREAM AT: ROAD CROSSING BRIDGE CROSSING THROUGH WOODS OTHER:_____

IS ACCESS ON PRIVATE PROPERTY OR PUBLIC PROPERTY? COMMENTS:

IF PRIVATE PROPERTY, ADDITIONAL INFORMATION NEEDED:

CAN YOU DRIVE UP TO THE STREAM? YES NO	**IF NOT, ESTIMATE DISTANCE FROM ROAD TO SITE_____**
CAN YOU DRIVE TO A BRIDGE CROSSING? YES NO	**IF NOT, ESTIMATE DISTANCE TO BRIDGE CROSSING_____**

ARE THERE PROPERTY BOUNDARIES OR FENCES TO CROSS? YES NO TYPE?_____

HAS A TRAIL BEEN HACKED TO STREAM SITE? YES NO	**HAS THE STREAM REACH BEEN FLAGGED?** YES NO

WHAT IS THE CONDITION OF THE NEAREST ROAD TO STREAM SITE? IS 4-WD REQUIRED?

DRIVING DIRECTIONS:

DIRECTIONS TO SAMPLE REACH:

COMMENTS:

WATERBODY RECONNAISSANCE REPORT

STREAM CHARACTERIZATION

STREAM SUBSYSTEM: PERENNIAL INTERMITTENT TIDAL
(May need to take a quick macroinvertebrate sample to determine tidal, also look at location)

STREAM TYPE: CLEARWATER BLACKWATER

IF UNSURE BLACKWATER (Need to do the following for determination): pH:_____DO mg/L:_____%DO:_____

TYPE OF VEGETATION PRESENT:_____ COLOR OF SAND:_____ Temp:_____ Conductivity:_____

FIELD MEASUREMENTS TAKEN AT TIME OF RECON (Take measurements if Reference Condition):

None pH:_____ DO mg/L:_____ %DO:_____ Temp:_____ Conductivity:_____

WEATHER CONDITIONS

NOW: STORM(HEAVY RAIN) RAIN(STEADY RAIN) SHOWERS(INTERMITTENT) _____%CLOUD COVER CLEAR SUNNY

PAST 24 HOURS: STORM(HEAVY RAIN) RAIN(STEADY RAIN) SHOWERS(INTERMITTENT) ____%CLOUD COVER CLEAR SUNNY

CHANNEL FLOW STATUS: WATER REACHES BASE OF BOTH LOWER BANKS >75% 25-75% LITTLE WATER

STREAM FLOW: RECENT FLOODING RECENT DROUGHT DRY CREEK BED NORMAL FLOW CONDITION

HEAVY RAIN IN LAST 7 DAYS? YES NO

INSTREAM: FEATURES/HABITAT/LAND USE

CHANNELIZED? YES NO	**DAM PRESENT?** YES NO TYPE:_____
WADEABLE? YES NO	**APPROXIMATE STREAM DEPTH(FT):_____ DEPTH:_____**

PREDOMINANT STREAM SUBSTRATE: BEDROCK COBBLE/GRAVEL GRAVEL/SAND SAND/SILT OTHER:_____

HABITAT TYPE(%): RIFFLES_____ WOODY DEBRIS/SNAGS_____ UNDERCUT BANKS/ROOTS_____

SOFT/SANDY SEDIMENTS_____ LEAF PACKS_____ AQUATIC VEGETATION/MACROPHYTES:_____

EXCESSIVE ALGAE PRESENT: YES NO

IS INSTREAM IMPACT VISIBLE? YES NO OTHER IMPACTS:_____

IF YES, WHAT ARE POSSIBLE SOURCES OF IMPACT IN STREAM REACH? (CIRCLE THOSE THAT APPLY) PETROLEUM

DAM AGRICULTURAL RUNOFF FJORD IN STREAM ILLEGAL DISCHARGE STORMWATER LANDFILL

PERMITTED DISCHARGE SOIL EROSION BEAVER ACTIVITY CONSTRUCTION LOGGING RUNOFF FROM ROAD CROSSING

STREAM ALTERATION LIVESTOCK EROSION

NEW IMPACTS (If Reference Condition, if dramatic changes, may not be able to sample as reference)

SILVICULTURAL: YES NO (1 to 2 years)	**CLEARCUTTING:** YES NO (1 to 2 years)
CONSTRUCTION: YES NO (1 to 2 years)	**COMMENTS:**_____
OTHER_____	**OTHER:**_____

LAND USE IN SURROUNDING AREA (Catchment) (CIRCLE FOR ALL THAT APPLY)

FORESTED AGRIC-PASTURE ACRIC-ROW CROPS RESIDENTIAL INDUSTRIAL LIVESTOCK WETLAND

SILVICULTURAL COMMERCIAL OTHER:_____ OTHER:_____

PHOTOS (IF TAKEN)

PHOTO(S) TAKEN? YES NO	**Photo Numbers:**

PHOTO DESCRIPTION(S):

HABITAT ASSESSMENT FIELD DATA SHEET—HIGH GRADIENT STREAMS (FRONT)

STREAM NAME:	**SITE (or ID) #:**
LAT (DD):	**LONG (DD):**
LAT (D,M,S):	**LONG (D,M,S):**
INVESTIGATORS:	**FORM COMPLETED BY:**
PROJECT:	**DATE** _____ **TIME** **AM PM** / **REASON FOR SURVEY:**
FIELD SEASON:	**COMMENTS:**

Parameters to be evaluated in sampling reach

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
1. Epifaunal Substrate/ Available Cover	Greater than 70% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e., logs/snags that are <u>not</u> new fall and <u>not</u> transient).	40-70% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	20-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 20% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
2. Embeddedness	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
3. Velocity/Depth Regime	All four velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (Slow is < 0.3 m/s, deep is > 0.5 m.)	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/ depth regime (usually slow-deep).
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 5% of the bottom affected by sediment deposition.	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5-30% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material, increased bar development; more than 50% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills >75% of the available channel; or <25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

HABITAT ASSESSMENT FIELD DATA SHEET – HIGH GRADIENT STREAMS (BACK)

Parameters to be evaluated broader than sampling reach

Habitat Parameter	Condition Category: Optimal	Suboptimal	Marginal	Poor
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
7. Frequency of Riffles (or bends)	Occurrence of riffles relatively frequent; percent of the reach covered by riffles 76-100%; variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstructions are important.	Occurrence of riffles less common; 51-75% of the reach covered by riffles.	Occasional riffle or bend; bottom contours provide some habitat; 26-50% of the reach covered by riffles.	Generally all flat water or shallow riffles; poor habitat; 25% or less of the reach covered by riffles.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank) Note: determine left or right side by facing downstream.	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank)	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
10. Riparian Vegetative Zone Width (score each bank riparian zone)	Width of riparian zone >18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone <6 meters: little or no riparian vegetation due to human activities.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0

Total Score _____

HABITAT ASSESSMENT FIELD DATA SHEET—LOW GRADIENT STREAMS (FRONT)

STREAM NAME:		**SITE (or ID) #:**
LAT (DD):	**LONG (DD):**	
LAT (D,M,S):	**LONG (D,M,S):**	
INVESTIGATORS:	**FORM COMPLETED BY:**	
PROJECT:	**DATE** _____ **TIME** **AM PM**	**REASON FOR SURVEY:**
FIELD SEASON:	**COMMENTS:**	

Parameters to be evaluated in sampling reach

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
1. Epifaunal Substrate/ Available Cover	Greater than 50% of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e., logs/snags that are <u>not</u> new fall and <u>not</u> transient).	30-50% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	10-30% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 10% stable habitat; lack of habitat is obvious; substrate unstable or lacking.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
2. Pool Substrate Characterization	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present.	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
3. Pool Variability	Even mix of large-shallow, large-deep, small-shallow, small-deep pools present.	Majority of pools large-deep; very few shallow.	Shallow pools much more prevalent than deep pools.	Majority of pools small-shallow or pools absent.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
4. Sediment Deposition	Little or no enlargement of islands or point bars and less than <20% of the bottom affected by sediment deposition.	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 20-50% of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 50-80% of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material, increased bar development; more than 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.	Water fills >75% of the available channel; or <25% of channel substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

HABITAT ASSESSMENT FIELD DATA SHEET – LOW GRADIENT STREAMS (BACK)

Parameters to be evaluated broader than sampling reach

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
6. Channel Alteration	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
7. Channel Sinuosity	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (Note - channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas.)	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line.	Channel straight; waterway has been channelized for a long distance.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
8. Bank Stability (score each bank)	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
9. Vegetative Protection (score each bank) Note: determine left or right side by facing downstream.	More than 90% of the streambank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0
10. Riparian Vegetative Zone Width (score each bank riparian zone)	Width of riparian zone >18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone.	Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.	Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.	Width of riparian zone <6 meters: little or no riparian vegetation due to human activities.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0

Total Score _____

HIGH GRADIENT HABITAT ASSESSMENT AVERAGE:

STREAM NAME:		**SITE (or ID) #:**
LAT (DD):	**LONG (DD):**	
LAT (D,M,S):	**LONG (D,M,S):**	
INVESTIGATORS:	**FORM COMPLETED BY:**	
PROJECT:	**DATE** _____ **TIME** **AM PM**	**REASON FOR SURVEY:**
FIELD SEASON:	**COMMENTS:**	

Habitat Parameter Scores	Habitat Parameter Scores	Habitat Parameter Scores	AVERAGE
ASSESOR :	**ASSESOR :**	**ASSESOR :**	
1. Epifaunal Substrate/ Instream Cover _____	1. Epifaunal Substrate/ Instream Cover _____	1. Epifaunal Substrate/ Instream Cover _____	_____
2. Embeddedness _____	2. Embeddedness _____	2. Embeddedness _____	_____
3. Velocity/Depth Combinations _____	3. Velocity/Depth Combinations _____	3. Velocity/Depth Combinations_____	_____
4. Sediment Deposition_____	4. Sediment Deposition_____	4. Sediment Deposition_____	_____
5. Channel Flow Status_____	5. Channel Flow Status_____	5. Channel Flow Status_____	_____
6. Channel Alteration_____	6. Channel Alteration_____	6. Channel Alteration_____	_____
7. Frequency of Riffles_____	7. Channel Flow Status_____	7. Channel Flow Status_____	_____
8. Bank Stability LB_____ RB _____	8. Bank Stability LB_____ RB _____	8. Bank Stability LB_____ RB _____	LB_____ RB_____ Total_____
9. Bank Vegetative Protection LB_____ RB _____	9. Bank Vegetative Protection LB_____ RB _____	9. Bank Vegetative Protection LB_____ RB _____	LB_____ RB_____ Total_____
10. Riparian Vegetative Zone LB_____ RB _____	10. Riparian Vegetative Zone LB_____ RB _____	10. Riparian Vegetative Zone LB_____ RB _____	LB_____ RB_____ Total_____
Total Score: _____	**Total Score:** _____	**Total Score:** _____	**TOTAL AVERAGE SCORE:**
Comments:	Comments:	Comments:	_____

LOW GRADIENT HABITAT ASSESSMENT AVERAGE:

STREAM NAME:		**SITE (or ID) #:**
LAT (DD):	**LONG (DD):**	
LAT (D,M,S):	**LONG (D,M,S):**	
INVESTIGATORS:	**FORM COMPLETED BY:**	
PROJECT:	**DATE** _____ **TIME** **AM PM**	**REASON FOR SURVEY:**
FIELD SEASON:	**COMMENTS:**	

Habitat Parameter Scores	Habitat Parameter Scores	Habitat Parameter Scores	AVERAGE
ASSESOR :	**ASSESOR :**	**ASSESOR :**	
1. Epifaunnal Substrate/ Available Cover _____	1. Bottom Substrate/ Available Cover _____	1. Bottom Substrate/ Available Cover _____	_____
2. Pool Substrate Characterization _____	2. Pool Substrate Characterization _____	2. Pool Substrate Characterization _____	_____
3. Pool Variability _____	3. Pool Variability _____	3. Pool Variability _____	_____
4. Sediment Deposition_____	4. Sediment Deposition_____	4. Sediment Deposition_____	_____
5. Channel Flow Status_____	5. Channel Flow Status_____	5. Channel Flow Status_____	_____
6. Channel Alteration_____	6. Channel Alteration_____	6. Channel Alteration_____	_____
7. Channel Sinuosity _____	7. Channel Flow Status_____	7. Channel Flow Status_____	_____
8. Bank Stability LB_____ RB _____	8. Bank Stability LB_____ RB _____	8. Bank Stability LB_____ RB _____	LB_____ RB_____ Total_____
9. Bank Vegetative Protection LB_____ RB _____	9. Bank Vegetative Protection LB_____ RB _____	9. Bank Vegetative Protection LB_____ RB _____	LB_____ RB_____ Total_____
10. Riparian Vegetative Zone LB_____ RB _____	10. Riparian Vegetative Zone LB_____ RB _____	10. Riparian Vegetative Zone LB_____ RB _____	LB_____ RB_____ Total_____
Total Score: _____	**Total Score:** _____	**Total Score:** _____	**TOTAL AVERAGE SCORE:**
Comments:	Comments:	Comments:	_____

Physical Characterization and Water Quality Field Data Sheet (Front)

STREAM NAME:	**SITE # (ID):**	
LATITUDE (DD):	**LONGITUDE (DD):**	
LATITUDE (D,M,S):	**LONGITUDE (D,M,S):**	
INVESTIGATORS:		
FORM COMPLETED BY:	**DATE:_____**	**REASON FOR SURVEY:**
PROJECT:	**TIME:_____ AM PM**	

SITE LOCATION/MAP	**Draw a map of the site and indicate the areas sampled:**	
STREAM CHARACTERIZATION	**Subsystem Classification** □ Perennial □ Intermittent □ Tidal **Stream Origin** □ Other_____ □ Swamp & Bog □ Spring Fed □ Unsure	**Stream Type (mark one from each group)** □ Coldwater □ Warmwater □ Clearwater □ Blackwater
WEATHER CONDITIONS	**Now** **Past 24 hours** □ □ storm (heavy rain) □ □ rain (steady rain) □ □ showers (intermittent) □ _____% □ _____%cloud cover □ □ clear/sunny	**Has there been a heavy rain in the last 7 days?** □ Yes □ No **Air Temperature**_____ ^{0} C **Other**_____

PHYSICAL CHARACTERIZATION/WATER QUALITY FIELD DATA SHEET (BACK)

RIPARIAN ZONE/ INSTREAM FEATURES

Predominant Surrounding Land Use
□ Forest □ Commercial
□Pasture □ Industrial
□Agricultural □ Planted Pine (Silviculltural)
□Residential □ Cropland_____
□ Clear Cut □ Other_____
□ Livestock_____

Local Watershed NPS Pollution
□No evidence □Some potential sources
□ Obvious sources □ _____

Canopy Cover
□ Open (0-10%) □ Partly open (11-45%)
□ Partly shaded (46-80%) □ Shaded (81-100%)

Local Water Erosion
□ None □ Moderate □ Heavy

BEAVER ACTIVITY BASED ON OBSERVATIONS:
□ Active Beaver Dam Affecting Stream
□ Inactive Beaver Dam Affecting Stream
□ Active Beaver Dam/Cutting Evident But Little Effect
□ Old Beaver Dam/ Activity, Little Effect
□ Inactive Beaver Dam, Little Effect on Stream
□ No Beaver Activity Evident

LIVESTOCK DAMAGE BASED ON OBSERVATIONS:
□ No Livestock Damage
□ Stable (0-25% Damage, Little/No Erosion)
□ Moderate (25-50%) Damage, < 50% Plant Biomass Remains)
□ High (51-75 % Damage, <25% Plant Biomass Remains)
□ Severe (76-100% Damage, Little/No Plant Biomass Remains)

High Water Mark _____m
Estimated Stream Width _____m
Estimated Stream Depth
□ Riffle _____m □ Run _____m
□ Pool _____m
Reach Morphological Types (% in reach):
□ Riffle _____m □ Run _____m
□ Pool _____m
Average Surface Velocity _____m/sec
(at thalweg)
Estimated Reach Length 100m
Channelized? □ Yes □ No
□ Full □ Partial
Dredging? □ Yes □ No

Dam Present? □ Yes □ No
If Present: □ Upstream □ Downstream □ In Reach

RIPARIAN VEGETATION (18 meter buffer)

Indicate the dominant type and record the dominant species present:
□ Trees □ Shrubs □ Grasses □ Herbaceous
dominant species present: _____
_____

Check one that best applies:
□ Brush – dominated by alders, willows, et.
□Forested – dominated by tress with brushy understory
□ Grass- bank covered with tall grasses, sedges, etc.
□ Exposed – bare rock, soil, rock, etc.

AQUATIC VEGETATION

Indicate the dominant type and record the dominant species present
□ Rooted emergent □ Rooted submergent □ Rooted floating □ Free Floating
□ Floating Algae □ Attached Algae

dominant species present _____

Portion of the reach with vegetative cover _____%

SEDIMENT/ SUBSTRATE

Odors
□ Normal □ Sewage
□ Chemical □ Anaerobic
□ Other_____

Oils
□ Absent □ Slight □ Moderate □ Profuse

Deposits
□ Petroleum □ Sludge □ Sawdust □ Paper fiber □ Sand
□ None □ Relict shells □ Other_____

Looking at stones which are not deeply embedded, are the undersides black in color?
□ Yes □ No

WATER QUALITY

Water Odors
□Normal/None □ Sewage
□ Petroleum □ Chemical
□ Fishy □ Other_____

Water Color
□ Tannic □ Clear
□ Green (Algae) □ Other _____

Water Surface Oils
□ Slick □ Sheen □ Globs □ Flecks
□ None □ Other_____

Turbidity (visual measurement)
□ Clear □ Slightly turbid □ Turbid
□ Opaque □ Water color □ Other_____

ORGANIC SUBSTRATE COMPONENTS (does not necessarily add up to 100%)

Substrate Type	Characteristic	% Composition in Sampling Area
Detritus	sticks, wood, coarse plant materials (CPOM)	
Muck-Mud	black, very fine organic (FPOM)	
Marl	grey, shell fragments	

INORGANIC SUBSTRATE COMPONENTS (should add up to 100%)

Substrate Type	Diameter	% Composition in Sampling Reach
Bedrock		
Boulder	> 256 mm (10")	
Cobble	64-256 mm (2.5" –10")	
Gravel	2-64 mm (0.1"-2.5")	
Sand	0.06-2mm (gritty)	
Silt	0.004-0.006mm	
Clay	<0.004mm (slick)	

Comments: _____

In-situ and Grab Sample Water Chemistry Field Sheet

STREAM NAME:	**SITE # (ID):**	
LATITUDE (DD):	**LONGITUDE (DD):**	
LATITUDE (D,M,S):	**LONGITUDE (D,M,S):**	
INVESTIGATORS:		
FORM COMPLETED BY:	**DATE:_____**	**REASON FOR SURVEY:**
PROJECT:	**TIME:_____ AM PM**	

Depth Calibration for Water Quality Multiprobe				
Initial Reading	Adjust To	Temperature	Final Reading	Δ Initial to Final

***In-situ* Field Chemistry Data**	
Unit used:	
Water Temperature: ° C	**Depth (m):**
Specific Conductance: (µmhos/cm)	**Salinity:**
Dissolved Oxygen (mg/L):	**Dissolved Oxygen: %**
pH:	**Air Temperature: ° C**

***In-situ* Turbidity Measurement**
Unit used:
Turbidity: NTU

Name of Lab to Send Grab Samples:
Sample ID #: # of Bottles Collected:
Parameters
Total Suspended Solids **Alkalinity** **No preservative Half-Gallon bottle**
Clean Metals (ICP/MS) Preservative: HNO_3, <2 pH 500mL plastic bottle
Metals blank collected at this site? Yes or No
Alkalinity No preservative 250 ml bottle
Total Kjeldahl Nitrogen (TKN) Ammonia (NH_3) **Nitrate-Nitrite (NO_2-NO_3) Total Organic Carbon (TOC)** **Preservative H_2SO_4, pH <2 250 ml bottle**
Ortho-phosphate Total Phosphorus **Preservative H_2SO_4, pH <2 250 ml bottle**

Sampled by (signature):	Date/Time:	Team Leader/Received (signature):	Date/Time:

Date/Time Delivered to Name of Lab Here:	Date/Time Delivered or Sent to Name of Lab Here:

DataSonde Calibration Form

Project: _____ **Unit:** _____

Sampling Date: _____ **Serial #:** _____

Calibration Date: _____ **Handheld#** _____

Calibrated By: _____

Set-up @_____ **on** _____ **Start Time:** _____

Warm-up @_____**on**_____ **End Time:** _____

Parameter	Initial Reading	Cal. Std. Value	Temp	Final Reading	Δ	Std. Lot#
% Saturation		100.00				
Dissolved Oxygen (mg/L)						
pH 4.00 (Standard Units)		4.00				
pH 7.00 (Standard Units)		7.00				
pH 10.00 (Standard Units)		10.00				
Conductivity (μmhos)		1412				
Battery (Volts)						
Barometric Pressure (mm Hg)						
Altitude						

Notes:

Initial When Completed

TURBIDIMETER CALIBRATION & TURBIDIMETER SECONDARY STANDARDS STANDARDIZATION SHEET

Project: _____
Calibration Date: _____
Calibrated By: _____
Standardized Date:_____
Standardized By:_____

Portable Turbidimeter Instrument
Model #:
Start Time: _____
End Time: _____

Turbidimeter Calibration

Primary Standards	Initial Reading	Cal. Std. Value	Final Reading	Δ
<0.1 NTU		0		
20 NTU		20		
100 NTU		100		
800 NTU		800		

Turbidimeter Standardization

Secondary Standards	Initial Reading	Determined Value	Final Reading	Δ
Range:				
Range:				
Range:				

Notes:

Initial When Completed _____

BENTHIC MACROINVERTEBRATE FIELD DATA SHEET

STREAM NAME:		**SITE # (or ID#):**
LATITUDE (DD):	**LONGITUDE (DD):**	
LATITUDE (D,M,S):	**LONGITUDE (D,M,S):**	
INVESTIGATORS:		
FORM COMPLETED BY:	**DATE** _____ **TIME**_____ **AM PM**	**REASON FOR SURVEY:**
PROJECT:		
Field Collector of Benthos:	**Field Processor of Benthos:**	

HABITAT TYPES	**Indicate the percentage of each habitat type present** Riffles _____% Snags_____% Banks/ root mats_____ % Soft/Sandy sediment_____% Leaf packs_____ % Submerged Macrophytes_____ %
SAMPLE COLLECTION	**Gear used**: D-frame, 500 µm net **How were the samples collected?** Wading **Indicate the number of jabs/kicks taken in each habitat type.** Riffles (fast)_____ Riffles (slow)_____ Snags_____ Banks/ root mats_____ Soft/Sandy sediment_____ Leaf packs_____ Submerged Macrophytes_____ Other ( )_____ **Total # of Jabs:**_____ **# of Jabs Reallocated (if any):**_____ **Total # of Bottles collected:**_____
GENERAL COMMENTS	

QUALITATIVE LISTING OF AQUATIC BIOTA
Indicate estimated abundance: 0 = Absent/Not Observed, 1 = Rare, 2 = Common, 3= Abundant, 4 = Dominant

Periphyton	0	1	2	3	4	Slimes	0	1	2	3	4
Filamentous Algae	0	1	2	3	4	Macroinvertebrates	0	1	2	3	4
Macrophytes	0	1	2	3	4	Fish	0	1	2	3	4

FIELD OBSERVATIONS OF MACROBENTHOS
Indicate estimated abundance: 0 = Absent/Not Observed, 1 = Rare (1-3 organisms), 2 = Common (3-9 organisms), 3= Abundant (>10 organisms), 4 = Dominant (>50 organisms)

Porifera	0	1	2	3	4	Anisoptera	0	1	2	3	4	Trichoptera	0	1	2	3	4
Hydrozoa	0	1	2	3	4	Zygoptera	0	1	2	3	4	Plecoptera	0	1	2	3	4
Platyhelminthes	0	1	2	3	4	Hemiptera	0	1	2	3	4	Megaloptera	0	1	2	3	4
Turbellaria	0	1	2	3	4	Coleoptera	0	1	2	3	4	Sialidae	0	1	2	3	4
Hirudinea	0	1	2	3	4	Lepidoptera	0	1	2	3	4	Corydalidae	0	1	2	3	4
Oligochaeta	0	1	2	3	4	Tipulidae	0	1	2	3	4	Other	0	1	2	3	4
Isopoda	0	1	2	3	4	Empididae	0	1	2	3	4						
Amphipoda	0	1	2	3	4	Simuliidae	0	1	2	3	4						
Decapoda	0	1	2	3	4	Tabanidae	0	1	2	3	4						
Gastropoda	0	1	2	3	4	Culicidae	0	1	2	3	4						
Bivalvia	0	1	2	3	4	Chironomidae	0	1	2	3	4						
						Ephemeroptera	0	1	2	3	4						

Channel Cross-Section Field Sheet (Front)

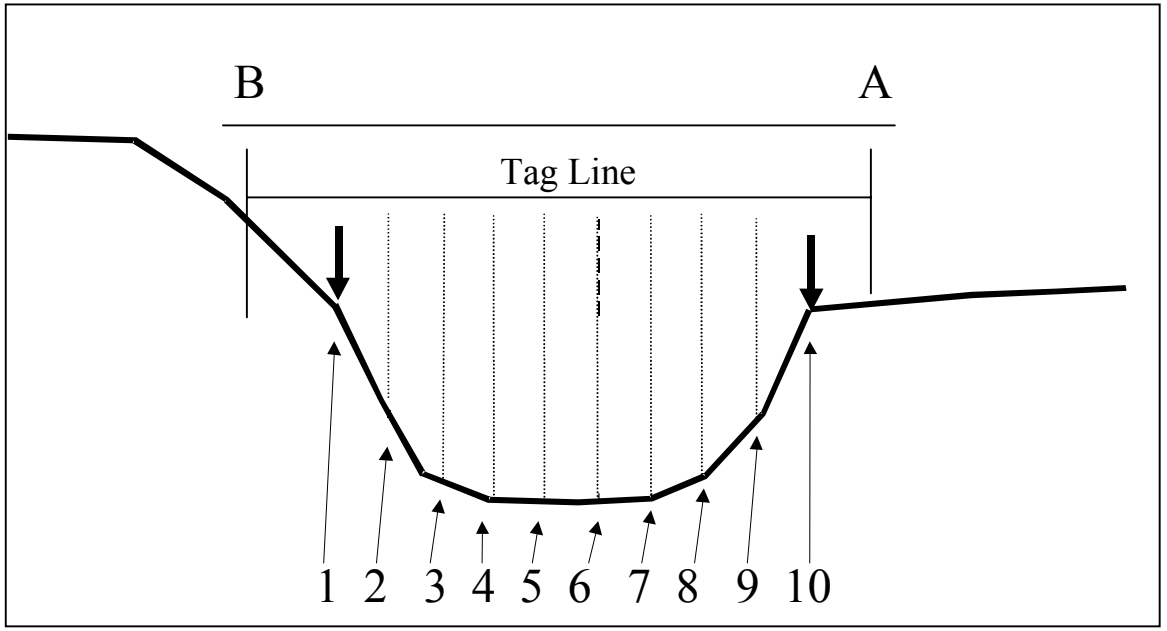
Stream Name: _____ **Site # (or ID #):** _____ **Date/Time:** _____/_____ **AM PM**
Project: _____ **Reason for Survey:**_____
Investigators: _____ **Recorder**: _____ **Measurer:** _____
Compass Direction of Tag Line _____° □ **LB to RB** □ **RB to LB** **Lower Bank Monument (Point A):** □ **Left Bank** □ **Right Bank**
Location Comments: _____
Distance from A to B: _____**(m) Active Stream Channel Width:** _____**(m) Distance from Top of monument (A) to tag line**_____**(m)**
Pin Height: _____**(m) Benchmark 1**_____ **Benchmark 2**_____
Benchmark 3_____ **Camera/Film #**_____ **Photo #'s: US**_____ **DS**_____
LB_____ **RB** _____ **Other/# /Location**_____ **Other/#/Location**_____

Station/Distance (m) (From Monument)	**Elevation (m)**	**Depth (m)**	**Remarks**	**Station/Distance (m)** (From Monument)	**Elevation (m)**	**Depth (m)**	**Remarks**

COMMENTS:_____
_____
_____

Channel Cross-Section Field Sheet (Back)

FIGURE 1: STREAM CHANNEL CROSS SECTIONAL DIAGRAM



ABBREVIATIONS (FOR REMARKS):

LB=Left Bank
RB=Right Bank
LTOB=Left Top of Bank
RPIN=Right Monument
BM=Benchmark
HI=Height of Instrument
TOP=Top of Pool

RTOB=Right Top of Bank
RBF=Right Bankfull
LBF=Left Bankfull
LPIN=Left Monument
FS=Fore Sight
TOR=Top of Riffle
BOP=Bottom of Pool

REOW=Right Edge of Water
LEOW=Left Edge of Water
TH=Thalwag
BPIN=Bank Pin
BS=Back Sight
BOR=Bottom of Riffle
WDJ=Woody Debris Jam

WA = Water
DCB=Dry Creek Bed
LBB=Left Bank Bedrock
SB= Channel (sand bar)
RK=Rock
BR=Bedrock
TR=Tree Root

CB=Cobble Bar
REOB=Right Edge of Bank
RBB=Right Bank Bedrock
EOC=Edge of Channel
LUW=Log Under Water
LL=Leaf Litter

LEOB=Left Edge of Bank
LBEOB=Left Bank Edge of Bedrock
RBEOB=Right Bank Edge of Bedrock
CB=Cobble
BD=Boulder
GB=Gravel Bar

Other(s):_____

_____

Velocity/Discharge Field Sheet

Stream Name:_____ **Site # (or ID#):**_____ **Date/Time:**_____/_____
Investigators:_____ **Recorder/Field Measurer**_____
Project: _____ **Reason for Survey:**_____
Latitude (DD): _____ **Longitude (DD):**_____ **Latitude (D,M,S):**_____ **Longitude (D,M,S):**_____
Weather:_____ **Rain in last 24 hrs? [ ] Y [ ] N** **Tape reading at Starting Edge:**_____ **Ending Edge:**_____

Standard: Discharge - ft^3/s Velocity – ft/s Width – ft Area – ft^2 Metric: Discharge – m^3/s Velocity – cm/s Width – m Area – m^2
Note: Units on measurements will depend on choice to use metric or standard.

Pre-Deployment Diagnostics							Setup Parameters				
Recorder Status/Memory	H2O T°	%Battery	Raw Data	Time Changed		Units: Metric Standard	Averaging time (sec)	Data Collection Mode		Salinity
				Y	N			General	Discharge	

	Stn1 Location:			Stn2 Location:			Stn3 Location:			Stn4 Location:			Stn5 Location:			Stn6 Location:		
Reading depth	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8
Depth																		
Velocity																		
Stn Disch																		
AveStnV																		
	Stn7 Location:			**Stn8 Location:**			**Stn9 Location:**			**Stn10 Location:**			**Stn11 Location:**			**Stn12 Location:**		
Reading depth	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8
Depth																		
Velocity																		
Stn Disch																		
AveStnV																		
	Stn13 Location:			**Stn14 Location:**			**Stn15 Location:**			**Stn16 Location:**			**Stn17 Location:**			**Stn18 Location:**		
Reading depth	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8
Depth																		
Velocity																		
Stn Disch																		
AveStnV																		
	Stn19 Location:			**Stn20 Location:**			**Stn21 Location:**			**Stn22 Location:**			**Stn23 Location:**			**Stn24 Location:**		
Reading depth	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8	0.2	0.6	0.8
Depth																		
Velocity																		
Stn Disch																		
AveStnV																		

Total Discharge:	**Mean Velocity:**	**Tot. Width:**	**Tot. Area:**

PEBBLE COUNT FIELD SHEET

Stream Name:_____ **Site # (or ID#):**_____**Date/Time:**_____/_____**AM PM**
Investigators:_____ **Recorder**_____ **Field Measurer**_____
Project: _____ **Reason for Survey:**_____
GPS UNIT: _____ **GPS [ ]Y [ ] N Differential Correction? [ ]Y [ ]N Pdop:** _____ **Error:** _____
Latitude (DD): _____**Longitude (DD):**_____
Latitude (D,M,S):_____ **Longitude (D,M,S):**_____ _____
Coordinate Location Description: _____
Weather:_____ **Rain in last 24 hrs? [ ] Y [ ] N**
Camera/Film No._____ **Photo #'s: US**_____**DS**_____**LB**_____**RB**_____**Other**_____**Location of Picture**_____

				Total Transects:		
Inches	**Particle**	**Millimeter**		**Particle Count**	**TOTAL #**	**% Cum**
	Silt/Clay	<.062	S/C			
	Very Fine	.062-.125	S			
	Fine	.125-.25	A			
	Medium	.25-.50	N			
	Coarse	.50-1.0	D			
	Very Coarse	1.0-2.0	S			
.08-.16	Very Fine	2-4				
.16-.24	Fine	4-6	G			
.24-.31		6-8	R			
.31-.47	Medium	8-12	A			
.47-.63		12-16	V			
.63-.94	Coarse	16-24	E			
.94-1.26		24-32	L			
1.26-1.9	Very Coarse	32-48	S			
1.9-2.5		48-64				
2.5-3.8	Small	64-96	C			
3.8-5.0		96-128	O			
5.0-7.6	Large	128-192	B			
7.6-10		192-256	L			
10-15	Small	256-384	B			
15-20		384-512	L			
20-40	Medium	512-1024	D			
40-160	Lgr -Very Lgr	1024-4096	R			
	Bedrock		BDRK			
				TOTALS		

Water Chemistry Analysis Chain of Custody

Laboratory Name: _____COLLECTED BY: _____ PROJECT CONTACTS: _____

Investigators: _____Latitude (DD): _____ Longitude (DD): _____

DATE COLLECTED	TIME COLLECTED	STATION NUMBER	**PROGRAM:**

PROJECT:_____

*** SAMPLE SITE DESCRIPTION:**

* Include site # and stream name in this section.

FIELD DATA:

PARAMETER		STORET CODE	Value
H2O Temperature	deg. C	00010	
Air Temperature	deg. C	00020	
Field Conductivity	umho/cm	00094	
Dissolved Oxygen	mg/L	00300	
Dissolved Oxygen	%		
pH	units	00400	
Depth	ft.	00003	
Gage Height	ft.	00065	
Photic Zone	m.	00204	
Secchi Disk	m.	00078	
Tapedown	ft.	82580	
Stream Flow	cfs	00061	
Salinity	mg/ml	00096	
Cl_2 (total)	mg/L	50060	
Cl_2 (free)	mg/L	50064	
Turbidity	NTU		

LABORATORY DATA:

	PARAMETER		STORET CODE
	Turbidity	NTU	00076
	Color	PCU	00080
	Specific Cond.	umho/cm	00095
	BOD – 5 day	mg/L	00310
	pH	units	00403
	Tot. Alkalinity	mg/L CaCO3	00410
	Hardness	mg/L CaCO3	00900
	Suspended Solids	mg/L	00530
	Tot. Dissolved Solids	mg/L	70300
	Ammonia	mg/L (N)	00610
	Nitrate + Nitrite	mg/L (N)	00630
	Tot. Kjeldahl Nitrogen	mg/L (N)	00625
	Tot. Phosphorous	mg/L (P)	00665
	Chloride	mg/L	00940
	COD	mg/L	00335
	Tot. Organic Carbon	mg/L	00680
	Fecal Coliform	MPN/100ml	31615

	PARAMETER
	CLEAN Metals (ICP/MS)
	MPP Metals
	Mercury
	Semi-Volatiles
	Volatiles
	Pesticides (chlorinated)
	PCB's
	Ortho Phosphorous
	Cyanide (total)
	Oil & Grease
	Total Phenols

Total Number of Bottles:	Delivered by & Date:	Received by & Date:	Lab Label

Contact Names:			Preservative (Y/N): **Ethanol**	Number of Containers	Type of Analysis Requested							Sample Check-in: Comments:	
Address: Fax #:					Recharged #1	Recharged #2	Sorted	QC of Sort Residue	Identification	QC of Identification (Internal)	QC of Identification (External)		
Macroinvertebrate Chain of Custody Project Name:												Collection Method	Log Number
Page ____ of _____		Stream Name & Site #:											
Date:	Time:	Sample Identification #			*Initials and Date*								
												D-frame net 20 Jab	
												D-frame net 20 Jab	
												D-frame net 20 Jab	
												D-frame net 20 Jab	
												D-frame net 20 Jab	
												D-frame net 20 Jab	
Sampled by (signature):		Date/Time:	Relinquished by Team Leader (signature):					Date/Time:	Relinquished Macros (signature):			Date/Time:	
Team Leader/Received (signature):		Date/Time:	Received by Lab (signature):					Date/Time:	Macros received by (signature):			Date/Time:	
Returned Macros (signature):		Date/Time:	Relinquished Macros (signature):					Date/Time:	Returned Macros (signature):			Date/Time:	
Lab Received (signature):		Date/Time:	Macros for Iding Received by:					Date/Time:	Lab received Macros (signature):			Date/Time:	
		Date/Time:						Date/Time:				Date/Time:	
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Appendix 2B
Supplemental Information

SUPPLEMENTAL INFORMATION: High Gradient (Riffle/Run Prevalent) Streams

(This supplemental information is to be used as a guideline for determining the habitat scores for each parameter.)

I. Epifaunal Substrate/Available Cover [High Gradient (Riffle/Run Prevalent) Streams]

Measures availability of actual substrates that are available as refugia, feeding, or sites for spawning & nursery functions for aquatic organisms. A wide variety and/or abundance of submerged structures in the stream provides macroinvertebrates with a large number of niches, thus increasing habitat diversity. Riffle areas are critical for maintaining a healthy variety of insects in most riffle prevalent streams.

Check habitat types which occur at this site: fallen trees/large woody debris, deep pools, shallow pools, large rocks, undercut banks, thick root mats, dense macrophyte beds, leaf packs, and riffles.

A. Stable and available habitat(s) expected for stream type make up >70% of reach. Stream exhibits a well-developed riffle-run complex.

1. 7 habitats common; stable substrate dominated by softball size cobble stones 20
2. 5 habitat types common, additional habitat types rare; stable substrate dominated by boulder stones 18
3. Less than 4 habitat types present; stable substrate dominated by a mixture of gravel stones and boulders/bedrock and/or stable woody debris 16

B. Stable and available habitat(s) expected for stream type make up >40-70% of reach

1. 7 habitats common; stable substrate dominated by softball size cobble stones 15
2. 5 habitat types common, additional habitat types rare; stable substrate dominated by boulder stones 13
3. Less than 4 habitat types present; stable substrate dominated by a mixture of gravel stones and boulders/bedrock and/or stable woody debris 11

C. Stable and available habitat(s) expected for stream type make up 20-40% of reach

1. 7 habitats common; stable substrate dominated by softball size cobble stones 10
2. 5 habitat types common, additional habitat types rare; stable substrate dominated by boulder stones 8
3. Less than 4 habitat types common, additional habitat types rare; stable substrate dominated by a mixture of cobble and gravel stones and/or stable woody debris 7

D. Stable and available habitat(s) expected for stream type make up <20% of reach. Riffles or runs are virtually nonexistent, no cobble substrate.

1. 2 habitat types present, additional habitat types rare; substrate dominated by large boulders, short runs 5
2. 1 habitat type common, additional habitat types rare; substrate dominated by rock and sand with long runs, no riffles 3
3. 1 habitat type; substrate dominated by rock and sand with short runs, no riffles 2
4. 1 habitat type rare; substrate dominated by rock and sand, no runs or riffles 1
5. 0 habitat types present; substrate dominated by sand with no riffles or runs 0

II. Embeddedness [High Gradient (Riffle/Run Prevalent) Streams]

Measures the degree to which cobble, boulders, and other rock substrate are surrounded by fine sediment. Embeddedness relates directly to the suitability of the stream substrate as habitat for macroinvertebrates.

Fine sediments/sands range from 0.062 mm to 2 mm in size. Silt particles measure less than 0.062 mm. Sediment and silt particles smaller than 2 mm can be distinguished using "texture by feel" techniques employed in soil surveys.

A. Little or no embeddedness present by fine silt and/or sediment surrounding and covering rocks

1. < 10% embeddedness 20
2. 10% embeddedness by sediment 19
3. 10% embeddedness by sediment and silt 18
4. 20% embeddedness by sediment 17
5. 20% embeddedness by sediment and silt 16

B. Fine sediment and silt surrounds and fills 25-50% of the living spaces around and in between gravel, cobble, and boulders

1. 30% embeddedness by sediment 15
2. 30% embeddedness by sediment and silt 14
3. 40% embeddedness by sediment 13
4. 40% embeddedness by sediment and silt 12
5. 50% embeddedness by sediment 11

C. Fine sediment and silt surrounds and fills 50-75% of the living spaces around and in between gravel, cobble, and boulders

1. 50% embeddedness by sediment and silt 10
2. 60% embeddedness by sediment 9
3. 60% embeddedness by sediment and silt 8
4. 70% embeddedness by sediment 7
5. 70% embeddedness by sediment and silt 6

D. Fine sediment and silt surrounds and fills more than 75% of the living spaces around and in between gravel, cobble, and boulders

1. 80% embeddedness by sediment 5
2. 80% embeddedness by sediment and silt 4
3. 90% embeddedness by sediment 3
4. 90% embeddedness by sediment and silt 2
5. 100% embeddedness by sediment 1
6. 100% embeddedness by sediment with a thick layer of silt on its surface 0

III. Velocity/Depth Regime [High Gradient (Riffle/Run Prevalent) Streams]

Measures a stream's characteristic velocity/depth regime. There are 4 combinations of velocity and depth that are characteristic to high quality riffle/run prevalent streams. These are: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The depth criteria used to distinguish shallow from deep is 0.5 meter; the velocity criteria used to distinguish slow from fast is 0.3 m/sec.

A. A complex stream system that exhibits a heterogeneous combination of all velocity/depth patterns.

1. All 4 velocity/depth regimes are present 20
2. All regimes present, but one or more may not be well-defined 18
3. All regimes present, but all may not be well-defined 16

B. Stream is less heterogeneous displaying some of the velocity/depth patterns.

1. Only 3 of the 4 velocity/depth regimes are present 15
2. 3 of the 4 regimes are present, but one or more may not be well-defined 13
3. All regimes present **except** for the fast-shallow regime 11

C. Stream becomes more homogeneous. Light sediment deposition is resulting in the loss of certain velocity/depth patterns.

1. Only 2 of the 4 velocity/depth regimes are present 10
2. 2 of the 4 regimes are present, but may not be well-defined 8
3. The fast-shallow or slow-shallow regime is missing 6

D. A simple stream system that is heavily affected by sediment deposition, restricting water flow, resulting in a monotonous velocity/depth pattern.

1. Only 1 of the 4 velocity/depth regimes is present, usually dominated by the slow-deep regime 5
2. Potential velocity/depth regime exists but is not clearly defined 3
3. None of the velocity/depth regimes are present 0

IV. Sediment Deposition [High Gradient (Riffle/Run Prevalent) Streams]

Relates to the amount of sediment that has accumulated and the changes that have occurred to the stream bottom as a result of deposition. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increases in size as the channel is diverted toward the outer bank), shoals, or result in the filling of pools. Depositional material comes from both the overall watershed, and bank erosion (Barbour and Stribling 1995). The growth or appearance of bars/islands where they did not previously exist may be an indication of upstream erosion. High levels of sediment deposition create an unstable and continually changing environment that becomes unsuitable for many organisms (FL DEP 1996).

A. No enlargements of islands/point bars present or less than 20% bottom affected by sand or silt accumulation.

1. No sediment deposition detected; especially in pools 20
2. Less than 20% sediment deposition with accumulation in pools only 18
3. Less than 20% sediment deposition with accumulation in runs and pools 17
4. Less than 20% sediment deposition with few, old, small point bars or islands made up of coarse gravel in stream channel 16

B. 20-50% bottom affected by sand or silt accumulation; slight deposition in pools; some new increase in bar and island formation.

1. 20-30% sediment deposition with gravel and/or sand 15
2. 20-30% sediment deposition with sand and/or silt 14
3. 40-50% sediment deposition with gravel and/or sand 12
4. 40-50% sediment deposition with sand and/or silt 11

C. 50-80% bottom affected with moderate deposition in pools. Number of shallow pools increases. Habitats smothered by sand, silt, and possibly coarse gravel. Deposits of fresh, fine, gravel, sand, and silt observed on old and new point bars, islands, and behind obstructions. Formation of few new bars/islands is evident and old bars are deep and wide; deposition at bends obvious.

1. 60-70% sediment deposition with gravel and/or sand 10
2. 60-70% sediment deposition with sand and/or silt 9
3. 70-80% sediment deposition with gravel and/or sand 7
4. 70-80% sediment deposition with sand and/or silt 6

D. More than 80% bottom affected with heavy deposition from coarse and fine gravel and sand at stream bends, constrictions, and/or pools. Extensive deposits of fine sand and/or silt on old and new bars, islands, and along banks in straight channels. Few pools are present due to siltation. Only larger rocks in riffle areas remain exposed.

1. 80-90% sediment deposition; pools almost absent due to substantial deposition; bottom silt may move with almost any flow above normal 3
2. 90-100% sediment deposition; pools almost absent 1
3. 100% sediment deposition; pools absent due to substantial deposition; bottom silt moves with almost any flow above normal 0

V. Channel Flow Status [High Gradient (Riffle/Run Prevalent) Streams]

This is the degree to which the channel is filled with water during base or average annual flow periods. This is a seasonal parameter. A decrease in water will wet smaller portions of the streambed, thus decreasing available habitat for aquatic organisms. Use the vegetation line on the lower bank as your reference point to estimate channel flow status.

A channel cross-section may help the investigator(s) estimate what percentage of the available channel is full. Stretch a tape very tightly across the channel. Level and secure tape at the base of both lower banks.

A. Water reaches the base of both lower banks and minimal amount of channel substrate is exposed (100% channel full) 20
   1. > 95% channel is full 18
   2. 90-95% channel is full 16

B. Water fills > 75% of the available channel (or <25% of channel substrate is exposed)
   1. 90% of channel is full 15
   2. 85% of channel is full 13
   3. 80% of channel is full 11

C. Water fills 25-75% of the available channel and/or riffle substrates are mostly exposed
   1. 75% of channel is full 10
   2. 60-65% of channel is full 9
   3. 50% of channel is full 8
   4. 35-40% of channel is full 7
   5. 25% of channel is full 6

D. Very little water in the channel and mostly present as standing pools
   1. 20% of channel is full 5
   2. 10% of channel is full 4
   3. < 10% of channel is full 3
   4. Water present as isolated standing pools 1
   5. Channel is dry 0

VI. Channel Alteration [High Gradient (Riffle/Run Prevalent) Streams]

Measurement of large-scale alteration of instream habitat that affects stream sinuosity and causes scouring. Channel alteration is present when: (1) artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; (2) when the stream is very straight for significant distances; (3) when dams and bridges are present; and, (4) when other changes have occurred.

A. Stream follows a normal and natural meandering pattern. Alteration is absent.

1. No evidence of disturbance with bends/runs frequent; bend angles average > 60° 20
2. No evidence of disturbance with bends/runs frequent; bend angles average between 60°- 40° 18
3. No evidence of disturbance with bends/runs frequent; bend angles average < 40° 16

B. Some stream straightening, dredging, artificial embankments, or dams present but NO evidence of recent alteration activities. May have been channelized in the past more than 20 years ago, but mostly recovered.

1. Bridge abutment present but disturbance is more than 20 years old; no other channel disturbance present 15
2. 10% of reach or less has channel disturbance other than bridge 14
3. 20% of reach has channel disturbance 13
4. 30% of reach has channel disturbance 12
5. 40% of reach has channel disturbance more than 20 years old 11

C. Somewhat channelized; 40-80% of the area has been straightened, dredged, or otherwise altered; disturbance may be less than 20 years old.

1. 40% of reach has channel disturbance 10
2. 50% of reach has channel disturbance 9
3. 60% of reach has channel disturbance 8
4. 70% of reach has channel disturbance 7
5. 80% of reach has channel disturbance 6

D. More than 80% of the stream site has been straightened, dredged, or otherwise altered; banks most likely box-cut; instream habitat highly altered.

1. 90% of reach has channel disturbance 5
2. Channel reach 100% disturbed; straight with no artificial embankments 3
3. Channel reach 100% disturbed; straight with artificial embankments 2
4. Channel reach 100% disturbed; straight with natural and artificial embankments 1
5. Banks 100% shored by gabion and/or cement 0

VII. Frequency of Riffles [High Gradient (Riffle/Run Prevalent) Streams]

Estimates the frequency or occurrence of riffles as a measure of sinuosity. Riffles are a source of high-quality habitat and diverse fauna; therefore, an increased frequency of occurrence enhances the diversity of the stream community. **Subtract the length of non-riffle areas from the length of the reach (or add the length of all the riffles, whichever is easiest). If the selected reach is unrepresentative of the rest of the stream please make a note of it on the habitat assessment sheets.** This measurement can be conducted while the reach is being delineated.

A. Occurrence of riffles relatively frequent to continuous. Deep pools may be present and riffles are deep enough to allow passage of fish.

1. Percent of reach made up of riffles 96-100% 20
2. Riffle percent = 91-95% 19
3. Riffle percent = 86-90% 18
4. Riffle percent = 81-85% 17
5. Riffle percent = 76-80% 16

B. Occurrence of riffles common; adequate depth in pools and riffles.

1. Riffle percent = 71-75% 15
2. Riffle percent = 66-70% 14
3. Riffle percent = 61-65% 13
4. Riffle percent = 56-60% 12
5. Riffle percent = 51-55% 11

C. Occasional or infrequent riffle; variable bottom contours may provide some habitat.

1. Riffle percent = 46-50% 10
2. Riffle percent = 41-45% 9
3. Riffle percent = 36-40% 8
4. Riffle percent = 31-35% 7
5. Riffle percent = 26-30% 6

D. Generally all flat water or some shallow riffles; essentially a straight and uniform depth stream; riffles are not deep enough to provide free passage for fish.

1. Riffle percent = 20-25% 4
2. Riffle percent > 20% with some shallow riffles 2
3. Riffle percent > 20% with no shallow riffles, water completely flat 0

VIII. Bank Stability [High Gradient (Riffle/Run Prevalent) Streams]

Measures the existence of, or the potential for, detachment of soil from the upper and lower stream banks and its movement into the stream. Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks and are considered unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Reinforcement of banks via rocks, artificial or natural, provides increased stability.

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. Bank stable; erosion absent or minimal. Side slopes are generally less than 30% and are stable. Bank may be reinforced by rock thus increasing slope >30% while providing stability.

1. No evidence of erosion or bank failure 10
2. Less than 5% bank affected by erosion 9

B. Moderately stable bank; small areas of erosion or bank slumping visible. Most areas are stable with only slight potential for erosion at flood stages. Side slopes up to 40% on one bank. Bank may be reinforced by rock thus increasing slope > 40% while providing stability.

1. 5% bank has erosional areas 8
2. 15% bank has erosional areas 7
3. 30% bank has erosional areas 6

C. Moderately unstable bank; frequency and size of raw areas are such that high water events have eroded some areas of the bank. Medium size areas of erosion or bank slumping visible. Side slopes up to 60% on some of the bank. High erosion potential during floods.

1. 40% - 50% bank has erosional areas 5
2. 50%- 60% bank has erosional areas 4
3. 60% - 70% bank has erosional areas 3

D. Unstable bank; mass erosion and bank failure is evident; erosion and pronounced undercutting present at bends and along some straight channel areas. Side slopes > 60% are common. Many raw areas present and 60-100% bank has erosional scars.

1. 70%- 80% bank has erosional areas 2
2. 80%-90% bank has erosional areas 1
3. > 90% streambank has eroded 0

IX. Bank Vegetative Protection [High Gradient (Riffle/Run Prevalent) Streams]

Measures the amount of the stream bank that is covered by vegetation. This parameter supplies information on the ability of the bank to resist erosion, the uptake of nutrients by existing vegetation, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than banks without vegetative protection or those shored up with concrete or riprap.

Four factors to consider when scoring bank vegetative protection: (1) Is the vegetation native and natural, or planted and introduced?; (2) Is the upperstory, understory, and ground cover vegetation well balanced?; (3) What is the standing crop biomass?; and ,(4) During which season are you conducting this assessment?

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. More than 90% of streambank surfaces are covered by healthy, living vegetation. A variety of vegetation present (e.g., trees, shrubs, understory, or nonwoody macrophytes). Any bare or sparsely vegetated areas are small and evenly dispersed.

1. 100% plant cover on streambank 10
2. > 90% plant cover on streambank 9

B. A variety of vegetation is present and covers 70-90% of streambank surface, but one class of plants is not well-represented. Some open areas with unstable vegetation are present. Disruption evident but not affecting full plant growth potential.

1. 90% plant cover but one class of plants is not well represented 8
2. 80% plant cover with a few barren or thin areas present 7
3. 70% plant cover with a few barren or thin areas present with fewer plant species 6

C. 50-70% of streambank surface covered by vegetation; typically composed of scattered shrubs, grasses, and forbes. Thin or bare spots visible and/or closely cropped vegetation with less than ½ plant stubble height remaining.

1. 70% vegetation cover; typically of shrubs, grasses, and forbes 5
2. 60% vegetation cover; typically of shrubs, grasses, and forbes 4
3. 50% vegetation cover; typically of shrubs, grasses, and forbes 3

D. Less than 50% of streambank surface covered by vegetation; 5 cm/2 in or less in average stubble height remaining. Any shrubs or trees on bank exist as individuals or widely scattered clumps.

1. 40% vegetation cover with many bare spots/rock 2
2. 20% vegetation cover with many bare spots/rock 1
3. No vegetation cover on streambank 0

X. Riparian Vegetation Zone Width [High Gradient (Riffle/Run Prevalent) Streams]

Measures the width of natural vegetation from the edge of the upper streambank out through the floodplain. The riparian vegetative zone serves as a buffer zone to pollutants entering a stream from runoff, controls erosion, and provides stream habitat and nutrient input into the stream.

Look for breaks in the riparian zone, which allow sediment to pass through the zone. When evaluating this parameter, walk around in the riparian area and pay close attention to the amount of natural vegetation present and how deep it extends through the floodplain.

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. Width of riparian zone > 18 meters (approx. 60 ft or more); human activities (e.g. parking lots, roadbeds, timber harvest activity resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have not impacted zone.

1. With no breaks 10
2. With breaks but rare 9

B. Width of riparian zone 12-18 meters (approx. 40-60 ft); human activities (e.g. parking lots, roadbeds, timber harvest activity resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have impacted zone only minimally.

1. With no breaks 8
2. With breaks but rare 7
3. With breaks common 6

C. Width of riparian zone 6-12 meters (approx. 20-40 ft); human activities (e.g. parking lots, roadbeds, timber harvest activity resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have impacted zone a great deal.

1. With no breaks 5
2. With breaks but rare 4
3. With breaks common 3

D. Width of riparian zone < 6 meters (approx. 20ft or less); little or no riparian vegetation due to human activities (e.g. parking lots, roadbeds, timber harvest activity resulting in removal of vegetation, active grazing fields, paths, lawns, or crops).

1. With no breaks 2
2. With breaks but rare 1
3. With breaks common 0

SUPPLEMENTAL INFORMATION: Low Gradient (Glide/Pool Prevalent) Streams

(This supplemental information is to be used as a guideline for determining the habitat scores for each parameter.)

I. Epifaunal Substrate/Available Cover [Low Gradient (Glide/Pool Prevalent) Streams]

Measures availability of actual substrates that are available as refugia, feeding, or sites for spawning & nursery functions for aquatic organisms. A wide variety and/or abundance of submerged structures in the stream provide macroinvertebrates with a large number of niches, thus increasing habitat diversity.

Check habitat types which occur at this site: fallen trees/large woody debris, deep pools, shallow pools, undercut banks, thick root mats, macrophyte beds, sand, and leafpacks.

A. Stable and available habitat(s) expected for stream type make up >70% of reach
   1. 7 habitats common 20
   2. 6 habitat types common, additional habitat types rare 19
   3. 5 habitat types common, additional habitat types rare 18
   4. 4 habitat types common, additional habitat types rare 17
   5. Less than 4 habitat types present 16

B. Stable and available habitat(s) expected for stream type make up >50% of reach
   1. 7 habitats common 15
   2. 6 habitat types common, additional habitat types rare 14
   3. 5 habitat types common, additional habitat types rare 13
   4. 4 habitat types common, additional habitat types rare 12
   5. Less than 4 habitat types present 11

C. Stable and available habitat(s) expected for stream type make up <50% of reach
   a. 7-3 habitats common
   1. 7 habitats common 10
   2. 6 habitat types common, additional habitat types rare 9
   3. 5 habitat types common, additional habitat types rare 8
   4. 4 habitat types common, additional habitat types rare 7
   5. 3 habitat types common, additional habitat types rare 6

   b. 2-0 habitats common
   1. 2 habitat types present, additional habitat types rare 5
   2. 2 habitat types only and common 4
   3. 1 habitat type common, additional habitat types rare 3
   4. 1 habitat type only and common 2
   5. 1 habitat type rare 1
   6. 0 habitat types present 0

II. Pool Substrate Characterization [Low Gradient (Glide/Pool Prevalent) Streams]

Evaluates the type and condition of bottom substrates found in pools. Firmer sediments and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants.

A. Mixture of substrate materials, with gravel and firm sand prevalent; root mats and/or submerged vegetation common. Substrate consists of:

1. Gravel, firm sand, root mats, and submerged vegetation 20
2. Gravel, root mats, and submerged vegetation 19
3. Gravel, root mats or submerged vegetation 18
4. Firm sand, root mats, and submerged vegetation 17
5. Firm sand, root mats or submerged vegetation 16

B. Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and/or submerged vegetation present. Substrate consists of:

1. Firm and soft sand, root mats, and submerged vegetation 15
2. Firm and soft sand, root mats or submerged vegetation 14
3. Soft sand, mud, clay, root mats and/or submerged vegetation common 13
4. Soft sand, mud, clay, root mats and/or submerged vegetation sparse 12
5. Soft sand/mud, soft sand/clay, or clay/mud with sparse root mats and/or submerged vegetation 11

C. All mud or clay or sand bottom; little or no root mat; no submerged vegetation. Substrate consists of:

1. All sand bottom with few root mats 10
2. All mud bottom with few root mats 9
3. All clay bottom with few root mats 8
4. All sand bottom with no root material 7
5. All mud or clay bottom with no root material 6

D. Hard pan clay or bedrock; lacking root mats and vegetation. Substrate consists of:

1. All hard pan clay with sparse root mats or vegetation 4
2. All bedrock with sparse root mats or vegetation 3
3. All hard pan clay with no root mats or vegetation 1
4. All bedrock with no root mats or vegetation 0

III. Pool Variability [Low Gradient (Glide/Pool Prevalent) Streams]

Rates overall mixture of pool types according to size and depth thus accommodating a diverse aquatic community consisting of a variety of species and age classes. In rivers with low sinuosity (few bends) and monotonous pool characteristics, very little instream habitat variety exists to support a diverse community. The four basic types of pools are: (1) large-shallow; (2) large-deep; (3) small-shallow; and, (4) small-deep.

Any pool dimension (e.g., length, width) greater than half the cross-section of the stream is a large pool. Small pools have length and width dimensions less than half the width of the stream. Pools with depths greater than 1.0 m are deep. Shallow pools are less than 1.0 m deep.

Reaeration is defined as the oxygen transfer from the atmosphere to the stream. Reaeration points are any areas where the stream surface is disturbed (e.g., dams, water falling over snags, logs, or other debris).

A. All pool sizes (area and depth) present and mixed.
   1. All sizes evenly mixed and below areas of reaeration 20
   2. All sizes evenly mixed but can be found below and above reaeration areas 18
   3. All sizes evenly mixed not below areas of reaeration 16

B. Majority of pools are large-deep; very few shallow.
   1. Large and small deep pools evenly mixed and all below areas of reaeration 15
   2. Majority of pools are large-deep and below areas of reaeration 14
   3. Large and small deep pools evenly mixed and above and below areas of reareation 13
   4. Majority of pools are large-deep and found above and below areas of reaeration 12
   5. Majority of pools are large-deep and not below areas of reaeration 11

C. Shallow pools are much more prevalent than deep pools.
   1. Large and small shallow pools evenly mixed and all below areas of reaeration 10
   2. Majority of pools are large-shallow and below areas of reaeration 9
   3. Large and small shallow pools evenly mixed and above and below areas of reareation 8
   4. Majority of pools are large-shallow and found above and below areas of reaeration 7
   5. Majority of pools are large-shallow and not below areas of reaeration 6

D. Majority of pools small-shallow or pools absent
   1. Majority of pools are small-shallow and all below areas of reaeration 5
   2. Majority of pools are small-shallow and above and below reaeration areas 3
   3. Majority of pools are small-shallow and all above areas of reaeration 2
   4. Pools absent 0

IV. Sediment Deposition [Low Gradient (Glide/Pool Prevalent) Streams]

Relates to the amount of sediment that has accumulated and the changes that have occurred to the stream bottom as a result of deposition. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increases in size as the channel is diverted toward the outer bank), shoals, or result in the filling of pools. Depositional material comes from both the overall watershed and bank erosion (Barbour and Stribling 1995). The growth or appearance of bars/islands where they did not previously exist is an indication of upstream erosion. High levels of sediment deposition create an unstable and continually changing environment that becomes unsuitable for many organisms (FL DEP 1996).

A. No enlargements of islands/point bars present or less than 20% bottom affected by sand or silt accumulation.

1. No sediment deposition detected; especially in pools 20
2. Less than 20% sediment deposition with accumulation in pools only 18
3. Less than 20% sediment deposition with accumulation in runs and pools 17
4. Less than 20% sediment deposition with few, old, small point bars or islands made up of coarse gravel in stream channel 16

B. 25-50% bottom affected by sand or silt accumulation; slight deposition in pools; some new increase in bar and island formation.

1. 20-30% sediment deposition with gravel and/or sand 15
2. 20-30% sediment deposition with sand and/or silt 14
3. 40-50% sediment deposition with gravel and/or sand 12
4. 40-50% sediment deposition with sand and/or silt 11

C. 50-80% bottom affected with moderate deposition in pools. Number of shallow pools increases. Habitats smothered by sand, silt, and possibly coarse gravel. Deposits of fresh, fine, gravel, sand, and silt observed on old and new point bars, islands, and behind obstructions. Formation of few new bars/islands is evident and old bars are deep and wide; deposition at bends obvious.

1. 60-70% sediment deposition with gravel and/or sand 10
2. 60-70% sediment deposition with sand and/or silt 9
3. 70-80% sediment deposition with gravel and/or sand 7
4. 70-80% sediment deposition with sand and/or silt 6

D. More than 80% bottom affected with heavy deposition from coarse and fine gravel and sand at stream bends, constrictions, and /or pools. Extensive deposits of fine sand and/or silt on old and new bars, islands, and along banks in straight channels. Few pools are present due to siltation. Only larger rocks in riffle areas remain exposed.

1. 80-90% sediment deposition; pools almost absent due to substantial deposition; bottom silt may move with almost any flow above normal 3
2. 90-100% sediment deposition; pools almost absent 1
3. 100% sediment deposition; pools absent due to substantial deposition; bottom silt moves with almost any flow above normal 0

V. Channel Flow Status [Low Gradient (Glide/Pool Prevalent) Streams]

This is the degree to which the channel is filled with water during base or average annual flow periods. This is a seasonal parameter. A decrease in water will wet smaller portions of the streambed, thus decreasing available habitat for aquatic organisms. Use the vegetation line on the lower bank as your reference point to estimate channel flow status.

A channel cross-section may help the investigator(s) estimate what percentage of the available channel is full. Stretch a tape very tight across the channel. Level and secure tape at the base of both lower banks.

A. Water reaches the base of both lower banks and minimal amount of channel substrate is exposed. (100% channel full) 20
   1. > 95% channel is full 18
   2. 90-95% channel is full 16

B. Water fills > 75% of the available channel (or <25% of channel substrate is exposed).
   1. 90% of channel is full 15
   2. 85% of channel is full 13
   3. 80% of channel is full 11

C. Water fills 25-75% of the available channel and/or riffle substrates are mostly exposed.
   1. 75% of channel is full 10
   2. 60-65% of channel is full 9
   3. 50% of channel is full 8
   4. 35-40% of channel is full 7
   5. 25% of channel is full 6

D. Very little water in the channel and mostly present as standing pools.
   1. 20% of channel is full 5
   2. 10% of channel is full 4
   3. < 10% of channel is full 3
   4. Water present as isolated standing pools 1
   5. Channel is dry 0

VI. Channel Alteration [Low Gradient (Glide/Pool Prevalent) Streams]

Measurement of large-scale alteration of instream habitat that affects stream sinuosity and causes scouring. Channel alteration is present when: (1) artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; (2) when the stream is very straight for significant distances; (3) when dams and bridges are present; and (4) when other changes have occurred.

A. Stream follows a normal and natural meandering pattern. Alteration is absent.

1. No evidence of disturbance with bends/runs frequent; bend angles average > 60° _____ 20
2. No evidence of disturbance with bends/runs frequent; bend angles average between 60°- 40° _____ 18
3. No evidence of disturbance with bends/runs frequent; bend angles average < 40° _____ 16

B. Some stream straightening, dredging, artificial embankments, or dams present but NO evidence of recent alteration activities. May have been channelized in the past more than 20 years ago, but mostly recovered.

1. Bridge abutment present but disturbance is more than 20 years old; no other channel disturbance present _____ 15
2. 10% of reach or less has channel disturbance other than bridge _____ 14
3. 20% of reach has channel disturbance _____ 13
4. 30% of reach has channel disturbance _____ 12
5. 40% of reach has channel disturbance more than 20 years old _____ 11

C. Somewhat channelized; 40-80% of the area has been straightened, dredged, or otherwise altered; disturbance may be less than 20 years old.

1. 40% of reach has channel disturbance _____ 10
2. 50% of reach has channel disturbance _____ 9
3. 60% of reach has channel disturbance _____ 8
4. 70% of reach has channel disturbance _____ 7
5. 80% of reach has channel disturbance _____ 6

D. More than 80% of the stream site has been straightened, dredged, or otherwise altered; banks most likely box-cut; instream habitat highly altered.

1. 90% of reach has channel disturbance _____ 5
2. Channel reach 100% disturbed; straight with no artificial embankments _____ 3
3. Channel reach 100% disturbed; straight with artificial embankments _____ 2
4. Channel reach 100% disturbed; straight with natural and artificial embankments _____ 1
5. Banks 100% shored by gabion and/or cement _____ 0

VII. Channel Sinuosity [Low Gradient (Glide/Pool Prevalent) Streams]

Measure of meandering or sinuosity. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding.

Divide the distance between bends by the average width of the stream to estimate the run-to-bend ratio. In general, low sinuosity suggests steeper channel gradient, uniform cross section shapes, limited bank cutting, and limited pools. High sinuosity is associated with lower gradients, asymmetrical cross sections, overhanging banks, and bank pools on the outside curves. Channel sinuosity should be determined over a channel reach long enough to make the value meaningful. **Use a distance of 20 times the bankfull width to determine sinuosity.**

Sinuosity can best be measured using aerial photography.

A. Occurrence of bends relatively frequent.
   1. Run-to-bend ratio = 1-2 ____ 20
   2. Run-to-bend ratio = 3-4 ____ 19
   3. Run-to-bend ratio = 5 ____ 18
   4. Run-to-bend ratio = 6 ____ 17
   5. Run-to-bend ratio = 7 ____ 16

B. Occurrence of bends infrequent.
   1. Run-to-bend ratio = 8 ____ 15
   2. Run-to-bend ratio = 10 ____ 14
   3. Run-to-bend ratio = 11 ____ 13
   4. Run-to-bend ratio = 13 ____ 12
   5. Run-to-bend ratio = 15 ____ 11

C. Occasional bend; variable bottom contours may provide some habitat.
   1. Run-to-bend ratio = 16 ____ 10
   2. Run-to-bend ratio = 18 ____ 9
   3. Run-to-bend ratio = 20 ____ 8
   4. Run-to-bend ratio = 22 ____ 7
   5. Run-to-bend ratio = 24 ____ 6

D. Essentially a straight and uniform depth stream.
   1. Run-to-bend ratio = 25 ____ 4
   2. Run-to-bend ratio = 30 ____ 2
   3. Run-to-bend ratio > 30 ____ 0

VIII. Bank Stability [Low Gradient (Glide/Pool Prevalent) Streams]

Measures the existence of or the potential for detachment of soil from the upper and lower stream banks and its movement into the stream. Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks and are therefore considered unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Reinforcement of banks via rocks, artificial or natural, provides stability.

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. Bank stable; erosion absent or minimal. Side slopes are generally less than 30% and are stable. Bank may be reinforced by rock thus increasing slope >30% while providing stability.

1. No evidence of erosion or bank failure 10
2. Less than 5% bank affected by erosion 9

B. Moderately stable bank; small areas of erosion or bank slumping visible. Most areas are stable with only slight potential for erosion at flood stages. Side slopes up to 40% on one bank. Bank may be reinforced by rock thus increasing slope > 40% while providing stability.

1. 5% bank has erosional areas 8
2. 15% bank has erosional areas 7
3. 30% bank has erosional areas 6

C. Moderately unstable bank; frequency and size of raw areas are such that high water events have eroded some areas of the bank. Medium size areas of erosion or bank slumping visible. Side slopes up to 60% on some of the bank. High erosion potential during floods.

1. 40% - 50% bank has erosional areas 5
2. 50%- 60% bank has erosional areas 4
3. 60% - 70% bank has erosional areas 3

D. Unstable bank; mass erosion and bank failure is evident; erosion and pronounced undercutting present at bends and along some straight channel areas. Side slopes > 60% are common. Many raw areas present and 60-100% bank has erosional scars.

1. 70%- 80% bank has erosional areas 2
2. 80%-90% bank has erosional areas 1
3. > 90% streambank has eroded 0

IX. Bank Vegetative Protection [Low Gradient (Glide/Pool Prevalent) Streams]

Measures the amount of the stream bank that is covered by vegetation. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the existing vegetation, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than banks without vegetative protection or those shored up with concrete or riprap.

Four factors to consider when scoring bank vegetative protection: (1) Is the vegetation native and natural, or planted and introduced?; (2) Is the upperstory, understory, and ground cover vegetation well balanced?; (3) What is the standing crop biomass?; and, (4) During which season are you conducting this assessment?

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. More than 90% streambank surfaces is covered by healthy, living vegetation. A variety of vegetation present (e.g., trees, shrubs, understory, or nonwoody macrophytes). Any bare or sparsely vegetated areas are small and evenly dispersed.

1. 100% plant cover on streambank_____10
2. > 90% plant cover on streambank_____9

B. A variety of vegetation is present and covers 70-90% of streambank surface, but one class of plants is not well-represented. Some open areas with unstable vegetation are present. Disruption evident but not affecting full plant growth potential.

1. 90% plant cover but one class of plants is not well represented_____8
2. 80% plant cover with a few barren or thin areas present_____7
3. 70% plant cover with a few barren or thin areas present with fewer plant species_____6

C. 50-70% of streambank surface covered by vegetation; typically composed of scattered shrubs, grasses, and forbes. Thin or bare spots visible and/or closely cropped vegetation with less than ½ plant stubble height remaining.

1. 70% vegetation cover; typically of shrubs, grasses, and forbes_____5
2. 60% vegetation cover; typically of shrubs, grasses, and forbes_____4
3. 50% vegetation cover; typically of shrubs, grasses, and forbes_____3

D. Less than 50% streambank surface covered by vegetation; 5 cm/2 in or less in average stubble height remaining. Any shrubs or trees on bank exist as individuals or widely scattered clumps.

1. 40% vegetation cover with many bare spots/rock_____2
2. 20% vegetation cover with many bare spots/rock_____1
3. No vegetation cover on streambank_____0

X. Riparian Vegetation Zone Width [Low Gradient (Glide/Pool Prevalent) Streams]

Measures the width of natural vegetation from the edge of the upper streambank out through the floodplain. The riparian vegetative zone serves as a buffer zone to pollutants entering a stream from runoff, controls erosion, and provides stream habitat and nutrient input into the stream.

Look for breaks in the riparian zone, which allow sediment to pass through the zone. When evaluating this parameter, walk around in the riparian area and pay close attention to the amount of natural vegetation present and how deep it extends through the floodplain.

Determine left or right bank by facing downstream. Score left and right banks separately.

Left Bank or Right Bank

A. Width of riparian zone > 18 meters (approx. 60 ft or more); human activities (e.g. parking lots, roadbeds, timber harvest activities resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have not impacted zone.

1. With no breaks 10
2. With breaks but rare 9

B. Width of riparian zone 12-18 meters (approx. 40-60 ft); human activities (e.g. parking lots, roadbeds, timber harvest activities resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have impacted zone only minimally.

1. With no breaks 8
2. With breaks but rare 7
3. With breaks common 6

C. Width of riparian zone 6-12 meters (approx. 20-40 ft); human activities (e.g. parking lots, roadbeds, timber harvest activities resulting in removal of vegetation, active grazing fields, paths, lawns, or crops) have impacted zone a great deal.

1. With no breaks 5
2. With breaks but rare 4
3. With breaks common 3

D. Width of riparian zone < 6 meters (approx. 20ft or less); little or no riparian vegetation due to human activities (e.g. parking lots, roadbeds, timber harvest activities resulting in removal of vegetation, active grazing fields, paths, lawns, or crops).

1. With no breaks 2
2. With breaks but rare 1
3. With breaks common 0

Chapter 3

Georgia Department of Natural Resources

Environmental Protection Division

Macroinvertebrate Biological Assessment
Sorting and Subsampling Procedures

3.1 Macroinvertebrate Laboratory Sorting and Subsampling

Upon receipt by the laboratory samples are logged in on the Macroinvertebrate "Log-in" sheets. Macroinvertebrate Chain-of-Custody (C-O-C) sheets are completed prior to sample delivery to the lab. These forms are filled out in the field and used to keep track of samples through field collection, laboratory processing (subsampling), taxonomic identification, and laboratory quality assurance/quality control. Logging in samples requires recording the sample name/number; project name/number; number of containers per sample; date collected; and, date received by the laboratory.

Equipment/Materials:

- Macroinvertebrate Log in sheets (p. 3A-2)
- Macroinvertebrate Chain-of-Custody (p. 3A-4)
- Macroinvertebrate Level of Effort Subsampling Sheet (p. 3A-10)
- Watch or clock
- 500 µ mesh screen sieve bucket (30 mesh)
- Two standardized gridded screens (595 micron screen, 30 squares, each 36 cm^2) [Caton, L. W. (1991)]
- Two White plastic holding trays for gridded screen [Caton, L. W. (1991)]
- Two 6 cm scoops [Caton, L. W. (1991)]
- Two 36 cm^2 metal dividing frames [Caton, L. W. (1991)] ("cookie cutters")
- Surgical Scissors
- Alcohol proof marker
- White plastic or enamel pan for sorting
- Light source
- Magnification source
- No. 80 sieve (8" diameter sieve)
- Aluminum foil and/or plastic wrap
- Spray bottle (for water to moisten sample)
- Sort residue labels
- Unsorted sample remains labels
- Duplicate subsample sort residue interior/exterior labels
- Complete subsample sort residue interior/exterior labels
- Interior subsampling vial labels
- Forceps
- Specimen vials with caps or stoppers
- 95 percent ethanol for storage of specimens
- Pencils, or India ink pens
- Sorting Efficiency for Benthic Macroinvertebrate Samples Sheet: *Quality Control Check* (p. 2A-12)

Procedures:

To facilitate processing and identification, a randomized 200-organism subsample (±20%) is sorted and preserved separately from the remaining sample. **The target**

number for a subsample is 200 organisms. Since each grid selected must be counted in its entirety there is an allowance of ± 20% that gives a range of 160-240 organisms to allow for counts that go over, or fall under, 200. **It must be stressed however that the target number is 200**; the closer a subsampler can get to 200 the better. Documentation for the level-of-effort, or proportion of sample processed is recorded on the Macroinvertebrate Level of Effort Subsampling Sheet.

1. All primary samples should be sorted in a single laboratory to enhance quality control. Samples should be preserved and stored at room temperature until ready for processing.

2. Thoroughly rinse sample in a sieve bucket (500µ mesh screen) to remove preservative and fine sediment. Any large organic material (whole leaves, twigs, algal or macrophyte mats) not removed in the field should be rinsed, visually inspected, and discarded. Rinse large organic material thoroughly and rub gently over sieve bucket before discarding to dislodge any organisms that may be attached to the material. Inspect material for attached organisms before discarding. If the samples have been preserved in alcohol, it will be necessary to soak the sample contents in water for approximately 15 minutes in order to hydrate the benthic organisms. This will prevent them from floating on the water surface during sorting. If the sample was stored in more than one container, the contents of all containers for a given sample should be combined at this time. Care must be taken to avoid crushing or damaging the invertebrates while rinsing. **Before removing sample from the container(s) verify internal and external labels.** The **original** sample external labels should be peeled off and affixed to the back of the macroinvertebrate C-O-C (p. 3A-4) form once subsampling has been completed.

3. A standardized gridded screen designed by Larry Caton, OR-DEQ (Caton 1991) can be divided into 30 marked squares, each square should be a uniform 36 cm^2. The grids are marked off with an alcohol proof marker in each of the trays. The gridded screen fits into another slightly larger tray (white plastic tray) so that water may be added to the sample to allow for even distribution. Place the gridded screen inside the tray and pour the sample onto the screen. Add enough water to spread the sample evenly over the screen then lift the screen out of the tray, the sample contents will settle onto the screen. Make sure the sample material spreads equally into the corners of the pan. Samples too large to be effectively sorted in a single pan may be thoroughly mixed in a container with some water, and half of the homogenized sample placed in each of two gridded pans. (Specifications for these trays can be found in the article Caton 1991.)

4. Note the presence of large (*i.e.* crayfish, mussels, snails,…etc.) or obviously abundant organisms on the Macroinvertebrate Level of Effort Subsampling Sheet (see p. 3A-10). **Do not remove as part of the subsample unless they occur in one of the randomly selected grids.** Also note the type of sample, collection date, project number, station number, log number, and any comments regarding

the sample (*i.e.*, description of material in sample -- sand, fine organics), preparation time (rinse time of subsample), and sorting time on the Macroinvertebrate Level of Effort Subsampling Sheet.

5. Use a random numbers generator to select numbers that correspond to squares within the gridded pan. When selecting random numbers, take into account the tray number (if one or two trays are used), column number, and row number. (*i.e.* Tray 1, Column 1, Row 5 = 1:1:5)

6. If the sample is large enough to be distributed onto two or more screens, each grid square should have a unique number such that all grid squares in all screens have an equal probability of being selected for sorting. For example, each tray is numbered and the columns and rows of the tray are numbered.

7. A minimum of four grids must be selected for each subsample. Remove all material (organisms and debris) from the four grid squares. Removal of material is accomplished by placing the 36-cm^2 metal dividing frame in the selected grid and using surgical scissors to cut around the edge (being careful not to cut organisms) of the frame. Then, the square is scooped out using the 6-cm scoop. Inspect the grid for any remaining organisms. Any organism that is lying over a line separating two grids is considered to be on the grid containing its head. In instances where it may not be possible to determine the location of the head (worms for instance), the organism is considered to be in the grid containing the majority of its body. Be careful not to disturb the subsampling device between each grid removal. This will aid in minimizing the accidental redistribution of organisms between each subsampling process.

8. **If** the density of organisms is high enough so that many **more** than 240 organisms are contained in the **first four grids** of the initial tray(s) (this is called the first level), transfer the contents of these four previously selected grids to a second gridded pan (this is called second level). Randomly select grids for this second level of sorting as was performed for the first level of sorting. **If** the density of organisms is again high enough that more than 240 organisms are contained in these four grids, transfer the contents of the grids to a third gridded pan (this would be the third level) and continue as before. This process would be continued until the target number ±20% (160-240) is obtained from **at least four grids**. When the targeted subsample amount, ± 20% organisms (160-240), is found, the subsample is completed. If less than 160 organisms are found, continue randomly selecting and sorting grids one at a time until the targeted subsample number, ± 20 percent organisms, is found. **All selected grids must be completely sorted.** Remove and count all the macroinvertebrates from these grids. Only count individuals that can be identified to family level. Exceptions to this rule are Copepods, Nemata, Hirudina, Cladocera, Ostrocoda, and Neoloricata; these should be counted even though they may not be identified down to family level. At times, it may be possible to identify these organisms to a lower taxonomic level. Do not discard any damaged macroinvertebrates. They must be accounted

for (these should be noted on the Macroinvertebrate Bench Sheet during identification).

9. The material is placed into a shallow white pan for sorting and a small amount of water or ethanol is added to facilitate sorting. Large debris can be visually inspected, under magnification and light, for organisms and removed at this time. Sorting of the remaining material pulled from the selected grids should be performed with a dissecting microscope; this ensures that small critical organisms such as Annelids or Chironomids are not missed. Decanting the material from the white pan into a Petri dish in small increments is probably the easiest way to methodically work through the material. Between each subsample, be careful not to disturb the subsample pan (this will cause a redistribution of specimens and could possibly change the probability of selection). It may be necessary to rinse individual squares in a sieve, depending on silt and organic material. Use an 8" diameter No. 80 sieve to prevent loss of very small organisms.

10. The total number of grids for each subsorting level should be noted on the Macroinvertebrate Level of Effort Subsampling Sheet. **Each grid selected for sorting must be <u>completely</u> sorted.**

    If a sorted subsample goes over 240 organisms read 11 and 12:

11. When a sample has been sorted and there are 241-260 individuals the sample is not resorted. This is to allow for the possibility that some organisms may be damaged or miscounted during sorting. The sample can then be identified. If after identification and recounting the subsample still contains 241-260 organisms, then random numbers are used to deselect down to 240 organisms. This is accomplished by starting at the top of the first bench sheet and working to the end of the bench sheets (*i.e.* the first taxa listed is number 1 through the total number of individuals for that taxa).

12. When a sample has been sorted and there are **<u>more than</u>** 260 individuals the sample is resorted. The macroinvertebrates are spread across the standardized gridded screen without debris. Using the random number generator select grids as before. Do not scoop the bare organisms from the grids because this will damage them. Use forceps to collect the organisms from each of the selected grids until the target number +/- 20% is obtained. Use great caution since organisms can be lost or damaged during this process. Use the same subsampling procedure as before, making sure that once a grid has been selected it is **completely** picked for organisms.

    Again, it must be stressed that the target is 200, not 240 organisms.

13. To help prevent desiccation of the sample and damage to specimens cover the sample with aluminum foil or plastic wrap. Periodically moisten the sample with water to prevent the top layer from drying out. At the end of the day, if the

subsample has not been completed, carefully place the gridded screen back in the white plastic tray and cover with aluminum foil or plastic wrap. Use caution so as not to mix debris in squares already removed. **Note:** The "cookie cutter" can be used (along with other items) to hold the spaces of the selected squares.

14. Save the sorted debris residue ("sort residue") in a separate container. Add internal and external labels that include the words "sorted residue" in addition to all prior sample label information. Preserve the sample in 95 percent ethanol and number the container (*i.e.* 1 of 2, 2 of 2). This will be saved for the subsample Quality Control check.

15. Save the remaining unsorted sample debris residue in a separate container labeled "unsorted sample remains". This container should include internal and external labels with the original sample label information. Also include length of storage information on the label. The laboratory or benthic section supervisor determines archival length. However, this material should be saved at least until the sample is completely finished (*i.e.* identified and the subsample Quality Control check performed).

16. Place specimens sorted as the subsample (200-organisms) into glass vials, and preserve in 95 percent ethanol. Organisms from the sorting process will be segregated into separate vials according to the categories: *midges, worms, insects, molluscs, and crustaceans* to be further identified later (See identification procedures). Label the vials inside with the project name, lot name, station identification, stream name, collection date, taxonomic group, sorter, and vial number if more than one vial is used. If more than one vial is required, each should be labeled separately and numbered (*i.e.* 1 of 2, 2 of 2). For convenience in reading the labels inside the vials, insert the labels left-edge first.

17. Once a subsample has been finished the level of effort is calculated.

    1st level = the initial subsample
    2nd level = the resubsample (Performed when over the target number + 20%)
    30 or 60 = Depends on if need 1 or 2 Caton trays for the sort; 30 grids per tray

(1st Level)		**(2nd Level)**	
$\frac{\text{\# Grids selected}}{30 \text{ or } 60}$	x	$\frac{\text{\# Grids selected}}{30 \text{ or } 60}$	= LOE

    The LOE is recorded on the Macroinvertebrate Level of Effort Subsampling Sheet (p. 3A-10).

18. Midges (Chironomidae) should be mounted on slides in cytoseal or other appropriate medium (e.g., Euperal, CMCP-10 or CMC-10). Slides should be labeled with the site identifier, date collected, and a space for the first initial and last name of the taxonomist responsible for identifying them. Slides will be stored in slide boxes. Label the slide boxes with site identifier, stream name, and

date collected. Oligochaetes should also be mounted on slides and should be appropriately labeled as with the midges.

Note: Any entirely sorted reference condition sample that falls 20% below the designated subsample size is excluded from further analyses (less than 160 organisms for 200-organism subsample). **This is not applicable for sampling done as a requirement for watershed assessments.**

3.1a Quality Control/Quality Assurance for Subsampling

3.1a1 % Sorting Efficiency

1. In each lot; which is defined as all the samples for a special study, basin study, ecoregion, subecoregion, bioregion, entire index period, etc.; ten percent of sorted samples are resorted by laboratory Quality Assurance personnel. Ten percent of the "sort residues" are randomly selected per individual sorter. The "sort residue" is completely checked for organisms. The "sort residue" is inspected under a dissecting microscope in the same manner the original material was sorted. Only sort small amounts of the "sort residue" at one time as not to overlook previously missed organisms. The number of organisms collected, comments, and taxa missed are recorded on the Sorting Efficiency for Benthic Macroinvertebrate Samples: *Quality Control Check* laboratory sheet (p. 3A-12).

2. The Quality Assurance personnel will calculate the sorting efficiency for each sample {[(number of organisms originally sorted) / (number of organisms recovered by Quality Control checker + number organisms originally sorted)] x 100 = %}.

3. The sorting efficiency should be ≥ 90% to pass. Subsamples with a sorting efficiency less than 90% are considered to fail. When a subsample fails, two more subsamples are Quality Control checked per individual sorter.

4. Sorters in-training will have 100 percent of their samples checked until sorting efficiency reaches 90%. A minimum of five samples will be Quality Control checked by the QC personnel. The results of the Quality Control check are entered on the Sorting Efficiency for Benthic Macroinvertebrates: *Quality Control Check* sheet (p. 3A-12). Then, once the sorter passes initial training samples, 10% of samples will be Quality Control checked per each lot.

5. Once a site has been Quality Control checked the level of effort is recalculated based on the new total of individuals for that site.

Evaluating consistency (=precision) *– evaluating consistency of the subsampling procedure* (**The following is not required for Watershed Assessments or Protection Plans**.)

3.1a2 Duplicate subsample (A second subsample conducted after the initial subsample)

1. A lot is defined as all the samples for a special study, basin study, ecoregion, subecoregion, bioregion, entire index period, etc. A duplicate subsample will need to be sorted from five percent of all sites (in each designated lot). A duplicate subsample is a second subsample from the same sample. Once the initial subsample has been completed, do not remove the remaining sample material from the subsample trays. The Duplicate sample is pulled from this remaining material.

2. Indicate on the random number sheet at what point the Duplicate subsample is started. Follow the procedure above (under Procedures of Subsampling) for subsampling. Make sure to keep vials from each of the subsamples separate and label the vials appropriately.

3. Complete a separate Macroinvertebrate Level of Effort Subsampling Sheet (see p. 3A-10) and please record that a duplicate subsample was conducted (indicate this on the "purpose" line on page 1).

4. Calculate the LOE as instructed in the subsampling procedure.

3.1a3 Complete sample (sorting of the remainder of the sample after the initial subsample)

1. A lot is defined as all the samples for a special study, basin study, ecoregion, subecoregion, bioregion, entire index period, etc. A complete sample will need to be sorted from five percent of all sites (in each designated lot). A complete sample is performed once the initial subsample has been completed. There is no need to fill out the random number sheet for the complete subsample as it consists of picking the remaining grids that are left in a sample after a normal subsample is conducted. However, make sure to fill out the random number sheet for the initial subsample like usual. Follow the procedure for subsampling outlined under Procedures of Subsampling. Make sure to keep vials separate and label vials appropriately.

2. Once the normal subsample has been conducted, completely subsample the rest of the entire tray. Make sure to keep vials and labeling information separate.

3. Complete a separate Macroinvertebrate Level of Effort Subsampling Sheet. On the Macroinvertebrate Level of Effort Subsampling Sheet please indicate complete subsample (write on the purpose line and on other, after subsample total organisms).

4. Calculate the LOE as instructed in the subsampling procedure.

3.1b Quality Assurance for Evaluating consistency

For duplicate samples and complete samples, include these samples in the total number of samples subsampled by an individual person for a particular lot. For each lot of samples, 10% are to be Quality Control checked. (Follow Quality Control procedure as previously outlined)

Note: Do not repeat the same 5% of samples for both Duplicate and Complete subsampling quality control. Randomly select sites for Duplicate subsamples first, then those samples can not be selected for Complete samples.

3.2 Sample Recharging

If the samples are to be held more than three weeks before sorting is conducted the samples need to be drained, rinsed with water, and recharged with fresh ethanol. If the samples are to be held more than six weeks then they should be recharged twice, once at three weeks after collection and again at six weeks after collection. Recharging removes excess silt, ethanol diluted from water leeching out of the sample debris, foul odors, and any discoloring compounds (such as chlorophyll). The samples should be drained through a number 30 sieve stacked on top of a number 80 sieve; this is to prevent the loss of any small organisms. After draining: fill the sample with water; tighten the lid; gently invert the sample a few times; drain as before; and, repeat once more. Once rinsing has been completed, wash any debris in the sieves back into the sample with a little bit of water then fill the sample bottle with ethanol. After the second recharge event the samples should not need recharging as long as there is a definite alcohol odor when the samples are opened.

3.3 References

Barbour, M. T., J. Gerritsen, B.D. Synder, and J. B. Stribling. 1999. Rapid Bioassessment Protocols for Use in Streams and Wadeable Rivers: Periphyton, Macroinvertebrates and Fish, 2nd Edit. EPA 841-B-99-002. U.S. Environmental Protection Agency; Office of Water; Washington, DC.

Caton, Larry W. 1991. Improved subsampling methods for the EPA "Rapid Bioassessment" benthic protocols. Bulletin of the North American Benthological Society, 8(3): 317-319.

Appendix 3A
Forms

Project Name: _____					**BENTHIC SAMPLE LOG-IN**		Page #: _____			
Sample ID Code	Date Collected	Collected By	Number of Containers	Site #	Stream Name	Date Received by Lab	***Date of Completion/ Initials***			
							Subsampling	Mounting	Identification	Chironomidae ID

Contact Names:			Preservative (Y/N): **Ethanol**	Number of Containers	Type of Analysis Requested								Sample Check-in: Comments:	
Address: Fax #:														
Macroinvertebrate Chain of Custody Project Name:														
Page ____ of _____		Stream Name & Site #:			Recharged #1	Recharged #2	Sorted	QC of Sort Residue (Internal)	QC of Sort Residue (External)	Identification	QC of Identification (Internal)	QC of Identification (External)	Collection Method	Log Number
Date:	Time:	Sample Identification #			*Initials and Date*									
													D-frame net 20 Jab	
													D-frame net 20 Jab	
													D-frame net 20 Jab	
													D-frame net 20 Jab	
													D-frame net 20 Jab	
													D-frame net 20 Jab	

Sampled by (signature):	Date/Time:	Relinquished by Team Leader (signature):	Date/Time:	Relinquished Macro's (signature):	Date/Time:
Team Leader/Received (signature):	Date/Time:	Received by Lab (signature):	Date/Time:	Macro's received for subsampling by (signature):	Date/Time:
Returned Macro's & sort residue (signature):	Date/Time:	Relinquished Macro's (signature):	Date/Time:	Returned Macro's (signature):	Date/Time:
Lab Received (signature):	Date/Time:	Macro's for ID-ing Received by:	Date/Time:	Lab received Macro's (signature):	Date/Time:
	Date/Time:		Date/Time:		Date/Time:
	Date/Time:		Date/Time:		Date/Time:

Contact Names:		Project Name: **Macroinvertebrate Chain of Custody** For **QA/QC (Internal only)** Project Name: _____		**Stream Name & Site #:**	
		Address: Fax #:		Page ____ of _____	
Relinquished Sort Residue (signature):	Date/Time:	Returned Sort Residue (signature):	Date/Time:	Relinquished Macro's For QC (signature):	Date/Time:
Sort Residue QCed By (signature):	Date/Time:	Received Sort Residue (signature):	Date/Time:	Macro's received for ID QC (signature):	Date/Time:
Returned Macro's after QC (signature):	Date/Time:		Date/Time:		Date/Time:
Lab Received Macro's after QC(signature):	Date/Time:		Date/Time:		Date/Time:

Additional Comments:

Contact Names:		Project Name: **Macroinvertebrate Chain of Custody** For **QA/QC (External only)** Project Name: _____		**Stream Name & Site #:**	
		Address: Fax #:		Page ____ of _____	
Relinquished Sort Residue (signature):	Date/Time:	Returned Sort Residue (signature):	Date/Time:	Relinquished Macro's For QC (signature):	Date/Time:
Sort Residue QCed By (signature):	Date/Time:	Received Sort Residue (signature):	Date/Time:	Macro's received for ID QC (signature):	Date/Time:
Returned Macro's after QC (signature):	Date/Time:		Date/Time:		Date/Time:
Lab Received Macro's after QC(signature):	Date/Time:		Date/Time:		Date/Time:

* Always attach a copy of the original Chain-of-Custody form, with the sample identification information with this form.

Additional Comments:

_____

_____

_____

_____

_____

_____

_____

_____

MACROINVERTEBRATE LEVEL OF EFFORT SUBSAMPLING SHEET(Page1)

Project:	**Purpose:**
Site #:	**Stream Name:**
Date Sampled : **By:**	**Target Number of Organisms: 200**
Date Subsampled: **Sorter(s):**	
Total # Organisms:	**Type of Sample (Gear):** D - Frame Net, 20 Jab Method
Total # Vials:	**Total # of Bottles for Sort Residue:**

Indicate the appropriate subsampling levels used and the number of grids picked in each (by marking with an X); and calculate the Level of Effort:

Initial Sample (1st level):

Tray 1 (30 grids)

1	2	3	4	5	6
7	8	9	10	11	12
13	14	15	16	17	18
19	20	21	22	23	24
25	26	27	28	29	30

Tray 2 (30 grids)

1	2	3	4	5	6
7	8	9	10	11	12
13	14	15	16	17	18
19	20	21	22	23	24
25	26	27	28	29	30

Total # organisms for initial sample: _____

_____

Re-subsample (2nd level):

Tray 1 (30 grids)

1	2	3	4	5	6
7	8	9	10	11	12
13	14	15	16	17	18
19	20	21	22	23	24
25	26	27	28	29	30

Tray 2 (30 grids)

1	2	3	4	5	6
7	8	9	10	11	12
13	14	15	16	17	18
19	20	21	22	23	24
25	26	27	28	29	30

Total # organisms for resubsample_____

_____

MACROINVERTEBRATE LEVEL OF EFFORT SUBSAMPLING SHEET (Page2)

Calculation:

(1st Level)		(2nd Level)	
# Grids selected / 30 or 60	x	# Grids selected / 30 or 60	**= LOE**

LOE = _____

Sorting Time:

Preparation Time (Rinse Time): _____

Date	Start Time	End Time	Hours: Minutes	Date	Start Time	End Time	Hours: Minutes	Total Time
Total Time for Subsample:								

Indicate the presence of large or obviously abundant organisms (add additional comments below):

Other comments (i.e., condition of specimens, odor of sample, condition of sample, organic material in sample, sand type, fine organics, etc.):

Sorting Efficiency for Macroinvertebrate Samples: *Quality Control Check*

Project & Reason: _____

Stream Name: _____ **Site #:** _____

Date Macro's sampled: _____ **By:** _____

Original Sorter(s): _____ **Sorting Date:** _____

QC Checker: _____ **QC Check Date:** _____

 # organisms originally sorted ÷ **# organisms recovered by checker + # organisms originally sorted** X 100=_____%

% Sorting Efficiency

90% or greater; sample passes _____

<90%, sample fails; action taken: _____

Missed Macroinvertebrates & Additional Comments: _____

Chapter 4

Georgia Department of Natural Resources

Environmental Protection Division

Macroinvertebrate Biological Assessment Identification Procedures

4.1 Macroinvertebrate Laboratory Identification

Equipment/Materials:

- Mounting Time Sheet (p. 4A-2)
- Macroinvertebrate Bench Sheet (p. 4A-4)
- Macroinvertebrate Identification Time Sheet (p. 4A-6)
- Chironomidae Macroinvertebrate Box Bench Sheet (p. 4A-10)
- Macroinvertebrate Bench Sheet – Chironomidae (p. 4A-8)
- Macroinvertebrate Chain-of-Custody (*i.e.* in Subsampling Procedures, p. 3A-4)
- Macroinvertebrate Log In Sheets (*i.e.* in Subsampling Procedures, p. 3A-2)
- Macroinvertebrate QA/QC Bench Sheet – re-identification (p. 4A-12)
- Macroinvertebrate Taxonomy: *Quality Control Check* form (p. 4A-14)
- Macroinvertebrate Taxonomy Quality Control/Quality Assurance Calculations form (p. 4A-16)
- Forceps
- Specimen vials with caps or stoppers
- Dissecting microscope for organism identification with magnification of 10-40x
- Fiber optic light source
- Compound microscope with phase contrast for identification of mounted organisms
- 95 percent ethanol for storage of specimens
- Appropriate taxonomic keys
- Scalpel and blades
- Slides and cover slips
- Slide boxes
- Internal/External Labels
- Clock or watch

Procedures:

1. Identification is to the lowest practical level (generally genus or species) by a qualified taxonomist using a dissecting microscope for most organisms. Midges (Family Chironomidae), Oligochaeta (*i.e.* Families Tubificidae and Naididae) and Polychchaeta (marine worms) are mounted on slides in an appropriate medium and identified using a compound microscope. If oligochatea and chironomidae are mounted, keep a record of the time necessary for mounting on the Mounting Time Sheet (p. 4A-2).

2. Each taxon found in a sample is recorded and enumerated on the Macroinvertebrate Bench Sheets (p. 4A-4). Any difficulties encountered during identification (*i.e.*, missing gills) are noted on these sheets. (Every specimen in the sample is to be identified to the lowest, practical taxonomic level. If a

specimen cannot be identified because it is too damaged, is an adult (except those groups that have aquatic adult stages), or pupa, they may be placed in a vial and labeled junk or unidentifiable.) The life-stage (adult, larval, or nymphal) for all taxa will be recorded for each site. The larval or nymphal stages will be identified for all taxonomic groups. Adult stages of Coleopterans, Hemipterans, Crustaceans, Mollusks, and Gastropodas will be identified, as well. Pupae, emergent and damaged, will be recorded, but not identified, and not counted in the total numbers. (A target level of identification will be provided on the EPD website as a separate document www.gaepd.org).

3. Only individuals that can be identified to the family level will be counted in the total. Copepods, Nemata, Hirudina, Cladocera, Ostrocoda, Neoloricata, and Planaria are the exceptions and are counted even when they cannot be identified to family level. However, at times it is possible to identify these organisms to a lower taxonomic level. (At this time GAEPD does not identify worms past class Oligochaeta. However, with proper training, worms will be identified in the future. If your lab has a qualified taxonomist, identify Oligochaeta and Polychaeta to lowest practical level.) Mites, ticks, fleas, and springtails are not counted in the sample. Make a note of these organisms on the appropriate bench sheet. However, if more than 10 water fleas are found in a sample, they are counted.

4. On the Macroinvertebrate Bench Sheet, each taxon is followed with a Taxonomic Certainty Rating (TCR) 1 to 5, with 1 being most certain and 5 being uncertain; any rating 2-5 must be explained (*i.e.*, missing legs, gills…etc.).

5. Record the taxonomic references used in the identification process on the Macroinvertebrate Bench Sheets. A list of primary and secondary taxonomic references will be provided on the EPD website as a separate document www.gaepd.org. Also record the time necessary for identification on the Macroinvertebrate Identification Time Sheet (see pg 4A-6).

6. After identification, place specimens into separate glass vials containing 95 percent ethanol according to the categories: worms, insects, molluscs, crustaceans, and junk. Label the vials internally and externally with the serial code, station descriptor (sample ID code and collection date), taxonomic group, sorter, vial number, taxonomist, and date identified (depending on what the laboratory requirements are and sample identification codes may depend on what is needed on the vial label). If more than one vial is needed, each should be labeled separately and numbered (*i.e.* 1 of 2, 2 of 2). For convenience in reading the labels inside the vials, insert the labels left-edge first.

i.e. Vial Label

Stream Name Site Number Date Sampled Sorter Date Sorted Taxonomic Group Taxonomist Name Date Identified

7. Chironomidae Mounting/Labeling

   A. Chironomidae will be mounted on slides and stored in slide boxes.
   B. Slide boxes will be labeled with Stream Name, Site Number, and Box number.
   C. Chironomidae will be mounted two per slide with either CMCP-10 or CMC-10 mounting solution.
   D. During the mounting process, track the time needed for mounting on the Mounting Time Sheet (see pg 4A-2).
   E. Tape will be used on the left hand of the slide to label the slide with the stream name, site identification number, collection date, taxonomist name, identification date, TCR (if not a TCR of 1), and slide number.

i.e. Chironomidae Label

Stream Name Site Number Collection Date Taxonomist Name Date Identified TCR Ranking Slide Number

   F. On the slide, the chironomidae on the left (the first chrionomidae) will be labeled number one and the second will be labeled number two. Use a black sharpie and circle the number.

   G. Label the slides prior to identification.

H. There are corresponding chironomidae forms to record the slide numbers and either number 1 or 2 chironomidae on the form. (Chironomidae Macroinvertebrate Box Bench Sheets p. 4A-10).
I. Once identification of Chironomidae is complete, then fill out the Macroinvertebrate Bench Sheets – Chironomidae (p. 4A-8) using the Chironomidae Macroinvertebrate Box Bench Sheets. This way the Chironomidae will be better organized for data entry.

*Note: The method used for mounting/labeling chironomidae can also be used for oligochaeta.

8. Once identification is completed for a sample, add the totals up and record these on the Macroinvertebrate Bench Sheet. (For sites over the 240 (target number of 200 + 20%), please refer to subsampling procedures (previous section) on how to deselect specimens using random numbers before entering data in the database or ecological condition worksheet for data analysis.)

9. For Watershed Assessments and Protection Plans use the metric spreadsheets and also enter taxa list in the Excel© spreadsheet provided on the EPD website (www.gaepd.org). These taxa lists are to be turned in along with the Watershed Assessment reports and every two years with the Annual Certification of the Watershed Protection Plans implementation reports.

10. (For **EPD employees** only enter the data into Ecological Data Application System (EDAS), an Access© database, for analysis.

4.1a Archive

1. For archival, the specimen vials, grouped by station and date, will be; placed in jars, filled with denatured 95 percent ethanol, and tightly capped. The ethanol level in these jars will be examined biannually and replenished as needed. This should be performed prior to ethanol loss from the specimen vials. A label will be placed on the vial indicating: (1) sample identifier; (2) date identified; (3) denatured 95 percent ethanol used as preservative; (4) stream name; (5) site number; (6) date sampled; (7) number of vials; (8) vial number; and, (9) archival date. **Samples are to be stored in a secure location for a minimum of five years.**

4.1b Voucher Collection

1. A voucher collection must be maintained to help resolve questions regarding the accuracy of taxonomic identifications. This collection should be used for quality assurance/quality control and should be identified by another laboratory for taxonomic proficiency. These specimens should be properly labeled, preserved, and stored in the laboratory for future reference. Specimen labels should include the name(s) of the verifying person(s). Senior taxonomists must QA/QC 10% of the sample identifications. (Voucher collections are not required for Watershed assessments and Protection Plans, but are very beneficial for any laboratory conducting identifications.)

4.2 Laboratory Quality Control/Quality Assurance

1. After the laboratory processing is completed for a given sample, all sieves, pans, petri dishes, trays…etc., that have come in contact with the sample will be rinsed thoroughly, examined carefully, and picked free of organisms or debris

2. Information on samples completed (through the taxonomic process) will be recorded on the Macroinvertebrate Log In Sheets and Macroinvertebrate Chain of Custody form to track the progress of each sample within the sample lot (see Subsampling Procedures). The tracking of each sample will be updated as each step is completed (*i.e.*, subsampling and sorting, mounting of midges and worms, and taxonomy).

3. A library of taxonomic literature is essential in aiding identification of specimens. References that should be available in the laboratory are provided on the EPD website (www.gaepd.org).

4. Samples are to be signed in and out using the Macroinvertebrate Chain-of Custody form. The mounting/identification date and name of the taxonomist should be filled in on the Macroinvertebrate Log In Sheet. Examples of these forms can be found in the subsampling section (previous section).

4.2a Taxonomic Quality Control/Quality Assurance:

All personnel performing macroinvertebrate identifications will be trained in a consistent manner. The taxa identifications will be standardized to ensure accuracy.

1. In each lot; which is defined as all the samples for a special study, basin study, ecoregion, subecoregion, bioregion, entire index period…etc., ten percent of the identified samples in each lot should be re-identified by laboratory QC personnel or a qualified co-worker.

2. Randomly select ten percent of the identified samples per individual sorter. The identified sample is completely re-identified ignoring the original identification of the original identifier, so as not to bias ones opinion. The identifications are recorded on Macroinvertebrate QA/QC Bench Sheets (p. 4A-12).

3. Identifiers in training will have all samples 100 percent QA/QC checked until each receives 90% pass for % Correctly Identified and 10% for Precision of counts & % difference in enumeration (see calculations below) for five samples consecutively. After an identifier in training passes five samples, then ten percent of the identified samples in each lot should be re-identified by laboratory QC personnel or a qualified co-worker.

4. Once a site has been QC checked, the **level of effort** is **recalculated** based on the new total of organisms for that site.

5. Ten percent of sites, for a lot, should be sent off to be QA/QC checked by a second laboratory.

4.2b Taxonomic Quality Control/Quality Assurance Calculations

First Calculation: Presence/Absence (ADEM 1996):

1. After re-identification, the Macroinvertebrate Bench Sheets and Macroinvertebrate QA/QC Bench Sheets are compared for missed taxa and recorded on the Macroinvertebrate Taxonomy: Quality Control Check form (p. 4A-14).

2. This calculation will compare taxa on presence/absence basis. Percentages of comparability between the two data sets will be calculated based on the number of taxa correctly identified divided by the total number of taxa.

   % Correctly Identified = [(Total Taxa - Incorrect Taxa) / Total Taxa] *100

   Results of the taxonomic verifications should be logged on the Macroinvertebrate Taxonomy: Quality Control Check form and the calculations are logged on the

Macroinvertebrate Taxonomy Quality Assurance/Quality Control Calculations Form (p. 4A-16).

3. The identification efficiency must be ≥ 90% to pass. Identifications less than 90% are considered to fail. When a sample fails, two more identification samples must be QC checked for the failing taxonomist.

Second Calculation: Precision of counts (Stribling *et al*. 2003):

1. One aspect of taxonomic data quality is the final count of specimens for each taxon in the sample; this is not the rough count during subsampling, but the final count after identification.

2. Precision of counts is determined by calculating % difference in enumeration (PDE):

$$PDE = [\,|\,n1 - n2\,|\,/\,(n1 + n2)\,] * 100$$

PDE = % difference in enumeration
$n1$ = number of specimens counted in a sample by the 1st taxonomist or laboratory
$n2$ = number of specimens counted in a sample by the 2nd taxonomist or laboratory

Results of the count verifications should be logged on the Macroinvertebrate Taxonomy: Quality Control Check form and the calculations should be logged on the Macroinvertebrate Taxonomy Quality Assurance/Quality Control Calculations form.

3. The purpose of this calculation is to emphasize substantial difference in samples where counts differ substantially and to determine the reason or reasons for the miscounts. Enumeration differences will affect the calculation of taxonomic precision. Some examples of enumeration errors are counting a mollusk shell when it is not occupied by a specimen, counting worms without their heads, etc.

4. The count efficiency must be ≤ 10% to pass. Counts greater than 10% are considered to fail. When a sample fails, two more identification samples must be QC checked for the failing taxonomist.

Third Calculation: % Taxonomic Disagreement (Stribling et al. 2003):

1. The number of agreements between two taxonomists or laboratories are compared to determine the % Taxonomic Disagreement (PTD).

2. PTD calculation:

$$PTD = [1 - (comp_{pos}/N)] * 100$$

PTD = % taxonomic disagreement
$comp_{pos}$ = the number of agreements (positive comparisons)
N = total number specimens in the larger of the 2 counts

3. If one taxonomist identifies the specimen at species level, but the other leaves the specimen at genus level this will be scored as an agreement. However if one taxonomist identifies the specimen at genus level and the re-identification is at family, this would not be counted as agreement because one identification met the target and the other did not.

4. The lower the percent taxonomic disagreement value, the greater the overall taxonomic precision of a sample. When a large number of specimens are in disagreement the samples may need to be isolated and evaluated further for corrective re-identifications.

5. The percent taxonomic disagreement efficiency should be ≤ 10% to pass. Percent taxonomic disagreements greater than 10% are considered to fail. When a sample fails, two more identification samples are QC checked.

4.3 References

Alabama Department of Environmental Management. 1996. Standard operating procedures and quality assurance manual for biological monitoring: Volume II-Freshwater macroinvertebrate biological monitoring. Field Operations Division, Montgomery, Alabama.

Stribling, James B., Stephen, R. Moulton II., Lester, Gary, T. 2003. Determining the quality of taxonomic data. Journal of North American Benthological Society. 22(4):621-631

Appendix 4A
Forms

MOUNTING TIME SHEET

Project: **Purpose:**

Site #: **Stream Name:**

Date Sampled : **By:**

 # Chironomidae Slides: **Chironomidae Slide Box #:**

 # Oligochaete Slides: **Oligochaete Slide Box #:**

Chironomidae Mounting Time:

Start Time	End Time	Hours: Minutes	Start Time	End Time	Hours: Minutes	Total Time
Total Time for Chironomidae Mounting:						

Oligochaete Mounting Time:

Start Time	End Time	Hours: Minutes	Start Time	End Time	Hours: Minutes	Total Time
Total Time for Oligochaete Mounting:						

Comments (i.e., condition of specimens, etc.):

PROJECT: ____________________

Macroinvertebrate Bench Sheet

Notes about this site: ____________________ Total # of Vials ____________________
____________________ Total # of Individuals for site ____________
____________________ Total # of Individuals for page ____________
Total # of Taxa for site ____________
Stream Name: ____________________ Site Number ____________ Chironomidae Box # ________
Taxonomist: ____________________ # of Taxa per page ____________ **Page** ________ of ________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

ORDER ____________ **Family** ____________ **Subfamily** ____________ **Genus** ____________ **Species** ____________ # in VC ______
Total Number ____________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** ____________ Life Stage **A L N or N/A** VC ______
Notes ____________________

Signature ____________________ **Completion Date** ____________ **Verified (QC)** ____________ **Date** __________

MACROINVERTEBRATE IDENTIFICATION TIME SHEET

Project: **Purpose:**

Site #: **Stream Name:**

Date Sampled : **By:**

Date Subsampled: **Sorter(s):**

Total # Organisms: **Type of Sample (Gear):** D - Frame Net, 20 Jab Method

Total # Vials:: **Identification By:**

Total Identification Time:

Start Time	End Time	Hours: Minutes	Start Time	End Time	Hours: Minutes	Total Time
					Total Time for Identification:	

Comments (i.e., condition of specimens, etc.):

PROJECT: ______________________________________

MACROINVERTEBRATE BENCH SHEET – ***Chironomidae***

*Notes about this site:*____ Total # of Vials_____
____ Total # of Individuals for site ________
____ Total # of Individuals for page_________
Total # of Taxa for site_____
Stream Name: ____________________________ Site Number __________________ # of Taxa per page_______ Chironomidae Box # ____
Taxonomist: ____ 1st 100 2nd 100 3rd 100 or 200 total **Page** _____ of _____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

ORDER *Diptera* **Family** *Chironomidae* Subfamily________________ Genus __________________ Species __________________ # in VC____
Total Number ____________________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date ID from** *Epler 2001*___ Life Stage **L** VC_________
Notes ____

Signature ___________________________ Completion Date __________ Verified (QC Mgr) ____________________ Date __________

CHIRONOMIDAE MACROINVERTEBRATE BOX BENCH SHEET

PROJECT:_____

*Notes about this site:*_____ Total # of Vials_____

_____ Total # of Individuals for site_____

_____ Total # of Individuals for page_____

Stream Name: _____Site Number _____ # of Taxa per page_____ Chironomidae Box # ____

Taxonomist: _____ Total # of Taxa for site_____ Page _____ of _____

Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species
Slide Number	Box Number	1.) Subfamily	Genus	Species	2.) Subfamily	Genus	Species

Quality Assurance/Quality Control Macroinvertebrate Bench Sheet – re-identification

PROJECT: ____________________________

Notes about this site: ____ Total # of Vials______________________
____ Total # of Individuals for site ___________
____ Total # of Individuals for page___________
Total # of Taxa for site__________
Stream Name: ____ Site Number _________________________ Chironomidae Box # ________
Taxonomist: ____ # of Taxa per page_______________ **Page** _________ of ________

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

ORDER ____________**Family** ______________**Subfamily** ____________ **Genus** _________________ **Species** ______________ # in VC_____
Total Number _______________ TCR 1 2 3 4 5 (Explain 2-5 in notes) **Book & Date** __________________ Life Stage **A L N or N/A** VC_____
Notes ____

Signature ______________________________ **Completion Date** _____________ **Verified (QC)** __________________ **Date** ___________

Macroinvertebrate Taxonomy: Quality Control Check (Page 1)

For Internal re-identification

Project:

Stream Name: _____ Site #: _____

Date Stream sampled: _____ By: _____

Original Sorter(s): _____ Sorting Date: _____

Subsample QC? Yes or No Subsample QC By Date: _____

Original Taxonomist: _____ Date of Id: _____

Taxonomy QC by: _____ Date of QC: _____

Count Errors:					
Taxa	**# Counted**	**Correct #**	**Taxa**	**# Counted**	**Correct #**
Total # of Count Errors					

Macroinvertebrate Taxonomy: Quality Control Check (Page 2)

Misidentification Errors:

Wrong Identification	Correct Identification	#	Wrong Identification	Correct Identification	#

Total # of Misidentification Errors	

Additional comments____

Macroinvertebrate Taxonomy Quality Assurance/Quality Control Calculations
For Internal re-identification

Project: _____

Stream Name: _____ **Site #:** _____

Date Stream sampled: _____ **By:** _____

Original Sorter(s): _____ **Sorting Date:** _____

Subsample QC? Yes or No **QC By/Date:** _____

Original Taxonomist: _____ **Date of Id:** _____

Taxonomy QC by: _____ **Date of QC**: _____

First Calculation: Presence/Absence

% Correctly Identified = [(Total Taxa - Incorrect Taxa) / Total Taxa] *100

%Correctly Identified = _____

90% or greater; sample passes _____

<90%, sample fails; action taken: _____
_____
_____
_____
_____

Second Calculation: Precision of counts

PDE = [$|n1 - n2|$ / $(n1 + n2)$] * 100

PDE = _____

PDE = % difference in enumeration
$n1$ = number of specimens counted in a sample by the 1st taxonomist or laboratory
$n2$ = number of specimens counted in a sample by the 2nd taxonomist or laboratory

10% or less; sample passes _____

>10%, sample fails; action taken:
_____
_____
_____
_____

Macroinvertebrate Taxonomy Quality Assurance/Quality Control Calculations

Third Calculation: % Taxonomic Disagreement

$PTD = [1 – (comp_{pos}/N)] * 100]$

PTD = _____

PTD = % taxonomic disagreement
$comp_{pos}$ = the number of agreements (positive comparisons)
N = total number specimens in the larger of the 2 counts

10% or less; sample passes _____

>10%, sample fails; action taken :
_____
_____
_____
_____

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Chapter 5

Georgia Department of Natural Resources

Environmental Protection Division

Macroinvertebrate Biological Assessment
Data Analysis Procedures

5.1 Ecoregional Divisions in Georgia

A critical element in the development of EPA approved bioassessment methodology is the selection of unimpaired/least impacted "reference" sites. Variations among natural surface waters throughout the country prevent the establishment of a single reference condition nationwide. Most states are too heterogeneous for single reference sites. It would be unrealistic to expect the biotic community of a South Georgia blackwater stream to be the same as a north Georgia trout stream. Therefore, some type of framework should be utilized or to standardize the recording of the complexity of biological and habitat information depicting reference conditions within the state.

Glenn Griffith and associates of the USDA – Natural Resources Conservation Service, developed the following information. Their project was financed in part through a grant from the US EPA under the provisions of Section 319(h) of the Federal Water Pollution Control Act as amended. A Level III and IV Ecoregions map of Georgia (Figure 5-1, p. 5-10), indicating the location of the subecoregions, follows the descriptions of the ecoregion and subecoregions. The map and information that follows is cited as:

> *Griffith, G.E., J.M. Omernik, T. Foster, and J.A. Comstock. 2001. Ecoregions of Georgia. U.S. Environmental Protection Agency, National Health and Environmental Effects Research Laboratory, Corvallis, OR.*

The following are descriptions of Georgia's ecoregions and subecoregions. The GA EPD multi-metric index uses subecoregions to determine the final index score and stream health rating. Table 5-1 is included on page 5-9 and lists all subecoregions that have metric indices developed for the state of Georgia.

5.1a Piedmont Ecoregion (45)

Considered the non-mountainous portion of the old Appalachian Highland by physiographers, the northeast-southwest trending Piedmont ecoregion comprises a transitional area between the mostly mountainous ecoregions of the Appalachians to the northwest, and the relatively flat coastal plain to the southeast. It is a complex mosaic of Precambrian and Paleozoic metamorphic and igneous rocks with moderately dissected irregular plains and some hills. The soils tend to consist of a finer texture than is commonly found in coastal plain regions. Once largely cultivated, much of this region has reverted to pine and hardwood woodlands, and, more recently, spreading urban- and suburbanization.

45a. The **Southern Inner Piedmont** contains more areas of higher elevation and greater relief than 45b, but is generally lower, has less relief, and contains different types of rocks and soils than 45d. The rolling to hilly, well-dissected upland contains mostly schist, gneiss, and granite bedrock. In the western portion, west of Atlanta, mica schist and micaceous saprolite are typical. To the east, biotite gneiss is more common. The region is now mostly forested, with major forest types of oak-pine and oak-hickory, and less loblolly-shortleaf pine forest than 45b. Open areas consist primarily of pasture,

although some small areas of cropland do exist. Hay, cattle, and poultry are the main agricultural products. Urban/suburban land cover has greatly increased within this ecoregion over the past twenty years.

45b. The **Southern Outer Piedmont** consists of lower elevations, less relief, and less precipitation than 45a. Loblolly-shortleaf pine is the major forest type, with less oak-hickory and oak-pine than in 45a. Gneiss, schist, and granite are the dominant rock types, covered with deep saprolite and mostly red, clayey subsoils. The majority of soils are Kanhapludults. The southern boundary of the ecoregion occurs at the Fall Line, where unconsolidated coastal plain sediments are deposited over the Piedmont metamorphic and igneous rocks.

45c. The **Carolina Slate Belt** originates in the Carolinas, and extends into Georgia. The mineral-rich metavolcanic and metasedimentary rocks with slatey cleavage are finer-grained and less metamorphosed than most Piedmont regions. It tends to be less rugged; less dissected, contains wider valleys than other Piedmont areas, and generally contains more silt and silt-clay soils.

45d. The **Talladega Upland** of the Georgia Piedmont consists of dissected hills and tablelands that are mostly forested and exist at higher elevations than 45a and 45b. The geology is distinctive, consisting of mostly phyllite, quartzite, slate, metasiltstone, and metaconglomerate. This represents a contrast to the high-grade metamorphic and intrusive igneous rocks of 45a and 45b. The climate of 45d is slightly cooler and wetter than the other ecoregions (45a, b, c) of the Georgia Piedmont. Oak-hickory-pine is the natural vegetation type.

45h. The **Pine Mountain Ridges**, a small, narrow region in the southwest portion of the Georgia Piedmont, contains quartzite-capped, steep-sloped ridges that rise 300-400 feet above the Piedmont surface to elevations over 1300 feet. Pine Mountain and Oak Mountain are the primary linear ridges trending southwest to northeast, and several other smaller ridges and mountains between these, including Bull Trail Mountain, Indian Grave Mountain, Salter Mountain, and Huckleberry Pinnacle, add to the region's more mountainous appearance. The Flint River has cut narrow, steep gorges, up to 400 feet deep, through the ridges. Streams in this region are often of higher gradient than surrounding areas of 45b, and contain more rock and gravel substrates.

5.1b Southeastern Plains Ecoregion (65)

These irregular plains with broad interstream areas contain a mosaic of cropland, pasture, woodland, and forest. Natural vegetation is composed primarily of oak-hickory-pine and Southern mixed forest. The Cretaceous or Tertiary-age sands, silts, and clays of the region contrast geologically with the Paleozoic limestone, shale and sandstone of ecoregions 67 and 68, and also with the even older metamorphic and igneous rocks of the Piedmont (45). Elevations and relief are greater than in the Southern Coastal Plain (75), but generally less than in much of the Piedmont. Streams in this area are relatively low gradient and are sandy-bottomed.

65c. The **Sand Hills** of Georgia form a narrow, rolling to hilly, highly dissected, coastal plain belt stretching across the state from Augusta to Columbus. The region is composed primarily of Cretaceous and some Eocene-age marine sands, as well as clays deposited over the crystalline and metamorphic rocks of the Piedmont (45). Many of the droughty, low-nutrient soils formed in thick beds of sand. However, soils in some areas consist of more loamy and clayey horizons. On the drier sites, turkey oak and longleaf pine are dominant, while shortleaf-loblolly pine forests and oak-pine type forests are common throughout the region.

65d. The dissected irregular plains and low, gently rolling hills of the **Southern Hilly Gulf Coastal Plain** developed over diverse bands of sand, clay, and marl formations. The heterogeneous region has a mix of clayey, loamy, and sandy soils. It has more rolling topography, higher elevations, and more relief than 65g and 65k, and streams exist on a higher gradient. The natural vegetation is mostly oak-hickory-pine forest. A transition into southern mixed forest begins to the south. Land cover is comprised primarily of mixed forest and woodland, pine plantations, and small areas of pasture and cropland.

65g. The **Dougherty Plain** is mostly flat to gently rolling and is influenced by the near-surface limestone. The karst topography contains sinkholes, springs, and consists of fewer streams in the flatter part of the plain. The northwestern boundary is gradational, with slopes becoming more gentle and lower relief found towards the center of the subecoregion. On the southeast, the Pelham escarpment marks the boundary with the Tifton Upland (65h). Landcover is primarily cropland and pasture, with small areas of mixed forest. Crops such as peanuts and pecans are common, and cotton production has increased dramatically in recent years. Natural forest cover consists of pines/hardwood mix, including; longleaf pine, red oaks, and hickories. Many shallow, flat-bottomed depressions are scattered throughout the subecoregion. These are a result of the dissolution of the underlying limestone (carbonate rock) by active groundwater. The wetter, poorly drained depressions contain blackgum, sweetgum, water oak, and a few pines and cypress. Many of the limesink ponds and marshes act as biological oases in the mostly agricultural landscape.

65h. The **Tifton Upland** of Georgia has more rolling, hilly topography than is found in 65g or 75e. It consists of a mosaic of agricultural land, pasture, and some mixed pine/hardwood forests. Soils are well drained, brownish, loamy, and are often found to contain iron-rich or plinthic layers. They support crops of cotton, peanuts, soybeans, and corn. On the west side of the region, the Pelham Escarpment has bluffs and deep ravines with cool microclimates that support several rare plants and animals, as well as species with more northern affinities.

65k. In contrast to the more forested Sand Hills (65c) that formed mostly on light-colored Cretaceous sands, the **Coastal Plain Red Uplands** formed on reddish Eocene sand and clay formations. Soils, for the most part, are well drained, have a brown or reddish brown, loamy or sandy surface layer, and have red subsoils. The majority of the area is cropland or pasture, with some woodland present on steeper slopes. The Fort

Valley Plateau falls within this subecoregion, a relatively small agricultural area with less relief, flat-topped interfluves, and less dissection than found in other parts of the 65k.

65l. Also called the Vidalia Upland in Georgia, the **Atlantic Southern Loam Plains** is generally lower, flatter, more gently rolling than 65k, has more cropland, and, consists of finer-textured soils than are found in 75f. Similar to 65h, it has an abundance of the agriculturally important Tifton soils, but the region also contains forested areas that are more sloping or are low, flat, and poorly drained. Parallel to some of the major stream courses are some excessively drained, dunal sand ridges with xeric vegetation such as longleaf pine/turkey oak forests, and some distinctive evergreen shrubs, such as rosemary and woody mints.

65o. The **Tallahassee Hills/Valdosta Limesink** ecoregion combines two slightly different areas, both influenced by underlying limestone. The Floridan aquifer is thinly confined in this region, and streams are often intermittent or in parts flow underground in the karst landscape. In the west, the Tallahassee Hills portion has rolling, hilly topography that is more forested than 65h. Clayey sands weathered to a thick red residual soil are typical. Relief decreases towards the east, and the Valdosta Limesink area has more solution basins with ponds, lakes, and swampy depressions, as well as areas with more cropland. The soils are typically brownish. Mixed hardwoods and pine are found on the clayhill upland soils, while longleaf pine/xerophytic oak types occur on the sandy, well-drained areas.

5.1c Blue Ridge Ecoregion (66)

The Blue Ridge varies from narrow ridges to hilly plateaus to more massive mountainous areas with high peaks. The mostly forested slopes, high-gradient, cool, clear streams, and rugged terrain occur on a mix of igneous, metamorphic, and sedimentary geology. Annual precipitation of over 80 inches can occur on the well-exposed high peaks.

The southern Blue Ridge is one of the richest centers of biodiversity in the eastern U.S. It is one of the most floristically diverse ecoregions, including; shrub, grass, heath balds, hemlock, cove hardwoods, and oak-pine communities. Black bear, whitetail deer, wild boar, turkey, grouse, songbirds, many species of amphibians and reptiles, thousands of species of invertebrates, and a variety of small mammals are found here.

66d. The **Southern Crystalline Ridges and Mountains** contain the highest and wettest mountains in Georgia. These occur primarily on Precambrian-age igneous and high-grade metamorphic rocks. The common crystalline rock types include gneiss, schist, and quartzite, covered by well-drained, acidic, brownish, loamy soils. Some mafic and ultramafic rocks also occur here, producing more basic soils. Elevations of this rough, dissected region range from 1800 to 4000 feet, with Brasstown Bald Mountain reaching 4,784 feet. Although there are a few small areas of pasture and apple orchards, the region is predominantly covered by forest.

66g. **The Southern Metasedimentary Mountains** contain rocks that are generally not as strongly metamorphosed as the gneisses and schists of 66d. The geologic materials are mostly late Pre-Cambrian and include slate, conglomerate, phyllite, metagraywacke, metasiltstone, metasandstone, and quartzite, with some schist and gneiss. Although the highest peaks are lower than in 66d, and parts of the region have more open, low hills, there are some isolated masses of rugged mountains, such as the biologically diverse Cohutta Mountains, Rich Mountains, and Fort Mountain.

66j. The **Broad Basins** is drier, and has lower elevations and less relief than the more mountainous Blue Ridge regions (66g, 66d). It also has less bouldery colluvium than those two surrounding regions and more saprolite. The primary soil type consists of deep, well drained, loamy to clayey Ultisols. Although this rolling foothills region is mostly forested, it has more pasture than adjacent regions, and some narrow areas of row crops and truck crops on terraces and floodplains. Much of the pasture and corn crops support local cattle, hog, or poultry operations.

5.1d Ridge and Valley Ecoregion (67)

Sometimes called the Great Valley in Georgia, this is a relatively low-lying region between the Blue Ridge (66) to the east and the Southwestern Appalachians (68) on the west. As a result of extreme folding and faulting events, the roughly parallel ridges and valleys come in a variety of widths, heights, and geologic materials, including limestone, dolomite, shale, siltstone, sandstone, chert, mudstone, and marble. Springs and caves are relatively numerous. Land cover is mixed and present-day forests cover about 50% of the region. Forested ridges, and valleys containing pasture and cropland, are typical in many parts of ecoregion 67. Its diverse habitats contain many unique terrestrial and aquatic flora and fauna.

67f. The **Southern Limestone/Dolomite Valleys and Low Rolling Hills** form a heterogeneous region composed predominantly of limestone and cherty dolomite. Landforms are primarily composed of undulating valleys and rounded ridges and hills containing many caves and springs. Soils vary in their productivity, and land cover includes: oak-hickory and oak-pine forests; pasture; intensive agriculture; and, urban and industrial area. Along the Coosa River floodplain, biota more typical of coastal plain regions can be found due to the valley and riverine connection to ecoregion 65 in Alabama. **& 67i.** The **Southern Dissected Ridges and Knobs** contain crenulated, broken, and hummocky ridges. Although shale is common, there is a mixture and interbedding of geologic materials, including cherts; siltstone, sandstone, and quartzose limestone. Oak forests and pine forests are typical for the higher elevations of the ridges, with oak-hickory and a number of more mesic forest species on the lower slopes, knobs, and draws. (**67f & i** were combined to form one subecoregion for the GA EPD multi-metric index.)

67g. The **Southern Shale Valleys** consist of undulating to rolling valleys and some low, rounded hills and knobs that are dominated by shale. The soils were formed from materials weathered from shale, shaly limestone, and clayey sediments, and tend to be

deep, acidic, moderately well drained, and slowly permeable. The steeper slopes are used for pasture or have reverted to brush and mixed forest land. Small fields of hay, corn, soybeans, tobacco, and garden crops are grown on the foot slopes and bottom land.

67h. The **Southern Sandstone Ridges** encompass the major sandstone ridges, but these ridges also have areas of shale, siltstone, and conglomerate. The steep, forested ridges tend to have narrow crests, and the soils are typically stony, sandy, and of low fertility. The chemistry of streams flowing down the ridges can vary greatly depending on the geologic material. In Georgia, most of the sandstone ridges are relatively narrow. Oak-hickory-pine forests are the dominant land cover.

5.1e Southwestern Appalachians Ecoregion (68)

These low mountains contain a mosaic of forest and woodland with some cropland and pasture. The eastern boundary of the ecoregion, along the abrupt escarpment next to the Ridge and Valley (67), is relatively smooth and only slightly notched by small, eastward flowing stream drainages. The mixed mesophytic forest is restricted mostly to the deeper ravines and escarpment slopes. The summit, or tableland, forests are dominated by mixed oaks with shortleaf pine.

68c. The **Plateau Escarpment** is characterized by steep, forested slopes and high velocity, high gradient streams. Local relief is often 1000 feet or more. The geologic strata include; Mississippian-age limestone, sandstone, shale, and siltstone; and, Pennsylvanian-age shale, siltstone, sandstone, and conglomerate. Streams have cut down into the limestone, but the gorge talus slopes are composed of colluvium with huge, angular, slabby blocks of sandstone. Vegetation community types in the ravines and gorges include; mixed oak and chestnut oak on the upper slopes, more mesic forests on the middle and lower slopes (beech-yellow poplar, sugar maple-basswood-ash-buckeye), some rare hemlock along rocky streamsides, and, river birch along floodplain terraces. **& 68d.** The **Southern Table Plateaus** include Sand Mountain and Lookout Mountain in northwest Georgia. Major characteristics include: 1. Pennsylvanian-age sandstone caprock; 2. shale layers; and, 3. coal-bearing strata. This ecoregion is lower in elevation, has a slightly warmer climate, and has more agriculture than the Cumberland Plateau (68a) to the north. Although the Georgia portion is mostly forested, primarily with mixed oak and oak-hickory communities, elevations decrease to the southwest in Alabama where there is more cropland and pasture. The plateau surface is less dissected with lower relief compared to the Plateau Escarpment (68c), and it is slightly cooler with more precipitation than in the nearby lower elevations of 67f. (**68 c & d** were combined to form one subecoregion for the GA EPD multi-metric index, due to the small size of these subecoregions.)

5.1f Southern Coastal Plain Ecoregion (75)

From a national perspective, the ecoregion appears to be mostly flat plains. However, it is a heterogeneous region also containing barrier islands, coastal lagoons, marshes, and swampy lowlands along Georgia's coast and the Atlantic coast. This ecoregion is

generally lower in elevation with less relief and wetter soils than ecoregion 65. Once covered by a variety of forest communities that included longleaf pine, slash pine, pond pine, beech, sweetgum, southern magnolia, white oak, and laurel oak, land cover in the region now consists primarily of slash and loblolly pine with oak-gum-cypress forest existing in some low lying areas. Citrus groves, pasture for beef cattle, and urban areas are also present.

75e. The **Okefenokee Plains** consist of flat plains and low terraces developed on Pleistocene-Pliocene sands and gravels. These plains have slightly higher elevations and less standing water than 75g (Okefenokee Swamps) although there are numerous swamps and bays. There are some highly acidic softwater lakes; with most containing low clarity, darkly colored water, but the color is variable depending on rainfall. Soils in the subecoregion are somewhat poorly to poorly drained. The subecoregion is predominantly covered by coniferous forest and young pine plantations. Areas of forested wetland are also present.

75f. The **Sea Island Flatwoods** are poorly-drained, flat plains with lower elevations and less dissection than 65l. Pleistocene sea levels rose and fell several times creating different terraces and shoreline deposits. Spodosols and other wet soils are common, although small areas of better-drained soils add some ecological diversity. Trail Ridge is located in this subecoregion, forming the boundary with 75g. Loblolly and slash pine plantations cover much of the region. Water oak, willow oak, sweetgum, blackgum and cypress occur in wet areas.

75g. The **Okefenokee Swamps** did have enough streams of the appropriate size and could have been evaluated (Olson 2002). However, because the subecoregion exists almost entirely within a national wildlife refuge, there was no need to find reference sites for determining the amount of human impact on streams within the refuge. Also, because the swamp is a unique landscape within Georgia, reference sites are not needed for comparison to other streams in different subecoregions. For these reasons, this sub-ecoregion was excluded from the study area.

75h. The **Bacon Terraces** include several relatively flat, moderately dissected terraces with subtle, east-facing scarps. The terraces, developed on Pliocene-Pleistocene sands and gravels, are dissected in a dendritic pattern by much of the upper Satilla River basin. Cropland is located on the well-drained soils found along the long, narrow, flat to gently sloping ridges paralleling many of the stream courses. The broad flats of the interfluves are often poorly drained and covered in pine, while bottomland forests, are found in the wet, narrow floodplains.

75j. The **Sea Islands/Coastal Marsh** contains the lowest elevations in Georgia and is a highly dynamic environment affected by ocean wave, wind, and river action. Mostly sandy soils occur on the barrier islands, while organic and clayey soils occur in the freshwater, brackish, and salt marshes. Maritime forests of live oak, red cedar, slash pine, and cabbage palmetto grow on parts of the sea islands, and various species of cordgrass, saltgrass, and rushes are dominant in the marshes. The coastal marshes, tidal creeks, and

estuaries are important nursery areas for fish, crabs, shrimp, and other marine species. Parts of the region have a long history of human alterations. Native Americans cultivated corn, melons, squash, and beans; a Spanish mission period during the 1500-1600's included crops of citrus, figs, peaches, olives, artichokes, and onions; and a plantation agriculture economy in the late 1700's through the 1800's produced indigo, rice, sugar cane, and sea island cotton.

Table 5-1: List of Ecoregions and Subecoregions for the State of Georgia (Griffith e*t al.* 2001)			
Ecoregion #	**Ecoregion Type**	**Subecoregion #**	**Subecoregion Type**
45	Piedmont	a	Southern Inner Piedmont
		b	Southern Outer Piedmont
		c	Carolina Slate Belt
		d	Talladega Upland
		h	Pine Mountain Ridge
65	Southeastern Plains	c	Sand Hills
		d	Southern Hilly Gulf Coastal Plain
		g	Dougherty Plain
		h	Tifton Upland
		k	Coastal Plain Red Uplands
		l	Atlantic Southern Loam Plains
		o	Tallahassee Hills/ Valdosta Limesink
66	Blue Ridge	d	Southern Crystalline Ridges and Mountains
		g	Southern Metasedimentary Mountains
		j	Broad Basins
67	Ridge and Valley	f & i	Southern Limestone /Dolomite Valleys and Low Rolling Hills & Southern Dissected Ridges and Knobs
		g	Southern Shale Valleys
		h	Southern Sandstone Ridges
68	Southwestern Appalachians	c & d	Plateau Escarpment & Southern Table Plateaus
75	Southern Coastal Plains	Tidal Condition	Tidal Sites – sites from f & j
		e	Okefenokee Plains
		f	Sea Island Flatwoods
		h	Bacon Terraces
		j	Sea Islands/Coastal Marsh – non tidal

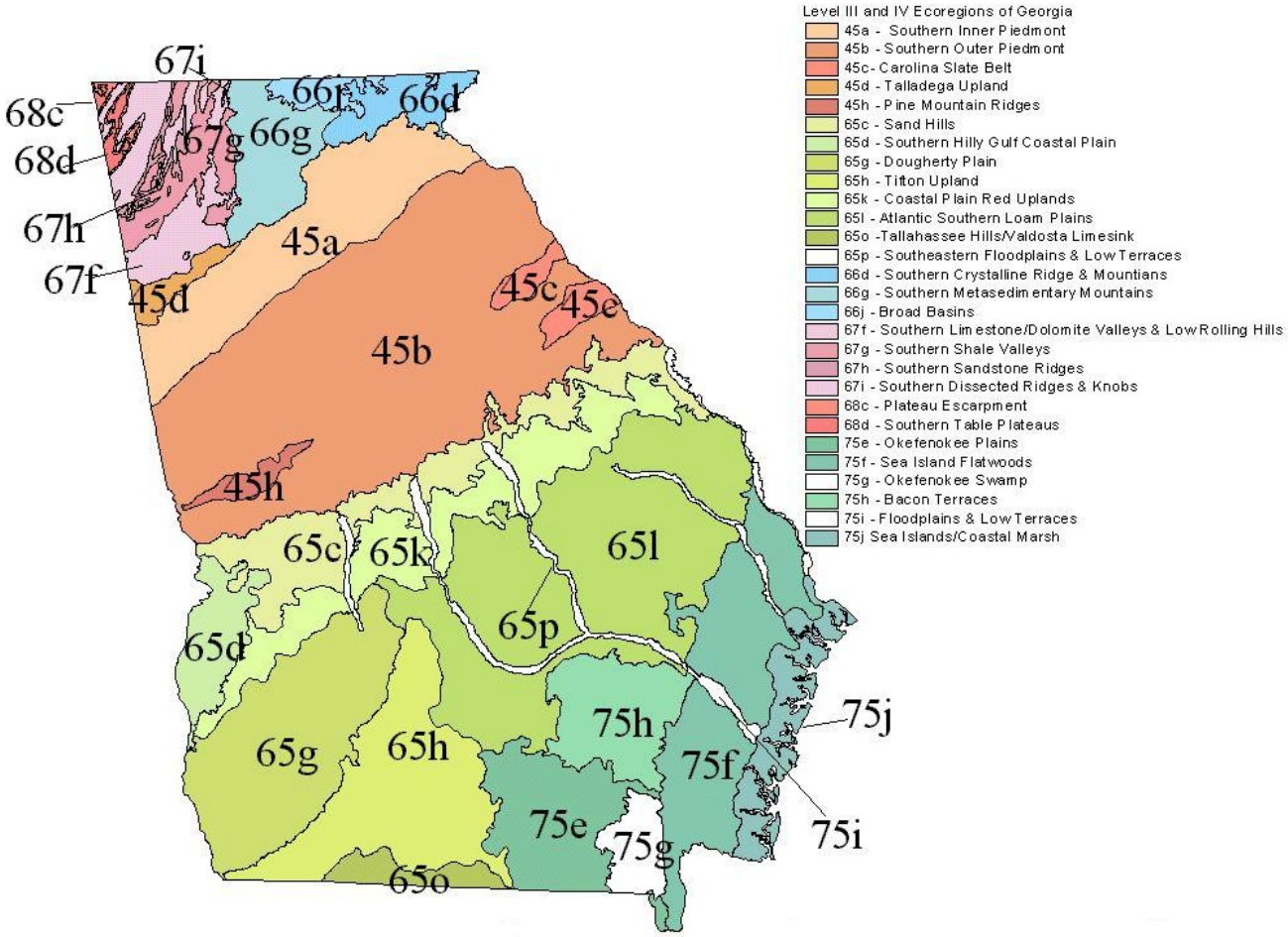

Figure 5-1: Level III and IV Ecoregions Map for the State of Georgia (Griffith e*t al.* 2001)

5.2 GA EPD Multi-Metric Index

For the macroinvertebrate biotic indices, Georgia is divided into 23 subecoregions. The tidal sites are also separated into a category, thus giving the state of Georgia 24 discrete macroinvertebrate indices. **GA EPD will continue to refine and calibrate the macroinvertebrate indices. As more data is collected and analyzed, the metrics may be adjusted.** The formulas for each individual metric can be found in Appendix 5A.

Data collected is entered into Ecological Data Application System (EDAS), an Access© based database, for analysis. The data analysis determines the index scores, thus determining the numeric rankings, narrative descriptions, stream health ratings, and management decisions (Table 5-2). Data analysis will be used in the near future to determine if streams should be on the 303(d) list. EPD will also use this data for many other projects.

For Watershed Assessments and Watershed Protection Plans an Excel© spreadsheet has been developed (and is available on the website www.gaepd.org) to calculate the index score; which determines the stream ranking, narrative description, and stream health rating of each of the sampling locations. Chemical, physical, and biological data for reference streams are also available on the website (www.gaepd.org). For Watershed Assessments and Protection Plans, use the metric spreadsheets and also enter the taxa list in the Excel© spreadsheet provided on the EPD website. These taxa lists, along with the metric spreadsheets for each site, are to be turned in along with the Watershed Assessment (WSA) report and every two years with the Annual Certification of the Watershed Protection Plans Implementation report.

In order to analyze the sample data collected, determine which index to use, based on the sample location in the state. To calculate the index, fill in the information on the metric calculation sheets (*i.e.* HBI, %Tolerant taxa, %Predator, EPT taxa, Simpson's Diversity Index) in the Excel© file for the subecoregion in question. A separate file will need to be completed for each sample location. On the first metric work sheet, fill in the site name or identification number. Fill in the numbers of individuals of a particular family, functional feeding group, habit, etc.; as well as taxa numbers, total number of individuals per site, tolerance values, etc. for each of the metric worksheets. There will be 5 to 8 metrics per index. These metrics combine to make up a multi-metric index comprised of five metric categories: Richness; Composition; Tolerance/Intolerance; Functional Feeding Group; and, Habit. Do not cut and paste data from other spreadsheets as this may include entries that have no relevance to the sheet and can influence the outcome of the metric calculation.

When calculating certain metrics, each taxon is counted even if it is possible they could be the same genus or species. This could result from not being able to identify the organisms to a lower taxonomic level due to damage to the organism or a lack of taxonomic proficiency. This may affect the metrics that use taxa numbers. For example, Perlodidae, Isoperla sp., and Isoperla clio are counted as three separate taxa.

Once data has been filled in for each of the metric worksheets, the metrics will be standardized and all calculations will be tabulated. Standardization scores are based on whether your metric increases or decreases with stress (Table 5-2). Each metric is then scored on a 100-point scale. The calculation for standardization was determined from the results of the GA EPD Ecoregion project (Gore *et al.* 2004 and Gore *et al.* 2006). The results can be found in the ranking classification worksheet in the metric spreadsheet (www.gaepd.org). Tolerance values, North Carolina tolerance values (for use with the NCBI metric), functional feeding groups, and habit can be found in the GA EPD Macroinvertebrate Taxa List, which is available on the website (www.gaepd.org) (Taxa list is formatted for legal size paper).

Table 5-2: Metric Stress Response (Hughes 2006)

Metric Category	Metric	Stress Response
Richness	Ephemeroptera, Plecoptera, & Trichoptera (EPT) Taxa	Decrease*
	Plecoptera Taxa	Decrease*
	Coleoptera Taxa	Decrease**
	Diptera Taxa	Decrease*
	Chironomidae Taxa	Decrease*
	Tanytarsini Taxa	Decrease**
	Margalef's Index	Decrease***
	Shannon-Wiener Index	Decrease**
	Simpson's Diversity Index	Increase****
Composition	% EPT	Decrease*
	% Amphipoda	Decrease**
	% Chironomidae	Increase*
	% Coleoptera	Decrease**
	% Diptera	Increase*
	% Gastropoda	Decrease**
	% Isopoda	Increase**
	% NonInsect	Increase*
	% Odonata	Increase**
	% Plecoptera	Decrease*
	% Tanytarsini	Decrease*
	% Oligochaeta	Increase*
	% Trichoptera	Decrease*
	% Orthocladiinae / TC	Decrease*
	% Tanypodinae / TC	Increase*
	% Hydropsychidae / Total Trichoptera	Increase*
	% Hydropsychidae / Total EPT	Increase*
	% Cricotopus sp. & Chironomus sp. / TC	Increase*

Metric Category	Metric	Stress Response
Tolerant /Intolerant	Tolerant Taxa	Increase*
	% Tolerant Individuals	Increase*
	Intolerant Taxa	Decrease*
	% Intolerant Individuals	Decrease*
	% Dominant Individuals	Increase*
	Dominant Individuals	Increase*
	Beck's Index	Decrease*
	Hilsenhoff's Biotic Index (HBI)	Increase*
	North Carolina Biotic Index (NCBI)	Increase*****
Functional Feeding Group	% Scraper	Decrease*
	Scraper Taxa	Decrease******
	% Collector	Decrease******
	Collector Taxa	Decrease******
	% Predator	Decrease******
	Predator Taxa	Decrease******
	% Shredder	Decrease*
	Shredder Taxa	Decrease****
	% Filterer	Increase*
	Filterer Taxa	Decrease*
Habit	Clinger Taxa	Decrease*
	% Clinger	Decrease*
	Burrower Taxa	Decrease*
	Sprawler Taxa	Decrease*
	Swimmer Taxa	Decrease*

* (Barbour *et. al.* 1999)

** (Barbour *et. al.* 1996)

*** (general literature)

**** (Jessup and Stribling 2002)

***** (Lenat 1993)

****** (Gerritsen and Leppo 2000)

The percentile, numeric ranking, narrative description, stream health rating, and management decision will remain constant throughout the subecoregions (Table 5-3). However, based on the reference condition, the index scores change based on the percentiles. All these values can be determined using the metric spreadsheet. Each subecoregion has a unique index associated with it based on the taxa identified in samples previously collected from the reference and impaired sites. These collections were taken

during the previously mentioned Ecoregions project. This index can be found on the website for each subecoregion (www.gaepd.org).

Table 5-3: Stream rating based on Numeric Ranking (Gore *et al.* 2006)				
Percentile	**Numeric Ranking**	**Narrative Description**	**Stream Health Rating**	**Management Decision**
Above 95th	1	Very good	**A**	Continue periodic monitoring to detect change baseline reference condition
Below 95th, Above 75th	2	Good		
Below 75th, Above 25th	3	Fair	**B**	Frequent monitoring critical to detect change in ecological status, lower range especially
Below 25th, Above 5th	4	Poor	**C**	Frequent monitoring necessary to determine remediation needs and if remediation has been successful
Below 5th	5	Very poor		

5.2a Protocol for Tolerance Values/Habit/FFG for GA EPD

A GA EPD taxa list is provided on the website (www.gaepd.org), which includes tolerance values, functional feeding groups, and habit. This information is needed to calculate metric scores. However, taxa may be encountered that are not on this list, but do occur in Georgia. This may be due to the level of identification or that it was not identified for the Ecoregions project.

A hierarchal priority list was developed to determine the tolerance values, habit, and functional feeding group of a taxon. If a taxon does not occur on the taxa list, follow the priority list depending on what taxonomic level you identified the taxon.

For example, if a specimen has been identified to genus level, start with the priority list of genus level. First, refer to the value in the Rapid Bioassessment Procedures (RBP) manual for SE (North Carolina), if that value is not available, then refer to the RBP Average Species in SE (North Carolina), if that value is not available then refer to the RBP (Mid-Atlantic Coastal Streams Workgroup) MACS Genus Value, if that value is not

available than refer to RBP Average species anywhere in US (If not listed in SE take average all species for that genus for all regions), if not available refer to RBP Nearest Geographically (genus value) Midwest (Ohio) and if this is available this is your tolerance value. The other taxonomic levels work in the same manner as well as habit and functional feeding group.

The complete, hierarchical priority list follows:

Order of Priority for Tolerance Values

Family Level

RBP SE (North Carolina)
↓
RBP MACS
↓
RBP Average SE genera and species average
(Take the average of all genera and species that have a SE value)
↓
RBP Average Countrywide genera/species
(If no SE values take the average of all genera and species for the other regions in the RBP)
↓
RBP Nearest Geographically (Family value) Midwest (Ohio)
↓
RBP If there are values for both Upper Midwest (Wisconsin) and Northwest (Idaho) then average the two
↓
RBP Upper Midwest (Wisconsin) genus value
↓
RBP Northwest (Idaho) genus value
↓
Best Professional Judgment

Genus Level

RBP SE (North Carolina)
↓
RBP Average Species in SE (North Carolina)
↓
RBP MACS Genus Value
↓
RBP Average species anywhere in US
(If not listed in SE take average all species for that genus for all regions)
↓
RBP Nearest Geographically (genus value) Midwest (Ohio)

↓
RBP If there are values for both Upper Midwest (Wisconsin) and Northwest (Idaho) then average the two
↓
RBP Upper Midwest (Wisconsin) genus value
↓

RBP Northwest (Idaho) genus value
↓
Tribe, Subfamily, Family value, or Superfamily (At this level, go to Family Level and follow that procedure)
↓
Best Professional Judgment

Species Level

RBP SE (North Carolina) species value
↓
RBP MACS species value
↓
RBP SE (North Carolina) genus value
↓
RBP SE Average Species Value
↓
RBP if MW, UM, or NW is listed for that particular species than use Nearest Geographically (see below)
↓
RBP average species value any region
(If not listed for SE, then take the average of all species of that genus for all other regions)
↓
RBP MACS genus value
↓
RBP Nearest Geographically (genus value) Midwest (Ohio)
↓
RBP If there are values for both Upper Midwest (Wisconsin) and Northwest (Idaho) then take the genus average
↓
RBP Upper Midwest (Wisconsin) genus value
↓
RBP Northwest (Idaho) genus value
↓
Tribe, Subfamily, Family value, or Superfamily (Once get to this level, go to Family Level and follow that procedure)
↓
Best Professional Judgment

Order Level

If nothing else use the Order Level (or Best Professional Judgment)

RBP SE (North Carolina)
↓
RBP MACS
↓
RBP Average SE family, genera, and species average
(Take the average of all family, genera, and species that have a SE value)
↓
RBP Average Countrywide family/genera/species
(If no SE values take the average of all genera and species for the other regions in the RBP)
↓
Best Professional Judgment

Complexes

Species complex

When listed as species complex or species group
RBP SE species value listed for that species use that value (RBP SE Species Value)
↓
RBP SE Genus Value
↓
RBP SE Average Species (take an Average all species for that genus for the SE)

Species/species complex

When listed as species/species complex or species/species group
RBP SE value, take the average of the two SE values (RBP SE Average Complex (or group) Value)
↓
When no SE value given for either species than take RBP SE Genus Value
↓
When no SE value given for either species and no SE Genus Value, then take the RBP SE species average for all species of that Genus

Exceptions for species/species complex:

When one listed for MAC and one not listed at all, then drop down to SE Genus value
And
If one listed and one not listed for SE 1st take Average species for that Genus, if several SE species values are listed

Genus/Genus complex

Listed as Genus/Genus complex or Genus/Genus Group

RBP SE Average, if have both Genera than take the average of them (RBP SE Average Genus (or group) Complex

Exceptions for Genus/Genus complex:

If one is listed for SE, but the other genus priority list would indicate to take MACS Family Value; then take the SE Average Genus/Species for entire Family (Average Genus/Species Complex) (Average for Family)

And

If one listed for SE, but other not listed and Average taken to get that value, which would include the other genus that is part of the complex, then use the RBP Average Genus/Species Complex

And

If there are two different regions (1 genus listed for 1 region and 1 genus listed for a different region), average the two different regions for those two genera (Average Genus Complex, don't specify regions).

Order of Priority for FFG and Habit

FFG and Habit/Behavior

RBP value
↓
Merritt and Cummins value
↓
If have species, but none listed can go to genus or family if everything else listed is the same in RBP, similarly for genus, - go to family, etc.
(Do not use if several different ones occur, leave as unidentified)
↓
If have species, but none listed can go to genus or family if everything else is the same in Merritt and Cummins, also can use that value if says generally; same for genus go to family, etc.
(Do not use if several different ones occur, leave as unidentified)

The above protocol is for the general tolerance value in the taxa list. The North Carolina tolerance value is the column in the RBP listed as Southeast (NC). This value is used for the North Carolina Biotic Index. The tolerance values cannot be modified for use with this index. If a value is not listed in the Southeast (NC) column, the specimen is not included as part of the North Carolina Biotic Index.

5.3 Guidelines for Using Old Macroinvertebrate Data with the New Metrics

Prior to 2005, WSAs and WPPs were conducted using the Ecological Condition Worksheet (GAEPD 2004 SOP). These metrics were best professional judgment and have been refined to decrease the variability within subecoregions. Instead of a single set of metrics for the whole state, it was found that each subecoregion had its own unique set of metrics that best reflected the health of the streams in that specific subecoregion. Now that subecoregion specific metrics are available, it is advisable that the original taxa list data be entered into the new metrics to get a "new" score for the sample. The new score would give a more comparable measurement of the health of the stream at the time of sampling and bring "older" data in line with newer/future data; thus allowing for easier assessment of trends at each study site over the span of the WSA and WPP.

Please note: re-evaluation is recommended, not required. Re-entry of the raw data into the new metrics will allow comparability with new/future data.

Requirements for re-evaluation of old site data with the new metrics:

Three requirements must be met in order to successfully re-evaluate older data using the new metrics.

These requirements are:

1.) The sampling method for the site must have followed the 20(+3) jab sampling procedures. If fewer than 20 jabs were collected, re-evaluation feasibility must be determined on a case-by-case basis.

2.) The subsampling procedures call for the selection of 200 organisms (±20%) to be picked from the sample for identification and use in the metrics. If more than the allowed number of specimens (240) for a site were identified, then use a random numbers method to either deselect, or select, 240 organisms. Ex: If the sample had less than 480 identified organisms it would be easier to randomly deselect individuals down to 240, if the sample has 480 or more identified organisms then it would be easier to randomly select 240 individuals from the taxa list.

3.) If metric spreadsheets are not available, reference data can be obtained from EPD. The Ecological Condition Worksheet (GA EPD 2004 SOP) can be used for metric analysis until the new metric spreadsheets become available, at which time the data should be recalculated. These spreadsheets will be posted on the EPD website (www.gaepd.org).

5.4 References

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Appendix 5A
Metric Equations

Metric Equations

Richness Metrics:

Ephemeroptera, Plecoptera, Trichoptera Taxa (EPT Taxa)

EPT Taxa = **#of Ephemer. taxa + #of Plecoptera taxa + #of Trichoptera taxa**

- The taxonomic level of Order is used to determine if an individual is considered to be Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa or not Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List (**This list can be found on the EPD website www.gaepd.gov**).

Plecoptera Taxa

Plecoptera Taxa = **# of Plecoptera taxa**

- The taxonomic level of Order is used to determine if an individual is considered to be Plecoptera taxa or not Plecoptera taxa.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

Coleoptera Taxa

Coleoptera Taxa = **# of Coleoptera taxa**

(note – do not count adult and larvae as separate taxa)

- The taxonomic level of Order is used to determine if an individual is considered to be Coleoptera taxa or not Coleoptera taxa.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

Diptera Taxa

Diptera Taxa = **# of Diptera taxa**

- The taxonomic level of Order is used to determine if an individual is considered to be Diptera taxa or not Diptera taxa.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

Chironomidae Taxa

Chironomidae Taxa = **# of Chironomidae taxa**

- The taxonomic level of Family is used to determine if an individual is considered to be Chironomidae taxa or not Chironomidae taxa.
- The taxonomic level of Family can be found in the GA EPD Macroinvertebrate Taxa List.

Tanytarsini Taxa

Tanytarsini Taxa = **# of Tanytarsini taxa**

- The taxonomic level of Tribe is used to determine if an individual is considered to be Tanytarsini taxa or not Tanytarsini taxa. Tanytarsini is a tribe in the family of Chironomidae.
- The taxonomic level of Tribe can be found in the GA EPD Macroinvertebrate Taxa List.

Margalef's Index

$$Dm = \frac{(S-1)}{LN(N)}$$

Dm = Margalef's Index (Diversity)
S = Number of Species in a site
N = Total number of Individuals in a sample
LN = natural log

- Do not count larvae and adult for Coleoptera as separate species.
- Species represent any level of taxonomic identification.

Shannon-Wiener Index (base-e)

Shannon-Wiener (base-e) = $- \Sigma ((p_i) * LN(p_i))$

- $p_i = n_i/N$ (relative abundance for each species)
- n_i = number of a species
- N = total number of all species
- LN = natural log (base e)

Simpson's Diversity Index:

$$D = \frac{\Sigma\, n(n-1)}{N(N-1)}$$

n = total number of organisms of a particular species (no matter what level of taxonomic identification)
N = total number of organisms of all species (total # of individuals in sample)

Composition Metrics:

% Ephemeroptera, Plecoptera, Trichoptera (%EPT)

$$\%\,EPT = 100 * \frac{\text{(\# of Ephemeroptera + \# of Plecoptera + \# of Trichoptera)}}{\text{Total Individuals in sample}}$$

- The taxonomic level of Order is used to determine if an individual is considered to be Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa or not Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Amphipoda

%Amp = 100 * **[# Individual Amphipods / Total Individuals in sample]**

- The taxonomic level of Order is used to determine if an individual is considered to be Amphipoda or not Amphipoda.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Chironomidae

%Chir = **100 * [# Individual Chironomidaes / Total Individuals in sample]**

- The taxonomic level of Family is used to determine if an individual is considered to be Chironomidae or not Chironomidae.
- The taxonomic level of Family can be found in the GA EPD Macroinvertebrate Taxa List.

% Coleoptera

%Coleoptera = 100 * [# Individual Coleoptera / Total Individuals in sample]

- The taxonomic level of Order is used to determine if an individual is considered to be Coleoptera or not Coleoptera.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Diptera

%Diptera = **100 * [# Individual Diptera / Total Individuals in sample]**

- The taxonomic level of Order is used to determine if an individual is considered to be Diptera or not Diptera.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Gastropoda

% Gastropoda = **100 * [# Individual Gastropoda / Total Individuals in sample]**

- The taxonomic level of Class is used to determine if an individual is considered to be Gastropoda individual or not a Gastropoda individual.
- The taxonomic level of Class can be found in the GA EPD Macroinvertebrate Taxa List.

% Isopoda

% Isopoda = 100 * [# Individual Isopoda / Total Individuals in sample]

- The taxonomic level of Order is used to determine if an individual is considered to be Isopoda individual or not an Isopoda individual.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Non-Insect

%NonIns = **100 * [# Individual Non-Insect / Total Individuals in sample]**

- The taxonomic level of Class is used to determine if an individual is considered to be an Insect or not Insect.
- The taxonomic level of Class can be found in the GA EPD Macroinvertebrate Taxa List.

% Odonata

%Odonata = **100 * [# Individual Odonata / Total Individuals in sample]**

- The taxonomic level of Order is used to determine if an individual is considered to be Odonata or not Odonata.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Plecoptera

%Plec = **100 * [# Individual Plecoptera / Total Individuals in sample]**

- The taxonomic level of Order is used to determine if an individual is considered to be Plecoptera or not Plecoptera.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% Tanytarsini

%Tanytarsini = **100 * [# Individual Tanytarsini / Total Individuals in sample]**

- The taxonomic level of Tribe is used to determine if an individual is considered to be Tanytarsini or not Tanytarsini. Tanytarsini is a tribe in the family of Chironomidae.
- The taxonomic level of Tribe can be found in the GA EPD Macroinvertebrate Taxa List.

% Oligochaeta

%Oligo = **100 * [# Individual Oligochaeta / Total Individuals in sample]**

- The taxonomic level of Subclass is used to determine if an individual is considered to be Oligochaeta or not Oligochaeta.
- The taxonomic level of Subclass can be found in the GA EPD Macroinvertebrate Taxa List.

% Trichoptera

%Tri = **100 * [# Individual Trichoptera / Total Individuals in sample]**

- The taxonomic level of Order is used to determine if an individual is considered to be Trichoptera or not Trichoptera.
- The taxonomic level of Order can be found in the GA EPD Macroinvertebrate Taxa List.

% (Orthocladiinae / Total Chironomidae)

$$\%(\text{Ortho/TC}) = 100 * \frac{\#\ \text{Individual Orthocladiinae}}{\text{Total Chironomidae in sample}}$$

- The taxonomic level of Subfamily is used to determine if an individual is considered to be Orthocladiinae or not Orthocladiinae.
- The taxonomic level of Family is used to determine if an individual is considered to be Chironomidae or not Chironomidae.
- The taxonomic level of Family and Subfamily can be found in the GA EPD Macroinvertebrate Taxa List.

% (Tanypodinae / Total Chironomidae)

$$\%(\text{Tany/TC}) = 100 * \frac{\#\ \text{Individual Tanypodinae}}{\text{Total Chironomidae in sample}}$$

- The taxonomic level of Subfamily is used to determine if an individual is considered to be Tanypodinae or not Tanypodinae.
- The taxonomic level of Family is used to determine if an individual is considered to be Chironomidae or not Chironomidae.
- The taxonomic level of Family and Subfamily can be found in the GA EPD Macroinvertebrate Taxa List.

% (Hydropsychidae / Total Trichoptera)

$$\%(\text{Hydro/TT}) = \mathbf{100} * \frac{\textbf{\# Individual Hydropsychidae}}{\textbf{Total Trichoptera}}$$

- The taxonomic level of Family is used to determine if an individual is considered to be Hydropsychidae or not Hydropsychidae.
- The taxonomic level of Order is used to determine if an individual is considered to be Total Trichoptera or not Trichoptera.
- The taxonomic level of Family and Order can be found in the GA EPD Macroinvertebrate Taxa List.

% (Hydropsychidae / Total Ephemeroptera + Plecoptera + Trichoptera)

$$\%(\text{Hydro/(EPT)}) = \mathbf{100} * \frac{\textbf{\# Individual Hydropsychidae}}{\textbf{(\# of Epheme. + \# of Plecoptera + \# of Trichoptera)}}$$

- The taxonomic level of Family is used to determine if an individual is considered to be Hydropsychidae or not Hydropsychidae.
- The taxonomic level of Order is used to determine if an individual is considered to be Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa or not Ephemeroptera taxa, Plecoptera taxa, & Trichoptera taxa.
- The taxonomic level of Order and Family can be found in the GA EPD Macroinvertebrate Taxa List.

% (Chironomus + Cricotopus / Total Chironomidae)

$$\%(\text{Chiro+Crico/TC}) = \mathbf{100} * \frac{\textbf{(\# Indiv. Chironomus + \# Indiv. Cricotopus)}}{\textbf{Total Chironomidae in sample}}$$

- The taxonomic level of genus is used to determine if an individual is considered to be Chironomus and Cricotopus or not Chironomus and Cricotopus.
- The taxonomic level of Family is used to determine if an individual is considered to be Chironomidae or not Chironomidae.
- The taxonomic level of Family and genus can be found in the GA EPD Macroinvertebrate Taxa List.

Tolerance/Intolerance Metrics:

Tolerant Taxa

Tolerant Taxa = **# of Tolerant taxa**

- Tolerant Individuals have a tolerance value ≥ 7
- Tolerance scores can be found in the GA EPD Macroinvertebrate Taxa List.

*** Please note it is the number of tolerant taxa not the number of tolerant individuals.** (Do not count adult and larvae for beetles as two separate taxa.)

% Tolerant Individuals

%TolInd = **100 * [# Tolerant Individuals / Total Individuals in sample]**

- Tolerant Individuals have a tolerance value ≥ 7
- Tolerance scores can be found in the GA EPD Macroinvertebrate Taxa List.

Intolerant Taxa

Intolerant Taxa = **# of Intolerant taxa**

- Intolerant Individuals have tolerance values ≤ 3.
- Tolerance scores can be found in the GA EPD Macroinvertebrate Taxa List.
- Please note it is the number of tolerant taxa not the number of tolerant individuals. (Do not count adult and larvae for beetles as two separate taxa.)

% Intolerant Individuals

%IntolInd = **100 * [# Intolerant Individuals / Total Individuals in sample]**

- Intolerant Individuals have a tolerance value ≤ 3.
- Tolerance values can be found in the GA EPD Macroinvertebrate Taxa List.

% Dominant Individuals

$$\% \text{ Dominant Individuals} = 100 * \frac{\text{\# Individual for Dominant Taxa}}{\text{Total Individuals in sample}}$$

- Determine the dominant taxa (max individuals per taxa) in a site.

Dominant Individuals

Dominant Individuals = # Individuals in sample for the Dominant taxa

- Determine the dominant taxa (largest number of individuals per taxa) in a site.

Beck's Index

Beck's Index = **[2*(C1 Taxa)] + (C2 Taxa)**

- C1 Taxa = # of Taxa with Tolerance values ≤ 1.
- C2 Taxa = # of Taxa with Tolerance values > 1 and ≤ 4.

Hilsenhoff Biotic Index

$$HBI = \frac{\Sigma\, n_i\, a_i}{N}$$

N = Number of total organisms
n_i = number of specimens in each taxonomic group
a_i = the pollution tolerance score for that taxonomic group

(Tolerance scores can be found in the GA EPD Macroinvertebrate Taxa List.)

North Carolina Biotic Index

$$NCBI = \frac{\Sigma\, n_i\, nc_i}{N}$$

N = Number of total organisms
n_i = number of specimens in each taxonomic group
nc_i = the North Carolina pollution tolerance score for that taxonomic group

- To calculate the NCBI only use the individuals that have a North Carolina tolerance value in the GA EPD Macroinvertabrate Taxa List. **Exclude all individuals that do not have a NC tolerance value when calculating this metric.**
- North Carolina tolerance scores can be found in the GA EPD Macroinvertebrate Taxa List under the column heading NCTV.

Functional Feeding Group Metrics:

% Scraper

%Scraper = **100 * [# Individual Scraper / Total Individuals in sample]**

- Scraper is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Scraper Taxa

Scraper Taxa = **# of Scraper taxa**

- The functional feeding group is used to determine if an individual is considered to be a Scraper taxa or not a Scraper taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

% Collector

%Coll = **100 * [# Individual Collector / Total Individuals in sample]**

- Collector is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Collector Taxa

Collector Taxa = **# of Collector taxa**

- The functional feeding group is used to determine if an individual is considered to be a Collector taxa or not a Collector taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

% Predator

%Pred = **100 * [# Individual Predator / Total Individuals in sample]**

- Predator is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Predator Taxa

Predator Taxa = **# of Predator taxa**

- The functional feeding group is used to determine if an individual is considered to be a Predator taxa or not a Predator taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

% Shredder

%Shed = **100 * [# Individual Shredder / Total Individuals in sample]**

- Shredder is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Shredder Taxa

Shredder Taxa = **# of Shredder taxa**

- The functional feeding group is used to determine if an individual is considered to be a Shredder taxa or not a Shredder taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

% Filterer

%Filt = **100 * [# Individual Filterer / Total Individuals in sample]**

- Filterer is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Filterer Taxa

Filterer Taxa = **# of Filterer taxa**

- The functional feeding group is used to determine if an individual is considered to be a Filterer taxa or not a Filter taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

Habit Metrics:

Clinger Taxa

Clinger Taxa = **# of Clinger taxa**

- The functional feeding group is used to determine if an individual is considered to be a Clinger taxa or not a Shredder taxa.
- The functional feeding group can be found in the GA EPD Macroinvertebrate Taxa List.

% Clinger

%Clinger = **100 * [# Individual Clingers / Total Individuals in sample]**

- Clinger is a functional feeding group.
- Functional feeding groups can be found in the GA EPD Macroinvertebrate Taxa List.

Burrower Taxa

Burrower Taxa = **# of Burrower taxa**

- The habit is used to determine if an individual is considered to be a Burrower taxa or not a Burrower taxa.
- The habit can be found in the GA EPD Macroinvertebrate Taxa List.

SprawlerTaxa

Sprawler Taxa = **# of Sprawler taxa**

- The habit is used to determine if an individual is considered to be a Sprawler taxa or not a Sprawler taxa.
- The habit can be found in the GA EPD Macroinvertebrate Taxa List.

Swimmer Taxa

Swimmer Taxa = # of Swimmer taxa

- The habit is used to determine if an individual is considered to be a Swimmer taxa or not a Swimmer taxa.
- The habit can be found in the GA EPD Macroinvertebrate Taxa List.

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PART 5B

Biological/Habitat Assessment -- Fish

This section contains guidance on evaluating fish communities for the biological/habitat assessment component of the District's Water Quality Monitoring Plan. These procedures and protocols were taken directly from Georgia DNR Wildlife Resource Division's Standard Operating Procedures for Conducting Biomonitoring on Fish Communities in Wadeable Streams in Georgia, 2005.

Appendix 5B-1 includes the Criteria for the Index of Biotic Integrity and the Index of Well-Being to monitor Fish Communities in Wadeable Streams in the Piedmont Ecoregion of Georgia, which is applicable to Cherokee, Clayton, Cobb, Coweta, DeKalb, Douglas, Fayette, Forsyth, Fulton, Gwinnett, Hall, Henry, Paulding, Rockdale, and Walton Counties.

Appendix 5B-2 includes Criteria for the Index of Biotic Integrity and the Index of Well-Being to monitor Fish Communities in Wadeable Streams in the Coosa and Tennessee Drainage Basins of the Ridge and Valley Ecoregion of Georgia, which is applicable to Bartow County.

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Part I: Standard Operating Procedures for Conducting Biomonitoring on Fish Communities in Wadeable Streams in Georgia

Georgia Department of Natural Resources
Wildlife Resources Division
Fisheries Management Section

June 1, 2005

Table of Contents

Introduction

Biotic integrity has been defined by Karr and Dudley (1981) as "the ability to support and maintain a balanced, integrated, adaptive community of organisms having a species composition, diversity, and functional organization comparable to that of the natural habitat of the region. " Since the passage of the Water Pollution Control Act of 1972, water regulatory agencies have been charged with restoring and maintaining the biological, or biotic, integrity of the nation's water resources (Karr, 1991). In the past, efforts to restore the biotic integrity of water resources have been directed primarily toward improving the chemical and physical water quality of point source effluents. Politically and logistically, monitoring point source discharges provided water regulatory agencies with an apparant means to satisfy the directives of the Water Pollution Control Act. The numeric pollution standards provided a certain degree of statistical validity and legal defensibility and were believed to be sufficient to protect water resources (Karr 1987). It was presumed that improvements in chemical/physical water quality would be followed by the restoration of biotic integrity.

While the implementation of effluent regulatory programs improved water quality from point source discharges, this approach allowed continued degradation of a variety of aquatic resources, particularly fish populations, from nonpoint sources (Karr et al 1985). Habitat alteration, flow regime modification, and changes in the trophic base of the stream biota are all detrimental impacts upon a stream that are not detected by point source monitoring programs (Karr 1987).

Continued decline in the biotic integrity of aquatic resources despite chemical/physical water quality monitoring programs has compelled some regulatory agencies to integrate a biological approach, or biomonitoring, into their water quality monitoring programs (Karr 1991). Karr (1987) used the term biomonitoring "to evaluate the health of a biological system to assess degradation from any of a variety of impacts of human society" rather than the traditional use of the term as it relates to toxicity testing. Since it is based on the direct observation of aquatic communities, for which traditional chemical/physical water quality monitoring programs have proved to be unreliable surrogates, biomonitoring explicitly addresses the directives of the Water Pollution Control Act to restore and maintain biotic integrity in the nation's water resources. Most of the biomonitoring programs that have been initiated by environmental regulatory agencies have consisted of sampling fish and/or macroinvertebrate communities (Ohio EPA 1987a; North Carolina Department of

Environment, Health, and Natural Resources 1997; Tennessee Valley Authority 1997; Roth et al 1998; Stribling et al 1998).

Besides the benefit of providing a direct measure of the biotic integrity of an aquatic community, adapting biomonitoring procedures into a water quality monitoring program has several other advantages:

1) Biomonitoring is more effective than chemical/physical water quality sampling in detecting the effects of nonpoint-source pollution and intermittent pollution events (Karr and Dudley 1981).

2) The cost of collecting biological data has been shown to be similar or less than the cost of collecting traditional water quality data. Considering the comparative usefulness of the data collected, Ohio EPA (1987a) found it less expensive to sample both fish and macroinvertebrates than to conduct either chemical sampling or bioassay evaluations.

Sampling fish communities as indicators of biotic integrity also provides the following additional benefits to a biomonitoring program (Fausch et al 1990):

3) Since most fish species are long lived (2-10 years or longer) they provide a direct measure of the long-term health of the aquatic community compared to chemical/physical water quality data which measures instantaneous conditions.

4) Fish communities are sensitive to a wide array of direct stresses, including the effects of point source and non-point source pollution, sedimentation, habitat loss, riparian zone disruption, and flow modification.

5) Fish occupy positions throughout the aquatic food web and use food resources from both aquatic and terrestrial environments, providing an integrative view of the entire watershed.

6) Fish communities can be used to evaluate societal costs of degradation more directly than other taxa because their economic and aesthetic values are widely recognized.

Despite the numerous advantages, biomonitoring should not be viewed as a cure-all for water quality monitoring. The purpose of biomonitoring should not be to replace traditional chemical/physical water quality sampling or bioassay testing, but rather to be incorporated as a part of an integrated system of water quality management. Biomonitoring should be used to provide insights

into the long-term biotic integrity of aquatic communities and to identify areas where chemical/physical water quality sampling and bioassay testing can be conducted more efficiently.

This document outlines the standard operating procedures (SOP) used by the Wildlife Resources Division of the Georgia Department of Natural Resources (GAWRD) to collect biomonitoring data on fish assemblages in wadeable streams in Georgia. Two indices of fish community health are used to assess the biotic integrity of streams in Georgia: the Index of Biotic Integrity (IBI) and the Index of Well-Being (Iwb). The IBI was developed by Karr (1981) to assess the health of aquatic communities based on the functional and compositional attributes of the fish population. The Iwb was developed by Gammon (1976) to measure the health of aquatic communities based on the structural attributes of the fish population. Both the IBI and the Iwb were developed to assess fish communities in the midwestern United States. Both indices required modification from their original formats to reflect the differences in fish fauna between the southeastern and midwestern United States. Together these two indices provide a direct and quantitative assessment of the biotic integrity of an aquatic community based on an overall evaluation of its fish population.

Ecoregions of Georgia

Traditionally, water quality standards have followed national guidelines, and the values established nationally did not recognize regional variations in water quality. Depending upon the natural variation of a region, the national water quality standards were often over- or under-protective of aquatic communities (Hughes and Larsen 1988; Hughes et al 1990). Over-protective criteria are needlessly expensive and a misuse of limited restoration funds. Under-protective criteria may not provide the minimal water quality needed to support aquatic communities, especially when the long-term effects of bioaccumulation and the indirect effects of changes to the trophic structure of a system are considered (Hughes et al 1990). Also, criteria for naturally occurring nontoxic pollutants, such as organic detritus and sediment, are difficult to establish with the traditional toxicological approach most water quality standards are based upon (Hughes and Larsen 1988; Hughes et al 1990).

Compounding the problem of using national water quality standards was the fact that most water quality assessments were conducted in a framework based upon administrative or political purposes and did not correspond to regional characteristics that controlled water quality (Omernik and Griffith 1991). Depending upon the regulatory agency or branch of government involved, water quality assessments were traditionally conducted in frameworks such as drainage basins, hydrologic units, or political boundaries and did not consider patterns of soil type, vegetation, land forms and land use. Changes in the patterns of fish assemblages and water quality often occur within individual river basins and hydrologic units. Traditional units tended to lump dissimilar land areas and water types together, concealing true spatial variations in water quality.

The need to address these problems, as well as satisfy the directives of the Water Pollution Control Act to maintain and restore the biotic integrity of the nation's aquatic resources, led to the concept of using natural regional patterns of ecosystems, or ecoregions, as a framework for assessing spatial variation in water quality (Omernik 1987). Ecoregions are generally considered to be regions of relative homogeneity in ecological systems or in relationships between organisms and their environments. Omernik (1987) established ecoregions throughout the conterminous United States by grouping naturally similar ecosystems based upon regional patterns in soil types, potential natural vegetation, land surface forms, and general land use. This approach provides a logical basis for characterizing ranges of ecoregion conditions or qualities that are realistically attainable. Realistic

attainment is a level of quality possible given a set of economically, culturally, and politically acceptable protective measures that are compatible with patterns of natural and anthropogenic characteristics within an ecoregion (Omernik 1987).

Studies throughout the United States have shown a marked correspondence between different ecoregions and patterns of biotic communities, physical habitat measures, and water quality. A study in Arkansas found that Omernik's classification reflected fundamental differences among streams in the six different ecoregions in patterns of fish assemblages, physical habitat, and water chemistry (Rohm et al 1987). Of these variables, changes in fish assemblage patterns provided the most significant differences between ecoregions. Patterns of fish assemblages, macroinvertebrate communities, physical habitat measures, and water chemistry were found to correspond with the eight ecoregions established in Oregon (Whittier et al 1988). Based on the results of over 9,000 fish collections, the eight ecoregions established in Oregon showed a much higher correspondence with fish assemblage patterns than either major river basins or physiographic regions (Hughes et al 1987). Spatial patterns in water quality variables, ionic water chemistry, and nutrient richness were found to correspond with five ecoregions established in Ohio (Larsen et al 1988). Another study used the Index of Biotic Integrity, species richness, and pollution tolerance guilds to establish significant differences in the fish assemblage patterns between ecoregions in Ohio (Larsen et al 1986). Patterns of fish assemblage distribution have also been found to correspond well with four ecoregions in southern and western Wisconsin (Lyons 1989).

The results of these studies depict the strong relationship between ecoregions and patterns in fish assemblages and water quality and demonstrate the value of an ecoregional approach for evaluating data on aquatic communities. By using ecoregions to establish biomonitoring criteria that are regionally appropriate, the problem of natural spatial variation is lessened. Most importantly, the use of ecoregions as a framework for establishing biomonitoring criteria directly addresses the mandates of the Water Pollution Control Act to maintain and restore the biotic integrity of the nation's water resources (Hughes and Larsen 1988; Hughes et al 1990).

Based upon the soil types, potential natural vegetation, geomorphology, and predominant land uses, six major ecoregions (Level III) have been mapped in Georgia (Griffith et al 2001). These include the Blue Ridge, Piedmont, Ridge and Valley, Southern Coastal Plain, Southeastern Plains, and

Southwestern Appalachians (Fig. 1). More detailed information on the physiographic characteristics of each ecoregion in Georgia can be found in Standard Operating Procedures Freshwater Macroinvertebrate Biological Assessment prepared by the Environmental Protection Division of the Georgia Department of Natural Resources, Water Protection Branch (2004).

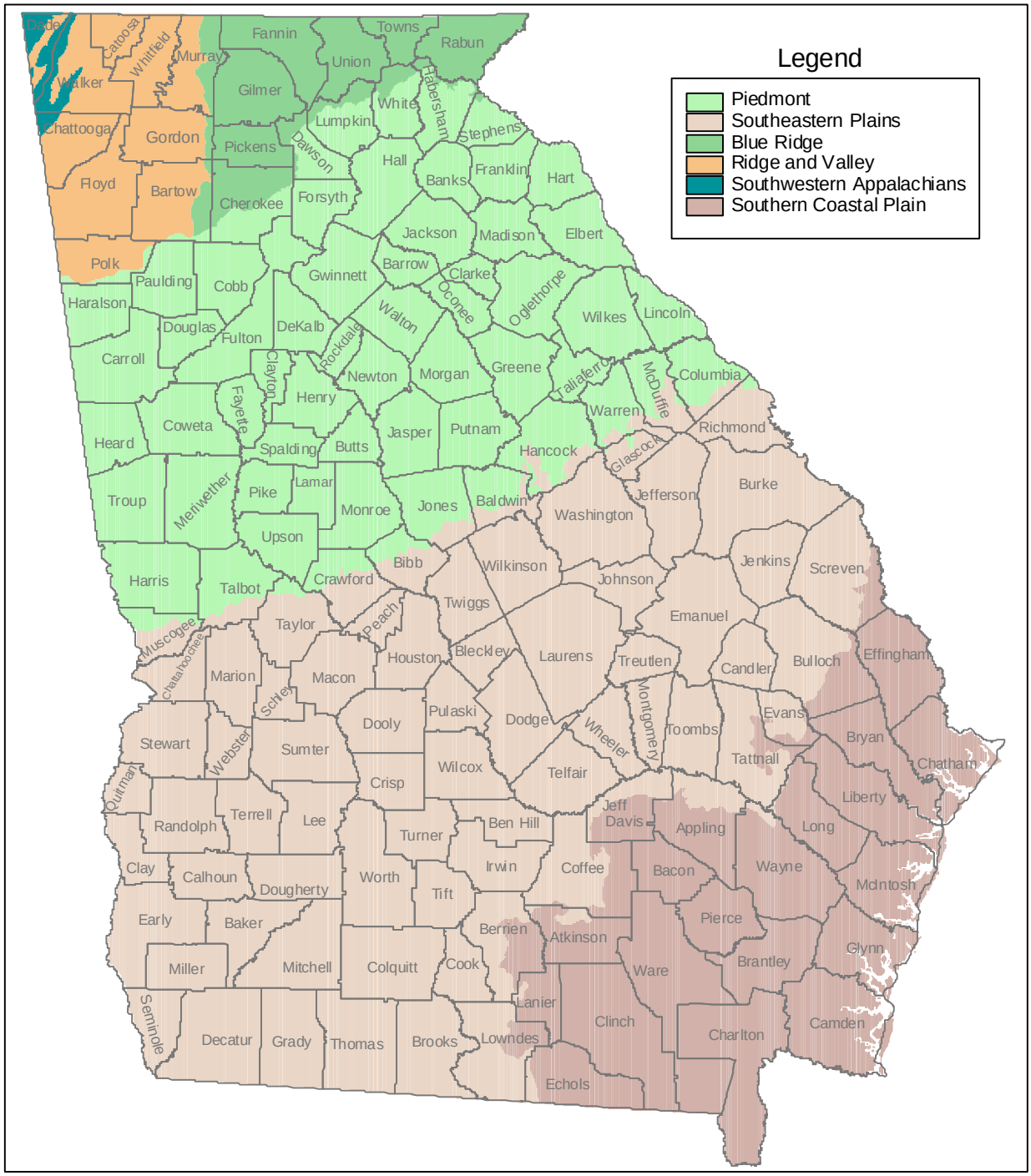

Figure 1. Level III ecoregions of Georgia (Griffith et al 2005).

Site Selection and Reconnaissance

Sample site selection is dependent upon the specific monitoring objectives to be addressed. Once identified, each potential sample site must undergo field reconnaissance to determine if the site is suitable for the collection of biomonitoring data. Sample sites must be accessible to the evaluators and equipment, be wadeable throughout the sample reach, and be representative of the stream under investigation. Sampling stations are usually located upstream of locally modified areas, such as bridges or small impoundments, unless it is desired to assess the effects of these modifications. Bridges and impoundments may alter water flow and sediment deposition, effecting major changes in the physical habitat and the fish community of the downstream area. The equipment list and data sheets needed for stream reconnaissance are included in Appendix 1.

Past studies have shown that biotic index values may show a notable decrease at and immediately below areas receiving point source discharges (Karr et al 1985; Karr et al 1986; Ohio EPA 1987a). When investigating areas of point source discharge, a control site should be located upstream from the discharge in question and at least one other sample site should be located downstream from the discharge area. The downstream site(s) should be located far enough from the point source discharge to characterize the fish community below the mixing zone where the discharged effluents enter the stream. The distance to locate the downstream site from the discharge area will depend on the size of the stream, amount of available macrohabitat, and amount of discharge into the stream (Ohio EPA 1987c). The control site should not be considered a reference site for the downstream sample site. Rather, the control site should provide the investigators with a comparison between the fish assemblages upstream and downstream of the point source. This comparison will allow investigators to determine if any detrimental effects to the downstream fish assemblage can be attributed to the discharge.

Once a sample site has been ascertained to be accessible to equipment and crew, the length of the sample site must be determined. The sample length must be long enough to include all the major habitat types present (e.g., riffle-run-pool sequences). Lyons (1992a) found that a single electrofishing pass at 35 times the mean stream width (MSW), covering a distance of approximately three riffle-run-pool sequences, provided meaningful estimates of species richness without the use of block nets. Lyons found stream widths easier to apply and less subjective than riffle-run-pool

sequences for determining the length of sample reaches. In a comparison of sampling techniques, Simonson and Lyons (1995) found that a single upstream electrofishing pass of 35 times the MSW adequately assessed fish species richness, abundance, and assemblage structure when compared to more intensive four-pass electrofishing removal at the same reach length. The GAWRD compared biomonitoring data collected from 125 sample reaches that were 15 times, 25 times, and 35 times the MSW. They found that standard deviations for IBI scores, species richness, and habitat replication were least for data collected from sample reaches 35 times MSW. Therefore, to fully replicate major habitat types throughout the sample site and decrease variability in IBI scores, a single electrofishing pass for a length of 35 times the MSW was adopted. Due to the constraints of time and resources, a maximum sample reach of 500 meters is employed for wadeable streams in Georgia.

MSW is determined by averaging the stream width measured at random transects along the stream. Initially, five random transects are selected between zero and one hundred meters from the start point using a random number table. Movement proceeds in an upstream direction, measuring the distance between each transect with a tape measure or hip chain. Upstream movement should be made in the midstream position, maintaining a close approximation to the contours of the stream. At each transect the stream width is measured from the water's edge on one bank to the water's edge on the other bank perpendicular to stream flow. Width measurements are recorded to the nearest tenth of a meter. If after five random transects the MSW is found to be greater than three meters, an additional five random transects are selected and the process is repeated. This process is repeated for each three-meter increment of MSW until the final sample length has been determined (i.e., measurements are taken at five random transects for sites with MSW less than 3m, at ten random transects for sites with MSW from 3 – 6 m, and so forth, up to a maximum or 25 random transects per sample site). Side channels should be included in the width measurement, but islands and sand and gravel bars should not, unless they have been exposed by drought and would be underwater at normal flow. When islands or bars are encountered, width measurements should be taken on each side and added. Backwaters, sloughs, and adjacent wetlands should not be included in width measurements (Lyons 1992b).

Besides stream width, stream depth is measured to the nearest hundredth of a meter at each random transect at 1/4, 1/2, and 3/4 of the stream width. The endpoints (beginning and ending) of the

sample reach should be demarcated with flagging tape.

Once the length of the sample site has been determined and marked off, the number of riffle and pool habitats in the sample site are counted. Riffles and pools provide important habitat for different types of fish species due to their characteristic differences in flow, depth, and substrate. Riffles tend to be areas of high energy, with faster water flows, shallower water depths, and coarser substrate material. Pools represent areas of less energy, with slower water flows, greater water depths, and finer substrate material. An abundance of riffle and pool habitats in a sample reach is an indication of a stream that can contain a diversity of fish species. For habitat counts in wadeable streams, any area where the water surface tension is continuously broken for more than one meter in length over a substrate of cobble, boulder, gravel, and/or stable woody debris is considered a riffle. To be considered a pool, an area must have a minimum depth of at least 0.5 meter. Any pool areas with a maximum depth greater than one meter are considered deep pools. Depth of deepest pool should also be recorded while conducting habitat counts.

A riffle frequency is calculated for stream located in the Blue Ridge, Piedmont, Ridge and Valley, and the Southwestern Appalachians ecoregions. Riffles represent a source of high quality habitat for macroinvertebrates and fish, and streams with a well developed riffle-run complex tend to support a more diverse biotic community. The riffle frequency ratio is determined by dividing the mean distance between consecutive riffles in the sample reach by the MSW (Barbour et al 1999). Distance between riffles is measured from the midpoint of the first riffle to the midpoint of the next riffle along the contour of the stream. The value for the riffle frequency is used to determine the score for the corresponding metric in the habitat assessment that is completed after the stream is sampled.

Channel sinuosity is calculated for streams located in the Southern Coastal Plain and Southeastern Plains ecoregions. Channel sinuosity is a measure of the bending or meandering in a stream channel. A high degree of channel sinuosity provides for diverse instream habitat fauna and better maintenance of stream flow fluctuations due to storm surges. The bends in the channel protect the stream from excessive erosion and flooding by absorbing the energy from storm surges. Bends also provide a refuge for the aquatic fauna during storm events. Channel sinuosity is determined by dividing the mean distance between consecutive bends in the sample reach by the MSW (Barbour et al

1999). Distance between bends is measured from the midpoint of the first bend to the midpoint of the next bend along the contour of the stream. The value for the channel sinuosity is used to determine the score for the corresponding metric in the habitat assessment.

Latitude and longitude are determined from a hand held Global Positioning System unit as close as possible to the downstream endpoint of the sample reach. Due to the effects of dense canopy cover at some sampling locations, latitude and longitude may need to be measured at the nearest downstream road crossing and the location noted on the reconnaissance data sheet. Conductivity and water temperature are measured at the sample site with a hand held water quality meter. Field investigators should also determine if seining would be an appropriate sampling technique. All prerequisite data are recorded on the Stream Reconnaissance Report, along with any observations on land use in the surrounding area and possible impacts to the stream and the adjacent riparian zone.

Sampling Procedures

A. Sampling Season

The length of the sampling season is a function of water level and temperature. Normally, biomonitoring samples in Georgia can be collected from early April until mid October, although the sampling season may be longer or shorter for a given year depending upon the local temperature and precipitation. Sampling in the early spring and late fall is normally precluded due to higher water levels and cooler water temperatures. Streams should be wadeable with a flow that allows the investigators to move in an upstream direction at a steady pace. Increased flows associated with elevated water levels decrease sampling efficiency by increasing the movement of stunned fish downstream before they can be captured. Higher turbidities associated with elevated water levels also decreases sampling efficiency by reducing the visibility of stunned fish to the netters. In general, sampling streams with a turbidity measurement greater than 35 NTUs should be avoided. However, not all elevated turbidities readings are related to increased water levels. Streams that have undergone changes to the flow regime, channel alterations, or riparian zone disruptions may have elevated turbidities unrelated to the channel flow status, and the sampling of these impacted streams is left to the best professional judgment of the investigators. At cooler water temperatures fish have a tendency to move into deeper water or under heavy cover where they will be less vulnerable to capture by electrofishing gear (Ohio EPA 1987b; Tennessee Valley Authority 1997). Sampling streams with a water temperature less than 10° Celsius should be avoided. Therefore, most sampling should occur during the summer months when water levels are generally lowest, fish populations tend to be most stable and sedentary, and pollution stresses are potentially the greatest (Ohio EPA 1987c).

B. Sampling Techniques

Electrofishing and seining techniques are used for sampling fish populations in wadeable streams in Georgia. The type of sampling gear to be used is dependent upon the size of the stream to be sampled. Streams with a MSW less than four meters can usually be sampled effectively using a single DC pulsed backpack electrofishing unit (BPEF). Streams with a MSW of five to ten meters are usually sampled with two BPEF units. Streams wider than ten meters are usually sampled with three

or more BPEF units or a barge electrofishing unit, or a combination of both, depending upon the width and depth of the stream to be sampled. These MSW bounds should be viewed as guidelines for sampling wadeable streams in Georgia. It will depend upon the individual investigator to determine the level of effort needed to adequately sample a site. For example, a small stream with an abundance of deep pool habitat may require a second or a third BPEF unit to effectively sample deeper waters. Likewise, a wide, heavily silted stream with shallow water and numerous sand bars may be sampled effectively with a lesser level of effort than the guidelines proposed above. In these instances, best professional judgment should be used when determining how to sample a stream reach most effectively.

Prior to sampling, the electrofishing unit should be tested outside of the sample area to determine the proper control settings needed to collect fish at that site. The ability to collect fish using electrofishing equipment varies between sample sites depending upon water temperature, conductivity, bottom substrate, turbidity, and stream morphology (Kolz et al 1998). Of these, water conductivity is the most important variable that affects electrofishing efficiency. Conductivity is the ability of the water to convey an electric charge, and is dependent upon water temperature and ionic concentration. MicroSiemens (µS) are the preferred units of measurement. Conductivity can be either ambient (at existing water temperature), or specific (adjusted to a reference temperature). For electrofishing purposes, the meter should be measuring ambient conductivity. In streams with higher conductivities, the voltage output from the electrofishing unit should be decreased. Generally, for high conductivity water (400 to 1,600 µS), use 100 to 300 volts, for medium conductivity water (100 to 400 µS), use 400 to 700 volts, and for low conductivity water (15 to 100 µS), use 800 to 1,100 volts (Smith-Root, Inc 1997). Sampling streams with conductivities less than 15 µS should be avoided due to decreases in sampling efficiency seen with most electroshocking equipment. To ascertain the proper control settings, the conductivity should be measured prior to testing the electrofishing unit. Control settings that produce amperages of 0.20 to 0.30 amps for the BPEF units and 1.5 to 2.5 amps for the tow barge can effectively sample fish populations without causing undue damage to the captured fish. The control settings, average amperage output, and total electrofishing time are recorded in the appropriate spaces on the stream collection report (Appendix 1).

1. Sampling with a single backpack electrofishing unit.

Sampling with a single DC pulsed backpack electrofishing unit requires a minimum of two people, although three is preferable. One individual operates the backpack electrofishing unit while the other(s) work the seine and dip nets, and carry the bucket used to transport captured fish. The backpack electrofishing operator should also carry a dip net. Sampling is conducted in an upstream direction to minimize the effect of substrate disturbance within the reach. The entire length of the site is sampled with the backpack unit. All habitats (pools, riffles, runs, woody debris, undercut banks, large rocks, thick root mats, etc.) should be thoroughly sampled to collect a representative sample of the fish population in the stream. An effective technique for sampling fish is to thrust the anode ring into or under the structure to be sampled, such as an undercut bank, thick root mat, or large woody debris, and then slowly withdraw the anode ring. This technique draws the fish out and simplifies their capture from under such structure. As the electrofishing unit operator moves upstream, he/she should apply intermittent power to the electrofishing probe. This technique will lessen the "herding" of fish in front of the operator and out of the range of the electrofishing unit. Two crew members with dip nets walk alongside and behind the electrofishing operator to collect the stunned fish. The collected fish should be frequently transferred from the dip nets to a bucket of water to lessen stress and mortality. This sampling method is not meant to provide an exhaustive survey of the fish fauna, but rather to provide a realistic sample of the fish population in that portion of the stream.

Riffle habitats are sampled by electrofishing downstream into a seine. A ten- to fifteen-foot long minnow seine is usually adequate for this purpose. The seine is positioned perpendicular to the stream flow so that the center section of the seine forms a bag where the flow is greatest. In order to prevent fish from escaping underneath the seine, crew members positioning the seine may find it necessary to stand on the lead line. The electrofishing operator then works in a downstream direction toward the seine. The stunned fish are carried downstream by the current into the seine. In riffles with a lot of cobble and rock substrate, it may be necessary for the backpack electrofishing unit operator to kick around the substrate to dislodge any stunned fish that may have become caught under the rocks. When the section of the stream covered by the seine has been passed through with the electrofishing unit, the seine should be scooped up and the fish removed and placed in a bucket.

Several consecutive sets using this method and moving in an upstream direction may be necessary to completely sample an entire area of riffle habitat.

2. Sampling with two or more backpack electrofishing units.

Sampling a larger stream with two backpack electrofishing units requires a minimum of four people, although five people is often better: two individuals to operate the backpack electrofishing units, two individuals to handle the dip nets and seine, and one individual to carry the bucket to transport the captured fish. Each electrofishing operator will sample an area ranging from one side of the stream bank to the center of the stream, so that each unit operator covers approximately one-half of the total stream area. At least one dip netter should accompany each electrofishing unit operator, following closely behind to gather any stunned fish.

When sampling a deep pool (one meter or deeper), one electrofishing unit operator should approach the pool from the upstream direction and one from the downstream direction. Keeping the pool between the electrofishing unit operators increases sampling efficiency by decreasing the avoidance of fish to a single electrofishing unit in deeper water. Large schools of fish can be sampled in a similar fashion, trapping the school between the electrofishing unit operators and lessening the effects of escape through upstream herding.

Sampling larger streams (> 10 meters MSW) with three BPEF units requires a minimum of seven people: three individuals to operate the BPEF units, three individuals to handle the dip nets and seine, and one individual to carry the buckets to transport the captured fish. In larger streams it may be possible to float a barge or small kayak with large fish containers rather than having individuals carry buckets. When using three BPEF units, a single BPEF unit operator should work each bank out to approximately 1/3 the width of the stream. The third BPEF unit operator should work the middle 1/3 of the stream. The middle operator should also assist in sampling large macrohabitats located along each bank, such as deep pools formed behind downed trees or in the bends of large streams. Each BPEF unit operator should carry a dip net and should also be followed by at least one dip netter.

Other procedures and electrofishing techniques are the same as when sampling a stream with a single backpack electrofishing unit.

3. Sampling with a barge electrofishing unit.

The barge electrofishing unit consists of a tow barge, pulsator, and a generator. The tow barge can be built or purchased directly from a manufacturer. The tote barge fabricated by the GAWRD consists of a PVC foam board core, two layers of fiberglass coating, and an outer gel coating. A stainless steel plate attached to the front and bottom of the barge acts as the cathode. A control box attached to the front of the barge provides plugs for up to three electrofishing probes. Probes are attached to the control box by 50-foot cables to allow for ample movement by the probe operators.

Sampling with the barge EF unit requires a minimum of five people: two people to operate the probes, two people to net the stunned fish, and one person to navigate the tow barge. Probe operators should also carry dip nets. When sampling large streams (MSW of 10 meters or greater), three probe operators and two to three netters should be employed, for a minimum crew of six or seven people. In very large streams (approximately 15 meters or greater) using an additional BPEF unit along one or both banks will increase the sampling efficiency of the barge EF unit. The probe operators sample the area in front of the barge, covering approximately equal portions of the stream area. Netters should stay behind the barge out of the electric field, netting the stunned fish that come up behind the probe operators. Stunned fish are placed in a storage container on the tote barge. An attempt should be made to sample the entire stream area in the sample reach, though this is often difficult in larger streams. As when using BPEF units, all micro- and macrohabitats should be thoroughly sampled to obtain a representative sample of the fish community in the stream. Other procedures and electrofishing techniques are the same as when sampling a stream with multiple BPEF units.

C. Sample Processing

All stunned fish are netted and placed in buckets of fresh water until the entire reach is sampled. Water in the buckets should be replaced frequently to reduce mortality of captured fish. For larger sites, it may be necessary to stop and process the sample several times until the entire site has been sampled. All readily identifiable fish are identified to species, counted, examined for external

anomalies, mass weighed by species, and released. All sample data is recorded on the stream collection data sheet. All field forms and sample tags should be printed on waterproof paper.

Fish less than 25 mm total length (approximately one inch) should be omitted during sample processing. The sampling techniques outlined in this document do not effectively sample fish less than 25 mm total length, and fish in this size range are often troublesome to identify in the field (Karr et al 1986). Most of the fish in the sample less than 25 mm total length are young-of-the-year (YOY) individuals. Populations dominated by highly variable pulses of YOY fish can lead to erroneous conclusions based on inflated IBI and species richness scores. Since YOY fish have not been subjected to the conditions of the sample site for a sustained period of time, they do not fully reflect the long-term conditions at that site. The presence of adult fish implies successful recruitment within a system and is a better indication of long-term conditions in a stream (Angermeier and Schlosser 1987; Angermeier and Karr 1986). Therefore, the exclusion of fish less than 25 mm in length from the sample analysis should significantly reduce bias. Juvenile individuals greater than 25 mm total length that may be YOY fish are included in the analysis since they reflect the attributes and trophic guilds of the adult species (Niemela et al 1998)

Any unidentifiable fish in the sample are counted, weighed, and examined for external anomalies at the streamside and returned to the laboratory in a plastic container of 10% formalin solution for identification. For individuals larger than 10 inches, the body cavity must be cut open to allow for adequate preservation. Each container returned to the lab should include a waterproof tag recording the stream name, sample identification number, collection date, total number of individuals returned, and their weight. Any new species of fish collected in a drainage basin should also be retained for addition to the reference collection. The number of individuals returned to the lab should be recorded on the stream collection data sheet.

Fish that are returned to the lab remain in the 10% formalin solution for approximately five days or until the fish are no longer floating in the preservative. The formalin solution is then decanted under a hood and disposed of in the proper manner and replaced with fresh water. The water should be replaced every day with fresh water for a minimum of three days or until the formaldehyde odor is gone. After the formaldehyde odor has dissipated, the water is replaced with a 70% ethanol solution and the sample is ready for identification. Any additions to the reference collection and problematic

identifications will require verification by a regional ichthyologist. After verification, additions to the reference collection should be stored in separate glass jars with a completed identification label showing the scientific name, common name, stream name, sample location, ecoregion, drainage basin, county, date of collection, and the sample identification number.

1. Presence of external anomalies.

All fish collected are examined for external anomalies. Each individual with an external anomaly and the type of anomaly are recorded on the stream collection data sheet. An external anomaly is defined as the presence of skin or subcutaneous disorders that are visible to the naked eye while processing the sample (Ohio EPA 1987c; O'Neil and Shepard 1998). A high incidence of individuals with external anomalies is a good indicator of a stream impacted by sublethal chemical stresses. Ohio EPA (1987b) has found that the highest incidence of external anomalies occurs in streams subjected to industrial and municipal waste water discharges, sewer outflows, and urban runoff. Some of the more common external anomalies are (Ohio EPA 1987b):

Deformities - Deformities can affect the head, fins, spinal column, and stomach shape. They have a variety of causes, including toxic chemicals, viral and bacterial infections, and protozoan parasites. Fish with extruded eyes, or popeye, a malady caused by fluid accumulation behind the eye due to the presence of certain parasites, are excluded, as are fish with obvious injuries.

Eroded fins - Eroded fins is a chronic condition principally caused by necrosis of the fin tissue due to a bacterial infection. Erosions on the opercle and preopercle are included in this category. In certain fish species, such as darters and suckers, care must be taken not to confuse fin damage caused by spawning activity with erosion due to disease.

Lesions and ulcers - Lesions and ulcers appear as open sores or exposed tissue and are usually caused by viral or bacterial infections. Prominent bloody areas on fish and physical injuries that have undergone secondary infection are included in this category.

Tumors - Tumors are the result of neoplastic diseases caused by viral infections or exposure to toxic chemicals. Certain parasitic infections may produce masses that appear as tumors but

should not be included in this category. Parasitic masses can be squeezed and broken whereas true tumors are firm and not easily broken.

Fungus - Fungus usually emerges as a secondary infection to an injured or open area on a fish and appears as a white cottony growth. Fungal infections often result in further disease or death.

Blindness - Blindness is indicated by a milky, opaque hue to one or both eyes. Fish with missing or grown over eyes are also included in this category.

The presence of parasites is not considered an external anomaly since the infestation could be natural and not related to environmental degradation. No consistent relationship has been established between the incidence of parasitism and environmental degradation (Leonard and Orth 1986; Ohio EPA 1987b). However, external anomalies, including deformities, lesions, and open sores, that may have been caused by the presence of parasites are included.

D. Habitat Assessment

Physical habitat has been shown to be an important factor in determining the structure of the biotic community residing in a body of water (Schlosser 1982; Fausch et al 1984; Karr et al 1987; Hughes and Gammon 1987). A habitat assessment is an evaluation of the quality of the physical habitat as it affects the biological communities, namely fish and macroinvertebrates, in the stream. A habitat assessment will be conducted at each sample site to supplement the findings of the biomonitoring data. It should be viewed as an explanatory tool that will help to clarify the results of the biotic indices.

The habitat assessment used by the GAWRD was developed by the Water Protection Branch of the Georgia Environmental Protection Division (2004). It was modified from the original version developed by Barbour et al (1999) for the EPA Rapid Bioassessment Protocols. This version incorporates different assessment parameters for riffle/run prevalent streams and glide/pool prevalent streams. The choice of which habitat assessment to use will depend upon where the stream is located. Streams located in the Blue Ridge, Piedmont, Ridge and Valley, and Southwestern Appalachians ecoregions are considered riffle/run prevalent streams. These ecoregions are areas of moderate to high gradient landscapes and under normal conditions can sustain water flow velocities of one foot

per second or greater. Streams located in the Southern Coastal Plain and the Southeastern Plains ecoregions are considered glide/pool prevalent streams. These ecoregions are areas of low to moderate gradient landscapes that have water flow velocities rarely greater than one foot per second, except during storm events.

The physical parameters for each habitat assessment are broken into primary, secondary, and tertiary levels. Primary parameters describe those instream physical characteristics that directly affect fish and macroinvertebrate communities. Primary parameters are measured by metrics that evaluate epifaunal substrate, available cover, embeddedness in runs, velocity and depth regimes, and pool substrate and variability. Secondary parameters describe the channel morphology that directly affects the behavior of stream flow and sediment deposition. Secondary parameters are measured by metrics that evaluate sedimentation and deposition, riffle frequency, channel sinuosity, channel alteration, and channel flow. Tertiary parameters describe the banks and riparian zone surrounding the stream, which indirectly affect the type of habitat and food resources available to the aquatic community. Tertiary parameters are measured by metrics that evaluate bank stability, bank vegetative cover, and vegetative riparian zone width (Barbour et al 1999).

The habitat assessment forms for riffle/run prevalent streams and glide/pool prevalent streams are included in Appendix 2. An explanation of each habitat metric and its scoring criteria is also included. Three crew members independently evaluate the habitat quality of the entire sample site. The habitat assessments are conducted after sampling has been completed to avoid disturbing the fish population at the sample site. The final habitat assessment score for a sample site is the average of the three independent scores. If one of the total habitat scores deviates 30 or more points from the middle score, the outlier score may be discarded from the calculation of the final habitat assessment score. If all three of the scores deviate from one another by 30 or more points, the crew members conducting the habitat assessment should review their individual parameter scores while at the station. Individual scores may be revised if appropriate after the review.

E. Water Quality Measurements

Water quality parameters measured at each sample site included turbidity, conductivity, concentration of dissolved oxygen, pH, total alkalinity, total hardness, and water temperature. One

factor determining the concentration of dissolved oxygen in water is the at the sample site. Elevation is estimated to the nearest 100-foot interval from USGS 7.5 minute topographic maps prior to leaving the office or from a GPS unit at the sample site. Conductivity, water temperature, and the concentration of dissolved oxygen are measured at the sample site with a handheld meter. Conductivity must be measured prior to sampling since it may be important in determining the settings on the electrofishing unit. After the fish collection is completed and the sample is processed, a grab sample of water is collected in a plastic bottle and returned to the vehicle where the remaining water quality measurements are conducted. The grab sample should be taken upstream of the sample site where the bottom substrate has not been disturbed to avoid distorting the water quality measures. Total alkalinity, total hardness, and pH are measured using standard Hach kits. A turbidity meter is used to measure turbidity in NTUs to the nearest tenth. At least one digital photograph is taken showing a representative view of the sample site. All water quality measurements and the numbers of photographs taken are recorded in the appropriate spaces on the stream collection data sheet.

Quality Assurance/Quality Control

In order to improve the precision, accuracy, comparability, and representativeness of biomonitoring data, a system of quality assurance and quality control (QA/QC) needs to be implemented. Quality control refers to the routine application of procedures for attaining prescribed standards of performance when collecting in the field, conducting habitat assessments, identifying fish species, and analyzing data. Quality assurance includes the quality control procedures and involves a totally integrated program for ensuring the reliability of monitoring and measurement data (United States EPA 1995). The QA/QC procedures described should ensure the utility of the biomonitoring data collected under the protocols outlined in this document.

A. Fish Identification and Sample Processing

All personnel involved with field identifications will be trained in a consistent manner in the identification of the fish species found throughout Georgia. Fish collections from approximately 10% of the sites should be retained as described in the section under fish processing and returned to the laboratory for verification of fish identifications, counts, and occurrence of external anomalies (Tennessee Valley Authority 1997). Retaining every tenth sample ensures that 10% of the sample sites undergo QA/QC procedures. If it is impractical to retain the entire sample, either due to the large size of certain individuals in the sample or the large total number of individuals collected in the sample, a voucher specimen from each species identified in the field may be returned to the lab for QA/QC purposes. If no fish are collected at the sample chosen for QA/QC, then the next sample should be retained for QA/QC purposes. Samples retained for QA/QC should be recorded in the appropriate space on the stream collection form.

In the laboratory, each crew member responsible for field identifications will independently identify and count all fish, and record the occurrence of anomalies. A follow-up will consist of a meeting between crew members to discuss their results and, if necessary, resolve any problems with sample processing or fish identification.

Every site sampled should be cataloged and tracked to link the sample with the field data sheets and to follow the sample through the final disposition of the data (O'Neil and Shepard 1998). The sample cataloging/tracking system used by the Wildlife Resources Division includes the following

information: sample identification number, stream name, major river basin, ecoregion, county, reconnaissance date, date of reconnaissance data entry, sample date, date of sample data entry, whether or not any portion of the sample was retained, and type of sample (QA/QC, point source, reference, or special project). An example of the sample-tracking log used by the GAWRD is included in Appendix 1.

B. Habitat Assessment

All personnel conducting habitat assessments will be trained in a consistent manner to ensure that the evaluations are conducted properly and to ensure standardization. Field validations comparing the independent habitat assessments of each crew member at a particular sample site will be conducted at least once a year. Any deviations, either between the individual metric scores or the total habitat assessment scores, will be discussed within the group to curtail future discrepancies.

C. Equipment Maintenance and Calibration

All sampling equipment and meters need to be maintained and calibrated in a manner consistent with the manufacturers' recommended schedules. All calibration standards and solutions need to be replaced according to the manufacturers' recommendations. A maintenance and calibration schedule should be posted in the work area where these procedures are performed. After each procedure is performed, the date and the initials of the individual that performed the procedure should be recorded on the maintenance and calibration form. If there is more than one meter of the same type (e.g., two turbidity meters), each meter should be marked and have its own space allotted on the calibration and maintenance form.

Prior to the sampling season, each scale should be checked with a standard set of weights and adjusted as needed to assure accuracy of fish weight data. Scales used in the field should be checked monthly with standard weights to assure their accuracy as they are often used in a more adverse environment than laboratory conditions.

D. Metric Calculations and Data Entry

Data collected in the field should be entered into the database as soon as possible upon returning to the lab. All data entries should be recorded in the appropriate spaces on the sample site log. All entries into the database must be verified to ensure the accuracy of the data from the field datasheets to the database. Two individuals should compare the database entries to the field datasheets, one reading off the field datasheet and the other checking the database entries. Any discrepancies between the two should be corrected and noted on the data entry QA/QC log, along with the date of the verification and the names of individuals conducting the verification. A second verification should be conducted in the same manner. A copy of the data entry QA/QC log used by the GAWRD is included in Appendix 1.

Any data calculations or counts for the IBI metrics or the Iwb should be conducted independently by two individuals who are familiar with the metric scoring criteria and fish guild assignments. A follow-up meeting should be held between the two individuals to determine the reason for any discrepancies and to resolve any future inconsistencies with the metric calculations.

Biotic Indices Used to Measure Fish Community Condition in Georgia

Two indices of fish community health are used to assess the biotic integrity of aquatic systems in Georgia: the Index of Biotic Integrity (IBI) and the Index of Well-Being (Iwb). The IBI was developed by Karr (1981) to assess the health of aquatic communities based on the functional and compositional attributes of the fish population. The Iwb was developed by Gammon (1976) to measure the health of aquatic communities based on the structural attributes of the fish population. Both the IBI and the Iwb were developed to assess fish communities in the midwestern United States. Both indices required modification from their original formats to reflect the differences in fish fauna between the southeastern and midwestern United States. Together these two indices provide a direct and quantitative assessment of the biotic integrity of an aquatic community based on an overall evaluation of its fish population.

A. Index of Biotic Integrity

Various methods using the structure of the fish population to assess the health of the aquatic community have been developed in the past (Fausch et al 1990; Karr 1991). Several of the most accepted approaches, including the presence or absence of indicator species or guilds and the use of species richness, evenness, and diversity indices, are no longer recommended because of their theoretical, statistical, and practical flaws. One of the approaches found to be most suited for identifying areas undergoing environmental degradation was the Index of Biotic Integrity. The IBI is a multimetric index that integrates characteristics of the fish community, population, and individual organism to assess biological integrity at a sample site (Karr 1987). The IBI offers several advantages over other approaches that use fish communities to determine environmental degradation (Fausch et al 1990; Karr 1991). These include: (1) it is a broadly based ecological index that assesses community structure and function at several trophic levels; (2) it gauges biotic integrity against an expectation based on minimal disturbance in that region; (3) it is a quantitative index; (4) there is no loss of information from the constituent metrics when the total score is determined since each metric contributes to the total evaluation of a site; (5) scores are reproducible; and (6) professional judgment is incorporated in the selection of metrics and the development of scoring criteria. Furthermore, the

IBI has been shown to be a statistically valid approach for evaluating water resources and establishing regulatory policies (Fore et al 1994).

The IBI offers several additional when incorporated into a biomonitoring program (Karr 1991). IBI scores can be used to evaluate current conditions at a site, detect trends over time at a specific site with repeated sampling, compare sites within the same ecoregion, and, to an extent, identify the sources of local degradation. Past studies have shown the IBI to be an effective tool in identifying areas suffering from numerous types of environmental degradation. Streams undergoing the negative impacts of effluent from wastewater treatment plants (Karr et al 1985; Hughes and Gammon 1987), mine drainage (Leonard and Orth 1986; Ahle and Jobsis 1996), sedimentation from agricultural and construction practices (Karr et al 1987; Crumby et al 1990; Rabeni and Smale 1995; Frenzel and Swanson 1996), flow modification (Bowen et al 1996), and urbanization and riparian zone destruction (Steedman 1988; Schleiger 2000) have all been identified using the IBI.

The original IBI was developed by Karr (1981) to assess the health of the aquatic community in wadeable streams in the midwestern United States. It consisted of 12 measures, or metrics, which assessed three facets of the fish population: species richness and composition, trophic composition and dynamics, and fish abundance and condition. Each of the 12 metrics was scored by comparing its value to expected values determined from regional reference sites. A regional reference site is a stream located in an area of minimal human impact or disturbance that represents the least impaired conditions for a stream in that ecoregion. The 12 metrics were scored based on whether they approximated, deviated somewhat, or deviated strongly from the values of the regional reference sites and were assigned values of 5, 3, or 1 accordingly, for a maximum score of 60 and a minimum score of 12.

Since regional reference conditions are used to define metric expectations, the IBI has proven to be adaptable to regions outside the midwestern United States while retaining the ecological framework of the original IBI (Fore et al 1994). Karr's original 12 metrics have been previously modified for use in other regions throughout the United States (Miller et al 1988) and North America (Steedman 1988; Lyons et al 1995), Europe (Oberdoff and Hughes 1992), Australia (Harris 1995), and Africa (Hugueny et al 1996). Due to regional differences in the fish fauna and community structure between the southeastern and midwestern portions of the United States, several of the

metrics originally proposed by Karr (1981) required modification for use in streams in the southeastern United States. Table 1 shows a comparison between Karr's original metrics and those developed for streams in Georgia.

Stream location was one of the most important natural factors to consider in adapting the IBI to Georgia. Georgia contains six major ecoregions (Level III, Fig. 1) and 14 major drainage basins as identified by the Environmental Protection Division of the Georgia Department of Natural Resources (Fig. 2). Within a single drainage basin, differences between ecoregions in gradient, soil type, vegetative cover, and mineral content can lead to significant differences in the species richness and composition of the fish community. For example, a stream located in the Blue Ridge Mountains ecoregion of the Chattahoochee drainage basin will differ significantly in the physical characteristics and fish fauna from a stream located in the Southeastern Plains ecoregion of the same drainage basin. Likewise, different drainage basins located in the same ecoregion can differ significantly in species richness and composition. For streams located in the Flint drainage basin in the Piedmont ecoregion, a maximum of four benthic invertivore species could be encountered. In comparison, 10 or more benthic invertivore species could be collected from a stream in the Coosa drainage basin in the Piedmont ecoregion. To address the differences in fish fauna and community composition found between ecoregions and drainage basins within Georgia, the GAWRD established scoring criteria for each major drainage basin or basin group within an ecoregion.

Stream size was another important natural factor to consider when investigating the structure and function of the fish community. In the past, stream order has been used frequently as a measure of stream size. However, due to a lack of consistency in map sizes and classification systems, stream order has not proven to be a universally applicable unit for comparing stream size (Huges and Omernik 1981). Upstream drainage basin area has been shown to be a better predictor of fish assemblage patterns (Hughes and Gammon 1987; Maret et al 1997), species diversity (Statzner and Higler 1985), and the physical and habitat characteristics of a stream (Hughes and Omernik 1981). Furthermore, the development of GIS computer programs allows for faster and more accurate delineation of drainage basin areas than in the past.

Streams with larger drainage basin areas naturally have increased species richness over streams with smaller drainage basin areas. To incorporate this trend in metric scoring, Maximum

Table 1. Comparison of the IBI metrics developed by Karr (1981) for wadeable streams in the midwestern United States and those developed by the Georgia Department of Natural Resources for wadeable streams in the Piedmont ecoregion of Georgia.

Karr (1981)	Georgia Department of Natural Resources
Species Richness	
1. Total number of fish species	1. Total number of native fish species
2. Total number of darter species	2. Total number of benthic invertivore species
3. Total number of sunfish species	3. Total number of native sunfish species (DBA < 15 sq. miles) Total number of native centrarchid species (DBA ≥ 15 sq. miles)
	4. Total number of native insectivorous cyprinid species
4. Total number of sucker species	5. Total number of native round-bodied sucker species
5. Total number of intolerant species	6. Total number of sensitive species (DBA < 15 sq. miles) Total number of intolerant species (DBA ≥ 15 sq. miles)
Species Composition and Trophic Dynamics	
6. Proportion of individuals as green sunfish	7. Evenness
7. Proportion of individuals as omnivores	8. Proportion of individuals as *Lepomis* species
8. Proportion of individuals as insectivorous cyprinid species	9. Proportion of individuals as insectivorous cyprinid species
9. Proportion of individuals as top carnivore species	10. Proportion of individuals as generalist feeders and herbivore species (DBA < 15 sq. miles) Proportion of individuals as top carnivore species (DBA ≥ 15 sq. miles)
	11. Proportion of individuals as benthic fluvial specialist species
Fish Abundance and Condition	
10. Total number of individuals in the sample	12. Number of individuals collected per 200 meters
11. Proportion of individuals as hybrids	
12. Proportion of individuals as diseased fish	13. Proportion of individuals with external anomalies

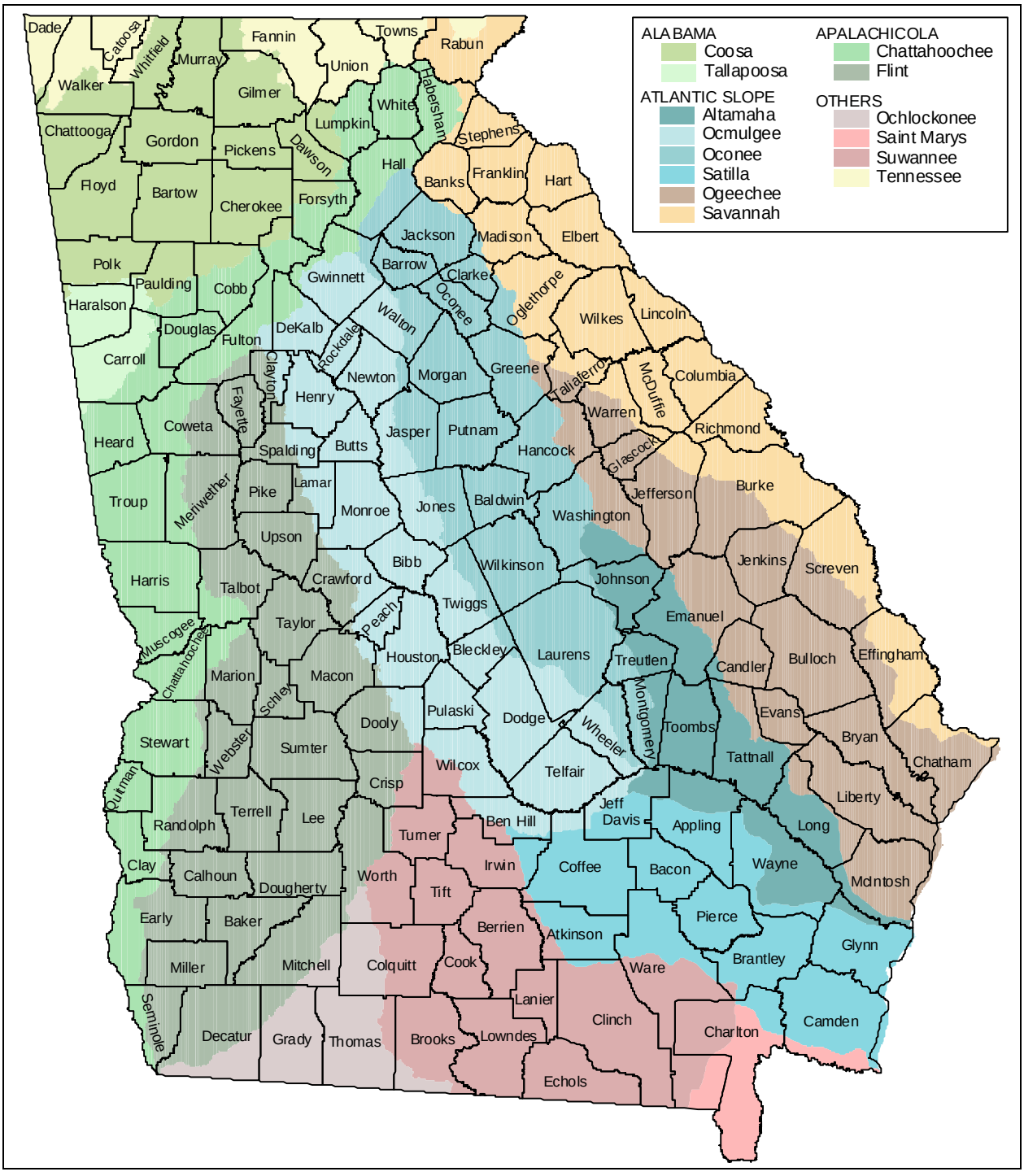

Figure 2. Major drainage basins of Georgia.

Species Richness (MSR) graphs were developed for the species richness metrics (metrics 1 – 6, Table 1). MSR graphs were derived by plotting the number of species collected for a given metric against the log (base 10) transformed values of the drainage basin area. A line delineating the 95th percentile was drawn by eye and, where data allowed, a line delineating the 5th percentile was also drawn. The area between the two lines was trisected using the method developed by Lyons (1992b). Data points falling above the middle trisection scored a 5, those falling in the middle trisection scored a 3, and those falling below the middle trisection scored a 1. Differences in species richness and composition required that separate MSR plots be developed for each major basin or basin group within an ecoregion.

Species composition is less reliant on stream size than species richness. Scoring for the species composition metrics (metrics 7 – 12, Table 1) was determined by plotting the data for a given metric against the log (base 10) transformed value of the drainage basin area. Horizontal lines delineating the 95th and the 5th percentiles were drawn by eye and the area between the lines was trisected.

Metrics 1- 6 evaluate species richness at a site. These metrics assess the health of the major taxonomic groups and habitat guilds of fishes, the availability of spawning habitat and food resources, and the diversity of the fish community. They include:

Metric 1. Total number of native fish species. This metric is a count of all the native fish species in the sample. The total number of native species collected is considered to be one of the most powerful metrics in determining stream condition because of the direct correlation between environmental conditions and the number of fish species present in warmwater assemblages (Ohio EPA 1987b). Highly diverse fish communities often contain intolerant species that are typically unable to cope with perturbations to habitat and water quality (Niemela et al 1998). Hybrids and non-native species are not included in this metric, as their presence does not give an accurate assessment of long-term biotic integrity. Rather, their abundance may indicate a loss of biotic integrity to the system. An abundance of hybrids in a sample indicates that reproductive isolation among species may have been altered by environmental degradation (Karr et al 1986). The prevalence of non-native species, especially top carnivores (gamefish) and cyprinids (baitfish) is

generally indicative of areas with high human population density and/or recreational use (Whittier et al 1997).

Metric 2. Total number of benthic invertivore species. This metric is a count of all the species of darters, madtoms, and sculpins in the sample. Benthic habitats are highly susceptible to degradation from the effects of siltation, flow modification, and reduction in dissolved oxygen levels from the accumulation of organic matter. Due to their specificity for feeding and reproducing in benthic habitats, benthic invertivore species tend to be highly sensitive to environmental degradation (Ohio EPA 1987b). The natural paucity of darter species in some drainage basins in Georgia required modification from Karr's (1981) original metric to include madtom and sculpin species (Table 2). Madtom and sculpin species display a benthic orientation similar to darters and their inclusion is in keeping with the concept of this metric as a measure of the benthic environment available for feeding and reproduction.

Metric 3. Total number of native sunfish / centrarchid species. Karr's (1981) original metric, the total number of sunfish species, required modification due to the increase in species richness of the centrarchid family in the southeastern United States and the abundance of sunfish species found in small streams in Georgia. In headwater streams, Karr's original metric was retained. In Georgia, the sunfish group includes all species of *Acantharchus, Ambloplites, Centrarchus, Enneacanthus,* and *Lepomis. Pomoxis* species are not included, as their presence in headwater streams is usually indicative of a stream impoundment. Sunfish hybrids and non-native species, such as the redbreast sunfish in the Tennessee and Alabama drainage basins, are also excluded from this metric. Sunfish species generally prefer quiet pool habitats near some form of instream cover. Preferred food items include terrestrial and aquatic insects, although some species of sunfish, such as the rock bass and shadow bass, feed predominately on fish as adults. The habitat and feeding preferences of most sunfish species make this metric an effective measure of the losses of instream cover and pool habitat and of the decreases in the terrestrial food supply due to the disruption of the riparian zone (Ohio EPA 1987b). Pools often act as sinks for the accumulation of

toxins and suspended sediments in streams, and are therefore highly susceptible to the effects of water quality and habitat degradations (Niemela et al 1998).

In wadeable streams with a drainage basin area greater than 15 square miles this metric was modified to include all species of native centrarchids. This includes all of the species in the sunfish group, plus all native species of *Micropterus* and *Pomoxis.* Centrarchids represent all levels of the food web, and the presence of a diverse centrarchid population is indicative of a healthy trophic structure within the aquatic community. Centrarchid species inhabit a variety of stream habitats from pools to shoals, and are generally collected near some form of instream cover. The centrarchid family also includes several species that are highly intolerant to habitat and water quality degradations, such as the smallmouth bass and the shoal bass. The presence of these species is indicative of healthy environmental conditions within a stream.

Metric 4. Total number of native insectivorous cyprinid species. This metric is a count of the number of species of the Cyprinidae family in the sample that feed extensively as insectivores. This group includes 64 species from 15 different genera in Georgia. Cyprinid species that feed extensively on plant material, such as the stoneroller species, or that regularly utilize both plant and animal food sources, such as the golden shiner and the bluehead chub, are not included in this metric. Insectivorous cyprinid species are abundant in all sizes of water bodies in Georgia, from the smallest streams to the largest rivers. Insectivorous cyprinid species are specialized feeders, whose presence provides a measure of the diversity of the aquatic macroinvertebrate community (Niemela et al 1998). Different species of insectivorous cyprinids also feed at different levels of the water column, so a variety of insectivorous cyprinid species in a sample is indicative of a diverse aquatic macroinvertebrate community and a healthy trophic structure of the fish community within a stream. Insectivorous cyprinid species can occur in many different types of habitats over a diverse array of substrates (O'Neil and Shepard 1998), thus providing a measure of the quality of instream cover and bottom substrates. Many insectivorous cyprinid species spawn by broadcasting their eggs over the stream bottom where they can develop in the interstices of sand, gravel, and cobble substrates, or by depositing their eggs in rocky crevices. Due to their specificity for clean substrates and a silt-free environment for successful reproduction, this metric also assesses the availability of

suitable spawning habitat in a stream. Insectivorous cyprinids also include several species that are highly intolerant to the effects of habitat and water quality degradation. Samples collected by the GAWRD displayed a marked decrease in the diversity of insectivorous cyprinid species at sites undergoing habitat and water quality degradation. Whittier et al (1997) found that minnow species richness declined in areas undergoing increased urbanization.

Metric 5. Total number of native round-bodied sucker species. This metric is a count of the number of round-bodied species in the Catostomidae family in the sample. In Georgia, round-bodied suckers include all species of *Catostomus, Erimyzon, Hypentelium, Minytrema, Moxostoma,* and *Scartomyzon.* Catostomids represent a small, but important, family of fishes in Georgia. Most catostomid species are sensitive to physical and chemical habitat degradation. In his study on the various effects of land use on fish communities, Schleiger (2000) found catostomids to be sensitive to habitat modification, sedimentation, and changes in water quality. Gammon (1976) found that species of *Moxostoma* and *Hypentilium* were better indicators of water quality in large rivers than any other species group. Most round-bodied sucker species reproduce as broadcast spawners over gravel or cobble substrates and feed extensively on benthic macroinvertebrates, thus providing another benthic-oriented species metric in the index. In addition, the relatively long life span of most Catostomid species provides a long-term assessment of past and present environmental conditions (Ohio EPA 1987b).

Metric 6. Total number of intolerant / sensitive species. A separate scoring criterion was developed for this metric between headwater streams and larger wadeable streams. At sample sites with an upstream drainage basin greater than 15 square miles, this metric is a count of all the species in the sample that have been designated as intolerant to the effects of environmental degradation. Environmental degradation includes the effects of chemical pollution, sedimentation, flow modification, habitat alteration, and riparian zone disruption. This metric distinguishes between sites of good and exceptional biotic integrity since species designated as intolerant should have disappeared by the time a stream has been degraded to the fair category (Karr et al 1986). Tolerance rankings were based upon mean IBI scores (minus metric 6) and Iwb scores for each species,

designations used by other IBI studies in the southeastern United States (Bowen et al 1996; Tennessee Valley Authority 1996; North Carolina DEHNR 1997; O'Neil and Shepard 1998; Schleiger 2000), regional ichthyological texts, and reviews from regional ichthyologists. Species ranked as intolerant include members of the families Cyprinidae, Ictaluridae, Catostomidae, Cyprinodontidae, Centrarchidae, and Percidae.

Since many of the species designated as intolerant do not naturally inhabit smaller streams, this metric was modified for use in headwaters streams to include all species that have been designated as either an intolerant or a headwater intolerant species, collectively termed sensitive species (Ohio EPA 1987b). Species designated as headwater intolerant are those species expected as part of the fish fauna normally found in smaller streams that are intolerant to the effects of environmental degradation and/or stream desiccation. Most headwater intolerant species require permanent pool or riffle habit. Thus the presence of headwater intolerant species at a site can help distinguish between permanent streams and those with ephemeral characteristics (Ohio EPA 1987b). The absence of headwater intolerant species at a site indicates a stream undergoing stress due to habitat or water quality degradations or loss of habitat due to lack of water. Species designated as headwater intolerants include members of the families Petromyzonidae, Cyprinidae, Ictaluridae, Cyprinodontidae, Centrarchidae, and Percidae. Species ranked as intolerants and headwater intolerants are indicated in the fish list for each ecoregion (Parts II – IV).

Metrics 7 – 11 measure the species composition and trophic dynamics at a site. These metrics assess the quality of the energy base and the flow of energy through a stream community and offer a means to quantitatively evaluate the shift toward more generalized foraging that occurs with increased habitat degradation. These metrics also provide a measure of the availability of suitable spawning habitat in the stream. They include:

Metric 7. Evenness. Evenness measures the equity of the proportion of each species in the sample. In general, the greater the equity between species in a sample, the more diverse and healthy the fish community should be. Evenness is measured by comparing the observed diversity in a sample to a theoretical maximum diversity. Evenness values approaching 100 indicate a more diverse community, while smaller evenness values indicate a less diverse community. Certain species, usually

the more pollution tolerant species, can dominate the fish community in degraded environments at the expense of other less tolerant species. As the proportions of the dominant species increase, the evenness of the fish community decreases. In these situations the total diversity of the fish community can be reduced even without a loss of species richness due to the increase in relative abundance of one or more species. Evenness is calculated by:

$$[H / \ln (S)] \times 100$$

Where H = Shannon-Wiener diversity index

S = total number of species collected.

The Shannon-Wiener diversity index is calculated by:

$$- ? \ (n_i/N) \ln (n_i/N)$$

Where n_i = number of individuals of a species

N = total number of individuals in the sample.

The evenness metric replaces Karr's original metric, the proportion of green sunfish in the sample. Most other regional studies have replaced the proportion of green sunfish metric with the proportion of tolerant species metric. Sampling by the GAWRD indicated that the proportion of tolerant species metric provided little utility in streams in Georgia, especially at larger sites. Often degraded sample sites were dominated by species that were not traditionally ranked as pollution tolerant species. Sites receiving nutrient enrichment and those located in highly urbanized areas were often dominated by *Lepomis* species. Degraded headwater sites were often dominated by omnivorous cyprinid species, such as the bluehead or dixie chub. Replacing the tolerant species metric with the evenness metric avoids awarding these degraded sites with a higher metric score. Some sites have been degraded to the point where few individuals, even pollution tolerant individuals, remain. Elevated evenness scores at these sparsely populated sites are not indicative of a highly diverse fish community. Therefore, to avoid awarding highly degraded sites with a high evenness score, if less than 100 individuals are collected, this metric automatically receives a score of one.

Metric 8. Proportion of individuals as *Lepomis* species. This metric measures the proportion of individuals in the sample that are *Lepomis* species. Non-native species and *Lepomis* hybrids are included in this metric. While the species richness of the sunfish population is used as a measure of instream cover and pool habitat (metric 3), an over abundance of *Lepomis* species is indicative of a site undergoing habitat and water quality degradation. Samples collected by the GAWRD show that *Lepomis* species can dominate sites undergoing anthropogenic perturbations, especially the effects of nutrient enrichment, urbanization, and flow modification. An aquatic community dominated by *Lepomis* species is indicative of a decrease in the diversity of the macroinvertebrate community and of suitable spawning habitat for broadcast spawners. At some severely stressed sites the proportion of individuals as *Lepomis* species exceeded 90% of the entire sample. O'Neil and Shepard (1998) also found that *Lepomis* species could dominate disturbed streams in Alabama, sometimes exceeding 50% of the sample. Paller et al (1996) found that the proportion of *Lepomis* species significantly differed between disturbed and undisturbed sample sites in coastal plain streams in South Carolina. This metric automatically receives a score of one if the number of native sunfish at a site equals zero.

Metric 9. Proportion of individuals as insectivorous cyprinids. This metric measures the proportion of the sample that is comprised of individuals that are insectivorous cyprinids. The majority of cyprinid species found in the southeastern United States are insectivores and they usually comprise the dominant trophic guild in surface waters (O'Neil and Shepard 1998). The abundance of insectivorous cyprinids in a sample is a reflection of the variability of the macroinvertebrate food base (Karr et al 1986). Increased degradation of habitat and water quality will lead to a decrease in the diversity of the aquatic insect community in a stream. When the aquatic insect community becomes dominated by only a few taxa, the specialized insectivorous species will be replaced by generalist species more suited to exploit the new food base (O'Neil and Shepard 1998). Sampling by the GAWRD indicates that at sites undergoing anthropogenic stress the proportion of insectivorous cyprinids markedly decreased, approaching zero percent at severely degraded sites. Sampling by the North Carolina Department of the Environment, Health, and Natural Resources (1997) found similar results at sites undergoing nutrient enrichment.

Metric 10. Proportion of individuals as generalist and herbivores / top carnivores. Due to natural variation in the trophic structure of aquatic communities related to stream size, a separate scoring criterion was developed for metric 10 between headwater and larger wadeable streams. At headwater streams, this metric measures the proportion of individuals in the sample that are designated as generalist feeders and herbivores. Generalist feeders are those species that consume both plant and animal materials (including detritus) and have the ability to utilize both types of food sources. This metric evaluates the shift in trophic composition of the fish community in streams with degraded physical and chemical habitat. As food resources become less reliable in degraded environments, generalist feeders frequently become the dominant members of the fish community since their opportunistic foraging habits convey a competitive advantage over more specialized feeders (Karr et al 1986). Degraded headwater streams in Georgia are often dominated by such generalist species as the bluehead chub, dixie chub, and mosquitofish. Nutrient enrichment is a primary disturbance that can cause a shift in the trophic composition of the fish community. Therefore, this metric also includes those species that feed primarily as herbivores, such as the stoneroller species, whose increased numbers in a sample are often associated with elevated nutrient levels (Tennessee Valley Authority 1997; O'Neil and Shepard 1998).

At wadeable sites with a drainage basin greater than 15 square miles, this metric measures the proportion of individuals in the sample that function as top carnivores in the fish community. Top carnivores include all species that feed primarily upon fish, other vertebrates, and crayfish as adults. Omnivores or generalist species that may opportunistically feed upon fish or crayfish are not included. An abundance of top carnivores is indicative of a healthy and trophically diverse fish community (Karr et al 1986). The presence of top carnivores also indicates the availability of instream cover and pool habitat at a sample site (Schleiger 2000). Samples collected by the GAWRD show that top carnivores usually comprise about four to ten percent of the fish population in a healthy, trophically diverse aquatic community. However, at some highly degraded sites the proportion of top carnivores may comprise 20 to 30% of the fish population. To reflect this trend of an over abundance of top carnivores at sites with a degraded aquatic community, the standard trisection method required modification. A pyramid scoring method was developed where an

increasing proportion of top carnivores resulted in a higher metric score up to a threshold proportion, beyond which an increase in the proportion of top carnivores resulted in a lower metric score.

Metric 11. Proportion of individuals as benthic fluvial specialists. This metric measures the proportion of the sample that is comprised of individuals that are ranked as benthic fluvial specialists. Benthic fluvial specialists include all species of benthic invertivores (darter, madtoms, and sculpins), round-bodied suckers, and subterminal mouth insectivorous cyprinid species. Benthic fluvial specialists are insectivorous species that forage on the stream bottom for benthic macroinvertebrates and species that may depend on specific benthic substrates for reproduction. An abundance of benthic fluvial specialists at a site is indicative of a diverse aquatic macroinvertebrate community. Many benthic fluvial specialist species reproduce by broadcasting their eggs over the stream bottom where they can develop in the interstices of sand, gravel, and cobble substrates without parental care. Due to their specificity of clean benthic substrates for foraging and reproduction, the proportion of benthic fluvial specialist species assesses the availability of suitable benthic habitat at a site. Bowen et al (1998) found that the proportion of benthic fluvial specialist species was an important indicator of the trophic diversity of the fish community in their study on the flow-regulated portion of the Tallapoosa River in Alabama.

Metrics 12 and 13 evaluate the population density and the condition of the fish community. These include:

Metric 12. Number of individuals collected per 200 meters. This metric evaluates population density as the number of individuals collected, standardized to 200 meters of sample reach. Population density is calculated by dividing the total number of fish collected by the reach length (35 times the mean stream width) and multiplying this value by 200. Environments that have sustained chemical and/or physical degradation generally contain fewer fish. A low abundance of fish is indicative of sites undergoing direct toxic effects or long-term disruptions in the normal trophic relationships of the fish community (Ohio EPA 1987b). However, samples collected by the GAWRD have shown that the effects of impoundments, urbanization, and nutrient enrichment, along with other types of perturbations, may lead to increases in the population of *Lepomis* species in a degraded

stream. Therefore, to avoid rewarding degraded sites with a higher metric score for the number of individuals collected, when metric 8 (the proportion of individuals as *Lepomis* species) scores a 1, all individuals of *Lepomis* species are excluded from the calculation of metric 12. Mosquitofish, a pollution tolerant species that can dominate fish samples from highly degraded headwater streams, are also excluded from metric 12, as are hybrids and any non-native species in the sample.

Metric 13. Correction Factor: Proportion of individuals with external anomalies. This metric measures the proportion of individuals in the sample that have deformities, eroded fins, lesions, and/or tumors (DELT anomalies). Bacterial, viral, and fungal infections, neoplastic diseases, and chemical pollution may cause DELT anomalies. A high proportion of individuals with DELT anomalies in a stream is indicative of an environment degraded by chemical pollution, excessive siltation, and overcrowding (Ohio EPA 1987b). A marked correspondence has been documented between the proportion of individuals with DELT anomalies and increasing stream degradation, making this metric useful in identifying impacted areas where other structural indices or metrics (e.g., species richness, CPUE, biomass) may indicate a higher quality environment (Leonard and Orth 1986; Ohio EPA 1987b). The presence of parasites is not included as a DELT anomaly since a consistent relationship has not been established between the incidence of parasitism and environmental degradation (Leonard and Orth 1986; Ohio EPA 1987b; Schleiger 2000). However, DELT anomalies that may have been caused by the presence of parasites are included. Individuals with fin or other external damage due to spawning activity are not included and professional judgment must be used when assessing DELT anomalies during the spawning season (North Carolina DEHNR 1997). Individuals that suffered physical damage due to collecting techniques (e.g., hemorrhaging due to electrofishing) are also excluded from this metric.

Sampling by the GAWRD indicates that a significant proportion of individuals in a sample with DELT anomalies is uncommon in Georgia. Lyons (1992b) found similar results in establishing an IBI for warmwater streams in Wisconsin. He retained the proportion of individuals with DELT anomalies as a metric by using it as a correction factor to the total score at sites that exceeded a maximum allowable proportion of DELT anomalies in the sample. We have incorporated Lyons's

Table 2. Total IBI scores, integrity classes, and the attributes of those classes (modified from Karr 1981 and Schleiger 2000).

Total IBI Score (sum of the 13 metric ratings)	Integrity Class	Attributes
60-52	Excellent	Comparable to the best ecoregional reference conditions; all regionally expected species for the habitat and stream size, including the most intolerant species are present with a full array of size classes; significant proportion of the sample composed of benthic fluvial specialist and insectivorous cyprinid species; number of individuals abundant, representing a balanced trophic structure.
50-44	Good	Species richness somewhat below expectation, especially due to the loss of the most intolerant forms; good number of individuals, with several species of suckers, minnows, and benthic invertivores present; trophic structure shows some signs of stress.
42-34	Fair	Species richness declines as some expected species are absent; few, if any, intolerant or headwater intolerant species present; trophic structure skewed toward generalist, herbivorous, and *Lepomis* species as the abundance of insectivorous cyprinid and benthic fluvial specialist species decreases.
32-26	Poor	Sample dominated by generalist, herbivorous, and *Lepomis* species; proportion of non-native species and hybrids increases; intolerant and headwater intolerant species absent; benthic fluvial specialist and insectivorous cyprinid species in low abundance or absent; growth rates and condition factors commonly depressed and diseased fish are often present; number of individuals in low abundance.
24-8	Very Poor	Few fish present, mostly generalist and *Lepomis* species; condition factors poor as unhealthy and juvenile individuals dominate the sample; fish with disease, eroded fins, lesions, and tumors common.
No Fish		No fish collected in the sample.

usage of the DELT metric as a correction. At sites where the proportion of individuals with DELT anomalies exceeds a maximum allowable proportion, four points are subtracted from the total of the previous 12 metrics. At sites where the proportion of individuals with DELT anomalies is less than a maximum allowable proportion, no change is made to the total of the previous 12 metrics. The 90th percentile from plots of the proportion of individuals with DELT anomalies against the log transformed drainage basin area was used to determine the maximum allowable proportion. The 90th percentile has previously been used (Ohio EPA 1987b) to determine the break between scores of 3 and 1 for the DELT metric.

Based on their total IBI score, sample sites are then assigned to one of five integrity classes, ranging from excellent to very poor. A sixth integrity class, no fish, was added for sites where no fish were collected. Integrity classes, along with their appropriate attributes and IBI scoring range, are listed in Table 2.

B. Index of Well-Being

The original Index of Well-Being (Iwb) was developed by Gammon (1976; 1980) as an assessment of the water quality of a river based on the density and diversity of its fish community. The basic premise of the Iwb is that least impacted stream segments will support a larger variety and greater abundance of fish than stressed segments of the same stream. The Iwb has been used to assess the detrimental effects of point source thermal, municipal, and industrial effluents and nonpoint source agricultural and urban runoff (Gammon 1976; 1980; 1983; Gammon and Reidy 1981; Gammon et al 1981).

The Iwb is a composite index that combines two parameters of fish diversity and two parameters of fish abundance in approximately equal measures to produce a single value reflective of the diversity and abundance of the fish community (Gammon 1976). The four community parameters comprising the Iwb include the relative density of fish, the relative biomass of fish, the Shannon-Wiener Index of Diversity based on numbers, and the Shannon-Wiener Index of Diversity based on biomass. These parameters have been used individually in the past as indicators of environmental stress on fish populations with disappointing results (Fausch et al 1990). However, when combined in the Iwb these individual community parameters work in a complementary manner.

The relative abundance parameters are standardized to a sample reach of 200 meters and are expressed as the number collected per 200 meters (No/200m) and the biomass collected per 200 meters (Kg/200m). The Shannon-Wiener diversity indices are calculated as follows:

$$H = - \Sigma(n_i/N) \ln (n_i/N)$$

Where n_i = numbers or biomass for individual species collected standardized to 200 meters sampled

N = total number of individuals (No./200m) or total weight (Kg/200m)

ln = natural logarithm.

The Iwb is calculated as follows:

$$Iwb = 0.5 \ln (No/200m) + 0.5 \ln (Kg/200m) + H_{(No)} + H_{(Kg)}$$

Where No/200m = number of individuals collected standardized to 200 meters sampled

Kg/200m = total biomass collected standardized to 200 meters sampled

$H_{(No)}$ = Shannon-Wiener Index of Diversity based on numbers of fish

$H_{(Kg)}$ = Shannon-Wiener Index of Diversity based on biomass of fish.

In comparisons of the coefficients of variation between the composite index and its individual community parameters, Gammon et al (1981) consistently found the Iwb to be the least variable parameter. Coefficients of variation for the Iwb were between 10 - 20%, compared to 20 – 50% for the two Shannon-Wiener indices of diversity and 40 – 80% for the relative abundance indices. The decreased variability of the Iwb enhances the chance of detecting a statistically significant difference between fish communities.

A shortcoming in the underlying theory of the original Iwb necessitated a modification in its computation to make it more sensitive to a wider array of environmental disturbances. In most cases of environmental degradation, an increase in the abundance of one or more tolerant species is offset by a concurrent decrease in the Shannon-Wiener indices of diversity. However, some environmental

perturbations, such as nutrient enrichment or channelization, can lead to a restructuring of the fish community without large decreases in species diversity. In these instances, the net increase in the abundance of species tolerant to the disturbance, combined with only a modest decrease in species diversity, can lead to an inflated Iwb score at environmentally degraded sites (Hughes and Gammon 1987; Ohio EPA 1987b).

To offset this bias of the original Iwb, a modified version of the Iwb was developed by the Ohio EPA (1987b). In the modified Iwb, any species designated as tolerant to the effects of pollution, hybrids, and non-native species are excluded from the relative abundance components of the Iwb, but retained in the calculations for the Shannon-Wiener indices of diversity. This modification eliminates the positive bias produced by increased abundance of tolerant species at degraded sites, but retains their influence on the Shannon-Wiener indices of diversity. Ohio EPA (1987b) compared the modified Iwb to the original Iwb in data collected from over 2,000 sampling sites and found that the original Iwb consistently overrated sites suffering from environmental degradation when compared to the modified Iwb. Through its treatment of tolerant species, the modified Iwb has proven to be a more accurate index for assessing the fish community at a sampling site.

The GAWRD has adapted a similar version of the modified Iwb developed by the Ohio EPA (1987b) for streams in Georgia. Samples collected by the GAWRD indicated that the abundance of individuals of *Lepomis* species was an important indicator of the health of an aquatic community. Streams undergoing some type of anthropogenic perturbation often see an increase in the abundance of individuals of *Lepomis* species in proportion to the rest of the fish population. This increase in the proportion of the *Lepomis* species population may offset decreases in the relative abundance and biomass of the rest of the fish population. Therefore, the Iwb for streams in Georgia was modified to offset the positive bias that the increase in the proportion of *Lepomis* species may have on the relative abundance parameters of the Iwb. At sites where metric 8 of the IBI (the proportion of individuals as *Lepomis* species) scored a one, all individuals of *Lepomis* species are excluded from the relative abundance components of the Iwb, but retained in the calculations for the Shannon-Wiener indices of diversity. Mosquitofish, hybrids, and non-native species are also excluded from the relative abundance components of the Iwb, but retained in the diversity calculations.

Due to the increased species richness and relative abundance expected in larger streams, it was necessary to develop scoring criteria for the Iwb between headwater sites (sites with an upstream drainage basin area less than 15 square miles) and larger wadeable sites. Scoring criteria were developed by plotting Iwb values against the log transformed (base 10) values of the drainage basin area. The 90^{th} and the 10^{th} percentiles were drawn by eye. Values above the 90^{th} percentile were considered excellent and those below the 10^{th} percentile were considered very poor. The area between the 90^{th} and the 10^{th} percentiles was divided into quarters. Values in the top quarter were considered good, those in the lowest quarter were considered poor, and those that fell into the middle two quarters were considered fair. Overall, the correlation between the modified Iwb and the IBI was highly significant across stream size and ecoregion ($r = 0.8019$, $p = 0.0000$, $N = 717$). The relationship was slightly stronger at larger wadeable streams ($r = 0.8225$, $p = 0.0000$, $N = 256$) than at headwater sites ($r = 0.7829$, $p = 0.0000$, $N = 461$).

The Iwb has proven most useful to aquatic resource managers when it is used as a complementary measure to the IBI for assessing fish communities (Ohio EPA 1987b; Schleiger 2000). In some rare instances the proportional metrics of the IBI do not follow their expected trends. This occurs at highly degraded sample sites where an extremely low number of fish are collected. A low number of individuals collected in a sample can lead to a low proportion or complete absence in the scoring criteria for the species composition metrics of the IBI. This may result in an elevated IBI score and an unrealistic assessment of the fish community at that site. In such instances, the Iwb will provide additional insight for assessing the quality of a sample site. Therefore, it is important for the investigator to consider several sources of information (i.e., IBI, Iwb, macroinvertebrate assessment, habitat assessment, and professional judgment) when assessing the biotic integrity of aquatic communities.

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Appendix 1– GAWRD Data Sheets and Logs

Stream Reconnaissance Equipment List

Stream List
Recon Data Sheets
Random Number Tables
Transect Table
County Maps
Delorme Atlas
Conductivity Meter
DO Meter
Flagging Tape
Waders
Clipboards
Backpack

Calculator
50m Measuring Tape (2)
Digital Camera (optional)
Depth Staff
Stakes (3)
Clamps (2)
GPS Unit
Extra Batteries (8 AA)
Pencils (4+)
Pencil Sharpener
Sun Block
Bug Spray
Hand Sanitizer

Stream Collection Equipment List (BPEF)

Backpack Electrofisher (BPEF) (3)
BPEF Batteries
BPEF Battery Chargers (3)
Battery Plugs (6)
BPEF Probes (4)
Anode Rings:
    11” Diamond Stainless Steel
    6” Diamond Stainless Steel
Seines (2):
    10 Foot and 15-Foot
Dipnets:
    4 Medium, 3 Small
Waders
5-Gallon Buckets (4)
Portable Aerators (2)
Fish Sorting Containers
Collection Jars (2 per site)
Collection Labels
Formalin
Face Shield
Rubber Gloves
Extra Batteries:
    AA (16)
    C (8)
Digital Scale (2)
Hanging Scale (for large fish)
Digital Camera
Water Quality Equipment
    Turbidity Meter
    Dissolved Oxygen Meter
    Conductivity Meter
    pH Test Kit
    Total Hardness Test Kit
    Alkalinity Test Kit
    DO Membrane Kit
Stream Collection Reports
Copy of Recon Reports
Habitat AssessmentReports:
    (3 per site)
Habitat Assessment Forms (3)
Metal Clipboards (4)
County Maps
Pencils (4+)
Pencil Sharpener
Fish Species List
Peterson’s Field Guide
Backpacks:
    2 Large, 1 Small
Collapsible Shovel
Sun Block
Bug Spray
Hand Sanitizer

Stream Collection Equipment List (Barge)

Barge
Generator:
- Spare Gas
- Oil (10W-30)

Pulsator Unit
BPEF Probes (4):
Extension Cables
Waist Belts
Anode Rings:
- 11” Diamond Stainless Steel
- 6” Diamond Stainless Steel

Seines (2):
- 10 Foot and 15 Foot

Dipnets:
- 3 Large, 4 Medium, 3 Small

Waders
Holding Container for Fish
Portable Aerators (2)
Fish Sorting Containers
Collection Jars (3 per site)
Collection Labels
Formalin
Face Shield
Rubber Gloves
Extra Batteries:
- AA (16)
- C (8)

Digital Scale (2)
Hanging Scale
Digital Camera
Water Quality Equipment
- Turbidity Meter
- Dissolved Oxygen Meter
- Conductivity Meter
- pH Test Kit
- Total Hardness Test Kit
- Alkalinity Test Kit
- DO Membrane Kit

Stream Collection Reports
Copy of Recon Reports
Habitat Assessment Reports:
- (3 per site)

Habitat Assessment Forms (3)
Metal Clipboards
County Maps
Pencil (4+)
Pencil Sharpener
Backpacks:
- 2 Large, 1 Small

Fish Species List
Peterson’s Field Guide
Collapsible Shovel
Sun Block
Bug Spray
Hand Sanitizer

Stream Reconnaissance Report

Site ID:	Lat:	Long:
Stream Name:		
Ecoregion:	County:	Basin:
Point of Assessment: _____		

Date:	Time:
Evaluators:	
Total Number of Pools in Reach:	Deepest Pool = m
Total Number of Riffles in Reach:	Total Number of Bends in Reach:
Sample Reach Length = Mean Stream Width _____m X 35 = _____m	
Riffle Frequency = Mean Distance Between Riffles _____ ÷ MSW _____ = _____	
Channel Sinuosity = Mean Distance Between Bends _____ ÷ MSW _____ = _____	
Reach Location: ı Upstream of Road Crossing ı Downstream of Road Crossing ı Combination	
ı Nonpoint Source ı Point Source ı QA/QC ı Potential Reference ı Special Project	
Shocker: ı 1BPEF ı 2BPEF ı 3BPEF ı Barge ı Other	Seine: ı Yes ı No

Watershed Impacts	Riparian Zone Impacts
ı Silviculture ı Row Crop Agriculture ı Animal Production Agriculture ı Landfill ı Urban / Suburban ı Land Application System (LAS) ı Land Disturbing Activity (LDA) ı Ponds/Lakes/Reservoirs	ı Silviculture ı Row Crop Agriculture ı Animal Production Agriculture ı Landfill ı Urban / Suburban ı Land Application System (LAS) ı Land Disturbing Activity (LDA) ı Ponds/Lakes/Reservoirs

Water Temp (°C):	Conductivity (µS):	Elevation (ft):
Comments: _____		

Sample Reach 0-3 Meters MSW

Random Transects	____ m	____ m	____ m	____ m	____ m	
Stream Width	____ m	____ m	____ m	____ m	____ m	Avg.____ m
Stream Depth	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	

Sample Reach 3-6 Meters MSW

Random Transects	____ m	____ m	____ m	____ m	____ m	
Stream Width	____ m	____ m	____ m	____ m	____ m	Avg.____ m
Stream Depth	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	

Sample Reach 6-9 Meters MSW

Random Transects	____ m	____ m	____ m	____ m	____ m	
Stream Width	____ m	____ m	____ m	____ m	____ m	Avg.____ m
Stream Depth	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	

Sample Reach 9-12 Meters MSW

Random Transects	____ m	____ m	____ m	____ m	____ m	
Stream Width	____ m	____ m	____ m	____ m	____ m	Avg.____ m
Stream Depth	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	

Sample Reach 12-15 Meters MSW

Random Transects	____ m	____ m	____ m	____ m	____ m	
Stream Width	____ m	____ m	____ m	____ m	____ m	Avg.____ m
Stream Depth	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	
	____ m	____ m	____ m	____ m	____ m	

Riffle/Bend Midpoint (meters)	Distance Between Riffles/Bends (meters)
	1:
	2:
	3:
	4:
	5:
	6:
	7:
	8:
	9:

Sum of the Distances:

_____

÷

Total Number of Distances

_____

=

Mean Distance Between Riffles/Bends: _____

Stream Collection Report

Site ID:	County:	Basin:
Stream Name:		
Date:	Time:	Ecoregion:
Collectors:		

Water Quality

Elevation (ft):	Water Temp (°C):	D.O. (mg/L):
Conductivity (µS):	pH:	Turbidity (NTU):
Total Hardness (ppm):	Alkalinity (ppm):	Camera: ı A ı B

Backpack Electrofisher

No. of Probes: ı 1 ı 2 ı 3	Total Shocking Time (sec): EF#1: +EF#2: +EF#3:	
Mode:	Voltage:	Avg. Output (Amps):

Barge Electrofisher

No. of Probes: ı 1 ı 2 ı 3	Shocking Time (sec):	
Mode:	Voltage:	Avg. Amps:

Species List

Number Retained	Species	Number	Weight (g)	External Anomalies

Number Retained	Species	Number	Weight (g)	External Anomalies

External Anomalies include: **AW** = Anchor Worm; **BL** = Blind; **BS** = Black Spot; **D** = Deformities; **EF** = Eroded Fin(s); F = Fungus; **I** = "Ich"; **L** = Lesions; **LE** = Leeches; **PE** = Popeye; **T =** Tumors

GAWRD QA / QC Data Entry Log

Site ID:		Entered	Date		QAQC 1	Date		QAQC 2	Date
Recon Data									
Transect Data									
Fish Data									
Habitat Data									

Comments:

Site ID:		Entered	Date		QAQC 1	Date		QAQC 2	Date
Recon Data									
Transect Data									
Fish Data									
Habitat Data									

Comments:

Site ID:		Entered	Date		QAQC 1	Date		QAQC 2	Date
Recon Data									
Transect Data									
Fish Data									
Habitat Data									

Comments:

Site ID:		Entered	Date		QAQC 1	Date		QAQC 2	Date
Recon Data									
Transect Data									
Fish Data									
Habitat Data									

Comments:

GAWRD Sample Tracking Log

Site ID	Stream Name	Basin	County	Ecoregion	Recon Date	Sample Date	Sample Retained (yes/no)	Sample Type (PS / NPS / QAQC Reference / Other)

Appendix 2– Habitat Assessments

Riffle / Run Habitat Assessment

Site ID:	Date:
Stream Name:	
Assessor:	

Habitat Parameter	Score	Notes
Epifaunal Substrate / Instream Cover		LWD DP SP OS LR UB TRM DMB DR
Embeddedness in Run Areas		
Velocity / Depth Combinations		
Channel Alteration		
Sediment Deposition		
Frequency of Riffles*		*measured during stream reconnaissance
Channel Flow Status		
Bank Vegetative Protection Left Bank Right Bank	LB RB	
Bank Stability Left Bank Right Bank	LB RB	
Riparian Vegetative Zone Left Bank Right Bank	LB RB	
Total Score →		

1. Epifaunal Cover / Instream Cover

Measures the amount of substrates that are available as cover for aquatic organisms. A wide variety and/or abundance of submerged structures in the stream provide fish and macroinvertebrates with a large number of niches, thus increasing the habitat diversity. As the variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases. Riffles and runs offer a variety of substrate sizes and flows and provide the most stable habitat in high-gradient streams. **Possible Habitat Types:** Fallen Trees / Large Woody Debris (**LWD**), Shallow Pools > 0.5 m (**SP**), Deep Pools > 1.0 m, Overhanging Shrubbery in water (**OS**), Large Rocks (**LR**), Undercut Banks (**UB**), Thick Root Mats (**TRM**), Dense Macrophyte Beds (**DMB**), Deep Riffles (**DR**), Long Runs with Cobble / Large Rock Substrate (**RU**)

A. Stable and available habitats expected for stream type make up > 70% of reach. Stream exhibits a well developed riffle-run complex.
   1. **Seven** habitat types common; stable substrate dominated by softball size cobble and boulder stones……..**20**
   2. **Five** habitat types common, additional habitat types rare; stable substrate dominated by boulder stones……..…**18**
   3. Less than **four** habitat types present, stable substrate dominated by gravel stones and boulders/bedrock and/or stable woody debris……..**16**

B. Stable and available habitats expected for stream type make up 40-70% of reach.
   1. **Seven** habitat types common; stable substrate dominated by softball size cobble and boulder stones……**15**
   2. **Five** habitat types common, additional habitat types rare; stable substrate dominated by gravel and boulder stones..... ..……..**13**
   3. Less than **four** habitat types present; stable substrate dominated by gravel stones and boulders/bedrock and/or stable woody debris……..**11**

C. Stable and available habitats expected for stream type make up 20-40% of reach.
   1. **Seven** habitat types common; stable substrate dominated by softball size cobble and boulder stones……..……**10**
   2. **Five** habitat types common, additional habitat types rare; stable substrate dominated by gravel and boulder stones……...……..…..**8**
   3. Less than **four** habitat types present, stable substrate dominated by gravel stones and boulders/bedrock and/or stable woody debris……....**6**

D. Stable and available habitats expected for stream type make up < 20% of reach. Riffles or runs are virtually nonexistent, no cobble substrate.
   1. **Two** habitat types common, additional habitat types rare; substrate dominated by gravel and sand/silt, short runs……..**4**
   2. **Two** habitat types only; substrate dominated by gravel and sand/silt, short runs……..……..**3**
   3. **One** habitat type common, additional habitat types rare; substrate . dominated by small gravel and sand/silt with short runs, no riffles…….…....**2**
   4. **One** habitat type only; substrate dominated by small gravel and sand/silt with short runs, no riffles……..…**1**
   5. **No** habitat types present; substrate dominated by sand/silt with no runs……**0**

.

2. Embeddedness in Run Areas

Measures the degree to which cobble, boulders, and other rock substrates are surrounded by **fine** sediment and silt. Embeddedness relates directly to the suitability of the stream substrate as habitat for macroinvertebrates and for fish spawning and egg incubation.

Fine sediments range from 0.062mm to 2mm in size. Silt particles measure less than 0.062mm. Sediment and silt particles smaller than 2mm can be distinguished using "texture by feel techniques" employed in soil surveys.

A. Little or no embeddedness present by fine sediment and/or silt surrounding and covering rocks.
   1. < 10% embeddedness ……..……**20**
   2. 10% embeddedness by sediment……..……**19**
   3. 10% embeddedness by sediment and silt……..…..**18**
   4. 20% embeddedness by sediment……..….**17**
   5. 20% embeddedness by sediment and silt……..…..**16**

B. Fine sediment and silt surrounds and fills 25 – 50 % of the living spaces around and in between gravel, cobble, and boulders.
   1. 30% embeddedness by sediment……..…**15**
   2. 30% embeddedness by sediment and silt……....**14**
   3. 40% embeddedness by sediment……....**13**
   4. 40% embeddedness by sediment and silt……..…**12**
   5. 50% embeddedness by sediment……..**11**

C. Fine sediment and silt surrounds and fills 50 - 75 % of the living spaces around and in between gravel, cobble, and boulders.
   1. 50% embeddedness by sediment and silt……**10**
   2. 60% embeddedness by sediment……...**9**
   3. 60% embeddedness by sediment and silt……..**8**
   4. 70% embeddedness by sediment……....**7**
   5. 70% embeddedness by sediment and silt……..**6**

D. Fine sediment and silt surrounds and fills more than 75 % of the living spaces around and in between gravel, cobble, and boulders.
   1. 80% embeddedness by sediment……....**5**
   2. 80% embeddedness by sediment and/or silt……...**4**
   3. 90% embeddedness by sediment……....**3**
   4. 90% embeddedness by sediment and/or silt ……...**2**
   5. 100% embeddedness by sediment……..……**1**
   6. 100% embeddedness by sediment and/or silt..……...**0**

3. Velocity / Depth Combinations

Measures a stream's characteristic velocity/depth regime. Patterns of velocity and depth are included for high-gradient streams as an important feature of habitat diversity. There are four combinations of velocity and depth that are characteristic of high quality riffle/run prevalent streams. These are: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The depth criterion used to distinguish shallow from deep is 0.5 meter; the velocity criterion used to distinguish slow from fast is 0.3 m/sec. The occurrence of these four patterns relates to a stream's ability to provide and maintain a stable aquatic environment.

A. A complex stream system that exhibits a heterogeneous combination of all velocity/depth patterns.
   1. All **four** velocity/depth patterns are present…..…… **20**
   2. All patterns present, but one may not be well defined……..…….. ..**18**
   3. All patterns present, but more than one may not be well defined……...**16**

B. Stream is less heterogeneous, displaying fewer of the velocity/depth patterns.
   1. Only **three** of the four velocity/depth patterns are present……**15**
   2. **Three** of the four patterns are present, but one may not be well defined... ...**13**
   3. **Three** of the four patterns are present, but more than one may not be well defined...……..….**11**

C. Stream becomes more homogeneous. Sediment deposition and/or channel alteration is resulting in the loss of certain velocity/depth patterns.
   1. Only **two** of the four velocity/depth patterns are present……**10**
   2. **Two** of the four patterns are present, but one is not be well defined..……..**8**
   3. The fast-shallow of the shallow regime is missing……..…**6**

D. A simple stream system that is heavily affected by the restriction of water flow due to sediment deposition and/or channel alteration, resulting in a monotonous velocity/depth pattern.
   1. Only **one** of the four velocity/depth patterns is present, usually dominated by the slow-deep pattern…….……..……..**5**
   2. Stream heavily affected by sediment; very little if any flow, dominated by the slow-shallow pattern…….**3**
   3. No flow regime present; stream nearly dry or pooled up ...……..…..**0**

4. Channel Alteration

Measures any large-scale alteration in stream morphology that affects flow, instream habitat, and/or sedimentation rates. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances due to dredging activities; when dams, culverts, or bridges are present; or when other morphological changes have occurred.

A. Stream flows a normal and natural meandering pattern with a well developed riffle/run complex. Alteration is absent.
   1. No evidence of disturbance; riffles as wide as the stream and extend twice the stream width; stable substrate dominated by cobble, boulders and/or bedrock……....**20**
   2. No evidence of disturbance; riffles as wide as stream but do not extend twice the stream width; stable substrate of cobble, boulder and/or bedrock…….**18**
   3. No evidence of disturbance; riffles not as wide as the stream……..**16**

B. Some stream straightening, dredging, artificial embankments, or dams present but NO evidence of recent alteration activities. Alteration probably occurred more than 20 years ago. Stream appears to be in the process of recovery.
   1. Less than 10% of reach has channel disturbance……....**15**
   2. 10% of reach has channel disturbance…..……**14**
   3. 10% - 20% of reach has channel disturbance……..……...…**13**
   4. 20% - 30% of reach has channel disturbance...……**12**
   5. 30% - 40% of reach has channel disturbance..…...……**11**

C. 40 to 80% of the stream reach has been altered or channelized. Alteration may have occurred less than 20 years ago.
   1. 40% - 50% of reach has channel disturbance..……**10**
   2. 50% - 60% of reach has channel disturbance…..……**9**
   3. 60% - 70% of reach has channel disturbance..……**8**
   4. 70% - 80% of reach has channel disturbance..……**7**
   5. 80% - 90% of reach has channel disturbance …..……...……**6**

D. Instream habitat highly altered. More than 80% of the stream reach has been altered. Alteration may be recent (<10 years).
   1. >90 % of reach has channel disturbance……..……..**5**
   2. Channel reach 100% disturbed; straight with no artificial embankments……...**3**
   3. Channel reach 100% disturbed; straight with some artificial embankments……...**1**
   4. Banks 100% shored by gabion, cement, and/or riprap....……...**0**

5. Sediment Deposition

Relates to the amount of sediment that has accumulated and the changes that have occurred to the stream bottom as a result of deposition. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually along the inner bank of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of pools and runs. High levels of sediment deposition are symptoms of an unstable environment that may be unsuitable for many organisms.

A. No enlargements of islands/point bars present; <20% of the stream bottom affected by gravel or sand accumulation.
   1. No deposition detected, especially in pool habitats……**20**
   2. <10% sediment deposition with accumulation in pools only……..…..**19**
   3. <10% sediment deposition with accumulation in pools and runs only……**18**
   4. 10% - 20% sediment deposition with gravel and/or coarse sand ……...**17**
   5. 10% - 20% sediment deposition with fine sand and/or silt ……..……**16**

B. 20% - 40% of the stream bottom affected by gravel, sand, and/or silt accumulation; increased deposition in pools and runs; some new increase in bar and island formation.
   1. 20% - 30% sediment deposition with gravel and/or coarse sand…..…. …**15**
   2. 20% - 30% sediment deposition with fine sand and/or silt……..……**14**
   3. 30% - 40% sediment deposition with gravel and/or coarse sand……...…**12**
   4. 30% - 40% sediment deposition with fine sand and/or silt……..…**11**

C. 40% - 60% of the stream bottom affected with increased deposition in pools. Number of shallow pools increases. Runs and riffles highly impacted by sand, silt, and fine gravel. Recent deposits of gravel, sand, and silt observed on old and new point bars, islands, and behind obstructions. Formation of few new bars/islands is evident and old bars are deep and wide; deposition at bends obvious.
   1. 40% - 50% sediment deposition with gravel and/or coarse sand…..……...**10**
   2. 40% - 50% sediment deposition with fine sand and/or silt……..……....**9**
   3. 50% - 60% sediment deposition with gravel and/or coarse sand…..……....**8**
   4. 50% - 60% sediment deposition with fine sand and/or silt……..……....**7**

D. >60% of the stream bottom affected with heavy deposition from fine gravel and sand at stream bends, obstructions, and/or pools. Extensive deposits of fine sand and/or silt on old and new bars, islands, and along banks in straight channels. Riffle and pool habitats are reduced or absent due to substantial deposition.
   1. 60% - 70% sediment deposition with gravel and/or coarse sand.……..……**5**
   2. 60% - 70% sediment deposition with fine sand and/or silt…….……..…**4**
   3. 70% - 80% sediment deposition with gravel and/or coarse sand……..…..**3**
   5. >80% sediment deposition with gravel and/or coarse sand……...**1**
   6. >80% sediment deposition with fine sand and/silt……**0**

6. Riffle Frequency

Estimates the frequency of occurrence of riffles and thus the heterogeneity occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna; therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. In some streams, a longer reach than that designated for sampling may need to be evaluated to adequately score this metric.

Riffle Frequency = Mean Distance Between Riffles / Mean Stream Width

Riffle frequency is determined during stream reconnaissance.

A. Occurrence of riffles relatively frequent. Deep pools may be present and riffles are deep enough to allow passage of fish.
   1. Riffles are continuous; run-to-riffle ratio = 1-2……..……**20**
   2. Run-to-riffle ratio = 3-4……..…**19**
   3. Run-to-riffle ratio = 5……..…**18**
   4. Run-to-riffle ratio = 6……..**17**
   5. Run-to-riffle ratio = 7……..…**16**

B. Occurrence of riffles less frequent; adequate depth in pools and riffles.
   1. Run-to-riffle ratio = 8……...**15**
   2. Run-to-riffle ratio = 9...…… ……..……....**14**
   3. Run-to-riffle ratio = 10……...……**13**
   4. Run-to-riffle ratio = 12……..……**12**
   5. Run-to-riffle ratio = 14……**11**

C. Occasional riffle; variable bottom contours may provide some habitat.
   1. Run-to-riffle ratio = 16……**10**
   2. Run-to-riffle ratio = 18……...**9**
   3. Run-to-riffle ratio = 20……...**8**
   4. Run-to-riffle ratio = 22……..…**7**
   5. Run-to-riffle ratio = 24……...**6**

D. Generally all flat water; any riffles present will be shallow; essentially a straight and uniform stream depth; riffles are not deep enough to provide free passage for fish.
   1. Run-to-riffle ratio = 25……...**4**
   2. Run-to-riffle ratio = 26 – 30……...**3**
   3. Run-to-riffle ratio > 30 with some shallow riffles and short runs.……**2**
   4. No riffles present within stream reach……..……..……**0**

7. Channel Flow Status

Evaluates the degree to which the channel is filled with water when the stream reach is sampled. The flow status will change as the channel enlarges or as flow decreases due to dams and other obstructions, diversion for irrigation, drought, or aggrading stream bottoms with actively widening channels. This is a seasonal parameter. A decrease in water will wet smaller portions of the streambed, thus decreasing available habitat for aquatic organisms. Use the vegetation line on the lower bank as your reference point to estimate channel flow status.

A. Water reaches the base of both lower banks and minimal amount of channel substrate is exposed.
   1. 100% of channel is full……**20**
   2. > 90% of channel is full……...**18**

B. Water fills > 50% of the available channel (or < 50% of channel substrate is exposed).
   1. 80% - 90% of channel is full ……..**17**
   2. 70% - 80% of channel is full ……..**15**
   3. 60% - 70% of channel is full ……..**13**
   4. 50% - 60% of channel is full ……..**11**

C. Water fills 20% - 50% of the available channel and/or riffle substrates are mostly exposed.
   1. 40% - 50% of channel is full……**9**
   2. 30% - 40% of channel is full ……**7**
   3. 20% - 30% of channel is full…….. .**5**

D. Very little water in the channel and mostly present as standing pools
   1. 10% - 20% of channel is full ……**3**
   2. < 10% of channel is full ……**2**
   3. Water present as isolated standing pools……...**1**
   4. Channel is dry……....**0**

8. Bank Vegetative Protection

Measures the amount of the stream bank that is covered by vegetation. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetation protection or those shored up with concrete or riprap.

Four factors to consider when scoring bank vegetative protection: (1) Is the vegetation native or introduced? (2) Is the vegetation planted or natural? (3) Is the upper story, understory, and ground cover vegetation well balanced? (4) During which season are you conducting this assessment?

Determine left or right bank by facing downstream. Score banks separately.

A. More than 90% of the stream bank surface is covered by healthy, living vegetation. A variety of different types of vegetation is present (e.g. trees, shrubs, understory, and nonwoody macrophytes). Any bare or sparsely vegetated areas are small and evenly dispersed.
   1. 100% plant cover on stream bank……...……...……..**10**
   2. >90% plant cover on stream bank...……...……**9**

B. A variety of vegetation is present and covers 70 - 90% of stream bank surfaces, but one class of plants is not well represented. Some open areas with unstable substrate are present. Disruption evident but not affecting full plant growth potential. Few barren or thin areas are present.
   1. 90% plant cover on stream bank.....……..…**8**
   2. 80% - 90% plant cover on stream bank…….……....**7**
   3. 70% - 80% plant cover on stream bank with fewer plant species…….…….**6**

C. 50 - 70% of stream bank surface is covered by vegetation; typically composed of scattered shrubs, grasses, and forbes. Disruption obvious, with patches of bare soil and/or closely cropped vegetation common.
   1. 60% - 70% vegetation cover; typically of shrubs, grasses, and forbes……..**5**
   2. 50% - 60% vegetation cover; typically of shrubs, grasses, and forbes……..**4**

D. Less than 50% of the stream bank surface covered by vegetation. Disruption of vegetation is prevalent. Any shrubs or trees on bank exist as individuals or widely scattered clumps.
   1. 40% - 50% vegetation cover with many bare spots/rock…….. ……**3**
   2. 30% - 40% vegetation cover with many bare spots/rock…….……..……**2**
   3. 20% - 30% vegetation cover with many bare spots/rock…..……..……**1**
   4. < 20% vegetation cover…….…….**0**

9. Bank Stability

Measures whether the stream banks are eroded or have the potential for erosion. Steep banks are more likely to collapse and suffer from erosion than gently sloping banks and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroding banks cause sediment deposition and may reduce instream cover.

Determine left or right bank by facing downstream. Score banks separately.

A. Bank stable; erosion absent or minimal, with little potential for future problems. Slopes are generally less than 30º. Banks may be reinforced by rock thus increasing the slope to >30º while providing stability.
   1. No evidence of erosion or bank failure……**10**
   2. Less than 10% of bank affected by erosion……..……**9**

B. Moderately stable bank; small areas of erosion or bank slumping visible. Most areas are stable with only slight potential for erosion at flood stages. Slopes up to 40º. Banks may be reinforced by rock thus increasing the slope to >40º while providing stability.
   1. 10% - 20% of bank has erosional areas……..……..**8**
   2. 20% - 30% of bank has erosional areas……**7**
   3. 30% - 40% of bank has erosional areas……**6**

C. Moderately unstable bank; frequency and size of raw areas are such that high water events have eroded some areas of the bank. Medium size areas of erosion or bank slumping visible. Slopes up to 60º. High erosion potential during floods.
   1. 40% - 50% of bank has erosional areas……**5**
   2. 50% - 60% of bank has erosional areas……**4**
   3. 60% - 70% of bank has erosional areas……**3**

D. Unstable bank; mass erosion and bank failure are evident; erosion and pronounced undercutting present at bends and along some straight channel areas. Slopes > 60º are common. Areas of distinct slumping visible. Many raw areas are present and 70% – 100% of bank has erosional scars.
   1. 70% - 80% of bank has erosional areas……**2**
   2. 80% - 90% of bank has erosional areas……**1**
   3. >90% of bank has erosional areas……..……**0**

10. Riparian Vegetation Zone Width

Measures the width of natural vegetation from the edge of the upper stream bank out through the floodplain. The riparian vegetative zone serves as a buffer zone to pollutants entering a stream from runoff; controls erosion; and provides habitat and nutrients to the stream. Narrow, far less useful zones occur when roads, parking lots, fields (currently in use), heavily used paths, lawns, bare soil, rocks, or buildings are near the stream bank. When evaluating this metric, look for breaks in the riparian zone that allow sediment to pass through the zone.

Human activities that impact the riparian zone include: Parking Lots (**PL**), Paved Roads (**PR**), Dirt Roads (**DR**), Row Crop Agriculture (**RCA**), Animal Production Agriculture (**APA**), Silviculture (**S**), Residential Activities (**RA**), and Commercial/Industrial Activities (**CIA**)

Determine left or right bank by facing downstream. Score banks separately.

A. Width of riparian vegetation zone > 18 m (> 60'). Human activities have not impacted the zone.
   1. With no breaks……..……..…**10**
   2. With breaks; breaks are narrow and widely spaced…..……**9**

B. Width of riparian vegetation zone 12 – 18 m (40 – 60'). Human activities have impacted the zone only minimally.
   1. With no breaks……..……**8**
   2. With breaks …….. ……**7**

C. Width of riparian vegetation zone 6 – 12 m (20 – 40'). Human activities have impacted the zone a great deal.
   1. With no breaks……..……**6**
   2. With narrow breaks widely spaced……**5**
   3. With breaks common throughout riparian zone…….……..…..**4**

D. Width of riparian zone < 6 m (<20'). Little or no riparian vegetation due to human activities.
   1. Riparian vegetation zone less than 20' wide with no breaks……..……**3**
   2. Riparian vegetation zone less than 20' wide with breaks……..……**2**
   3. No riparian vegetation zone present. Canopy cleared to the edge of the stream bank. Surrounding area covered with grass/pasture……**1**
   4. Riparian vegetation zone absent. Vegetation cleared to the edge of the stream bank and the surrounding area is covered with pavement, concrete or some other artificial covering……**0**

Glide / Pool Habitat Assessment

Site ID:	**Date:**
Stream Name:	
Assessor:	

Habitat Parameter	Score	Notes
Bottom Substrate / Available Cover		_____ _____ _____
Pool Substrate Characterization		_____ _____ _____
Pool Variability		_____ _____ _____
Channel Alteration		_____ _____ _____
Sediment Deposition		_____ _____ _____
Channel Sinuosity*		***measured during stream reconnaissance**
Channel Flow Status		_____ _____
Bank Vegetative Protection **Left Bank** **Right Bank**	**LB** **RB**	_____ _____ _____ _____
Bank Stability **Left Bank** **Right Bank**	**LB** **RB**	_____ _____ _____ _____
Riparian Vegetative Zone **Left Bank** **Right Bank**	**LB** **RB**	_____ _____ _____ _____
Total Score →		

1. Bottom Substrate / Available Cover

Measures availability of substrates that can be used as refugia for aquatic organisms. A wide variety and/or abundance of submerged structures in the stream provide macroinvertebrates w/ a large number of niches, thus increasing the diversity of the aquatic community. As the variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases.

Possible Habitat Types:
Fallen Trees / Large Woody Debris (**LWD**), Deep Pools (**DP**), Shallow Pools (**SP**), Overhanging Shrubbery in stream (**OS**), Large Rocks (**LR**), Undercut Banks (**UB**), Thick Root Mats (**TRM**), Dense Macrophyte Beds (**DMB**), Deep Riffles with lots of turbulence (**DR**), Long Runs with cobble / large rock substrate (**RU**)

A. Stable and available habitats make up > 70% of reach
   1. **Seven** habitat types common……..**20**
   2. **Six** habitat types common, additional habitat types rare……..**19**
   3. **Five** habitat types common, additional habitat types rare……**18**
   4. **Four** habitat types common, additional habitat types rare……**17**
   5. Less than **four** habitat types present……..**16**

B. Stable and available habitats make up > 50% of reach
   1. **Seven** habitat types common……..**15**
   2. **Six** habitat types common, additional habitat types rare……..**14**
   3. **Five** habitat types common, additional habitat types rare……**13**
   4. **Four** habitat types common, additional habitat types rare……**12**
   5. Less than **four** habitat types present……..**11**

C. Stable and available habitats make up < 50% of reach
   1. **Seven** habitat types common……**10**
   2. **Six** habitat types common, additional habitat types rare……**9**
   3. **Five** habitat types common, additional habitat types rare……..**8**
   4. **Four** habitat types common, additional habitat types rare……..**7**
   5. **Three** habitat types common, additional habitat types rare……**6**

D. Two habitats or less common
   1. **Two** habitat types common, additional habitat types rare……**5**
   2. **Two** habitat types only and common……**4**
   3. **One** habitat type common, additional habitat types rare……**3**
   4. **One** habitat type only and common……**2**
   5. **One** habitat type rare……..**1**
   6. **No** available habitat in the reach……**0**

2. Pool Substrate Characterization

Evaluates the type and condition of bottom substrates found in pools. Firmer sediments and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants

A. A mixture of predominately firm substrate material, including gravel and firm sand; root mats and/or submerged vegetation common. Substrate consists of:
   1. Gravel, firm sand, root mats, and/or submerge vegetation……..**20**
   2. Gravel, root mats, and/or submerged vegetation……**19**
   3. Firm sand, root mats and/or submerge vegetation……..**18**

B. A heterogeneous mixture of soft substrates, including soft sand, mud, or clay; root mats and/or submerged vegetation present. Substrate consists of:
   1. Soft sand, mud, clay, root mats, and/or submerged vegetation……..**15**
   2. Soft sand, mud, root mats, and/or submerged vegetation……**14**
   3. Soft sand, clay, root mats, and/or submerged vegetation……**12**
   4. Clay, mud, root mats, and/or submerged vegetation……..**11**

C. Homogeneous substrate consisting of sand, mud, or clay; root mats sparse; submerged vegetation lacking. Substrate consists of:
   1. All sand bottom with few root mats……**10**
   2. All mud bottom with few root mats……..**8**
   3. All clay bottom with few root mats……..……..**6**

D. Homogeneous substrate consisting of sand, mud, clay, or bedrock with no root material. Substrate consists of:
   1. All sand bottom with no root material……..**5**
   2. All mud bottom with no root material……..**3**
   3. All clay bottom with no root material……**1**
   4. All bedrock or hardpan clay bottom……**0**

3. Pool Variability

Rates the overall mixture of pool types according to size and depth. Increased pool variability in a stream accommodates a diverse aquatic community consisting of a variety of species and age classes. In streams with low sinuosity and monotonous pool characteristics, very little instream habitat variety exists to support a diverse community. The four basic types of pools are **large-shallow, large-deep, small-shallow, and small-deep**. Any pool dimension greater than half the width of the stream is a large pool. Small pools have length and width dimensions less than half the width of the stream. Pools with depths greater than 1.0m are considered to be deep pools. Shallow pools are 0.5m to 1.0m deep. Aeration occurs at any area where the stream surface is broken (e.g. dams, water falling over woody debris, riffles).

A. All pool sizes (area and depth) present and mixed.
   1. All sizes evenly mixed and below areas of aeration ……...**20**
   2. All sizes evenly mixed; found below and above aeration areas ……**18**
   3. All sizes evenly mixed above areas of aeration or aeration lacking ……...**16**

B. Majority of pools are deep; very few shallow pools present.
   1. Large and small deep pools evenly mixed and below areas of aeration ...**15**
   2. Majority of pools are large-deep and below areas of aeration ..……..……..**14**
   3. Large and small deep pools evenly mixed above and below areas of aeration ……...……...**13**
   4. Majority of pools are large-deep; found above and below areas of a aeration ……...……..……**12**
   5. Majority of pools are large-deep above areas of aeration or aeration lacking……..……..**11**

C. Shallow pools are more prevalent than deep pools.
   1. Large and small shallow pools evenly mixed and all below areas of aeration ..……..……...**10**
   2. Majority of pools are large-shallow and below areas of aeration ..…….….**9**
   3. Large and small shallow pools evenly mixed above and below areas of aeration ……..……**8**
   4. Majority of pools are large-shallow and found above and below areas of aeration ……..…**7**
   5. Majority of pools are large-shallow above areas of aeration or aeration lacking ……..**6**

D. Majority of pools small-shallow or pools absent.
   1. Majority of pools are small-shallow and below areas of aeration ……**5**
   2. Majority of pools are small-shallow above and below aeration areas ……..**4**
   3. Majority of pools are small-shallow above areas of aeration or aeration lacking ..……**2**
   4. Pools absent from sample reach……..……..**0**

4. Channel Alteration

Measures any large-scale alteration of instream habitat that affects stream sinuosity and causes scouring. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances due to dredging activities; when dams, culverts, or bridges are present; or when other morphological changes have occurred.

A. Stream flows a normal and natural meandering pattern. Alteration is absent.
   1. No evidence of disturbance with bends/runs frequent; bend angles average >60°.. . ……...…**20**
   2. No evidence of disturbance with bends/runs frequent; bend angles average 40° - 60°……**18**
   3. No evidence of disturbance with bends/runs frequent; bend angles average <40°……..……**16**

B. Some stream straightening, dredging, artificial embankments, or dams present but NO evidence of recent alteration activities. Alteration probably occurred more than 20 years ago. Stream appears to be in the process of recovery.
   1. Less than 20% of reach has channel disturbance……..…**15**
   2. 20% - 40% of reach has channel disturbance…..…….……**14**
   3. 40% - 60% of reach has channel disturbance……..……...…**13**
   4. 60% - 80% of reach has channel disturbance...……...**12**
   5. 80% - 100% of reach has channel disturbance..…..……..…**11**

C. Stream has been altered or channelized. Alteration probably occurred less than 20 years ago.
   1. Less than 20% of reach has channel disturbance……...……**10**
   2. 20% - 40% of reach has channel disturbance…..……**9**
   3. 40% - 60% of reach has channel disturbance..……..**8**
   4. 60% - 80% of reach has channel disturbance..……..**7**
   5. 80% - 100% of reach has channel disturbance...…..……...……**6**

D. Instream habitat highly altered. More than 80% of the stream reach has been altered. Alteration may be recent (<10 years).
   1. >90 % of reach has channel disturbance……..……..**5**
   2. Channel reach 100% disturbed; straight with no artificial embankments……...**3**
   3. Channel reach 100% disturbed; straight with some artificial embankments……...**1**
   4. Banks 100% shored by gabion, cement, and/or riprap....……...**0**

5. Sediment Deposition

Relates to the amount of sediment that has accumulated and the changes that have occurred to the stream bottom as a result of deposition. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or results in the filling of pools and runs. High levels of sediment deposition are symptoms of an unstable environment that may be unsuitable for many organisms.

A. No enlargements of islands/point bars present; <30% of the stream bottom affected by sand or silt accumulation.
   1. <20% sediment deposition with accumulation in pools only……**20**
   2. <20% sediment deposition with accumulation in pools only……**19**
   3. 20% - 30% sediment deposition with gravel and/or coarse sand……**18**
   4. 20% - 30% sediment deposition with fine sand and/or silt……**17**

B. 30% - 60% of the stream bottom affected by sand and/or silt accumulation; increased deposition in pools and runs; some new increase in bar and island formation.
   1. 30% - 40% sediment deposition with gravel and/or coarse sand……**15**
   2. 30% - 40% sediment deposition with fine sand and/or silt……**14**
   3. 40% - 50% sediment deposition with gravel and/or coarse sand……**13**
   4. 40% - 50% sediment deposition with fine sand and/or silt……**12**
   5. 50% - 60% sediment deposition with gravel and/or coarse sand……**11**

C. 60% - 80% of the stream bottom affected with increased deposition in pools. Number of shallow pools increases. Instream habitats smothered by sand, silt, and fine gravel. Deposits of gravel, sand and silt observed on old and new point bars, islands, and behind obstructions. Formation of few new bars/islands is evident and old bars are deep and wide; deposition at bends obvious.
   1. 50% - 60% sediment deposition with fine sand and/or silt……**10**
   1. 60% - 70% sediment deposition with gravel and/or coarse sand……**9**
   2. 60% - 70% sediment deposition with fine sand and/or silt……**8**
   3. 70% - 80% sediment deposition with gravel and/or coarse sand……**7**
   4. 70% - 80% sediment deposition with fine sand and/or silt……**6**

D. >80% of the stream bottom affected with heavy deposition from fine gravel and sand at stream bends, constrictions, and/or pools. Extensive deposits of fine sand and/or silt on old and new bars, islands, and along banks in straight channels. Few pools are present due to siltation.
   1. 80% - 90% sediment deposition with gravel and/or coarse sand……**4**
   2. 80% - 90% sediment deposition with fine sand and/or silt……**3**
   3. >90% sediment deposition; pools almost absent……**1**
   4. 100% sediment deposition; pools absent due to substantial deposition; bottom silt moves with almost any flow above normal……**0**

6. Channel Sinuosity

Evaluates the meandering or sinuosity of the stream. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding. In some streams, a longer reach than that designated for sampling may need to be evaluated to adequately score this metric.

Channel Sinuosity = Mean Distance Between Bends / Mean Stream Width

Channel sinuosity is determined during stream reconnaissance.

A. Occurrences of bends relatively frequent. Pools and other instream habitats abundant throughout the sample reach.
   1. Run-to-bend ratio = 1-2 ……**20**
   2. Run-to-bend ratio = 3-4……**19**
   3. Run-to-bend ratio = 5……**18**
   4. Run-to-bend ratio = 6……**17**
   5. Run-to-bend ratio = 7……**16**

B. Occurrence of bends infrequent. Adequate pool and other instream habitats throughout reach.
   1. Run-to-bend ratio = 8……**15**
   2. Run-to-bend ratio = 9……**14**
   3. Run-to-bend ratio = 10……**13**
   4. Run-to-bend ratio = 12……**12**
   5. Run-to-bend ratio = 14……**11**

C. Occasional bends; variable bottom contours may provide some habitat.
   1. Run-to-bend ratio = 16……**10**
   2. Run-to-bend ratio = 18……**9**
   3. Run-to-bend ratio = 20……**8**
   4. Run-to-bend ratio = 22……**7**
   5. Run-to-bend ratio = 24……**6**

D. Essentially a straight stream of uniform depth. Sample reach has most likely been straighten or channelized. Instream cover and pool habitat lacking.
   1. Run-to-bend ratio = 25……**4**
   2. Run-to-bend ratio = 26 – 30……**3**
   3. Run-to-bend ratio = 30 ……**2**
   4. No bends within stream reach ……**0**

7. Channel Flow Status

Evaluates the degree to which the channel is filled with water when the stream reach is sampled. The flow status will change as the channel enlarges or as flow decreases due to dams and other obstructions, diversion for irrigation, drought, or aggrading stream bottoms with actively widening channels. This is a seasonal parameter. A decrease in water will wet smaller portions of the streambed, thus decreasing available habitat for aquatic organisms. Use the vegetation line on the lower bank as your reference point to estimate channel flow status.

A. Water reaches the base of both lower banks and minimal amount of channel substrate is exposed.
   1. 100% of channel is full……**20**
   2. > 90% of channel is full……...**18**

B. Water fills > 50% of the available channel (or < 50% of channel substrate is exposed).
   1. 80% - 90% of channel is full ……..**17**
   2. 70% - 80% of channel is full ……..**15**
   3. 60% - 70% of channel is full ……..**13**
   4. 50% - 60% of channel is full ……...**11**

C. Water fills 20% - 50% of the available channel and/or riffle substrates are mostly exposed.
   1. 40% - 50% of channel is full……**9**
   2. 30% - 40% of channel is full ……**7**
   3. 20% - 30% of channel is full…….. .**5**

D. Very little water in the channel and mostly present as standing pools
   1. 10% - 20% of channel is full ……**3**
   2. < 10% of channel is full ……**2**
   3. Water present as isolated standing pools……...**1**
   4. Channel is dry…….……....**0**

8. Bank Vegetative Protection

Measures the amount of the stream bank that is covered by vegetation. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetation protection or those shored up with concrete or riprap.

Four factors to consider when scoring bank vegetative protection: (1) Is the vegetation native or introduced? (2) Is the vegetation planted or natural? (3) Is the upper story, understory, and ground cover vegetation well balanced? (4) During which season are you conducting this assessment?

Determine left or right bank by facing downstream. Score banks separately.

A. More than 90% of the stream bank surface is covered by healthy, living vegetation. A variety of different types of vegetation are present (e.g. trees, shrubs, understory, and nonwoody macrophytes). Any bare or sparsely vegetated areas are small and evenly dispersed.
   1. 100% plant cover on stream bank……...……...……..**10**
   2. >90% plant cover on stream bank...……...……**9**

B. A variety of vegetation is present and covers 70 - 90% of stream bank surfaces, but one class of plants is not well represented. Some open areas with unstable substrate are present. Disruption evident but not affecting full plant growth potential. Few barren or thin areas are present.
   1. 90% plant cover on stream bank....……...…**8**
   2. 80% - 90% plant cover on stream bank…….……....**7**
   3. 70% - 80% plant cover on stream bank with fewer plant species…….……**6**

C. 50 - 70% of stream bank surface is covered by vegetation; typically composed of scattered shrubs, grasses, and forbes. Disruption obvious, with patches of bare soil and/or closely cropped vegetation common.
   1. 60% - 70% vegetation cover; typically of shrubs, grasses, and forbes……..**5**
   2. 50% - 60% vegetation cover; typically of shrubs, grasses, and forbes……..**4**

D. Less than 50% of the stream bank surface covered by vegetation. Disruption of vegetation is prevalent. Any shrubs or trees on bank exist as individuals or widely scattered clumps.
   1. 40% - 50% vegetation cover with many bare spots/rock…….. ……**3**
   2. 30% - 40% vegetation cover with many bare spots/rock…….……..……**2**
   3. 20% - 30% vegetation cover with many bare spots/rock….……..……**1**
   4. < 20% vegetation cover…….…….**0**

9. Bank Stability

Measures whether the stream banks are eroded or have the potential for erosion. Steep banks are more likely to collapse and suffer from erosion than gently sloping banks and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroding banks cause sediment deposition and may reduce instream cover.

Determine left or right bank by facing downstream. Score banks separately.

A. Bank stable; erosion absent or minimal, with little potential for future problems. Slopes are generally less than 30º. Banks may be reinforced by rock thus increasing the slope to >30º while providing stability.
   1. No evidence of erosion or bank failure……**10**
   2. Less than 10% of bank affected by erosion……..……..……**9**

B. Moderately stable bank; small areas of erosion or bank slumping visible. Most areas are stable with only slight potential for erosion at flood stages. Slopes up to 40º. Banks may be reinforced by rock thus increasing the slope to >40º while providing stability.
   1. 10% - 20% of bank has erosional areas……..……..**8**
   2. 20% - 30% of bank has erosional areas……**7**
   3. 30% - 40% of bank has erosional areas……**6**

C. Moderately unstable bank; frequency and size of raw areas are such that high water events have eroded some areas of the bank. Medium size areas of erosion or bank slumping visible. Slopes up to 60º. High erosion potential during floods.
   1. 40% - 50% of bank has erosional areas……**5**
   2. 50% - 60% of bank has erosional areas……**4**
   3. 60% - 70% of bank has erosional areas……**3**

D. Unstable bank; mass erosion and bank failure are evident; erosion and pronounced undercutting present at bends and along some straight channel areas. Slopes > 60º are common. Areas of distinct slumping visible. Many raw areas are present and 70% – 100% of bank has erosional scars.
   1. 70% - 80% of bank has erosional areas……**2**
   2. 80% - 90% of bank has erosional areas……**1**
   3. >90% of stream bank has eroded……...……**0**

10. Riparian Vegetation Zone Width

Measures the width of natural vegetation from the edge of the upper stream bank out through the floodplain. The riparian vegetative zone serves as a buffer zone to pollutants entering a stream from runoff; controls erosion; and provides habitat and nutrients to the stream. Narrow, far less useful zones occur when roads, parking lots, fields (currently in use), heavily used paths, lawns, bare soil, rocks, or buildings are near the stream bank. When evaluating this metric, look for breaks in the riparian zone that allow sediment to pass through the zone.

Human activities that impact the riparian zone include: Parking Lots (**PL**), Paved Roads (**PR**), Dirt Roads (**DR**), Row Crop Agriculture (**RCA**), Animal Production Agriculture (**APA**), Silviculture (**S**), Residential Activities (**RA**), and Commercial/Industrial Activities (**CIA**)

Determine left or right bank by facing downstream. Score banks separately.

A. Width of riparian vegetation zone > 18 m (> 60'). Human activities have not impacted the zone.
   1. With no breaks……..……..**10**
   2. With breaks; breaks are narrow and widely spaced…..……**9**

B. Width of riparian vegetation zone 12 – 18 m (40 – 60'). Human activities have impacted the zone only minimally.
   1. With no breaks……..……**8**
   2. With breaks …….. ……**7**

C. Width of riparian vegetation zone 6 – 12 m (20 – 40'). Human activities have impacted the zone a great deal.
   1. With no breaks……..……**6**
   2. With narrow breaks widely spaced……**5**
   3. With breaks common throughout riparian zone…….……..….**4**

D. Width of riparian vegetation zone < 6 m (<20'). Little or no riparian vegetation due to human activities.
   1. Riparian vegetation zone less than 20' wide with no breaks…..…….……..**3**
   2. Riparian vegetation zone less than 20' wide with breaks……..……**2**
   3. No riparian vegetation zone present. Canopy cleared to the edge of the stream bank. Surrounding area covered with grass/pasture…….....**1**
   4. Riparian vegetation zone absent. Vegetation cleared to the edge of the stream bank and the surrounding area is covered with pavement, concrete, or some other artificial covering……..……...**0**

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APPENDIX 5B-1
Scoring Criteria for the Index of Biotic Integrity and the Index of Well-Being to Monitor Fish Communities in Wadeable Streams in the Piedmont Ecoregion of Georgia

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Part II: Scoring Criteria for the Index of Biotic Integrity and the Index of Well-Being to Monitor Fish Communities in Wadeable Streams in the Piedmont Ecoregion of Georgia

Georgia Department of Natural Resources
Wildlife Resources Division
Fisheries Management Section

June 1, 2005

Table of Contents

Introduction

The Piedmont ecoregion is one of six Level III ecoregions found in Georgia (Part 1, Figure 1). The Piedmont ecoregion covers most of the north central portion of Georgia, between the Southeastern Plains ecoregion to the south and the Ridge and Valley and Blue Ridge Mountains ecoregions to the north. It is the second largest ecoregion in Georgia, covering over 17,000 square miles (United States Census Bureau 2000) and including all or portions of 65 counties (Fig. 1).

The biotic indices developed by the GAWRD are based on the Level III ecoregion delineations (Griffith et al 2001). The metrics and scoring criteria adapted to the Piedmont ecoregion were developed from biomonitoring samples collected in the eight major drainage basins located in the Piedmont ecoregion. These major drainage basins include the Chattahoochee, Coosa, Flint, Ocmulgee, Oconee, Ogeechee, Savannah, and Tallapoosa. Based on similarities in species richness and composition, the eight major drainage basins were aligned into three basin groups. The Alabama Drainage Basin (ACT) includes the Coosa and Tallapoosa drainage basins; the Apalachicola Drainage Basin (ACF) includes the Chattahoochee and Flint drainage basins; and the Atlantic Slope Drainage Basins (AS) include the Ocmulgee, Oconee, Ogeechee, and Savannah drainage basins. A total of 378 biomonitoring samples have been collected by the GAWRD in the Piedmont ecoregion since 1998.

The Alabama drainage basin was the most species rich in the Piedmont ecoregion, with a total of 64 native species collected. Fifty-seven native species were collected in the Atlantic Slope drainage basins and 55 native species were collected in the Apalachicola drainage basin. A total of 14 state listed species were collected in the Piedmont ecoregion. The state listed fish collected in the Piedmont ecoregion were ranked as endangered (E), threatened (T), or rare (R) based on the Endangered Wildlife Act of 1973 (Georgia Department of Natural Resources, Nongame – Endangered Wildlife Program, 1999). Endangered species collected in the Piedmont ecoregion include the Altamaha shiner (*Cyprinella xaenurus*), collected in the Ocmulgee and Oconee drainage basins, and the lipstick darter (*Etheostoma chuckwachatte*), found in the Tallapoosa drainage basin. Threaten species included the bluestripe shiner (*Cyprinella callitaenia*) collected in the Flint drainage basin, the pretty shiner (*Lythrurus bellus*), collected in the Tallapoosa drainage basin, the highscale shiner (*Notropis hypsilepis*), collected in the

Chattahoochee and Flint drainage basins, and the holiday darter (*Etheostoma brevirostrum*), Etowah darter (*Etheostoma etowahae*), and Cherokee darter (*Etheostoma scotti*), all collected in the Coosa drainage basin. Species ranked as rare included the Tallapoosa shiner (*Cyprinella gibbsi*), found in the Tallapoosa drainage basin, the sandbar shiner (*Notropis scepticus*), found in the Savannah drainage basin, the black madtom (*Noturus funebris*), found in the Chattahoochee and Tallapoosa drainage basins, the goldstripe darter (*Etheostoma parvipinne*), collected in the Ocmulgee drainage basin, and the Tallapoosa darter (*Etheostomna tallapoosae*) and the muscadine bridled darter (*Percina sp.*), found in the Tallapoosa drainage basin. The Etowah darter (endangered) and the Cherokee darter (threatened) are both federally listed under the Endangered Species Act of 1973. Table 1 shows a complete list of state listed fish found in the Piedmont ecoregion of Georgia.

Based on the IBI integrity classes (Part I, Table 2), 25 sites scored in the excellent class, 54 scored in the good class, 108 scored in the fair class, 87 scored in the poor class, and 104 scored in the very poor class. IBI scores in the Piedmont ecoregion ranged from a maximum of 58 to a minimum of 10. Based on the IBI scoring criteria, over 50% of the streams sampled in the Piedmont ecoregion scored in the poor and very poor integrity class ([191/378] * 100 = 50.5). Major impacts to streams in the Piedmont ecoregion include the effects of erosion and sedimentation, impoundments, point source pollution, and urban / suburban development. The Piedmont ecoregion is the most densely populated area in Georgia, averaging nearly 316 individuals per square mile (United States Census Bureau 2000). Approximately 63% of the total population of Georgia lives in the Piedmont ecoregion, an area covering only 29.5% of the entire state. Most of the major metropolitan areas in Georgia are located in the Piedmont ecoregion, including Atlanta, Gainesville, Athens, and portions of Columbus, Macon, and Augusta.

Table 2 shows the scoring criteria for the IBI metrics in the Piedmont ecoregion. The Maximum Species Richness (MSR) graphs for each basin group within the Piedmont ecoregion are included in Appendix 1. Figures ACF1 – PDT through ACF6b - PDT depict the MSR graphs used to score the species richness metrics (metrics 1- 6b) in the Apalachicola drainage basin. Figures ACT1 - PDT through ACT6b - PDT depict the MSR graphs used to score the species richness metrics in the Alabama drainage basin. Figures AS1 - PDT through AS6b - PDT depict the MSR graphs used to score the species richness metrics in the Atlantic Slope drainage basins.

The fish list for the Piedmont ecoregion showing the water quality tolerance rankings, feeding guilds, and species categories used in calculating the IBI score is also included in Appendix 1.

Based on the modified Index of Well-Being integrity classes for the Piedmont ecoregion (Table 3), 35 sites scored in the excellent class, 65 scored in the good class, 153 scored in the fair class, 40 scored in the poor class, and 62 scored in the very poor class. Modified Iwb scores in headwater streams ranged from a maximum score of 9.98 to a minimum of 0.21. At larger wadeable streams modified Iwb scores ranged from a maximum of 10.58 to a minimum of 3.83. There was a significant relationship between the indices across the Piedmont ecoregion ($r = 0.8051$, $p = 0.0000$, $N = 355$), although the relationship was stronger in the larger wadeable streams ($r = 0.8701$, $p = 0.0000$, $N = 95$) compared to the headwater streams ($r = 0.7797$, $p = 0.0000$, $N = 260$).

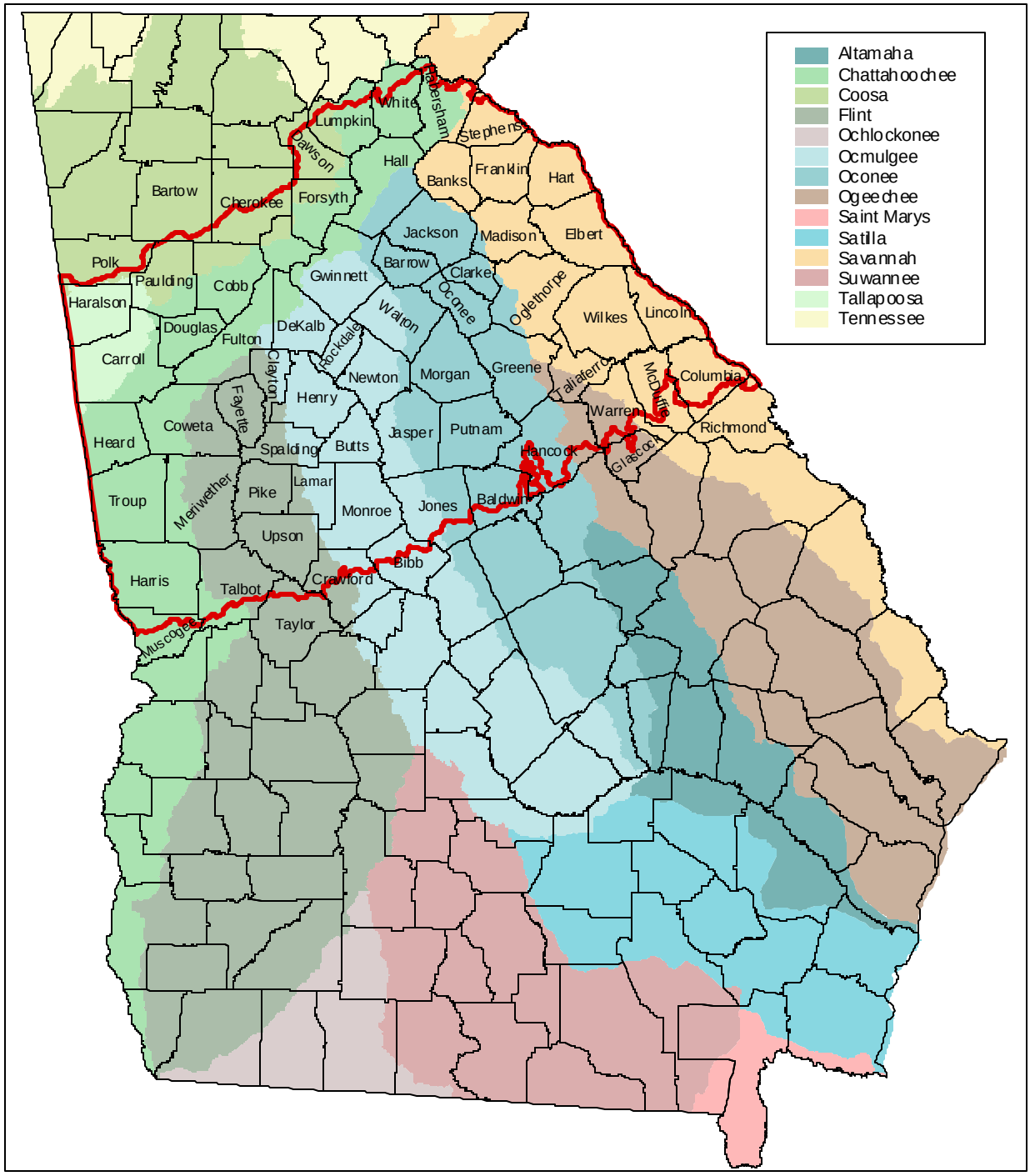


Figure 1. Level III Piedmont ecoregion (outlined in bold red) in Georgia. Major drainage basins include the Coosa, Tallapoosa, Chattahoochee, Flint, Ocmulgee, Oconee, Ogeechee, and Savannah.

Table 1. State listed fish found in the Piedmont ecoregion of Georgia (Georgia Department of Natural Resources, Nongame – Endangered Wildlife Program, 1999).

Species	State Status	Federal Status	Basin
Bluestripe Shiner (*Cyprinella callitaenia)*	T	None	CHA, FLI
Tallapoosa Shiner *(Cyprinella gibbsi)*	R	None	TAL
Altamaha Shiner *(Cyprinella xaenura)*	E	None	OCM, OCO
Holiday Darter *(Etheostoma brevirostrum)*	T	None	COO
Lipstick Darter *(Etheostoma chuckwachatte)*	E	None	TAL
Etowah Darter *(Etheostoma etowahae)*	T	E	COO
Goldstripe Darter *(Etheostoma parvipinne)*	R	None	CHA, FLI, OCM
Cherokee Darter *(Etheostoma scotti)*	T	T	COO
Tallapoosa Darter *(Etheostoma tallaposae)*	R	None	TAL
Stippled Studfish *(Fundulus bifax)*	E	None	TAL
Pretty Shiner *(Lythrurus bellus)*	T	None	TAL
Robust Redhorse *(Moxostoma robustum)*	E	None	OCO, SAV
Highscale Shiner *(Notropis hypsilepis)*	T	None	CHA, FLI
Sandbar Shiner *(Notropis scepticus)*	R	None	SAV
Black Madtom *(Noturus funebris)*	R	None	CHA, TAL
Frecklebelly Madtom *(Noturus munitus)*	E	None	COO
Amber Darter *(Percina antesella)*	E	E	COO
Freckled Darter *(Percina lenticula)*	E	None	COO
Muscadine Bridled Darter *(Percina* sp.*)*	R	None	TAL
Upland Bridled Darter *(Percina* sp.*)*	R	None	COO

Status: E = endangered; R = rare; T = threatened
Basin: CHA = Chattahoochee; COO = Coosa; OCM = Ocmulgee; OCO = Oconee; SAV = Savannah; TAL = Tallapoosa

Table 2. Index of Biotic Integrity metrics for wadeable streams in the Piedmont ecoregion of Georgia. ACF includes the Chattahoochee and Flint drainage basins, ACT includes the Coosa and Tallapoosa drainage basins, and AS includes the Ocumulgee, Oconee, Ogeechee, and Savannah drainage basins.

Metric	Basin Group	Scoring Criteria		
1. Number of native species	ACF/ACT/AS	MSR Graphs		
2. Number of benthic invertivore species	ACF/ACT/AS	MSR Graphs		
3a. Number of native sunfish species[a]	ACF/ACT/AS	MSR Graphs		
3b. Number of native centrarchid species[b]	ACF/ACT/AS	MSR Graphs		
4. Number of native insectivorous cyprinid species	ACF/ACT/AS	MSR Graphs		
5. Number of native round-bodied sucker species	ACF/ACT/AS	MSR Graphs		
6a. Number of sensitive species[a]	ACF/ACT/AS	MSR Graphs		
6b. Number of intolerant species[b]	ACF/ACT/AS	MSR Graphs		
		5	3	1
7. Evenness	ACF	≥ 72	72 - ≥ 62	< 62
	ACT	≥ 79	79 - ≥ 69	< 69
	AS	≥ 68	68 - ≥ 57	< 57
8. % of individuals as *Lepomis* species	ACF	≤ 27	27 - ≤ 53	> 53
	ACT	≤ 23	23 - ≤ 46	> 46
	AS	≤ 23	23 - ≤ 45	> 45

		5	3	1
9. % of individuals as insectivorous cyprinids	ACF	≥ 42	42 - ≥ 21	< 21
	ACT	≥ 32	32 - ≥ 16	< 16
	AS	≥ 50	50 - ≥ 27	< 27
10a. % of individuals as generalist feeders and herbivores[a]	ACF	≤ 22	22 - ≤ 40	> 40
	ACT	≤ 15	15 - ≥ 28	> 28
	AS	≤ 20	20 - ≤ 36	> 36
10b. % of individuals as top carnivores[b]	ACF/ACT/AS	≥ 3.8 - ≤ 9.5	≥ 1.9 – < 3.8 or > 9.5 - ≤ 11.4	< 1.9 or > 11.4
11. % of individuals as benthic fluvial specialist	ACF	≥ 38	38 - ≥ 19	< 19
	ACT	≥ 36	36 - ≥ 21	< 21
	AS	≥ 28	28 - ≥ 14	< 14
12. Number of individuals per 200 meters	ACF	≥ 670	670 - ≥ 335	< 335
	ACT	≥ 450	450 - ≥ 225	< 225
	AS	≥ 640	640 - ≥ 320	< 320
13. % of individuals with external anomalies	ACF/ACT/AS		> 1.2 – subtract 4 points from total score	

[a] used at sites with an upstream drainage basin area < 15 square miles
[b] used at sites with an upstream drainage basin area ≥ 15 square miles

Table 3. Index of well-being scoring criteria and integrity classes for wadeable streams in the Piedmont ecoregion of Georgia.

Iwb Score	DBA (Sq. miles)	Integrity Class	Attributes
≥ 8.1	< 15	Excellent	Comparable to the best regional reference conditions; all regionally expected species for the habitat and stream size, including the most intolerant species, are present with a full array of size classes; healthy species diversity within the fish community, indicated by elevated evenness scores; number of individuals abundant; total biomass is high, with each level of the food web represented, indicating a balanced trophic structure.
≥ 9.6	≥ 15		
8.1 - ≥ 7.3	< 15	Good	Species richness somewhat below expectation; evenness scores decrease as species diversity falls, especially due to the loss of the most intolerant forms; good number of individuals in the sample, with several species of benthic fluvial specialists and insectivorous cyprinids present; some decreases in total biomass as trophic structure shows some signs of stress.
9.6 - ≥ 8.6	≥ 15		
7.3 - ≥ 5.7	< 15	Fair	Species richness and diversity decline as some expected species are absent; abundance of individuals declines; total biomass continues to decline as some levels of the food web in low abundance or missing; trophic structure skewed toward generalist feeders and/or *Lepomis* species as the abundance of insectivorous cyprinid and benthic fluvial specialist species decreases.
8.6 - ≥ 6.6	≥ 15		
5.7 - ≥ 4.9	< 15	Poor	Number of individuals is low; species richness and diversity are very low, with benthic fluvial specialist and insectivorous cyprinid species in low abundance or absent; sample dominated by generalist feeders, herbivores, and *Lepomis* species; increase in the proportions of non-native species and hybrids; growth rates depressed as sample is heavily skewed to the smaller size classes; total biomass low.
6.6 - ≥ 5.6	≥ 15		
< 4.9	< 15	Very Poor	Sample represented by few individuals, mainly generalist feeders and *Lepomis* species; some sites dominated by non-native species; total biomass very low.
< 5.6	≥ 15		

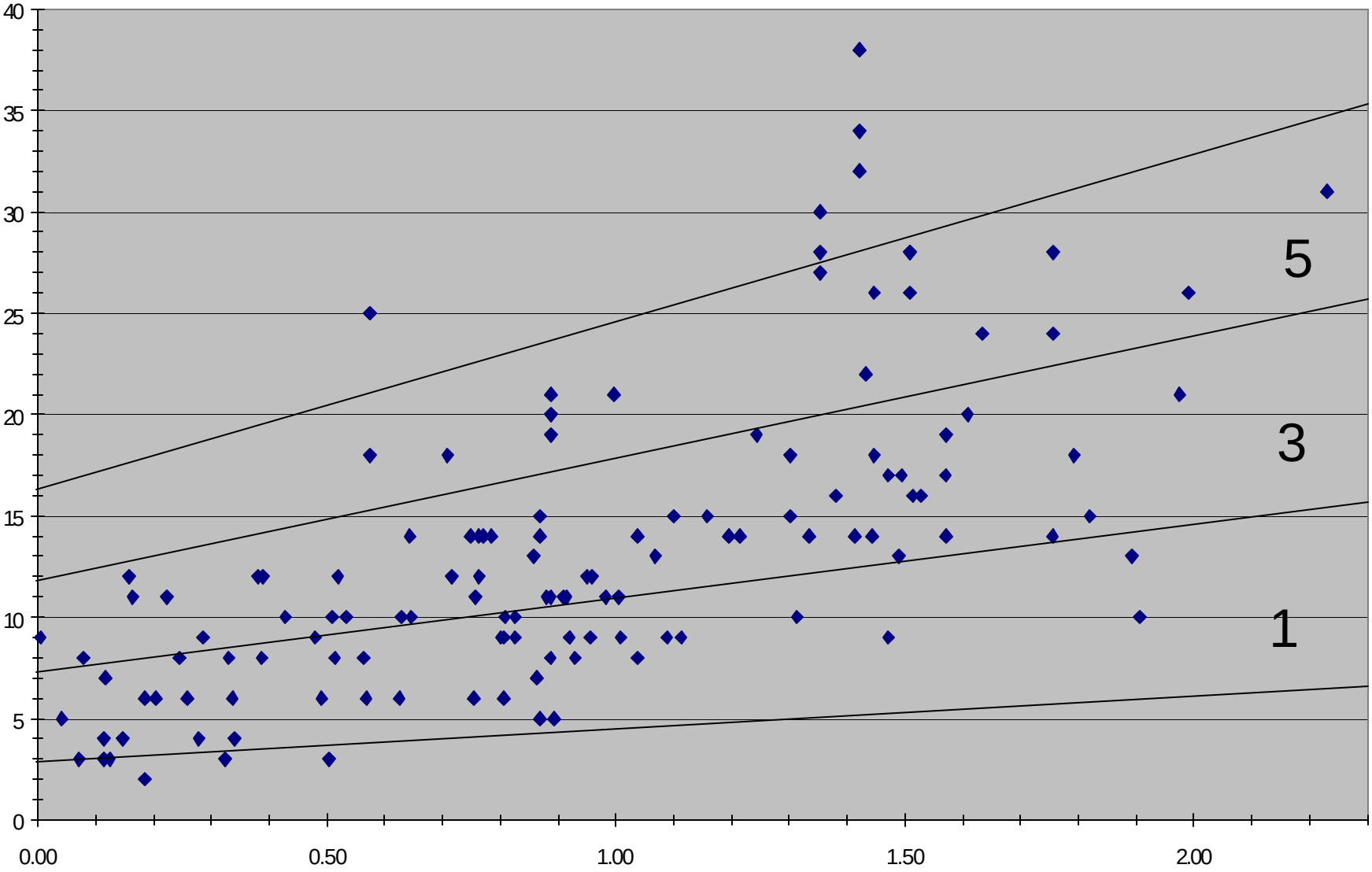
References

Georgia Department of Natural Resources, Wildlife Resources Division. 1999. Protected Animals of Georgia. Nongame Wildlife – Natural Heritage Section, Forsyth, Georgia.

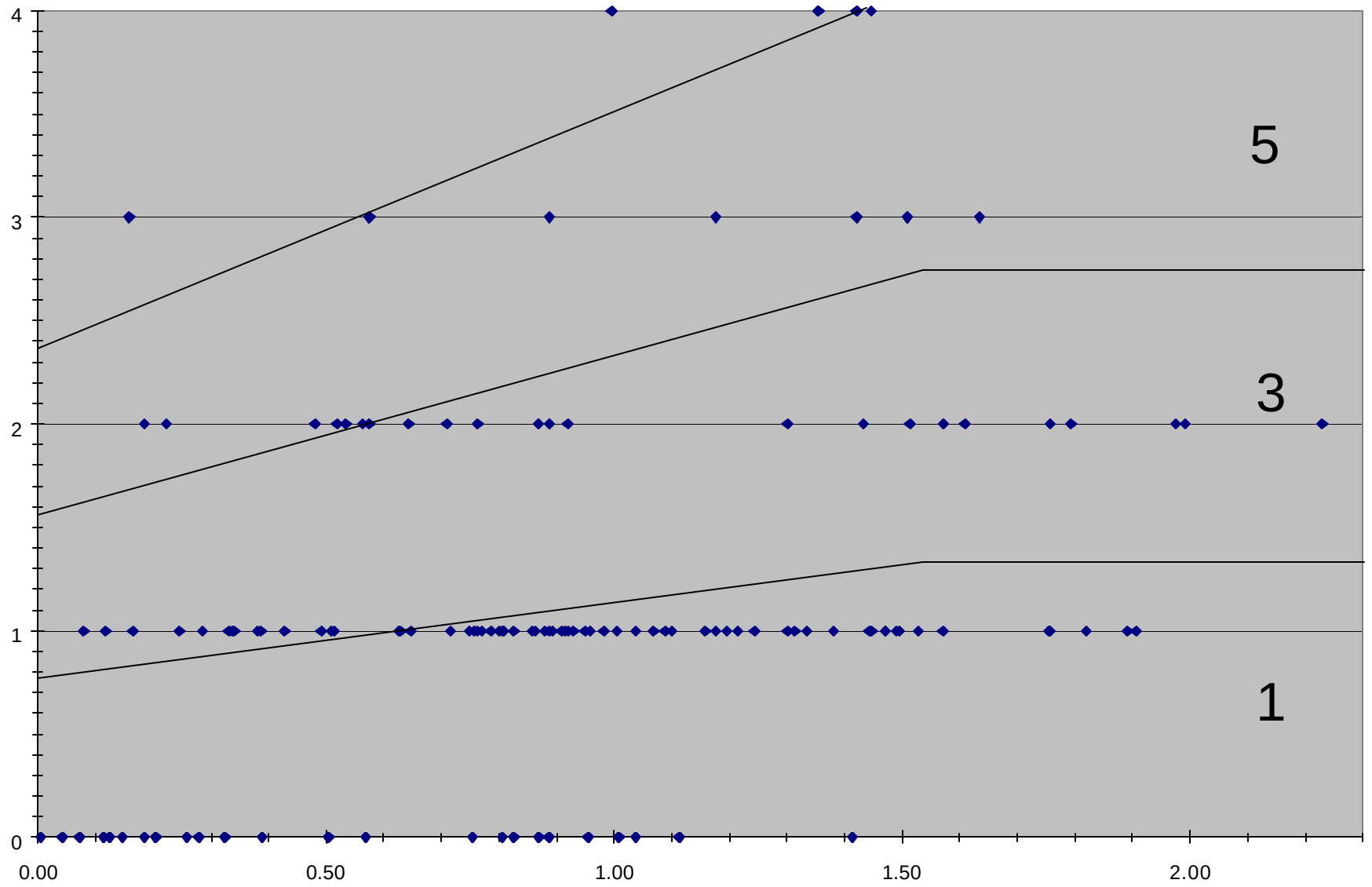
Griffith, G.E., J.M. Omernik, J.A. Comstock, S. Lawrence, and T. Foster. 2001. Level III and IV Ecoregions of Georgia, (color poster with map, descriptive text, summary tables, and photographs). Reston, Virginia, U.S. Geological Survey.

United States Census Bureau. 2000. 2000 Census of Population and Housing. United States Census Bureau, Washington, D.C.

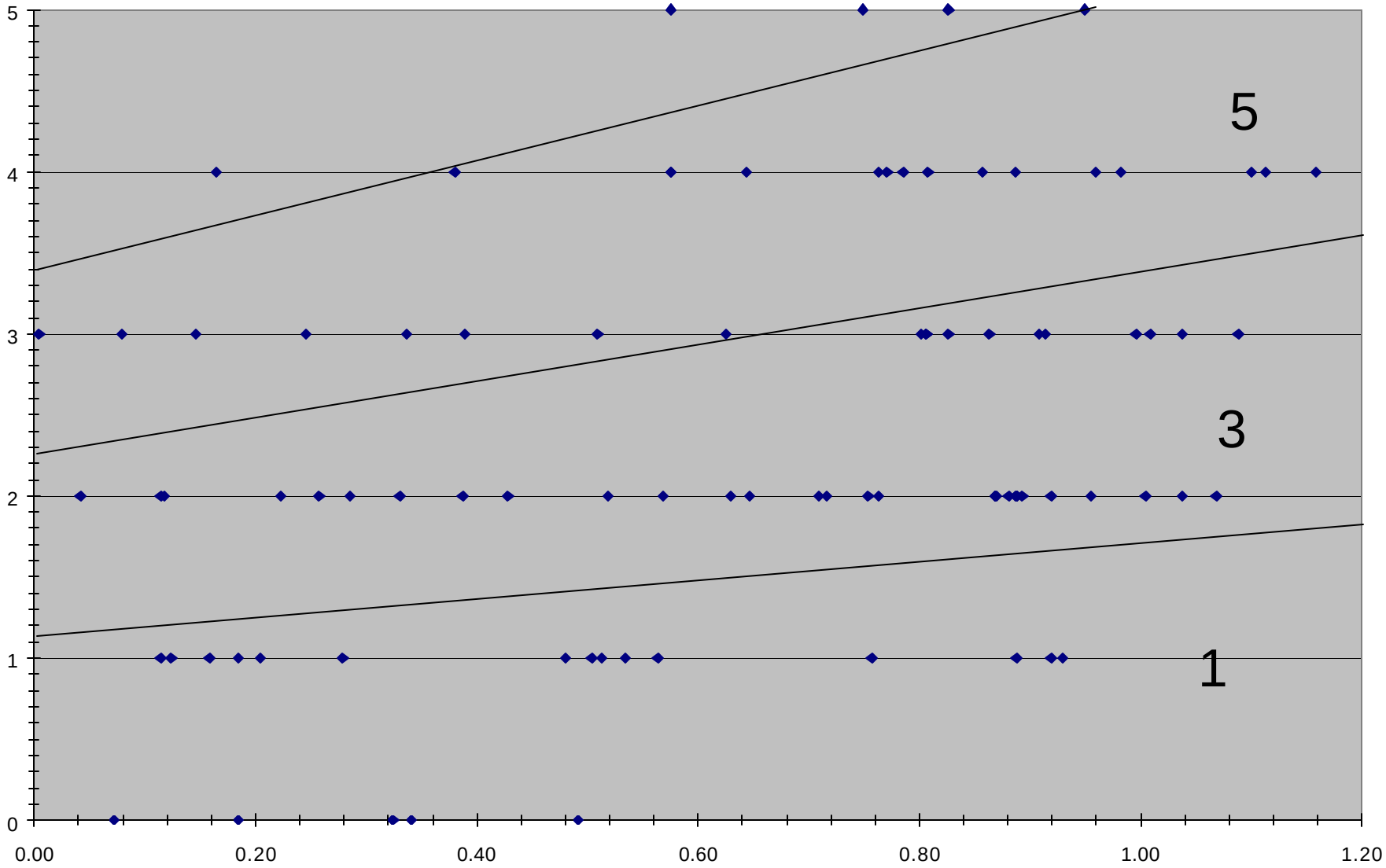
Appendix 1



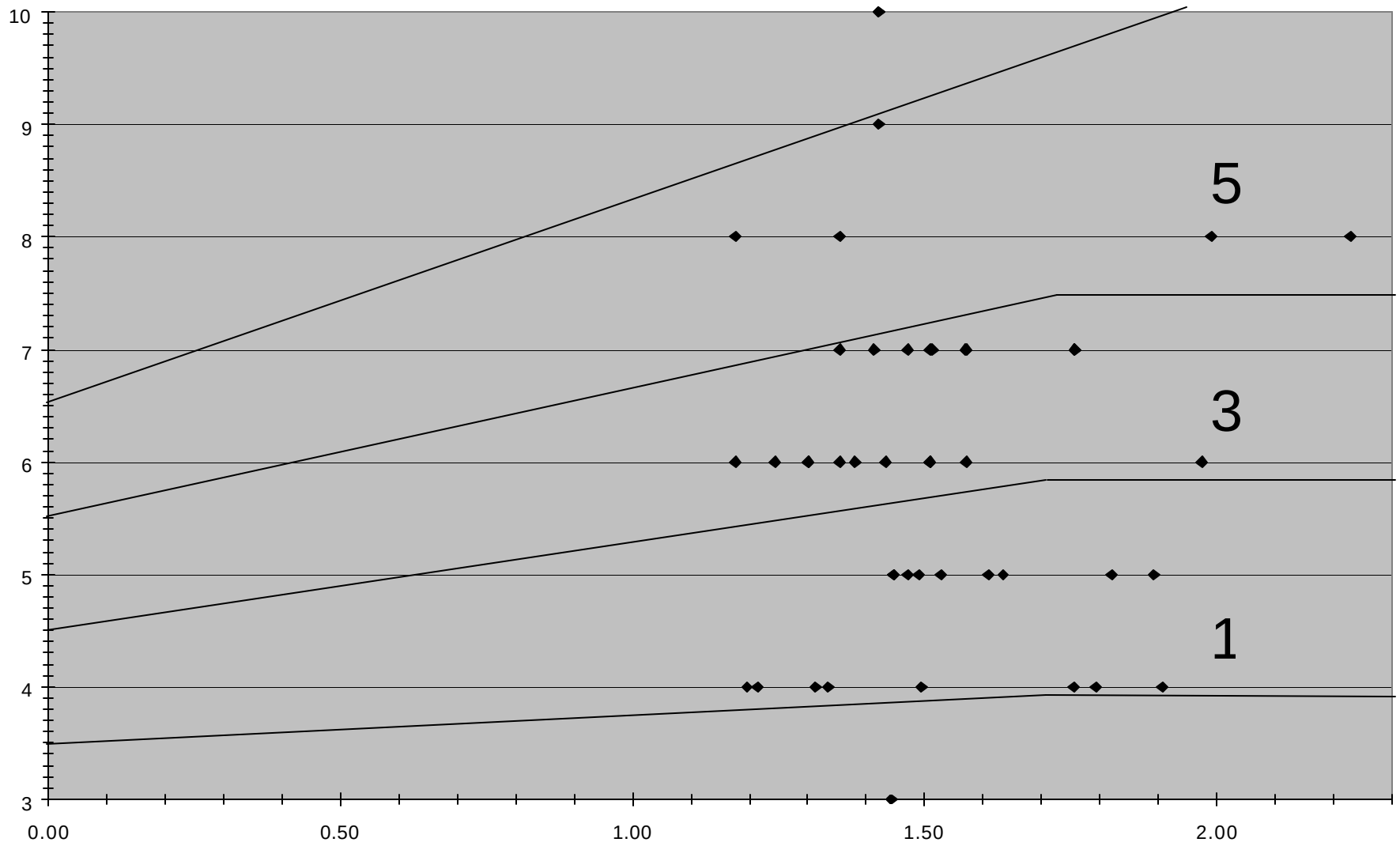
ACF 1 – PDT. Total number of native species in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 141.



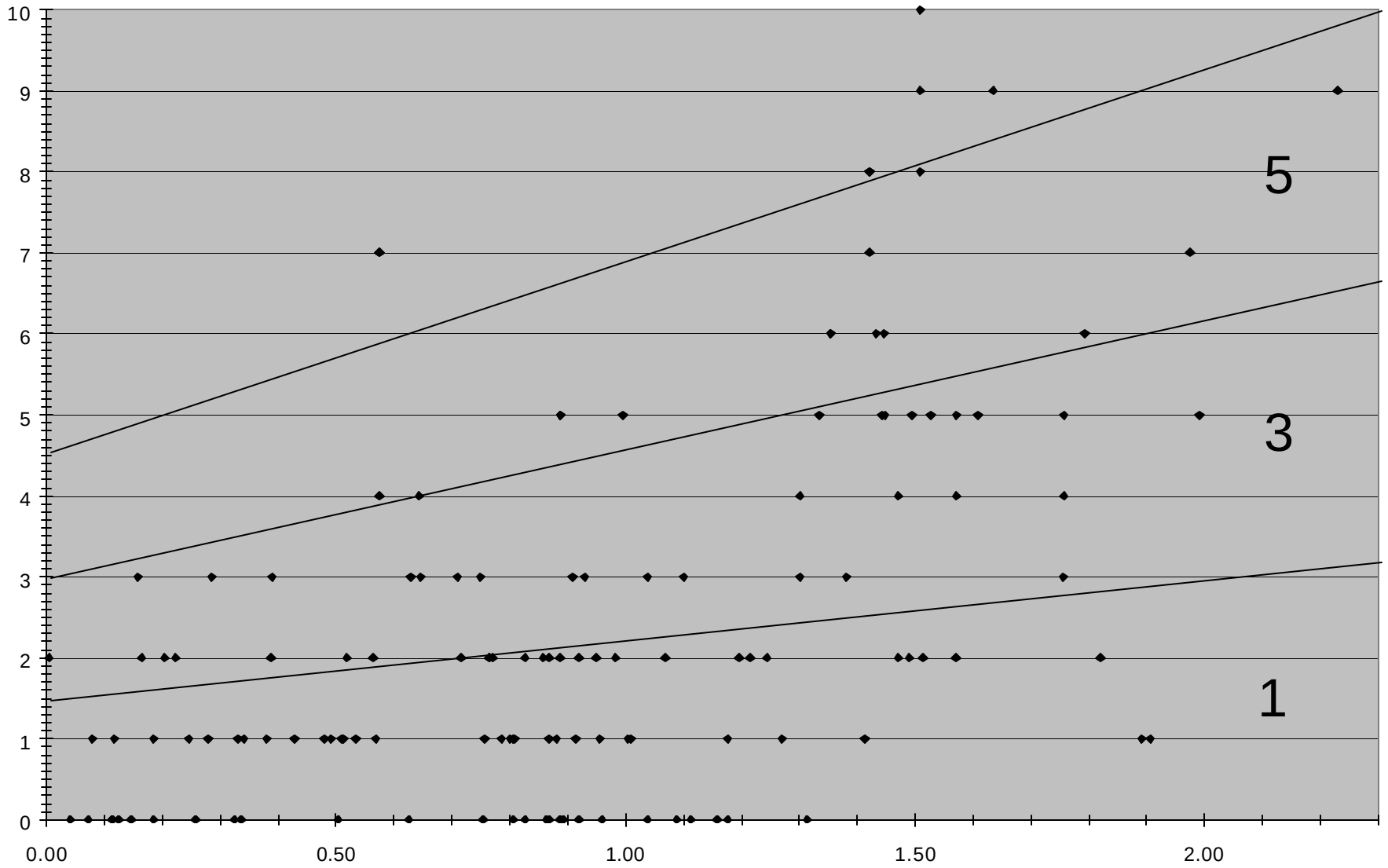
ACF2 – PDT. Number of benthic invertivore species in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 30 square miles. Total samples equal 141.



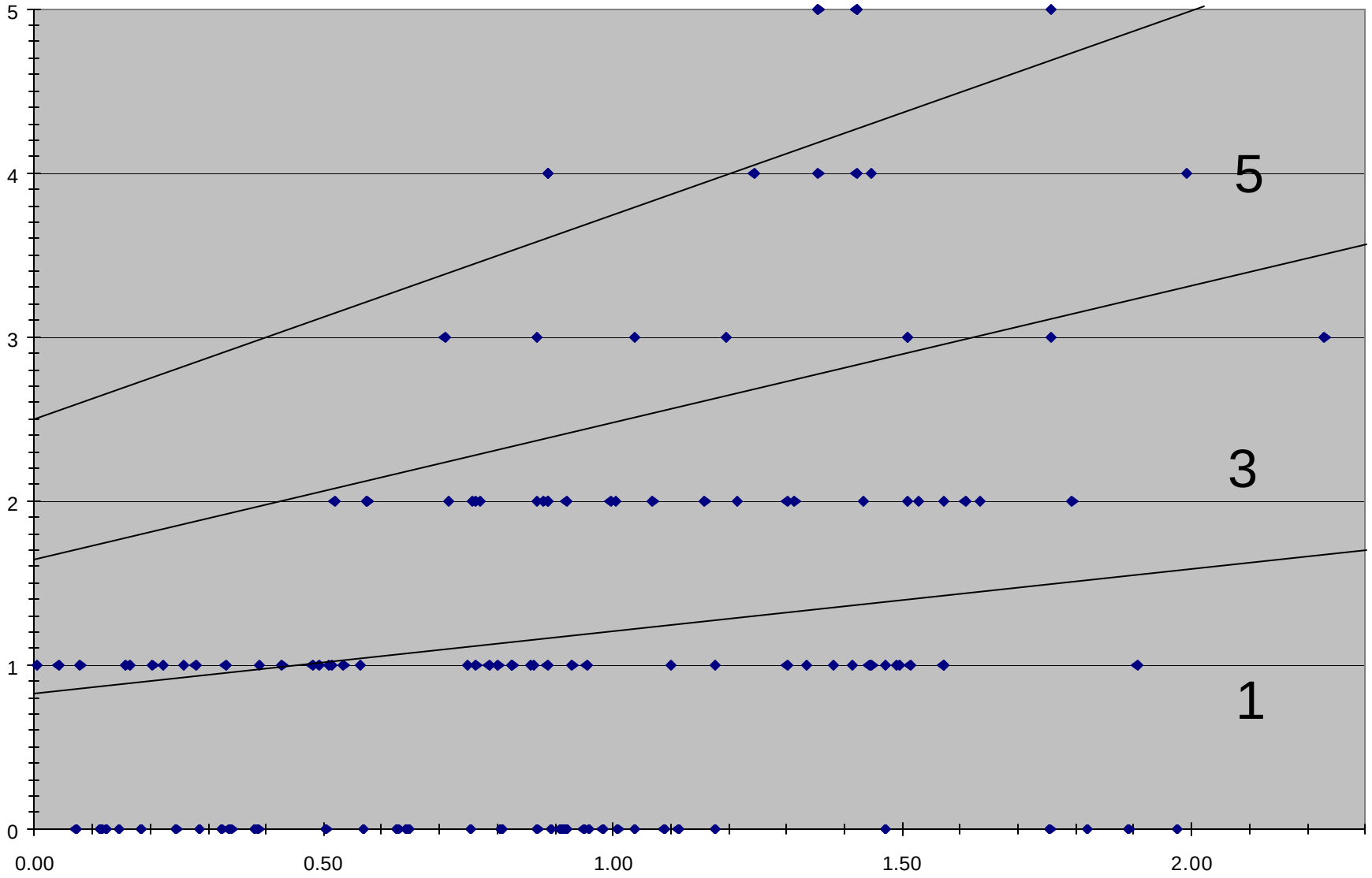
ACF3a – PDT. Number of native sunfish species in headwater streams (<15 square miles drainage basin area) in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 96.



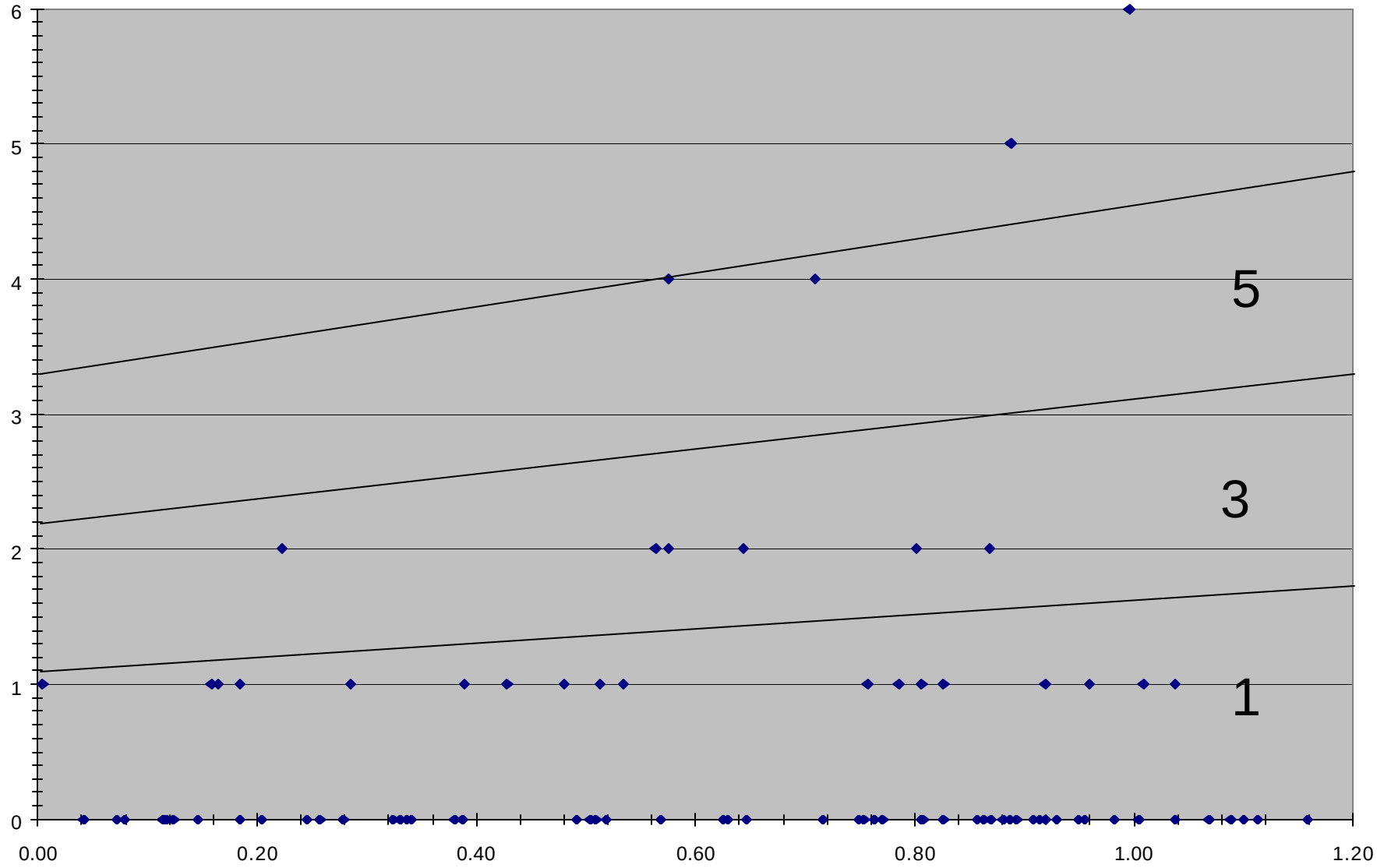
ACF3b – PDT. Number of native centrarchid species in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 50 square miles. Total samples equal 45.



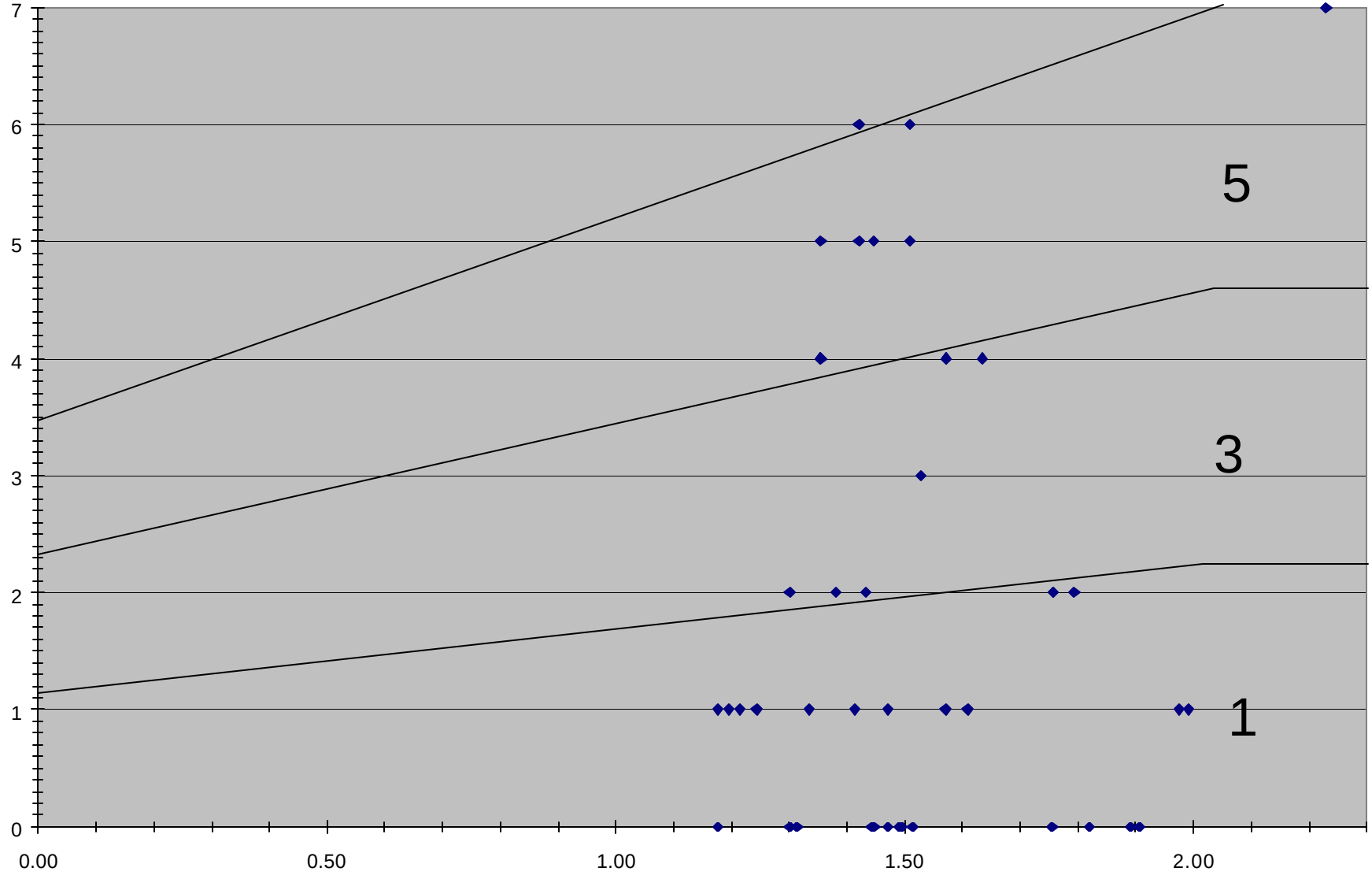
ACF4 – PDT. Number of native insectivorous cyprinid species in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 141.



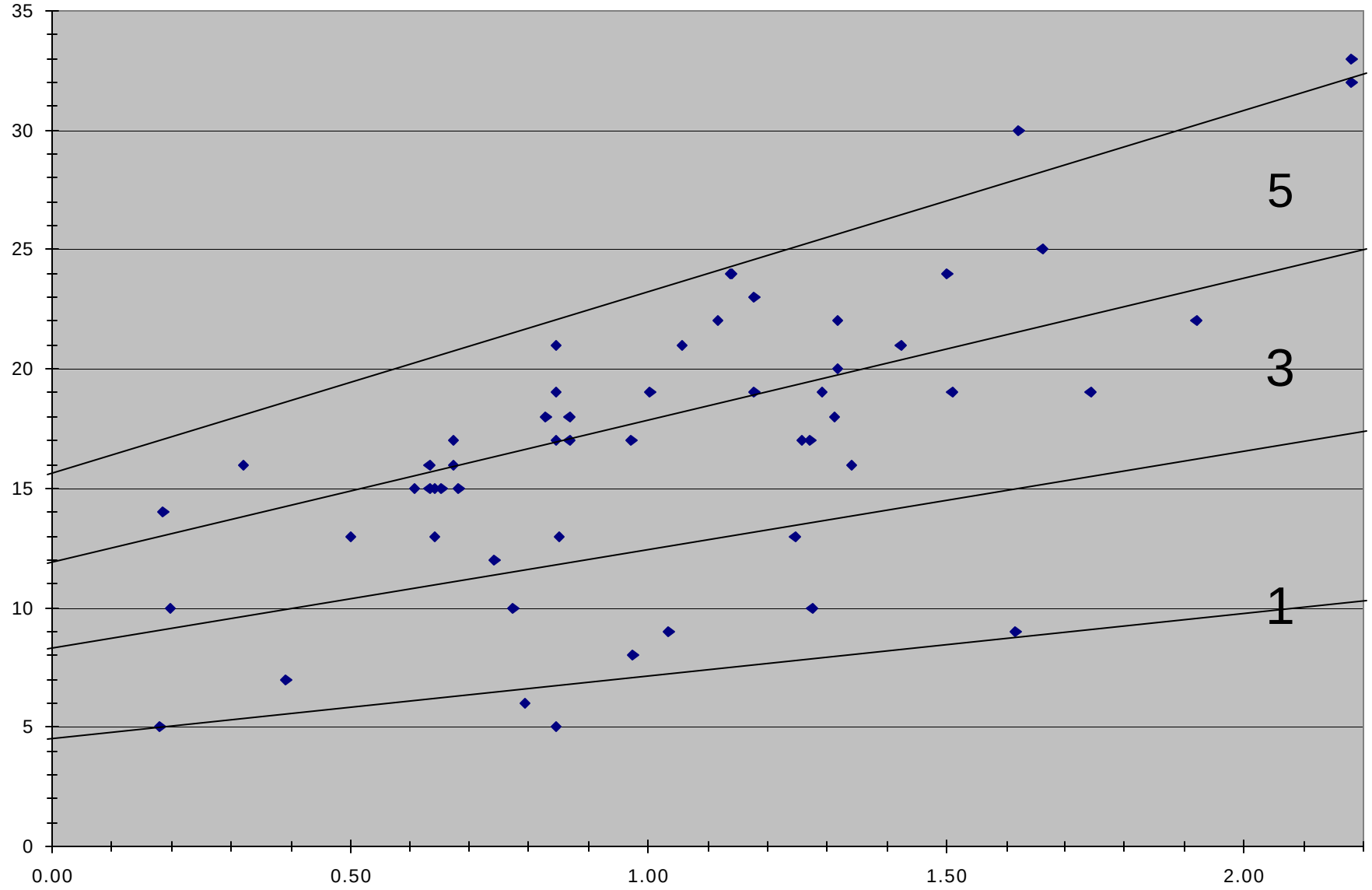
ACF5 – PDT. Number of native round-bodied sucker species in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 141.



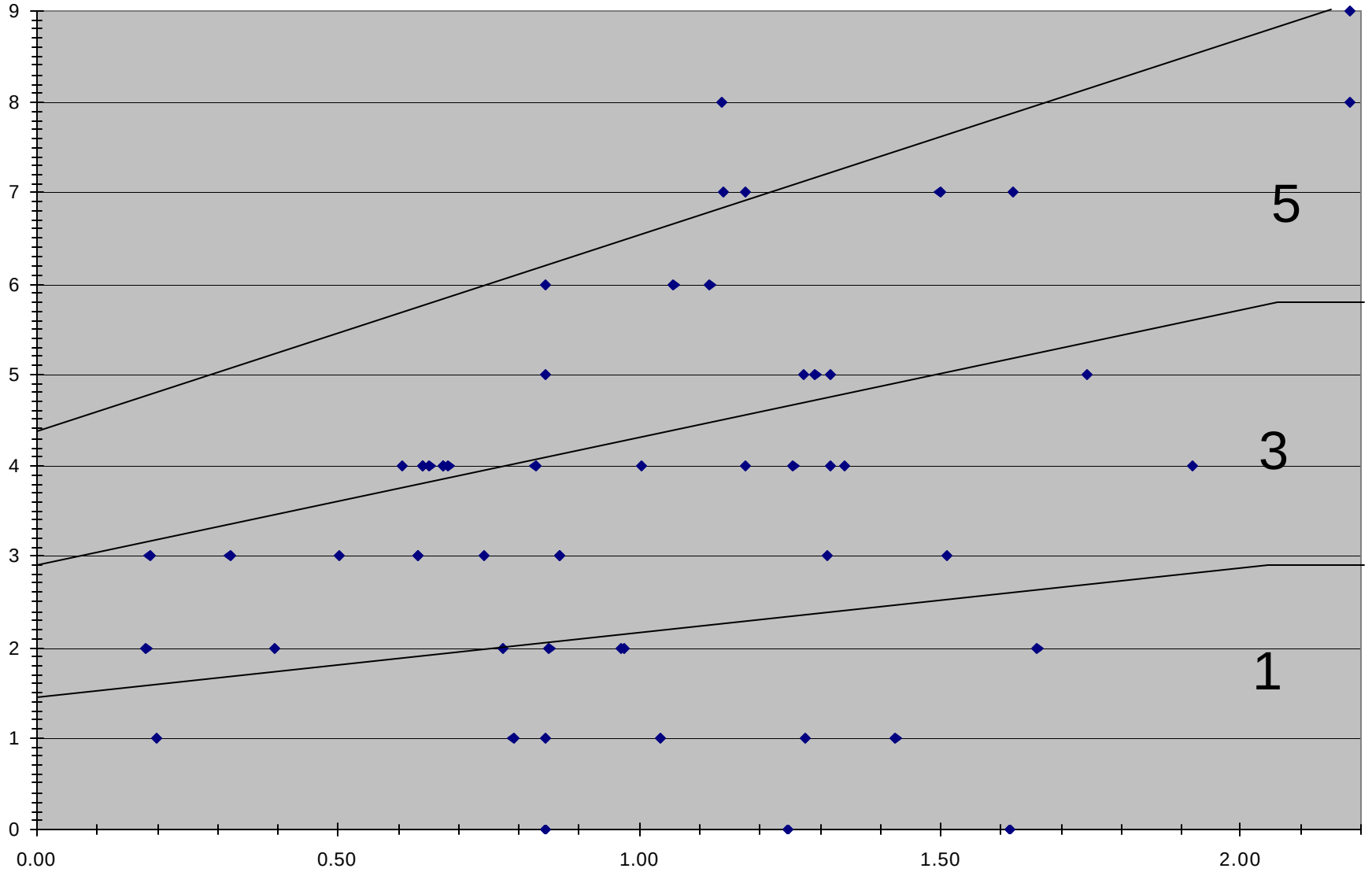
ACF6a – PDT. Total number of species ranked as sensitive at headwater sites (<15 square miles drainage basin area) in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 96.



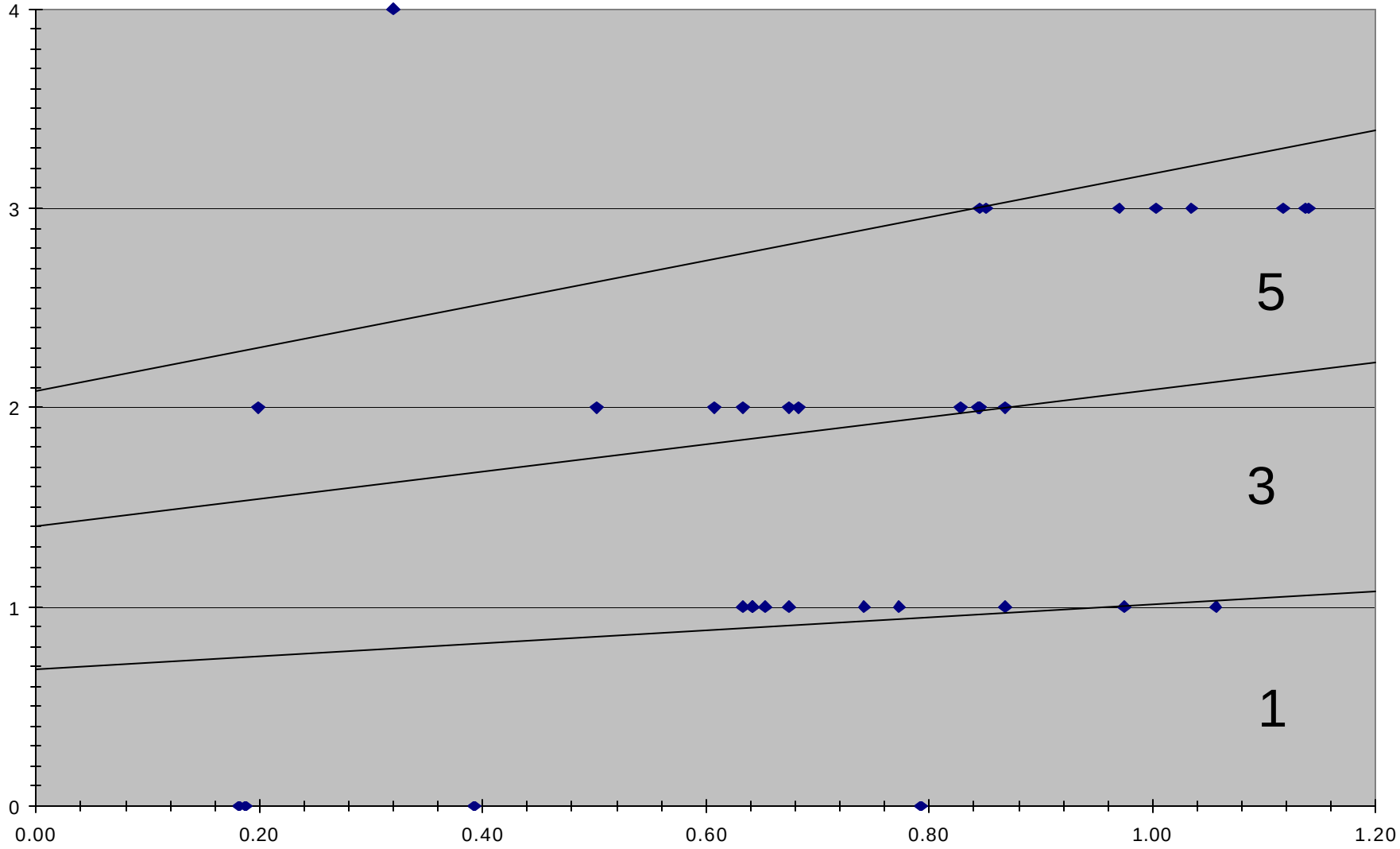
ACF6b – PDT. Number of species ranked as intolerant in the Piedmont ecoregion of the Apalachicola drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 100 square miles. Total samples equal 45.



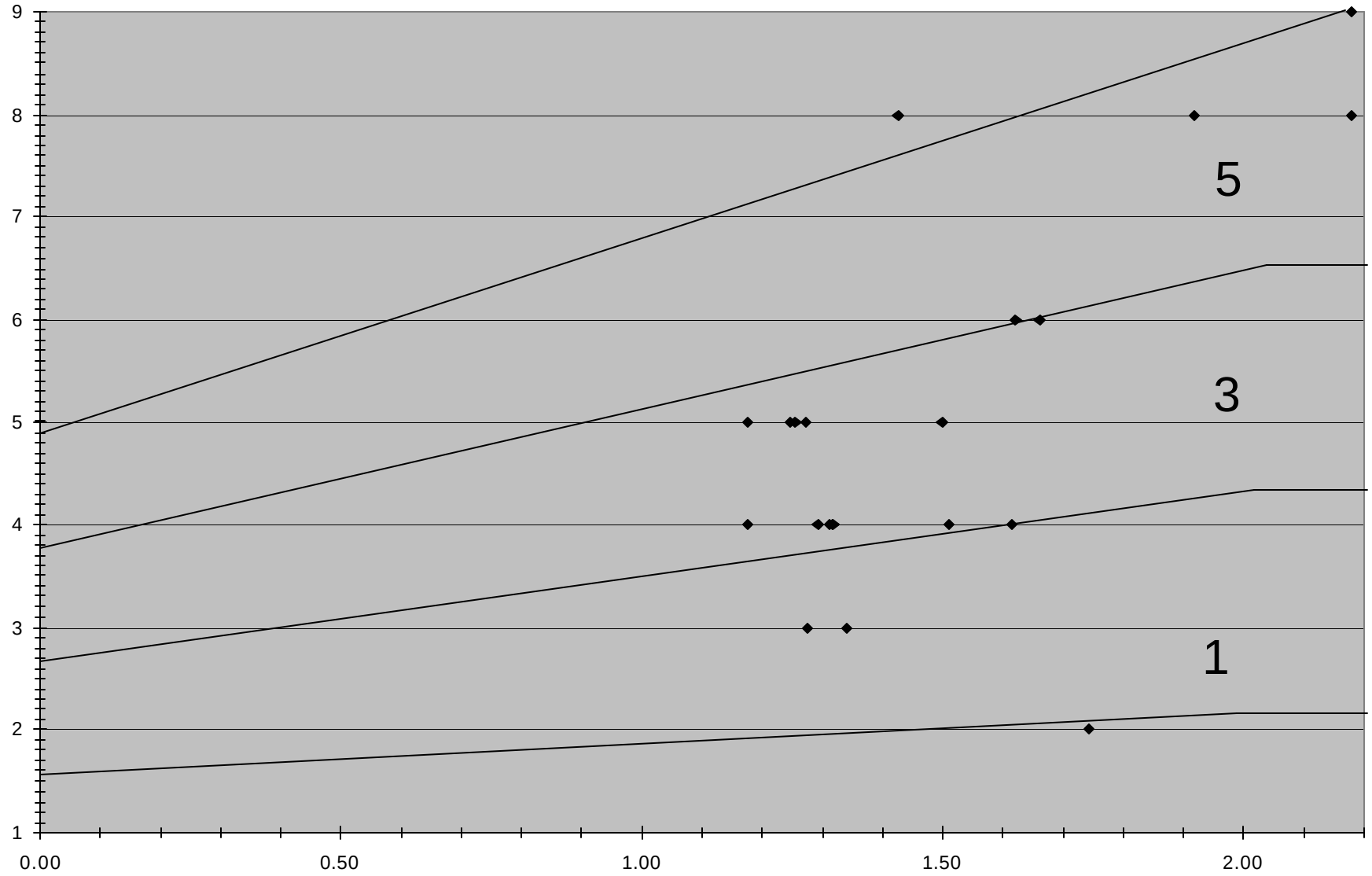
ACT1 – PDT. Total number of native species in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 55.



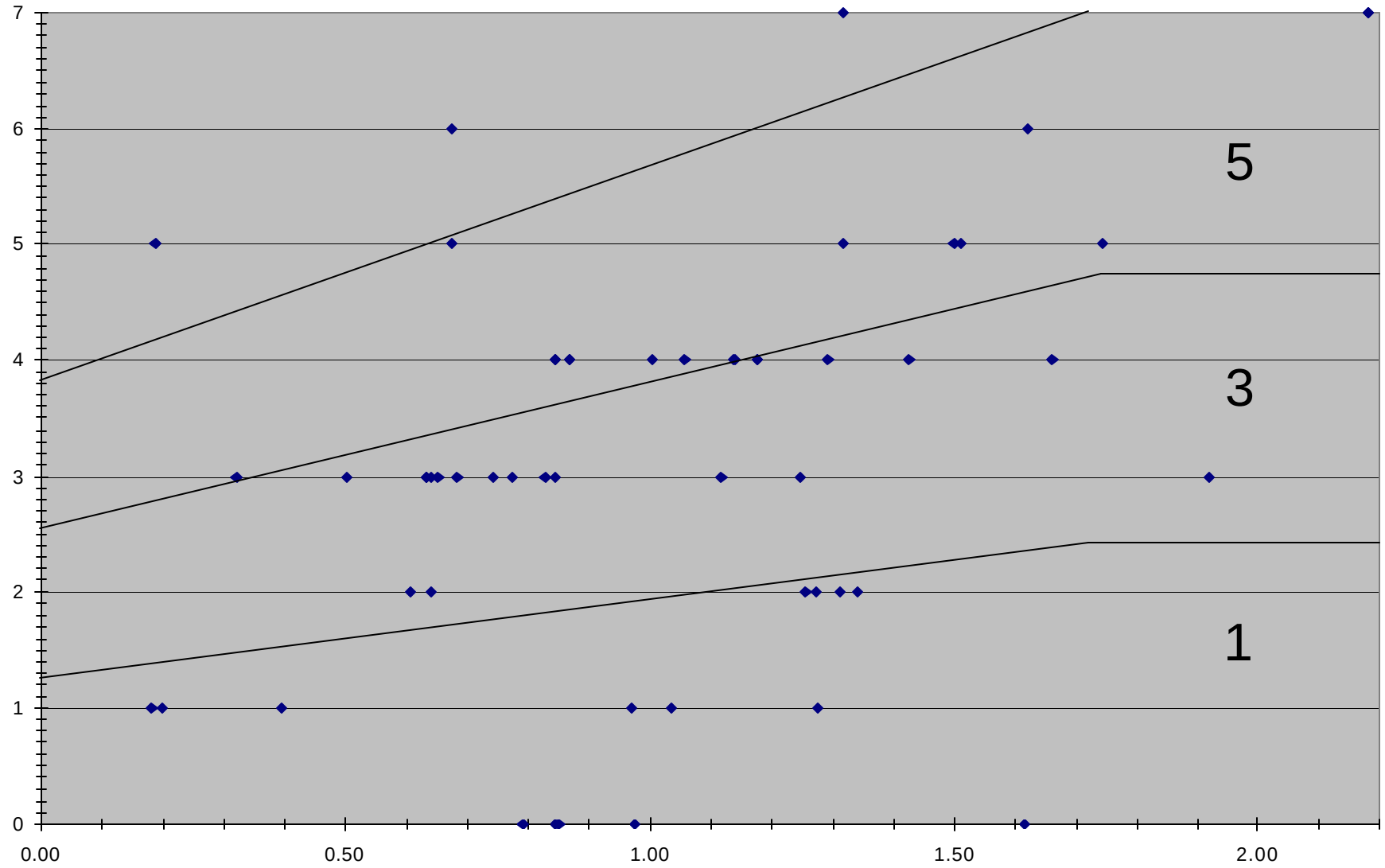
ACT2 – PDT. Number of benthic invertivore species in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 100 square miles. Total samples equal 55.



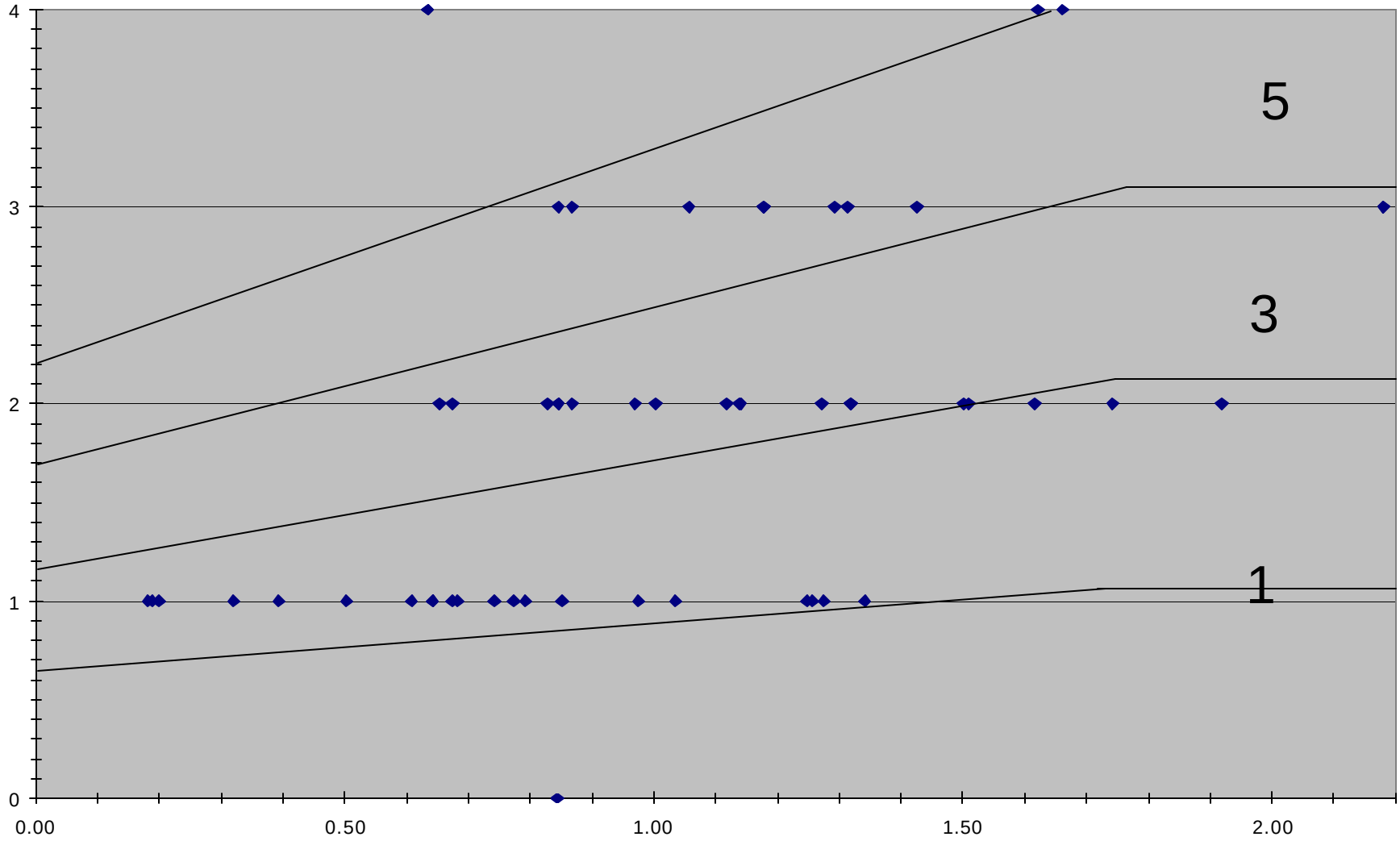
ACT3a – PDT. Number of native sunfish species in headwater streams (<15 square miles drainage basin area) in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 34.



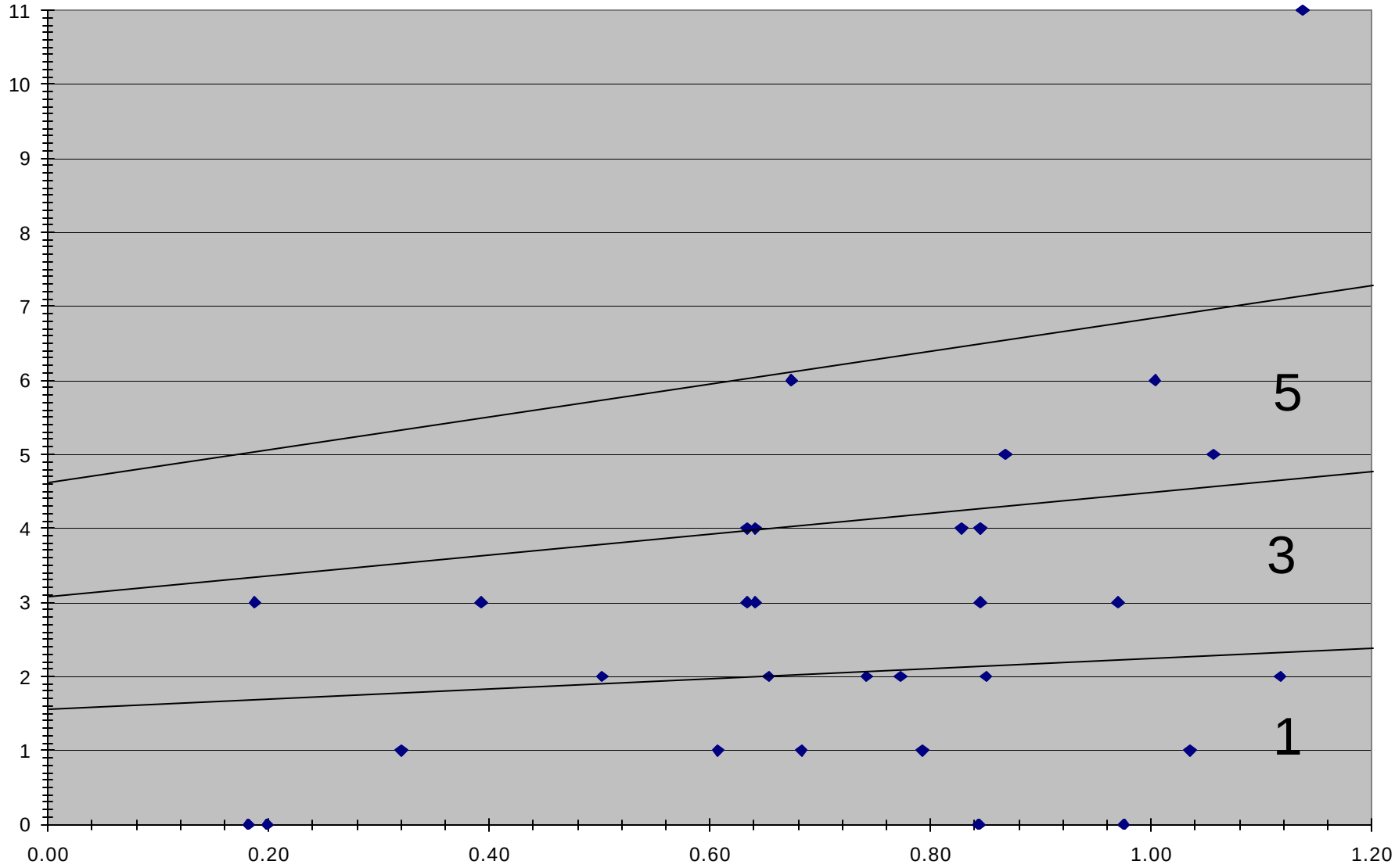
ACT3b – PDT. Number of native centrarchid species in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 100 square miles. Total sites equal 21.



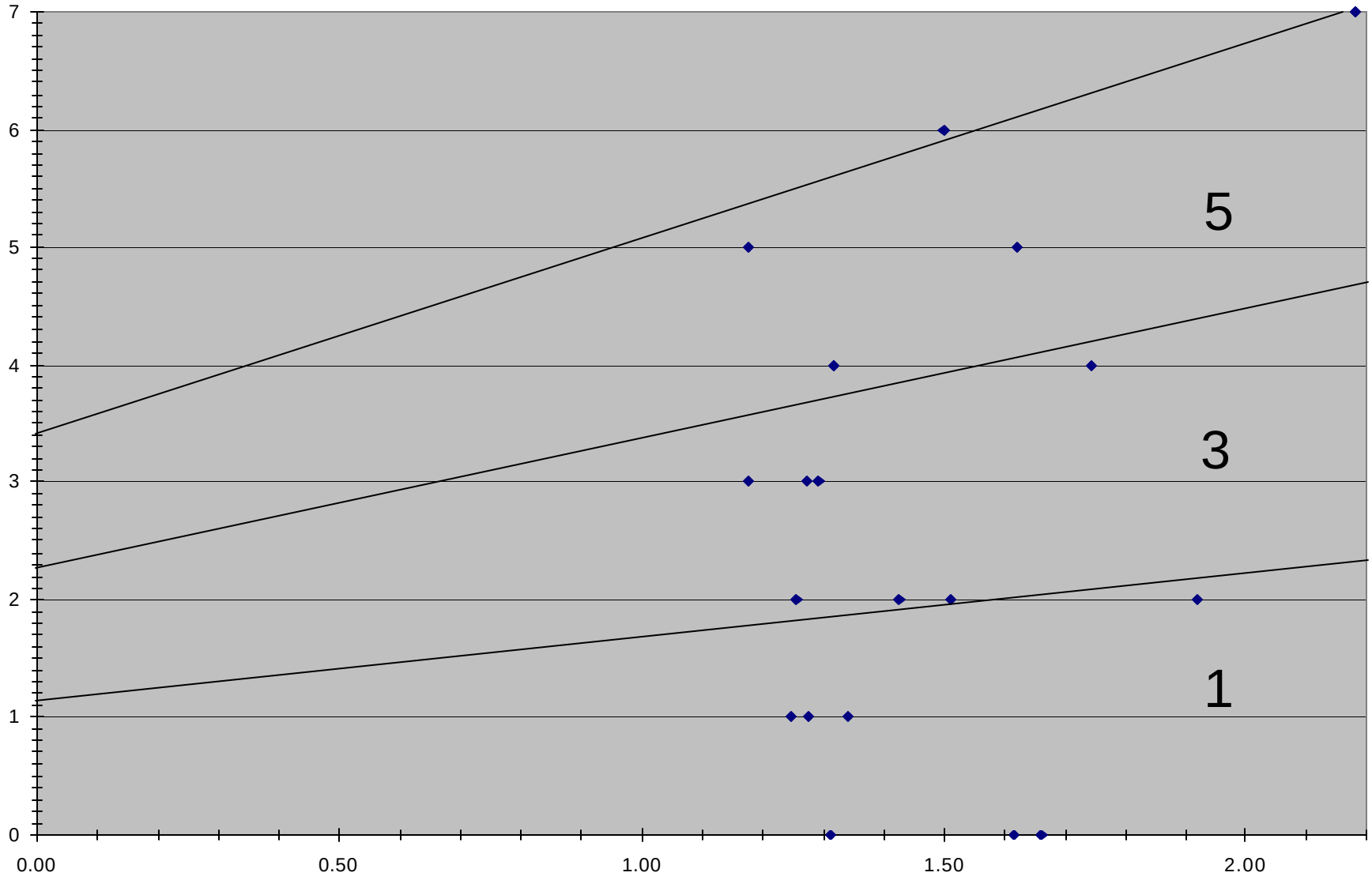
ACT4 – PDT. Number of native insectivorous cyprinid species in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 50 square miles. Total samples equal 55.



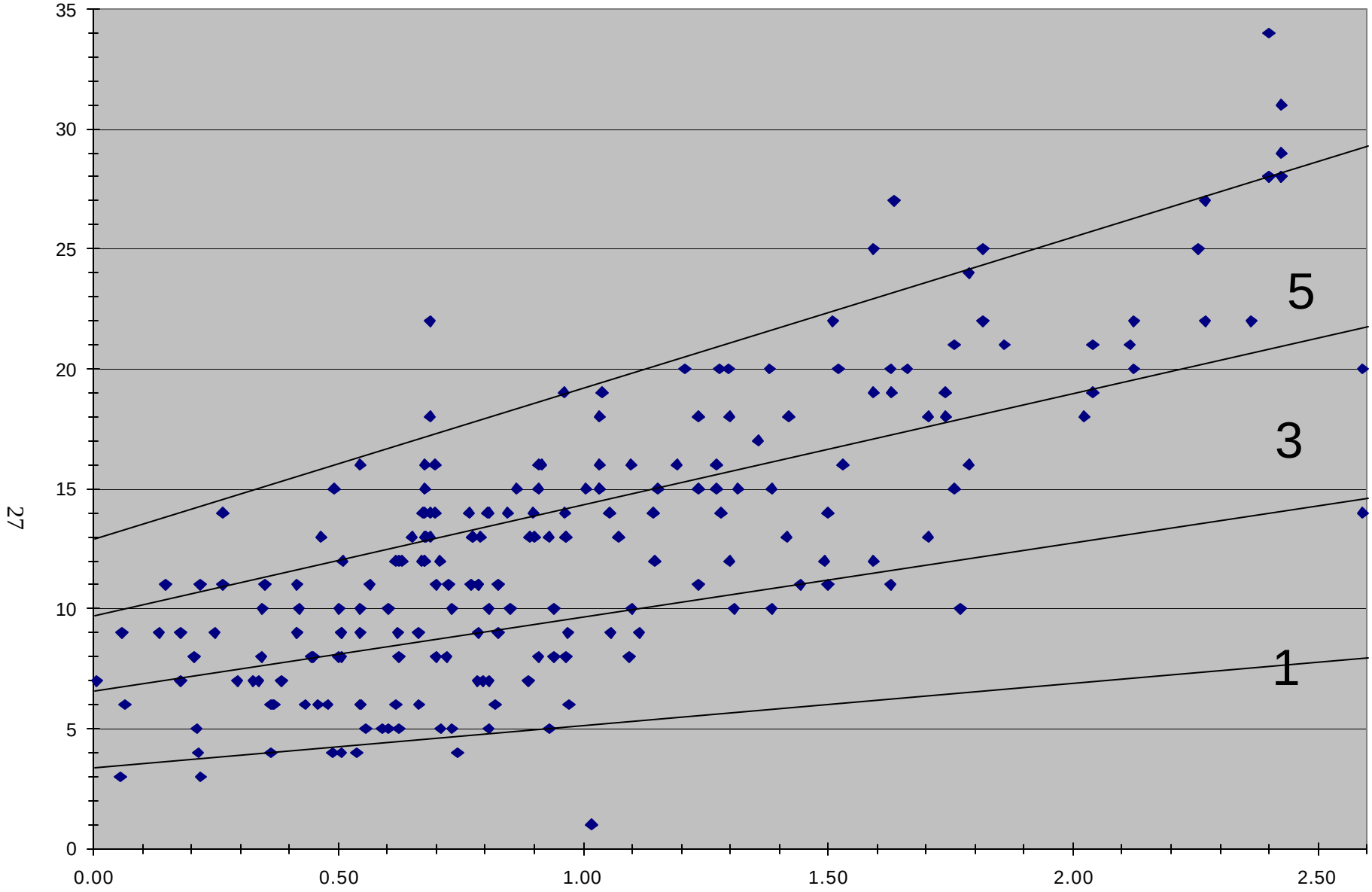
ACT5 – PDT. Number of native round-bodied sucker species in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 50 square miles. Total samples equal 55.



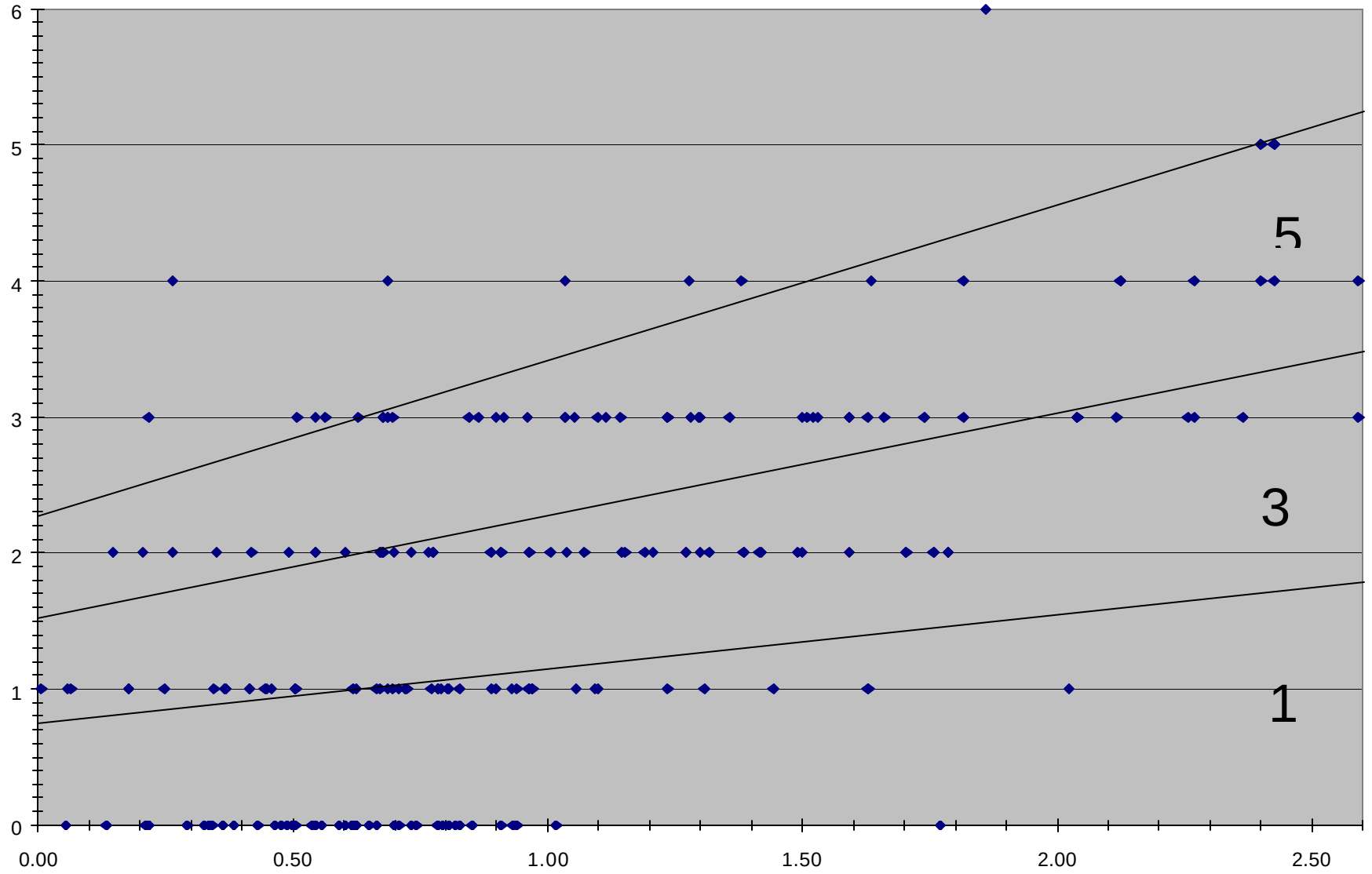
ACT6a – PDT. Total number of species ranked as sensitive at headwater sites (<15 square miles drainage basin area) in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 34.



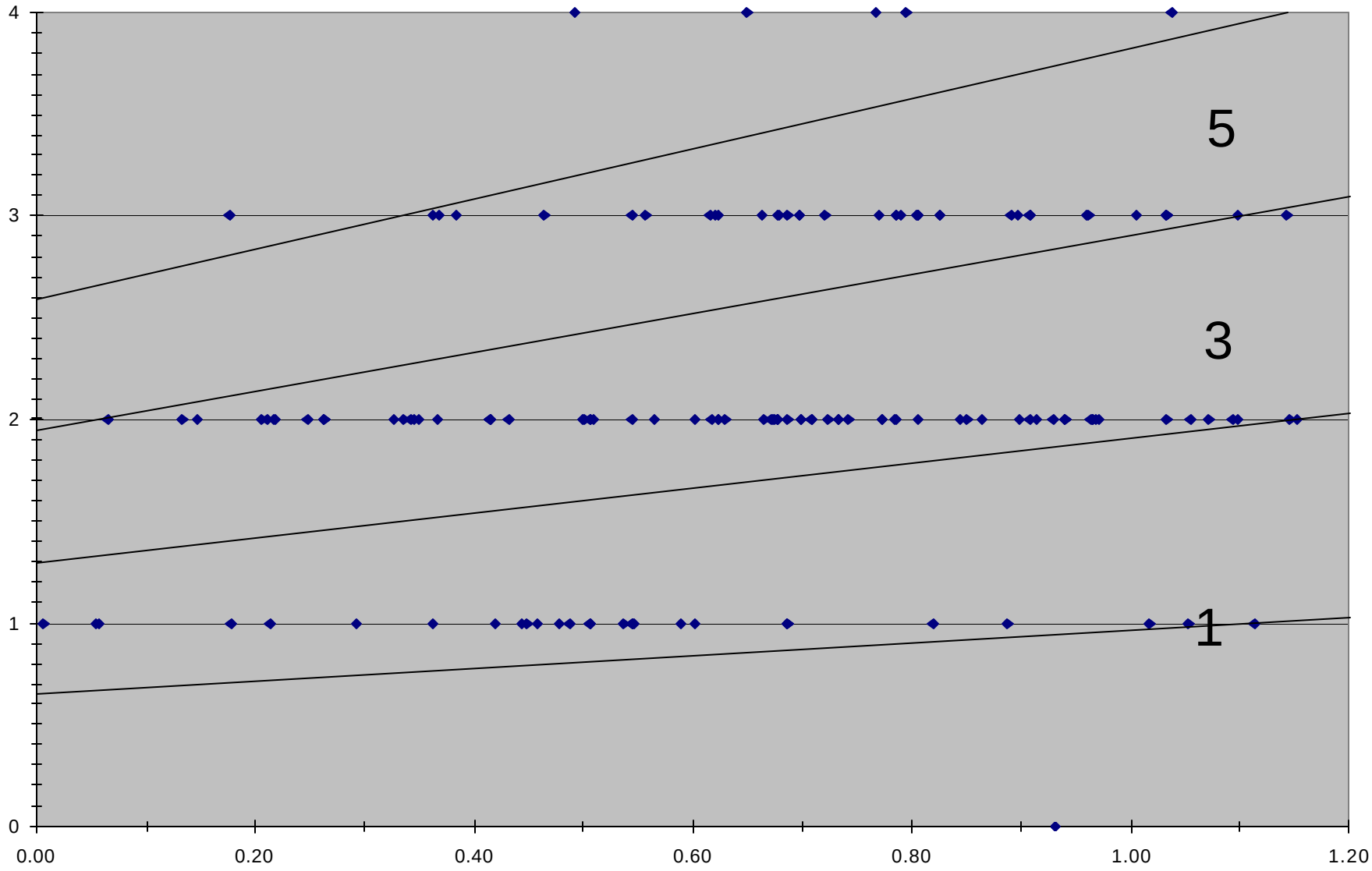
ACT6b – PDT. Number of species ranked as intolerant in the Piedmont ecoregion of the Alabama drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 21.



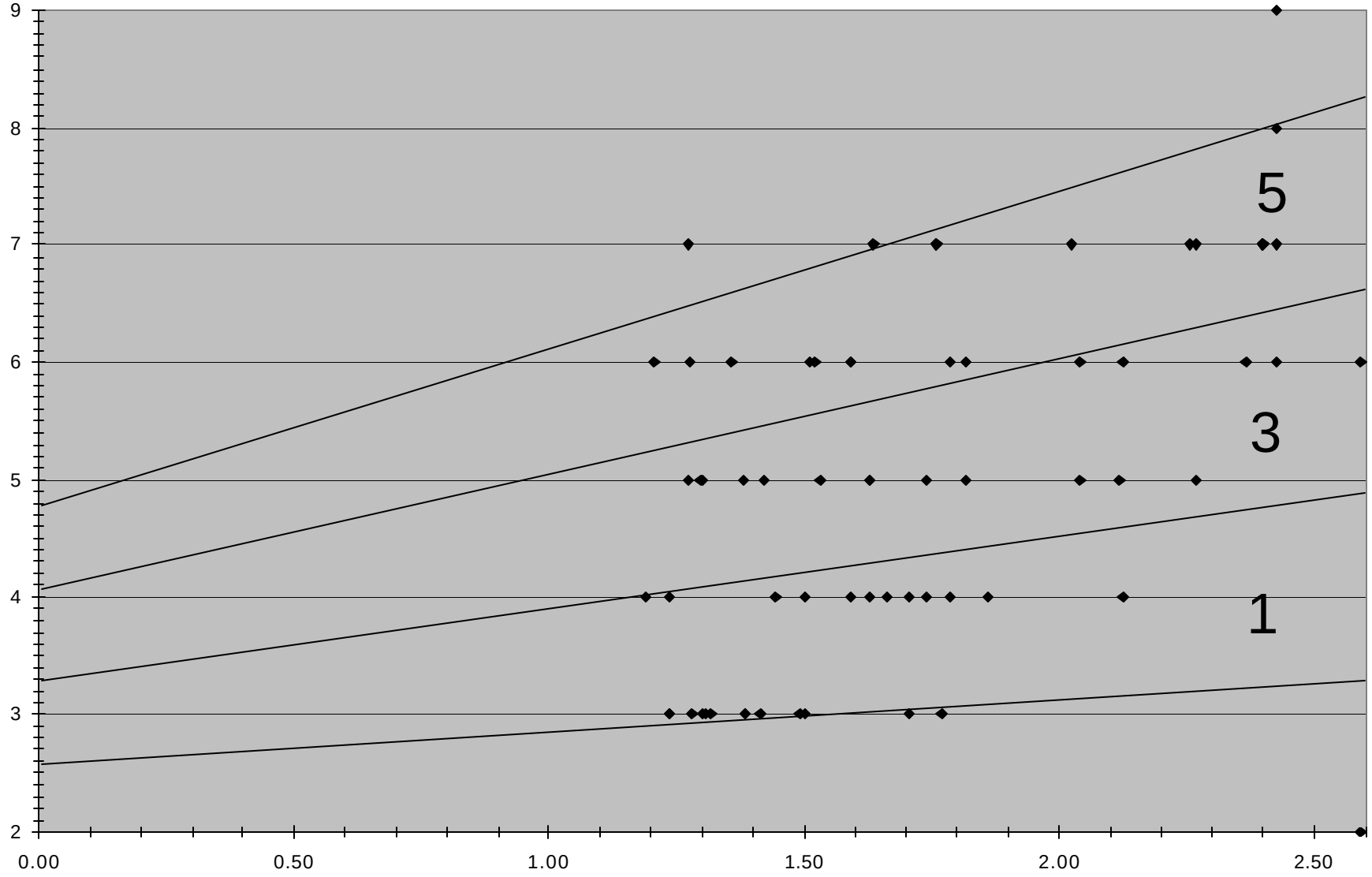
AS1 – PDT. Total number of native species in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 233.



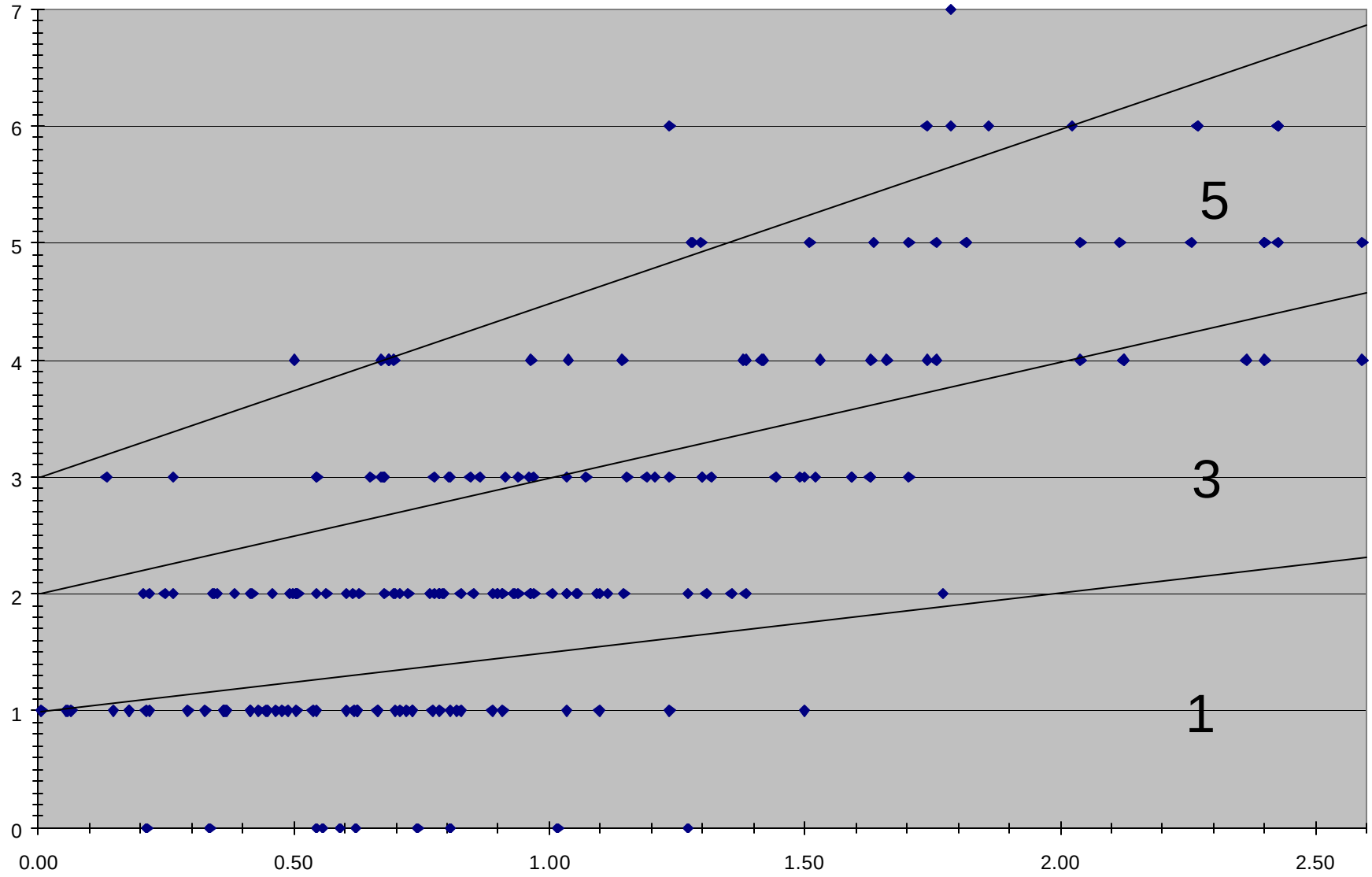
AS2 – PDT. Number of benthic invertivore species in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 233.



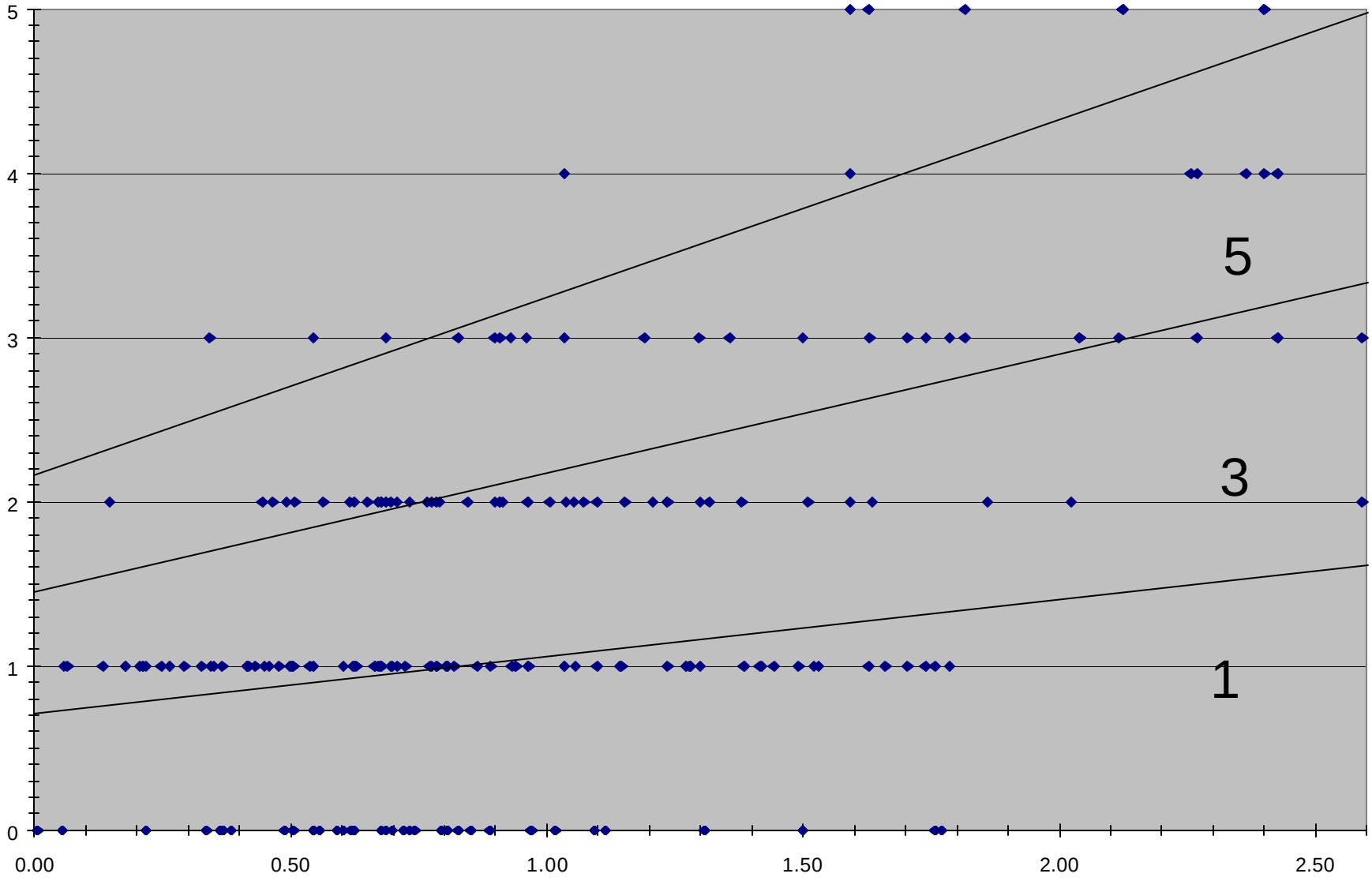
AS3a – PDT. Number of native sunfish species in headwater streams (<15 square miles drainage basin area) in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 167.



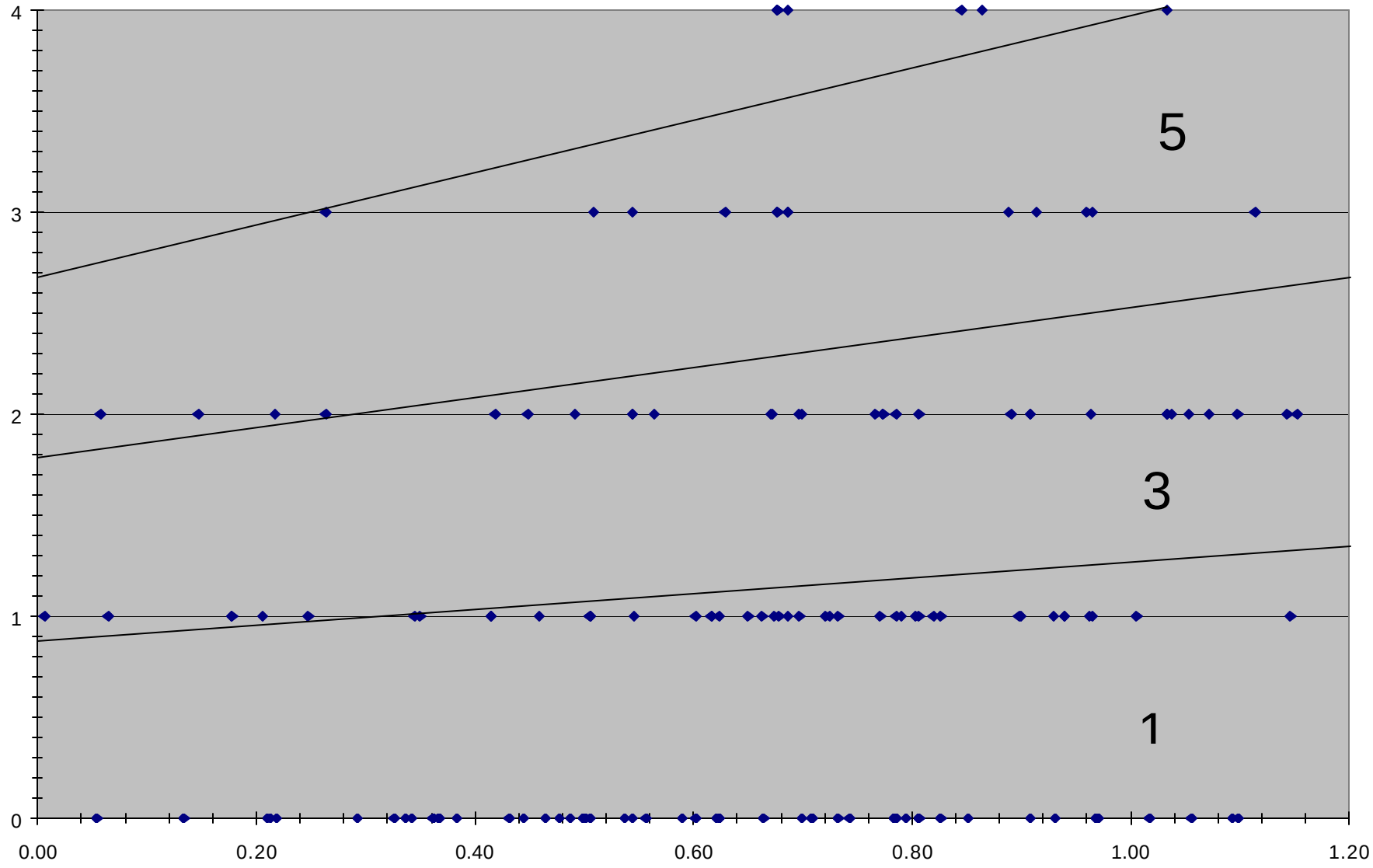
AS3b – PDT. Number of native centrarchid species in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 66.



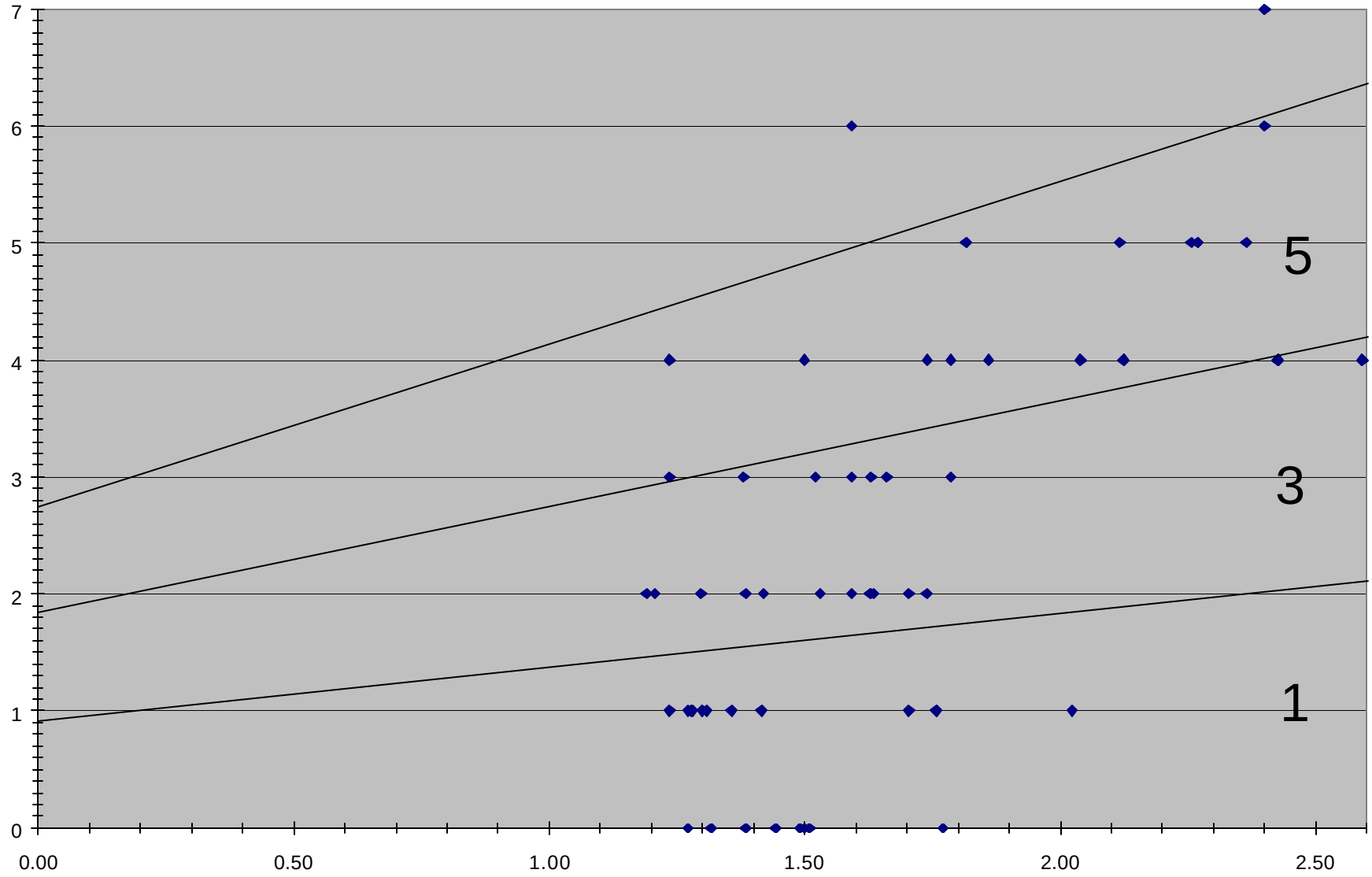
AS4 – PDT. Number of native insectivorous cyprinid species in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin (square miles). Total samples equal 233.



AS5 – PDT. Total number of native round-bodied sucker species in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 233.



AS6a – PDT. Total number of species ranked as sensitive in headwater streams (< 15 square miles drainage basin area) in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 167.



AS6b - PDT. Number of species ranked as intolerant in the Piedmont ecoregion of the Atlantic Slope drainage basins plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 66.

Fish List for the Piedmont Ecoregion of Georgia. (Updated May 11, 2005)

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Petromyzontidae				
Chestnut Lamprey *Ichthyomyzon castaneus*		PR		COO
Southern Brook Lamprey *Ichthyomyzon gagei*		HB		CHA, COO, FLI, TAL
Least Brook Lamprey *Lampetra aepyptera*		HB		COO
Lepisosteidae				
Longnose Gar *Lepisosteus osseus*		CR		CHA, COO, FLI, OCM, OCO, OGE, SAV
Amiidae				
Bowfin *Amia calva*		CR		CHA, FLI, OCM, OCO, OGE, SAV, TAL
Anguillidae				
American Eel *Anguilla rostrata*		CR		OCM, OCO, OGE, SAV
Clupeidae				
Blueback Herring *Alosa aestivalis*		IN		CHA**, OCM, OCO
American shad *Alosa sapidissima*		IN		OCM, OCO, OGE, SAV
Gizzard Shad *Dorosoma cepedianum*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Threadfin Shad *Dorosoma petenense*		HB		CHA, COO, FLI, OCM**, OCO**, SAV**

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Cyprinidae				
Largescale Stoneroller *Campostoma oligolepis*		HB		COO, TAL
Bluefin Stoneroller *Campostoma pauciradii*		HB		CHA, FLI, OCM, OCO, TAL
Goldfish *Carassius auratus*		GE		EXOTIC
Rosyside Dace *Clinostomus funduloides*		IC		SAV
Grass Carp *Ctenopharyngodon idella*		HB		EXOTIC
Ocmulgee Shiner *Cyprinella callisema*	INT	IC	SMM	OCM, OCO, OGE
Alabama Shiner *Cyprinella callistia*	INT	IC	SMM	COO, TAL
Bluestripe Shiner *Cyprinella callitaenia*	INT	IC	SMM	CHA, FLI
Tallapoosa Shiner *Cyprinella gibbsi*		IC		TAL
Red Shiner *Cyprinella lutrensis*		GE		EXOTIC
Whitefin Shiner *Cyprinella nivea*		IC	SMM	SAV

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Tricolor Shiner *Cyprinella trichroistia*	INT	IC		COO, TAL
Blacktail shiner *Cyprinella venusta*		IC		CHA, COO, FLI, OCM, TAL
Altamaha Shiner *Cyprinella xaenura*	INT	IC		OCM, OCO
Common Carp *Cyprinus carpio*		GE		EXOTIC
Silverjaw Minnow *Ericymba buccata*		IN	IC	CHA, COO, FLI, OCM, OCO
Eastern Silvery Minnow *Hybognathus regius*	INT	HB		OCM, OCO, OGE, SAV
Lined Chub *Hybopsis lineapunctata*		IC	SMM	COO, TAL
Rosyface Chub *Hybopsis rubrifrons*		IC	SMM	OCM, OCO, OGE, SAV
Coastal Chub *Hybopsis sp.*	INT	IC	SMM	CHA, COO, FLI
Striped Shiner *Luxilus chrysocephalus*		IC		COO, TAL
Bandfin Shiner *Luxilus zonistius*		IC		CHA, COO**, FLI, OCO, TAL**
Blacktip Shiner *Lythrurus atrapiculus*	INT	IC		CHA, FLI

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Pretty Shiner *Lythrurus bellus*	INT	IC		TAL
Speckled Chub *Macrhybopsis aestivalis*		IC	SMM	COO, TAL
Bluehead Chub *Nocomis leptocephalus*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
River Chub *Nocomis micropogon*		IC	SMM	COO**, SAV**
Golden Shiner *Notemigonus crysoleucas*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Rough Shiner *Notropis baileyi*		IC		CHA**
Rainbow Shiner *Notropis chrosomus*	HWI	IC		COO
Dusky Shiner *Notropis cummingsae*		IC		FLI, OCM, OCO, OGE, SAV
Spottail Shiner *Notropis hudsonius*		IC	SMM	CHA, FLI, OCM, OCO, OGE, SAV
Highscale Shiner *Notropis hypsilepis*		IC	SMM	CHA, FLI
Longnose Shiner *Notropis longirostris*		IC	SMM	CHA, COO, FLI, OCM
Yellowfin Shiner *Notropis lutipinnis*		IC		CHA, COO, FLI, OCM, OCO, OGE, SAV

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Coastal Shiner *Notropis petersoni*		IC		OCM, OCO, OGE, SAV
Sandbar Shiner *Notropis scepticus*	INT	IC		SAV
Silverstripe Shiner *Notropis stilbius*	INT	IC		COO, TAL
Weed shiner *Notropis texanus*		IC		CHA, FLI, OCM, TAL
Coosa Shiner *Notropis xaenocephalus*		IC		COO
Pugnose Minnow *Opsopoeodus emiliae*	INT	IC		FLI
Riffle Minnow *Phenacobius catostomus*		IC	SMM	COO, TAL
Fathead Minnow *Pimephales promelas*		GE		EXOTIC
Bullhead Minnow *Pimephales vigilax*		GE		COO, TAL
Sailfin Shiner *Pteronotropis hypselopterus*		IC		OCO
Creek Chub *Semotilus atromaculatus*		GE		CHA, COO, OCM, OCO, OGE, SAV, TAL

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Dixie Chub *Semotilus thoreauianus*		GE		CHA, FLI, TAL
Catostomidae				
White Sucker *Catostomus commersoni*		IN	RBS	CHA**
Creek Chubsucker *Erimyzon oblongus*		IN	RBS	CHA, FLI, OCM, OCO, OGE, SAV
Alabama Hogsucker *Hypentelium etowanum*		IN	RBS	CHA, COO, TAL
Northern Hogsucker *Hypentelium nigricans*		IN	RBS	OCO, SAV
Spotted Sucker *Minytrema melanops*		IN	RBS	CHA, COO, FLI, OCM, OCO, OGE, SAV
V-lip Redhorse *Moxostoma collapsum*	INT	IN	RBS	OCM, OCO, OGE, SAV
Black Redhorse *Moxostoma duquesnei*		IN	RBS	COO, TAL
Golden Redhorse *Moxostoma erythrurum*		IN	RBS	COO, TAL
Blacktail Redhorse *Moxostoma poecilurum*		IN	RBS	COO, TAL
Robust Redhorse *Moxostoma robustum*		IN	RBS	OCO, OGE, SAV

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Apalachicola Redhorse *Moxostoma* sp.	INT	IN	RBS	CHA, FLI
Greater Jumprock *Scartomyzon lachneri*	INT	IN	RBS	CHA, FLI
Striped Jumprock *Scartomyzon rupiscartes*		IN	RBS	CHA, FLI, OCM, OCO, OGE, SAV
Brassy Jumprock *Scartomyzon* sp.	INT	IN	RBS	OCM, OCO, OGE, SAV
Ictaluridae				
Snail Bullhhead *Ameiurus brunneus*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV
White Catfish *Ameiurus catus*		GE		CHA, FLI, OCM, OCO, OGE, SAV
Black Bullhead *Ameiurus melas*		GE		CHA**, COO, TAL
Yellow Bullhead *Ameiurus natalis*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Brown Bullhead *Ameiurus nebulosus*		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Flat Bullhead *Ameiurus platycephalus*		GE		OCM, OCO, OGE, SAV, TAL**
Blue Catfish *Ictalurus furcatus*		CR		OCO**, SAV**

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Channel Catfish *Ictalurus punctatus*		GE		CHA, COO, FLI, OCM**, OCO**, OGE**, SAV**, TAL
Black Madtom *Noturus funebris*	INT	IN	BI	CHA, TAL
Tadpole Madtom *Noturus gyrinus*	HWI	IN	BI	OCM, OCO, OGE, SAV
Margined Madtom *Noturus insignis*	INT	IN	BI	OCM, OCO, OGE, SAV
Speckled Madtom *Noturus leptacanthus*	HWI	IN	BI	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Frecklebelly Madton *Noturus munitus*		IN	BI	COO
Flathead Catfish *Pylodictis olivaris*		CR		COO, FLI**, OCM**, OCM**, OGE**, SAV**
Esocidae				
Redfin Pickerel *Esox americanus*		CR		CHA, FLI, OCM, OCO, OGE, SAV, TAL
Chain Pickerel *Esox niger*		CR		CHA, COO, FLI, OCM, OCO, OGE, SAV
Salmonidae				
Rainbow Trout *Oncorhynchus mykiss*		CR		EXOTIC
Brown Trout *Salmo trutta*		CR		EXOTIC

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Aphredoderidae				
Pirate Perch *Aphredoderus sayanus*		IN		FLI, OCM, OCO, OGE, SAV
Fundulidae				
Stippled Studfish *Fundulus bifax*		IN		TAL
Blackspotted Topminnow *Fundulus olivaceus*		IN		CHA, COO, TAL
Southern Studfish *Fundulus stellifer*	HWI	IN		CHA, COO
Poeciliidae				
Mosquitofish *Gambusia* sp.		GE		CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Atherinidae				
Brook Silversides *Labidesthes sicculus*		IN		CHA, FLI, OCM, OCO, OGE, SAV
Cottidae				
Mottled Sculpin *Cottus bairdi*		IN	BI	COO
Banded Sculpin *Cottus carolinae*		IN	BI	CHA, COO, TAL
Percichthyidae				
White Bass *Morone chrysops*		CR		CHA**, COO**, FLI**, OCM**, OCO**, SAV**
Striped Bass *Morone saxatalis*		CR		CHA, COO, FLI, OCM, OCO, OGE, SAV

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Centrarchidae				
Shadow Bass *Ambloplites ariommus*	INT	CR	SF	CHA, COO, FLI, TAL
Flier *Centrarchus macropterus*		IN	SF	CHA, FLI, OCM, OCO, OGE, SAV
Redbreast Sunfish *Lepomis auritus*		IN	SF	CHA, COO**, FLI, OCM, OCO, OGE, SAV, TAL**
Green Sunfish *Lepomis cyanellus*		IN	SF	CHA**, COO, FLI**, OCM**, OCO**, OGE**, SAV**, TAL
Pumpkinseed *Lepomis gibbosus*		IN	SF	SAV
Warmouth *Lepomis gulosus*		CR	SF	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Bluegill *Lepomis macrochirus*		IN	SF	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Dollar Sunfish *Lepomis marginatus*		IN	SF	CHA, OGE, SAV
Longear Sunfish *Lepomis megalotis*		IN	SF	CHA**, COO, OCM**, OCO**, SAV**, TAL
Redear Sunfish *Lepomis microlophus*		IN	SF	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Spotted Sunfish *Lepomis punctatus*		IN	SF	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Shoal Bass *Micropterus cataractae*	INT	CR	CENT	CHA, FLI, OCM**

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Redeye Bass *Micropterus coosae*	HWI	CR	CENT	CHA, COO, OCM, OCO, OGE, SAV, TAL
Spotted Bass *Micropterus punctulatus*		CR	CENT	CHA**, COO, FLI**, OCO**, OCM**, TAL
Largemouth Bass *Micropterus salmoides*		CR	CENT	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
White Crappie *Pomoxis annularis*		CR		CHA**, COO, FLI**, OCM**, OCO**, OGE**, SAV**
Black Crappie *Pomoxis nigromaculatus*		CR	CENT	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Percidae				
Holiday Darter *Etheostoma brevirostrum*		IN	BI	COO
Lipstick Darter *Etheostoma chuckwachatte*	INT	IN	BI	TAL
Coosa Darter *Etheostoma coosae*		IN	BI	COO
Etowah Darter *Etheostoma etowahae*	INT	IN	BI	COO
Swamp Darter *Etheostoma fusiforme*		IN	BI	FLI, OCM, OCO, SAV
Christmas Darter *Etheostoma hopkinsi*	HWI	IN	BI	OCM, OCO, OGE, SAV

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Turquoise Darter *Etheostoma inscriptum*	HWI	IN	BI	OCM, OCO, OGE, SAV
Greenbreast Darter *Etheostoma jordani*	INT	IN	BI	COO
Tessellated Darter *Etheostoma olmstedi*	INT	IN	BI	OCM, OCO, OGE, SAV
Goldstripe Darter *Etheostoma parvipinne*		IN	BI	CHA, FLI, OCM
Rock Darter *Etheostoma rupestre*		IN	BI	COO
Cherokee Darter *Etheostoma scotti*	HWI	IN	BI	COO
Speckled Darter *Etheostoma stigmaeum*	INT	IN	BI	COO, TAL
Gulf Darter *Etheostoma swaini*	INT	IN	BI	CHA, FLI
Tallapoosa Darter *Etheostoma tallapoosae*		IN	BI	TAL
Trispot Darter *Etheostoma trisella*		IN	BI	COO
Amber Darter *Percina antesella*		IN	BI	COO
Mobile Logperch *Percina kathae*	INT	IN	BI	COO, TAL

Fish List for the Piedmont Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Freckled Darter *Percina lenticula*		IN	BI	COO
Blackbanded Darter *Percina nigrofasciata*		IN	BI	CHA, COO, FLI, OCM, OCO, OGE, SAV, TAL
Bronze Darter *Percina palmaris*	HWI	IN	BI	COO, TAL
Upland Bridled Darter *Percina* sp.		IN	BI	COO
Muscadine Bridled Darter *Percina* sp.		IN	BI	TAL
Yellow perch *Perca flavescens*		CR		EXOTIC
Sauger *Stizostedion canadense*		CR		CHA**, SAV**
Walleye *Stizostedion vitreum*		CR		CHA**, COO, OCO**, SAV**
Sciaenidae				
Freshwater Drum *Aplodinotus grunniens*		CR		COO

Water Quality Tolerance: **HWI** = headwater intolerant; **INT** = intolerant
Feeding Guild: **CR** = top carnivore; **GE** = generalist; **HB** = herbivore; **IC** = insectivorous cyprinid; **IN** = insectivore/invertivore; **PR** = parasitic
Species Category: **BI** = benthic insectivore species; **CENT** = centrarchid species; **RBS** = round-bodied sucker species; **SF** = sunfish species; **SMM** = subterminal mouth minnow species;
Drainage Basin: **CHA** = Chattahoochee; **COO** = Coosa; **FLI** = Flint; **OCM** = Ocmulgee; **OCO** = Oconee; **OGE** = Ogeechee; **SAV** = Savannah; **TAL** = Tallapoosa
EXOTIC = introduced to Georgia; ****** = species introduced to that drainage basin

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APPENDIX 5B-2

Scoring Criteria for the Index of Biotic Integrity and the Index of Well-Being to Monitor Fish Communities in Wadeable Streams in the Coosa and Tennessee Drainage Basins of the Ridge and Valley Ecoregion of Georgia

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Part IV: Scoring Criteria for the Index of Biotic Integrity and the Index of Well-Being to Monitor Fish Communities in Wadeable Streams in the Coosa and Tennessee Drainage Basins of the Ridge and Valley Ecoregion of Georgia

Georgia Department of Natural Resources
Wildlife Resources Division
Fisheries Management Section

June 1, 2005

Table of Contents

Introduction

The Ridge and Valley ecoregion is one of the six Level III ecoregions found in Georgia (Part 1, Figure 1). It is contained within two major drainage basins, the Coosa and the Tennessee, in the northwestern corner of Georgia. The Ridge and Valley ecoregion covers nearly 3,000 square miles (United States Census Bureau 2000) and includes all or portions of 10 counties (Fig. 1), bordering the Piedmont ecoregion to the south and the Blue Ridge ecoregion to the east. A small portion of the Southwestern Appalachians ecoregion is located in the upper northwestern corner of the Ridge and Valley ecoregion.

The biotic indices developed by the GAWRD are based on the Level III ecoregion delineations (Griffith et al 2001). The metrics and scoring criteria adapted to the Ridge and Valley ecoregion were developed from biomonitoring samples collected in the two major river basins that drain the Ridge and Valley ecoregion, the Coosa (ACT) and the Tennessee (TEN). A total of 169 biomonitoring samples have been collected by the GAWRD in the Ridge and Valley ecoregion since 2001.

A total of 57 native species were collected from samples in the Coosa drainage basin, while 52 native species were collected from samples in the Tennessee drainage basin. Six species on Georgia's list of protected animals of Georgia list were collected in the Ridge and Valley ecoregion. The state listed fish were ranked as endangered, threatened, or rare based on the Endangered Wildlife Act of 1973 (Georgia Department of Natural Resources, Nongame – Endangered Wildlife Program, 1999). The flame chub (*Hemitremia flammea*), ranked as endangered, was collected in the Tennessee drainage basin. Three species were ranked as threatened: the stargazing minnow (*Phenacobius uranops*) and the northern studfish (*Fundulus catenatus*), which were collected in the Tennessee drainage basin, and the trispot darter (*Etheostoma trisella*), which was found in the upper Coosa drainage basin. Three species ranked as rare were collected from the Tennessee drainage basin: the bigeye chub (*Hybopsis amblops*), the black darter (*Etheostoma duryi*), and the dusky darter (*Percina sciera*). Table 1 shows a complete list of state listed fish found in the Ridge and Valley ecoregion of Georgia.

IBI scores were generally higher in the Ridge and Valley ecoregion than in the Piedmont and Southeastern Plains ecoregions. Based on the IBI integrity classes (Part I, Table 2), 22 sites scored in the excellent class, 47 scored in the good class, 41 scored in the fair class, 29 scored in

the poor class, and 30 scored in the very poor class. IBI scores in the Ridge and Valley ecoregion ranged from a maximum of 58 to a minimum of 12. Unlike the Piedmont ecoregion, more sites scored in the excellent and good integrity classes ([69/169] * 100 = 40.8) than in the poor and very poor integrity classes ([59/169] * 100 = 34.9). Major impacts in the Ridge and Valley ecoregion include the effects of animal agriculture production and urban / suburban development.

Table 2 shows the scoring criteria for the IBI metrics in the Ridge and Valley ecoregion. The Maximum Species Richness (MSR) graphs for each basin group within the Ridge and Valley ecoregion are included in Appendix 1. Figures ACT1 - RGV through ACT6b - RGV depict the MSR graphs used to score the species richness metrics (metrics 1 – 6b) in the Coosa drainage basin. Figures TEN1 - RGV through TEN6b - RGV depict the MSR graphs used to score the species richness metrics in the Tennessee drainage basin. The fish list for the Ridge and Valley ecoregion showing the water quality tolerance rankings, feeding guilds, and species categories used in calculating the IBI is also included in Appendix 1.

Based on the modified Iwb integrity classes for the Ridge and Valley ecoregion (Table 3), 16 sites scored in the excellent class, 49 scored in the good class, 68 scored in the fair class, 14 scored in the poor class, and 22 scored in the very poor class. Modified Iwb scores in headwater streams ranged from a maximum score of 10.04 to a minimum of 0.89. At larger wadeable streams, modified Iwb scores ranged from a maximum of 10.24 to a minimum of 5.86. There was a significant relationship between the indices across the Ridge and Valley ecoregion ($r = 0.8379$, $p = 0.0000$, $N = 169$), although the relationship was stronger in larger wadeable streams ($r = 0.8838$, $p = 0.0000$, $N = 44$) than in headwater streams ($r = 0.8322$, $p = 0.0000$, $N = 169$).

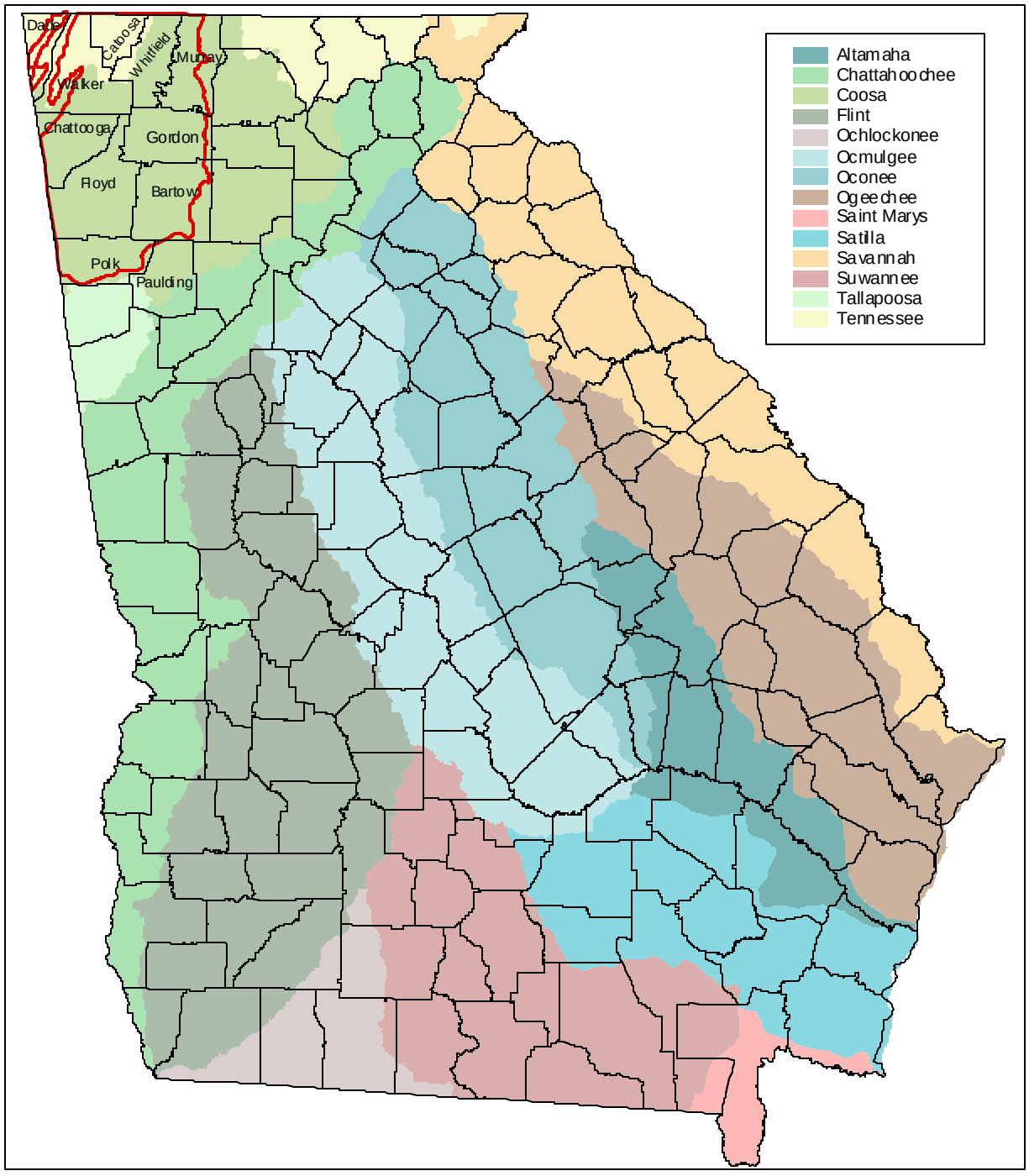


Figure 1. Level III Ridge and Valley ecoregion (outlined in bold red) in Georgia. Major drainage basins include the Coosa and the Tennessee.

Table 1. State listed fish found in the Ridge and Valley ecoregion of Georgia (Georgia Department of Natural Resources, Nongame – Endangered Wildlife Program, 1999).

Species	State Status	Federal Status	Basin
Blue Shiner *(Cyprinella caerulea)*	E	T	COO
Holiday Darter *(Etheostoma brevirostrum)*	T	None	COO
Coldwater Darter *(Etheostoma ditrema)*	T	None	COO
Black Darter *(Etheostoma duryi)*	R	None	TEN
Trispot Darter *(Etheostoma trisella)*	T	None	COO
Northern Studfish *(Fundulus catenatus)*	T	None	TEN
Flame Chub *(Hemitremia flammea)*	E	None	TEN
Bigeye Chub *(Hybopsis amblops)*	R	None	TEN
Ohio Lamprey *(Ichthyomyzon bdellium)*	R	None	TEN
River Redhorse *(Moxostoma carinatum)*	R	None	COO, TEN
Popeye Shiner *(Notropis ariommus)*	T	None	TEN
Mountain Madtom *(Noturus eleutherus)*	T	None	TEN
Frecklebelly Madtom *(Noturus munitus)*	E	None	COO
Amber Darter *(Percina antesella)*	E	E	COO
Goldline Darter *(Percina aurolineata)*	T	T	COO
Conasauga Logperch *(Percina jenkinsi)*	E	E	COO
Freckled Darter *(Percina lenticula)*	E	None	COO
Dusky Darter *(Percina sciera)*	R	None	TEN
River Darter *(Percina shumardi)*	E	None	COO, TEN
Upland Bridled Darter *(Percina* sp.*)*	R	None	COO
Snail Darter *(Percina tanasi)*	T	T	TEN
Stargazing Minnow *(Phenacobius uranops)*	T	None	TEN

Status: E = endangered; R = rare; T = threatened
Basin: COO = Coosa; TEN = Tennessee

Table 2. Index of Biotic Integrity metrics for wadeable streams in the Ridge and Valley ecoregion of Georgia.

Metric	Basin Group	Scoring Criteria		
1. Number of native species	COO / TEN	MSR Graphs		
2. Number of benthic invertivore species	COO / TEN	MSR Graphs		
3a. Number of native sunfish species[a]	COO / TEN	MSR Graphs		
3b. Number of native centrarchid species[b]	COO / TEN	MSR Graphs		
4. Number of native insectivorous cyprinid species	COO / TEN	MSR Graphs		
5. Number of native round-bodied sucker species	COO / TEN	MSR Graphs		
6a. Number of sensitive species[a]	COO / TEN	MSR Graphs		
6b. Number of intolerant species[b]	COO / TEN	MSR Graphs		
		5	3	1
7. Evenness	COO	≥ 77	77 - ≥ 69	< 69
	TEN	≥ 73	73 - ≥ 65	< 65
8. % of individuals as *Lepomis* species	COO	≤ 30	30 - ≤ 54	> 54
	TEN	≤ 28	28 - ≤ 53	> 53
9. % of individuals as insectivorous cyprinids	COO	≥ 28	28 - ≥ 14	< 14
	TEN	≥ 34	34 - ≥ 17	< 17

		5	3	1
10a. % of individuals as generalist feeders and herbivores	COO	≤ 25	25 - ≤ 44	> 44
	TEN	≤ 21	21 - ≤ 40	> 40
10b. % of individuals as top carnivores[b]	COO	≥ 3.5 - ≤ 8.75	≥ 1.75 – 3.5 or 8.75 - ≤ 10.5	< 1.75 or > 10.5
	TEN	≥ 3.8 - ≤ 9.5	≥ 1.9 – 3.8 or 9.5 - ≤ 11.4	< 1.9 or > 11.4
11. % of individuals as benthic fluvial specialist	COO	≥ 27	27 - ≥ 15	< 15
	TEN	≥ 26	26 - ≥ 13	< 13
12. Number of individuals per 200 meters	COO	≥ 720	720 - ≥ 360	< 360
	TEN	≥ 800	800 - ≥ 400	< 400
13. % of individuals with external anomalies	COO / TEN	> 1.2 – subtract 4 points from total score		

[a] used at sites with an upstream drainage basin area < 15 square miles
[b] used at sites with an upstream drainage basin area ≥ 15 square miles

Table 3. Index of well-being scoring criteria and integrity classes for wadeable streams in the Ridge and Valley ecoregion of Georgia.

Iwb Score	DBA (Sq. miles)	Integrity Class	Attributes
$\geq$ 9.5	< 15	Excellent	Comparable to the best regional reference conditions; all regionally expected species for the habitat and stream size, including the most intolerant species, are present with a full array of size classes; healthy species diversity within the fish community, indicated by elevated evenness scores; number of individuals abundant; total biomass is high, with each level of the food web represented, indicating a balanced trophic structure.
$\geq$ 9.85	$\geq$ 15		
9.5 - $\geq$ 8.6	< 15	Good	Species richness somewhat below expectation; evenness scores decrease as species diversity falls, especially due to the loss of the most intolerant forms; good number of individuals in the sample, with several species of benthic fluvial specialist and insectivorous cyprinids present; some decreases in total biomass as trophic structure shows some signs of stress.
9.85 - $\geq$ 9.25	$\geq$ 15		
8.6 - $\geq$ 6.8	< 15	Fair	Species richness and diversity decline as some expected species are absent; abundance of individuals declines; total biomass continues to decline as some levels of the food web in low abundance or missing; trophic structure skewed toward generalist feeders and/or *Lepomis* species as the abundance of insectivorous cyprinid and benthic fluvial specialist species decreases.
9.25 - $\geq$ 8.05	$\geq$ 15		
6.8 - $\geq$ 5.9	< 15	Poor	Number of individuals is low; species richness and diversity are very low, with benthic fluvial specialist and insectivorous cyprinid species in low abundance or absent; sample dominated by generalist feeders, herbivores, and *Lepomis* species; increase in the proportions of non-native species and hybrids; growth rates depressed as sample is heavily skewed to the smaller size classes; total biomass low.
8.05 - $\geq$ 7.45	$\geq$ 15		
< 5.9	< 15	Very Poor	Sample represented by few individuals, mainly generalist feeders and *Lepomis* species; some sites dominated by non-native species; total biomass very low.
< 7.45	$\geq$ 15		

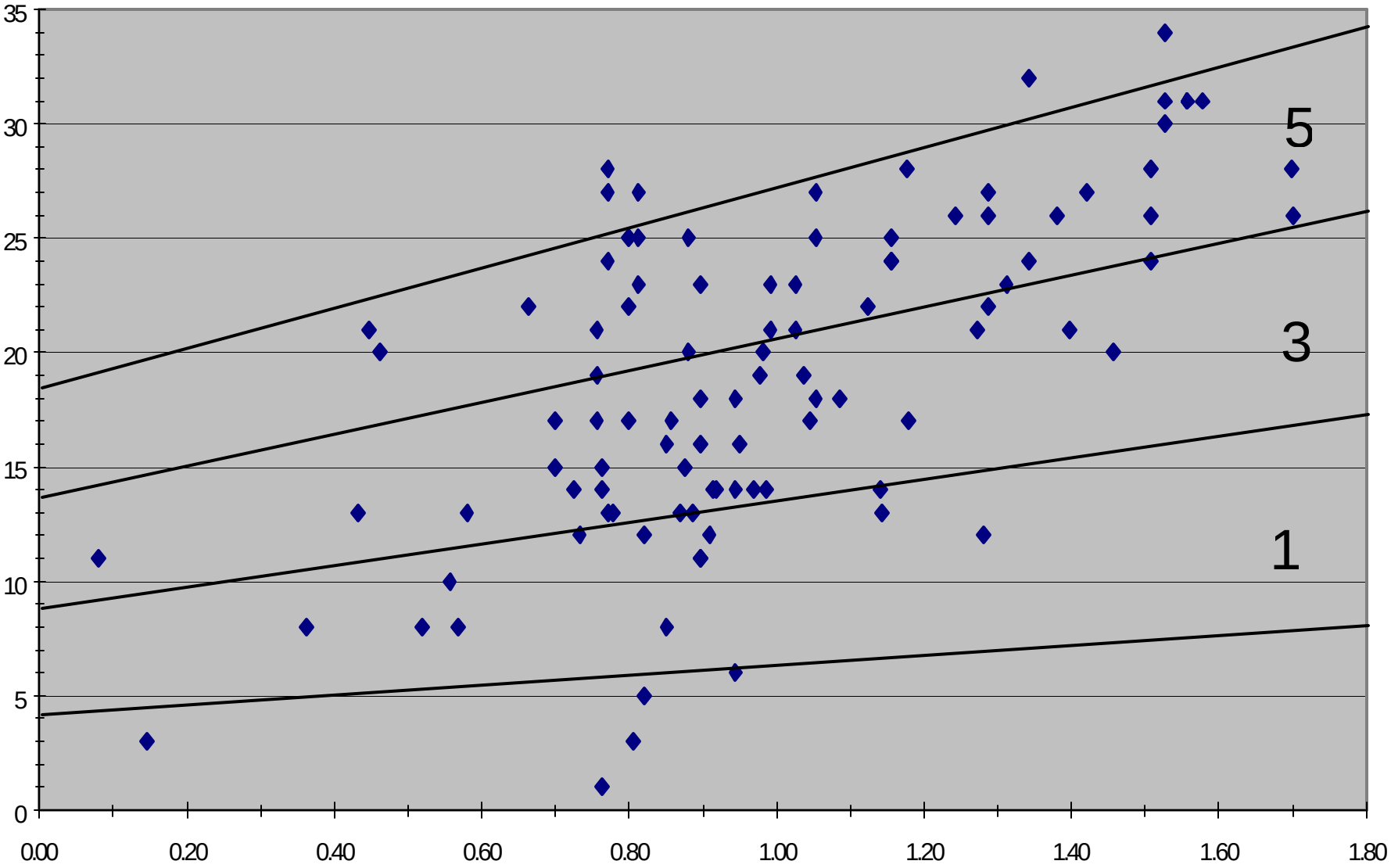
References

Georgia Department of Natural Resources, Wildlife Resources Division. 1999. Protected Animals of Georgia. Nongame Wildlife – Natural Heritage Section, Forsyth, Georgia.

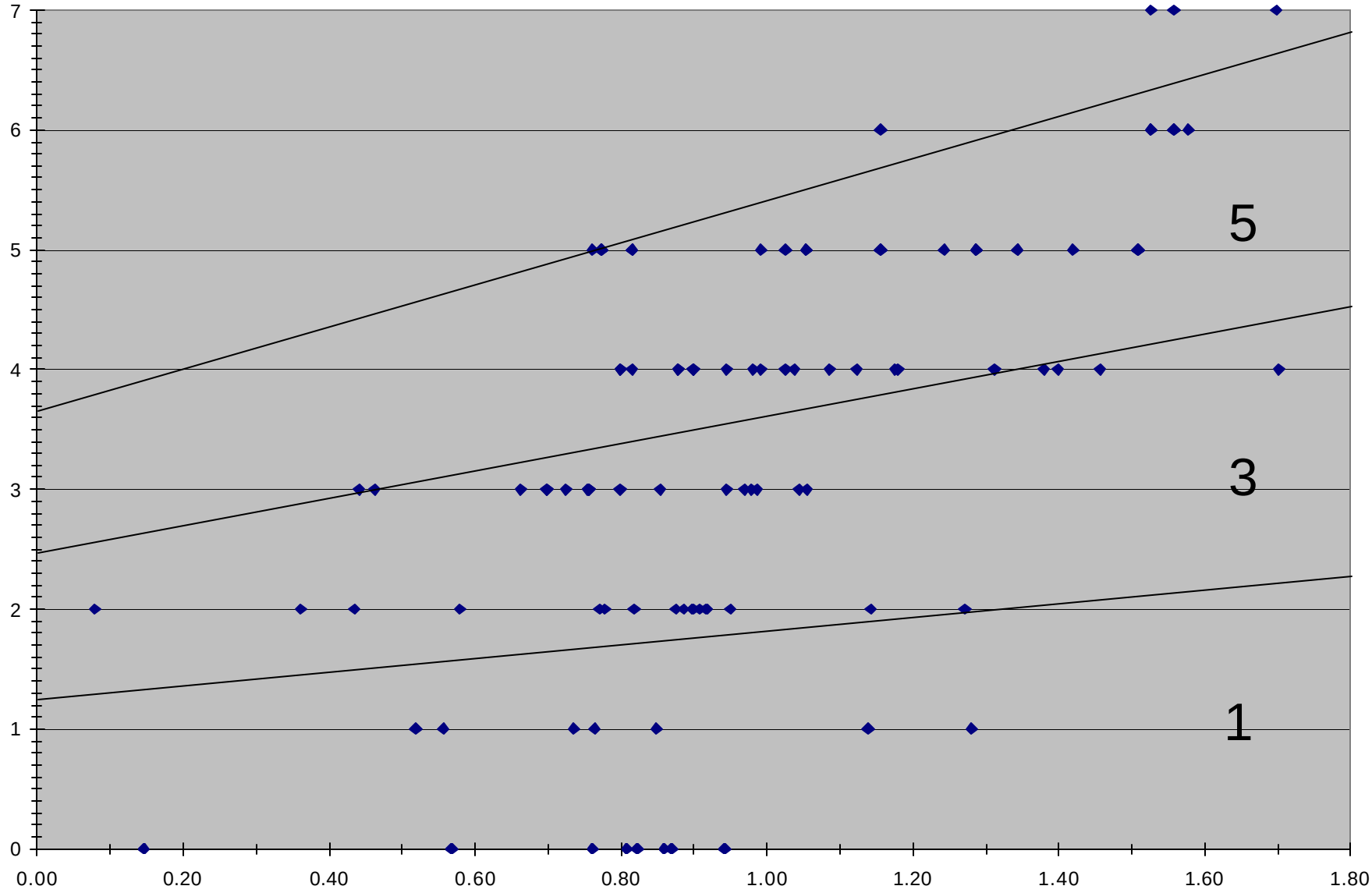
Griffith, G.E., J.M. Omernik, J.A. Comstock, S. Lawrence, and T. Foster. 2001. Level III and IV Ecoregions of Georgia, (color poster with map, descriptive text, summary tables, and photographs). Reston, Virginia, U.S. Geological Survey.

United States Census Bureau. 2000. 2000 Census of Population and Housing. United States Census Bureau, Washington, D.C.

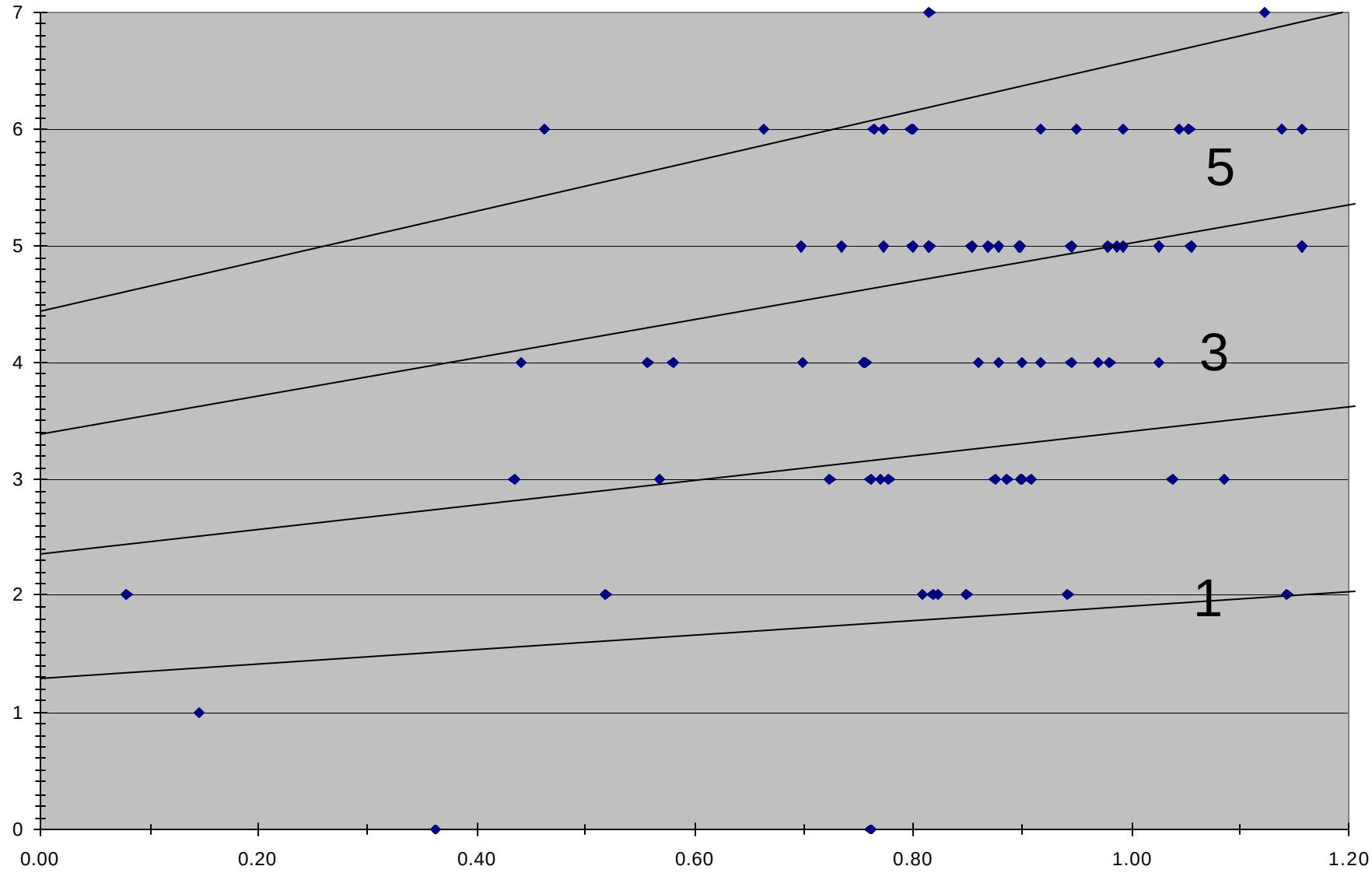
Appendix 1



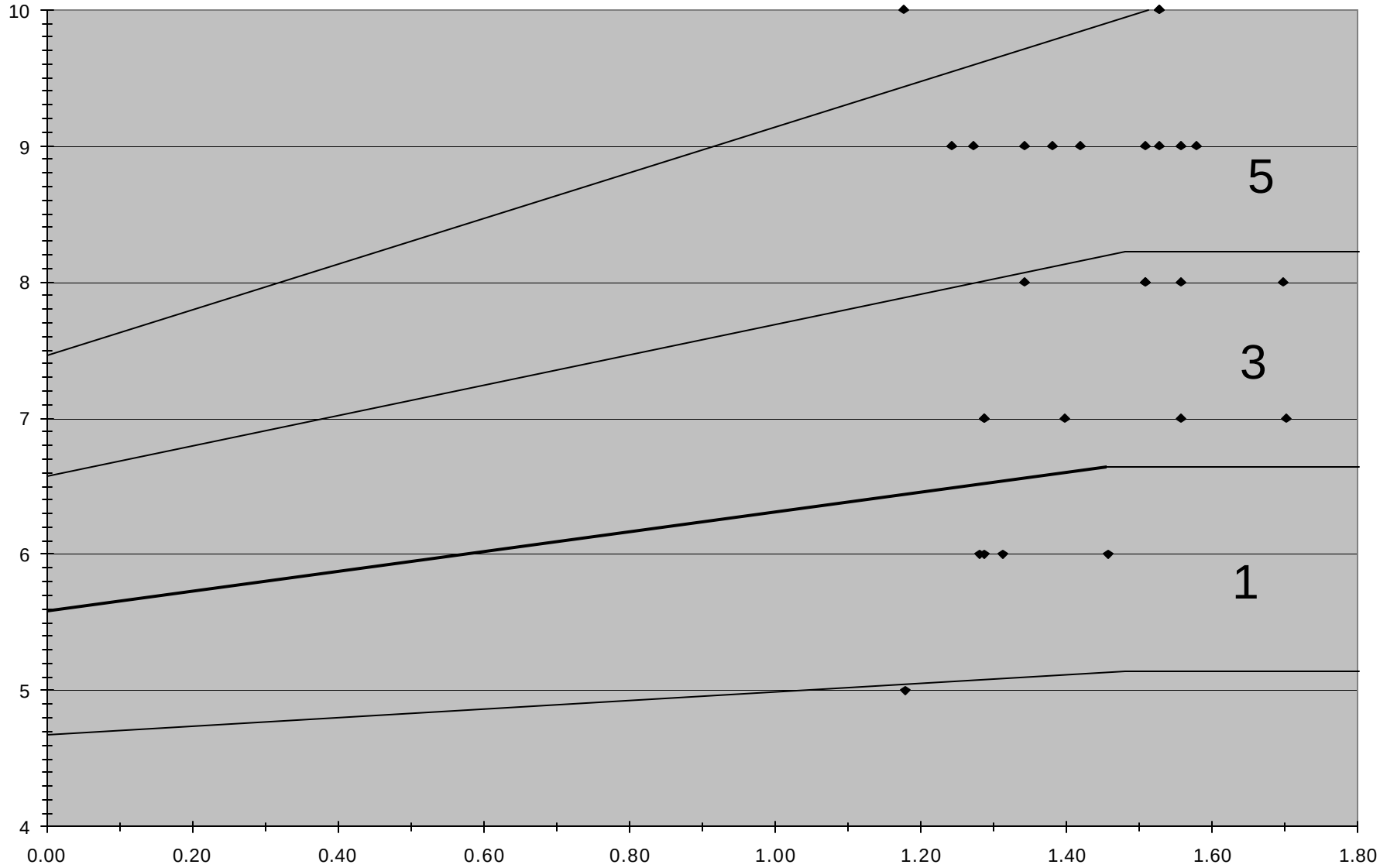
ACT1 - RGV. Total number of species in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 102.



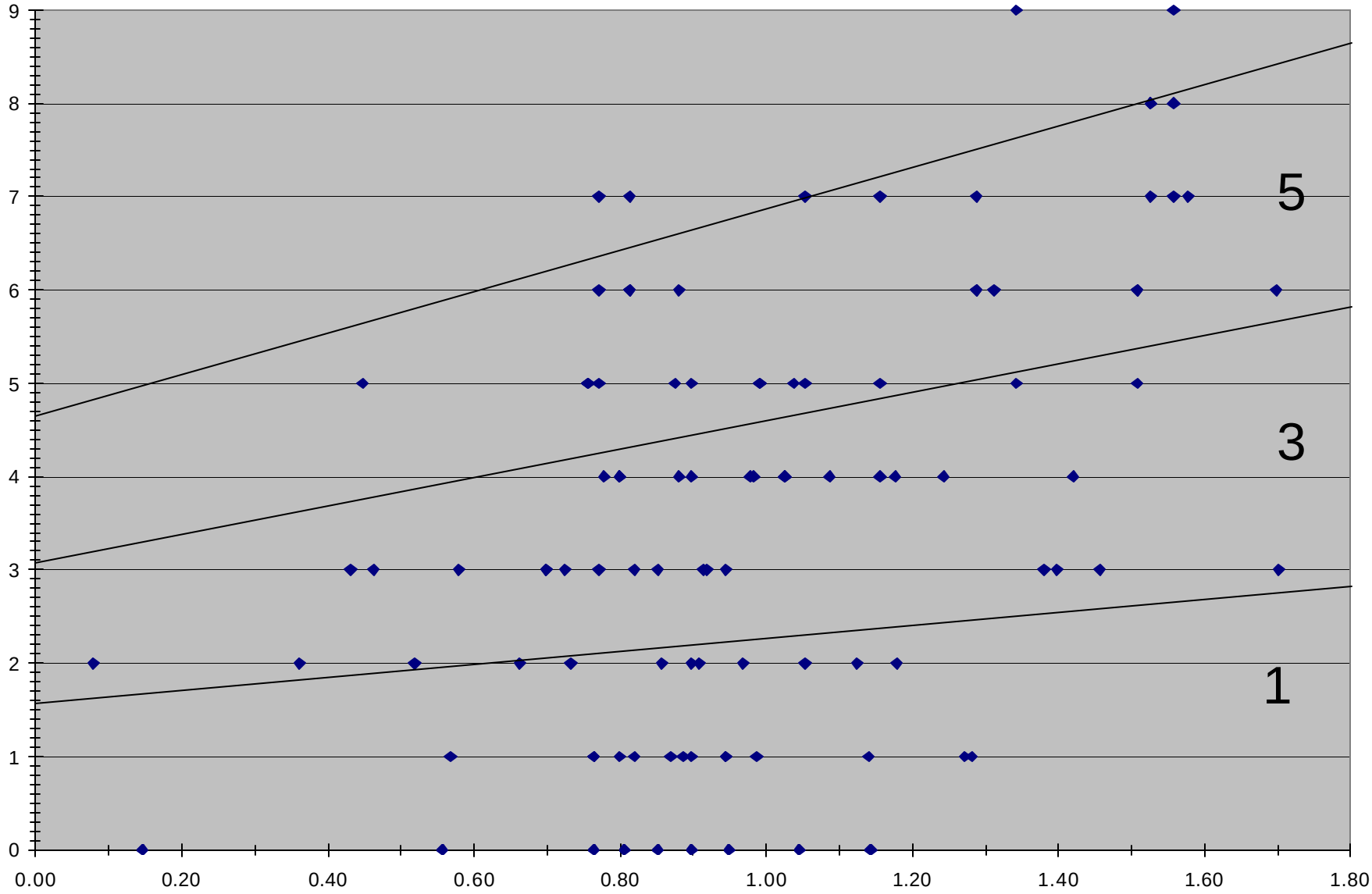
ACT2 - RGV. Number of benthic invertivore species in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 102.



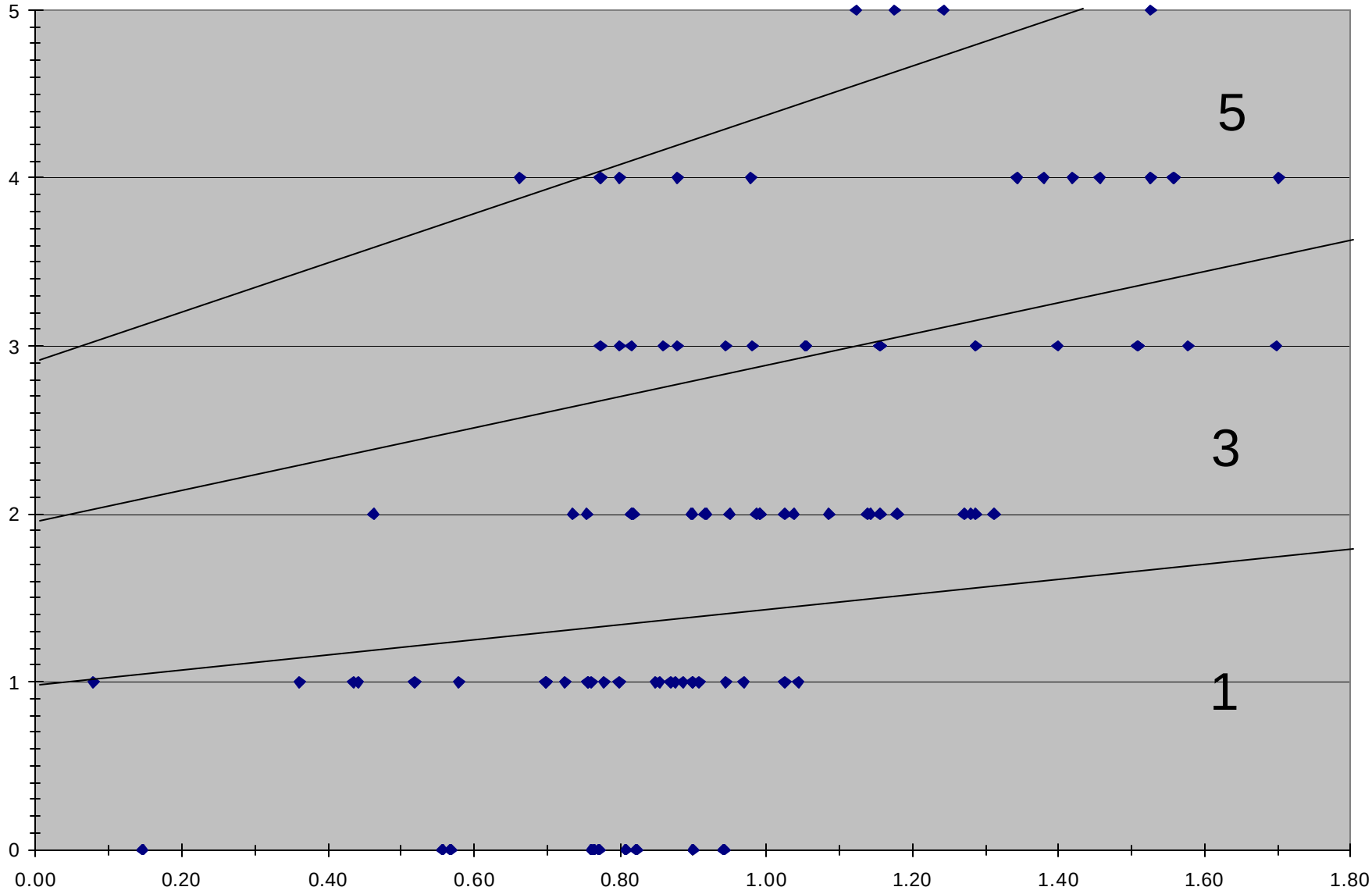
ACT3a - RGV. Number of native sunfish species in headwater streams (<15 square miles drainage basin area) in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 75.



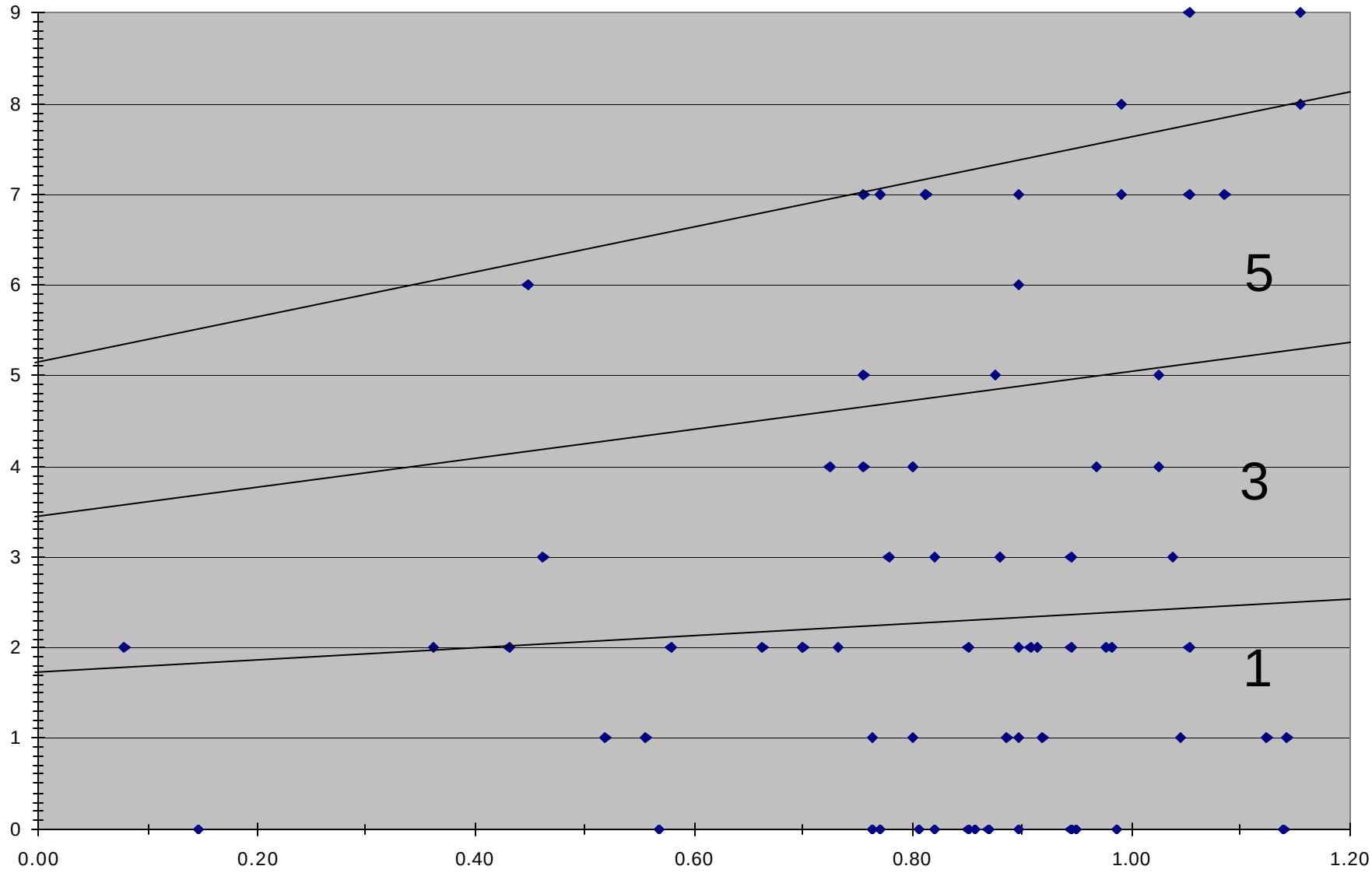
ACT3b - RGV. Number of native centrarchid species in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 30 square miles. Total samples equal 27.



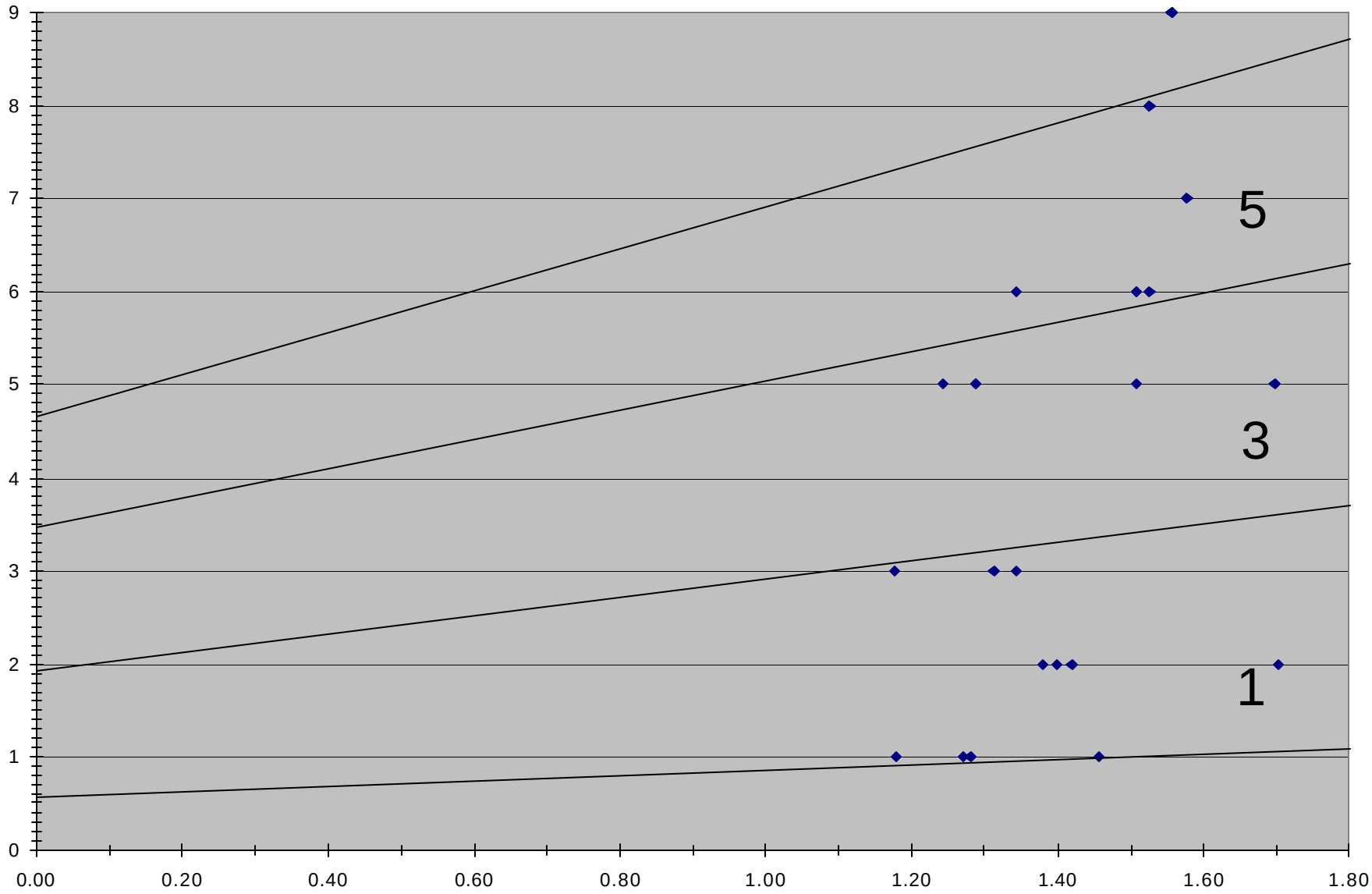
ACT4 - RGV. Number of native insectivorous cyprinid species in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 102.



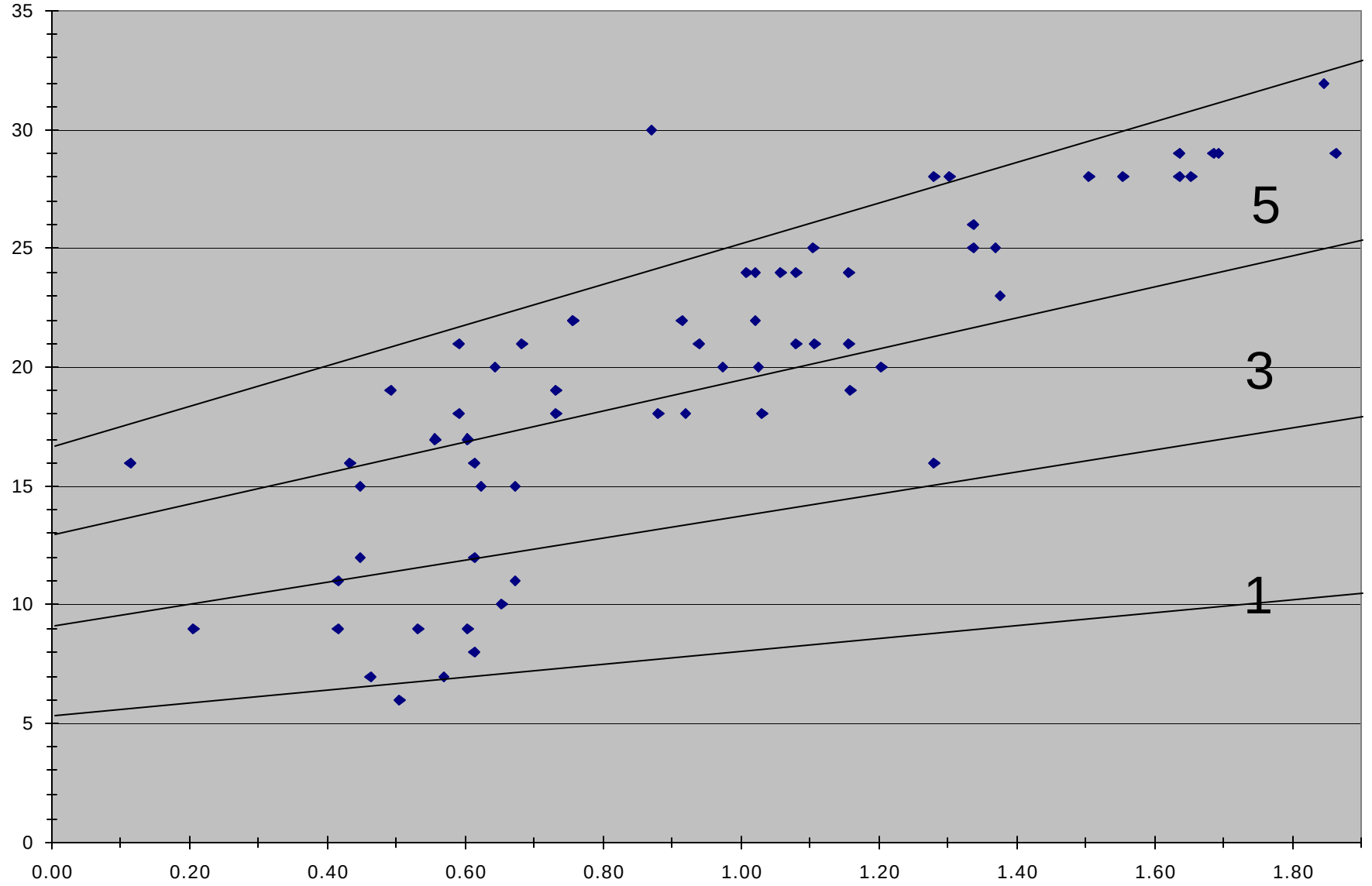
ACT5 - RGV. Number of native round-bodied sucker species in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 102.



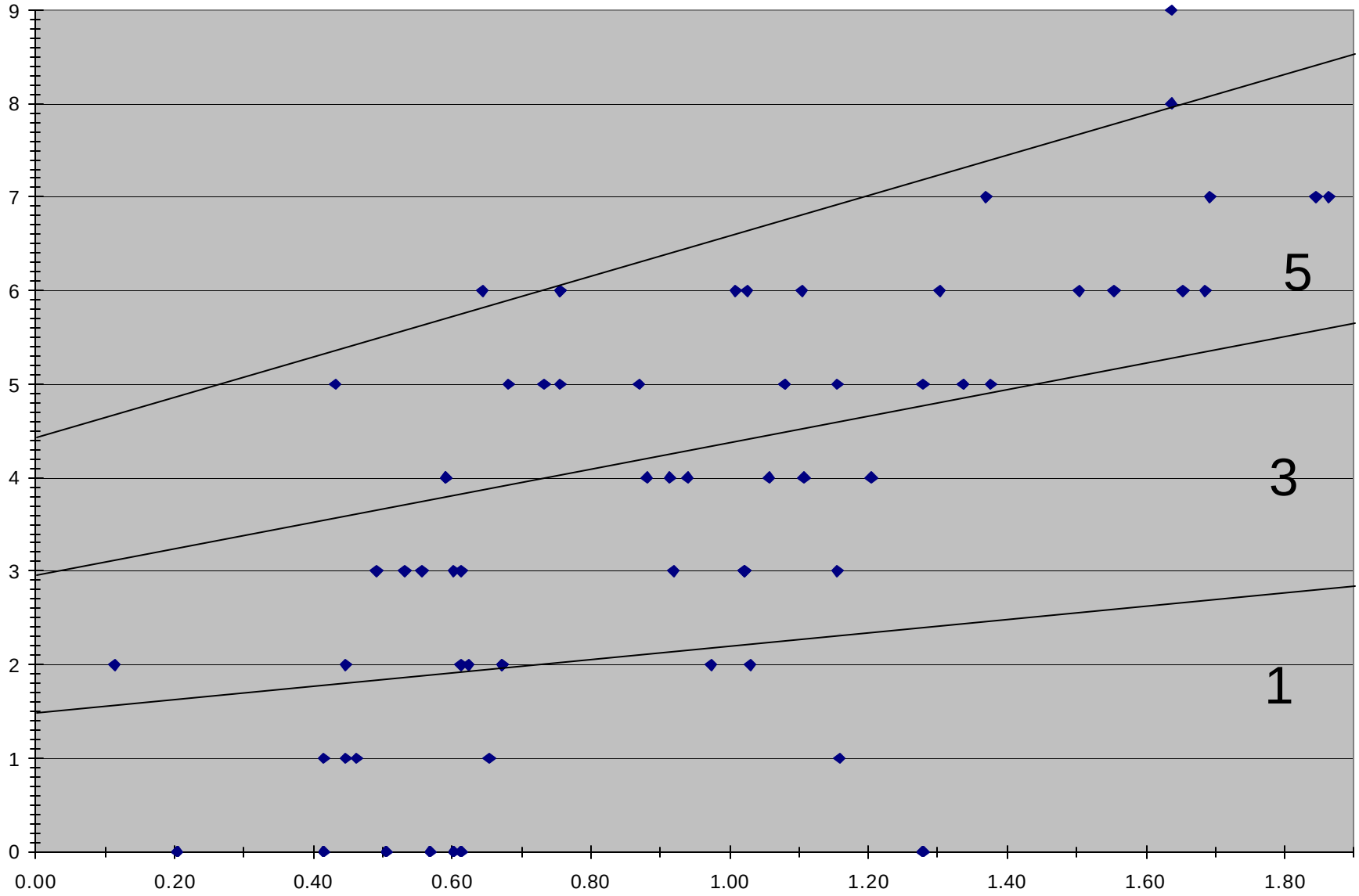
ACT6a - RGV. Total number of species ranked as sensitive at headwater sites (<15 square miles drainage basin area) in the Ridge and Valley ecoregion of the Coosa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 75.



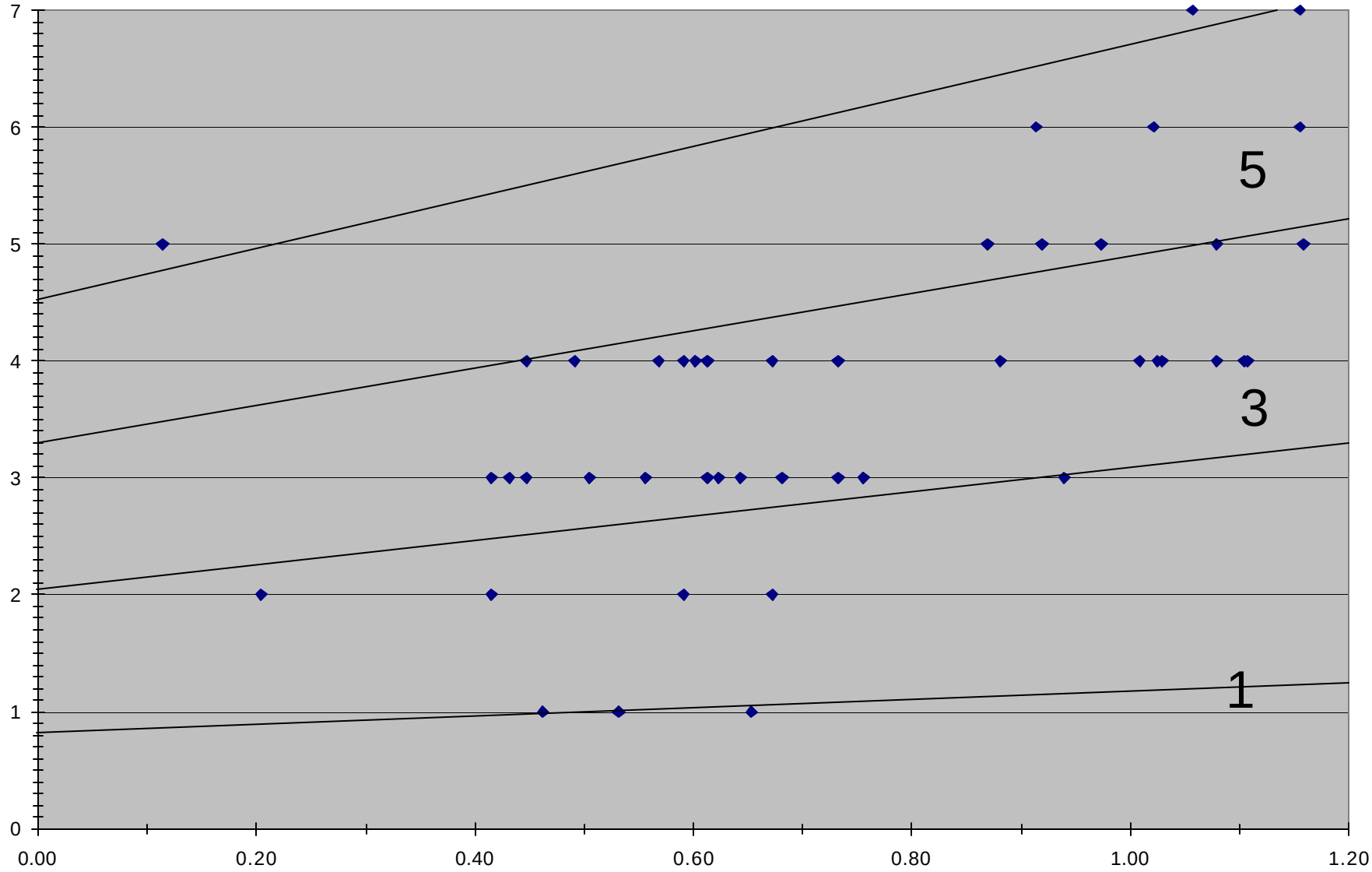
ACT6b - RGV. Number of species ranked as intolerant in the Ridge and Valley ecoregion of the Coosaa drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 27.



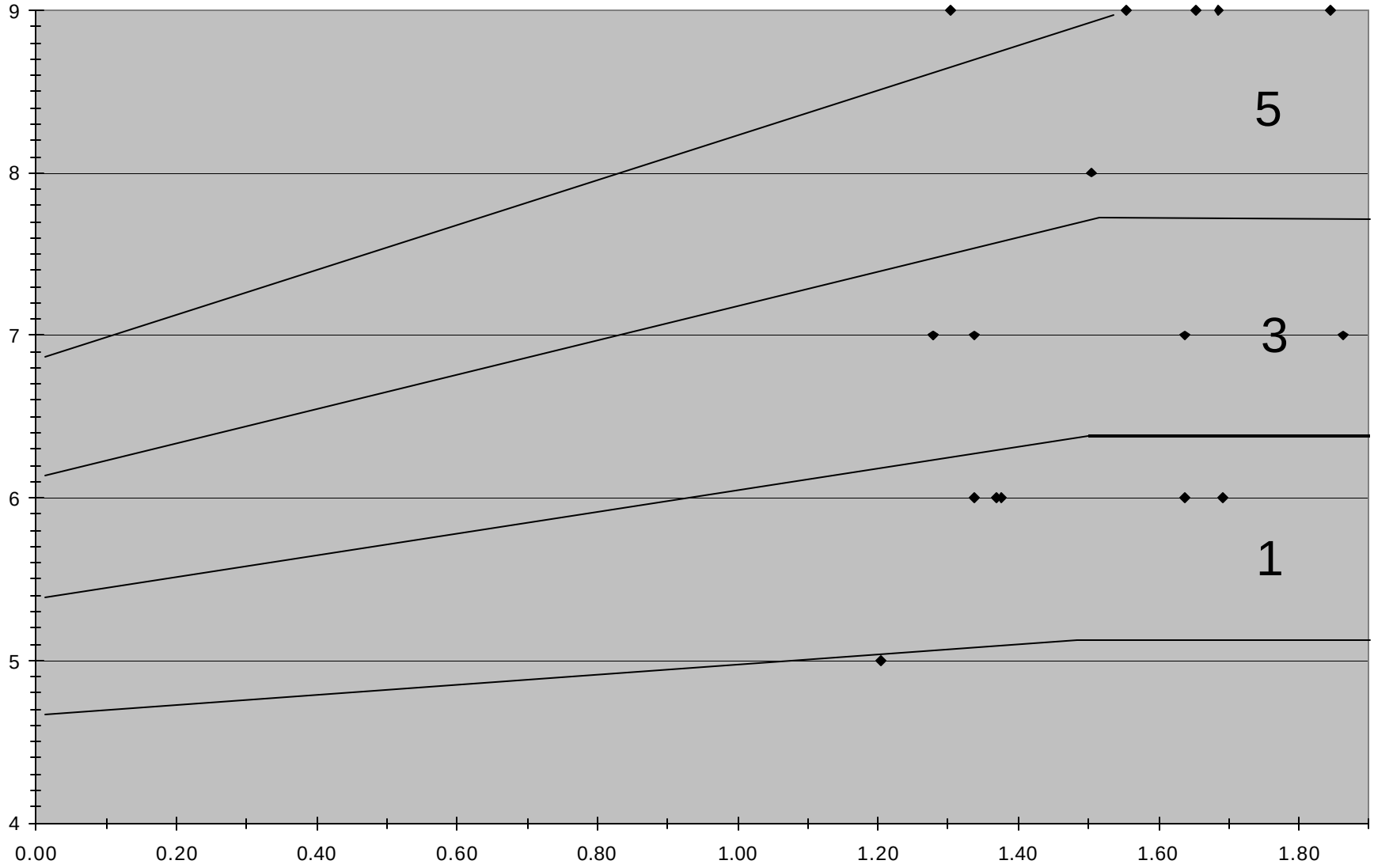
TEN1 - RGV. Total number of native species in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 67.



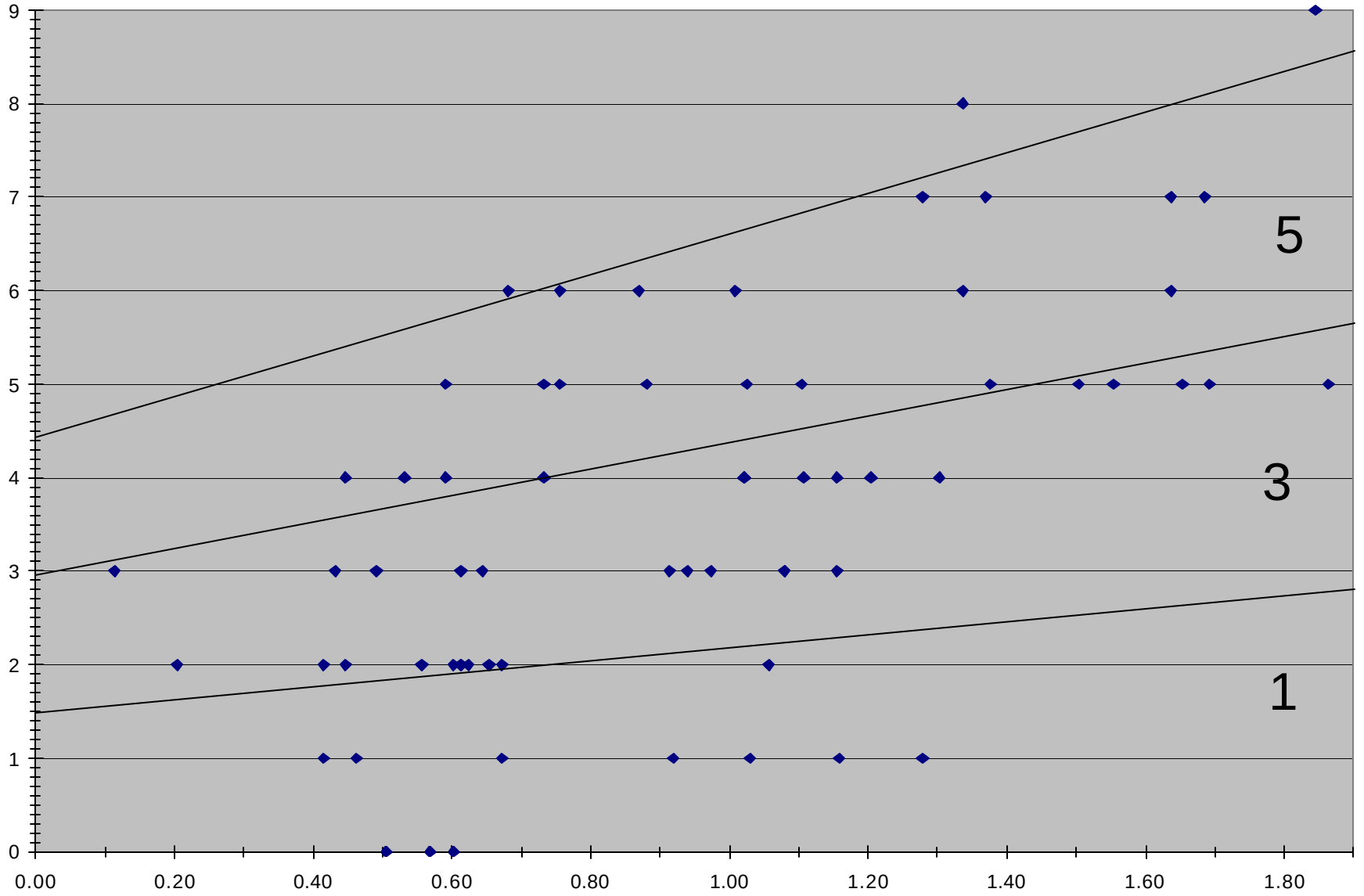
TEN2 - RGV. Number of benthic invertivore species in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 67.



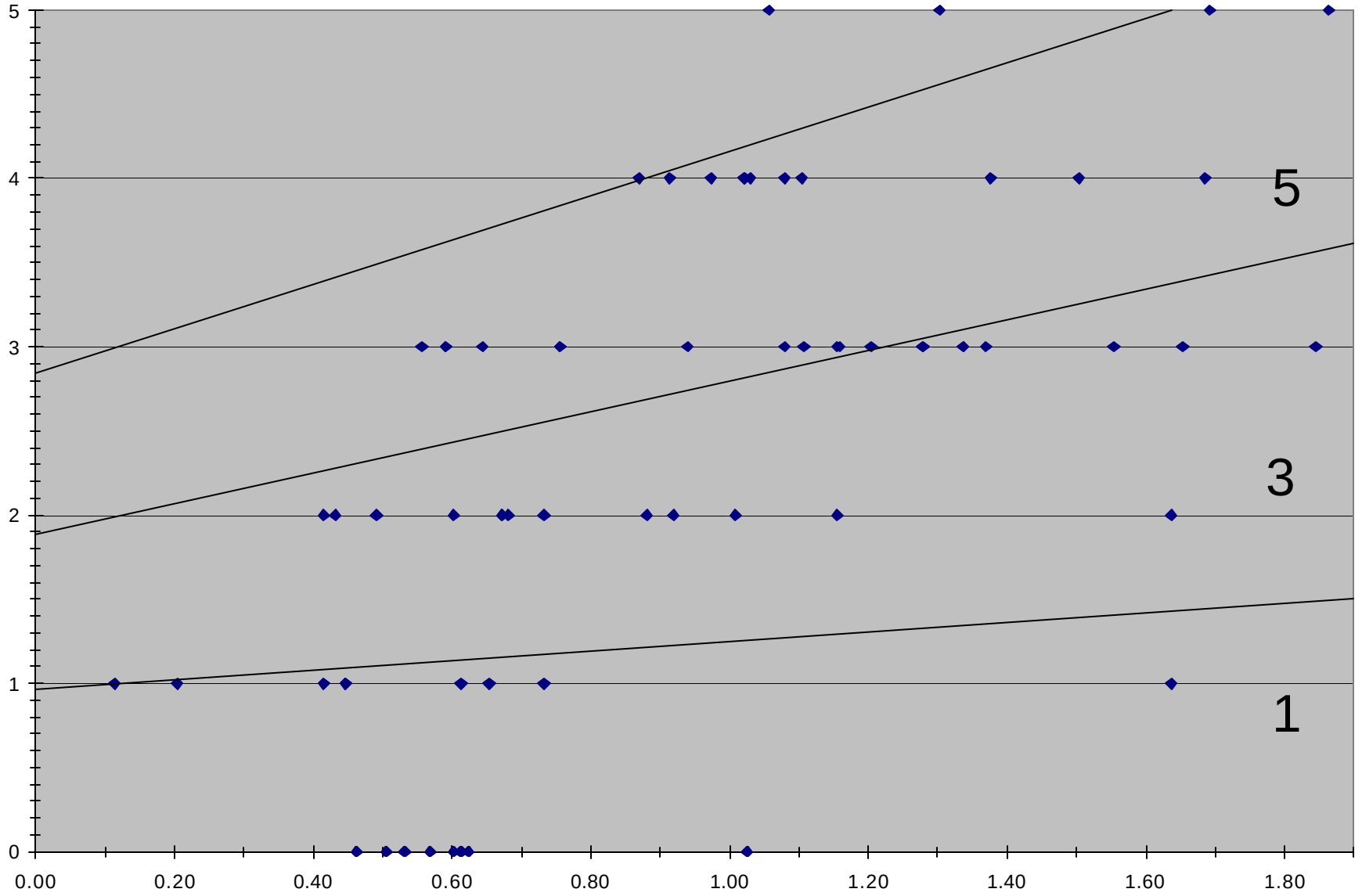
TEN3a - RGV. Number of native sunfish species in headwater streams (<15 square miles drainage basin area) in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 50.



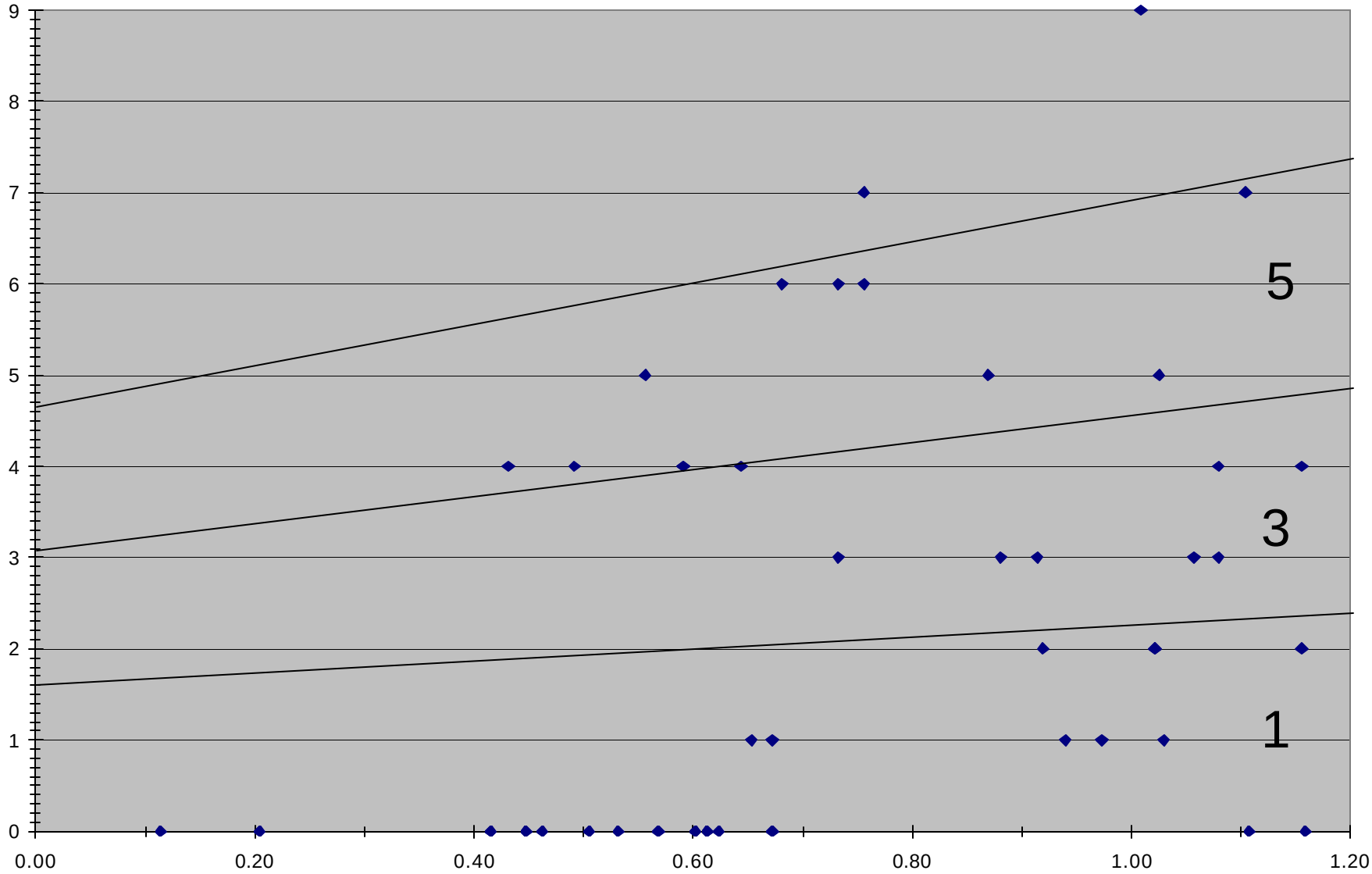
TEN3b - RGV. Number of native centrarchid species in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Flatlines at 30 square miles. Total samples equal 17.



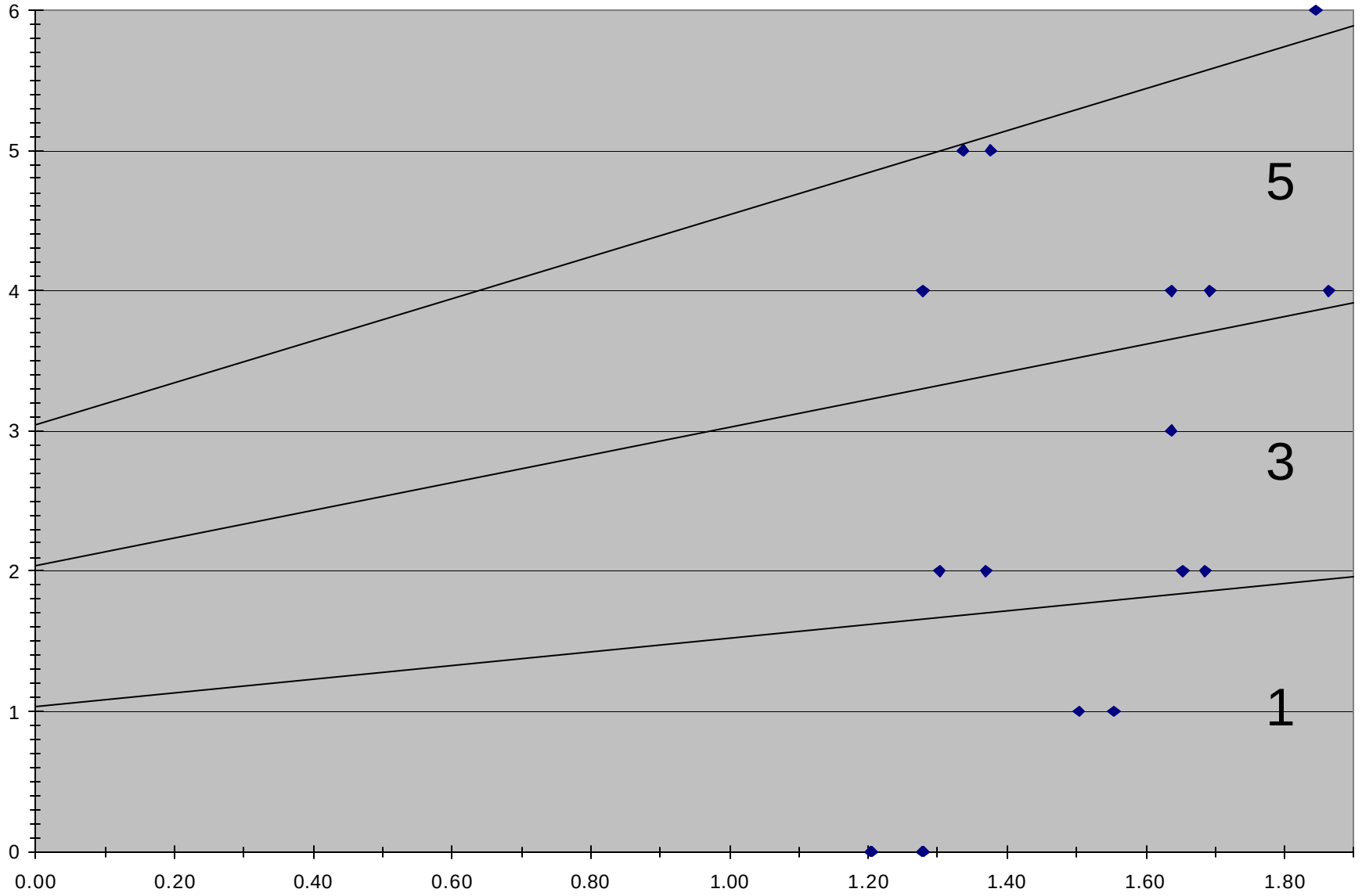
TEN4 - RGV. Number of native insectivorous cyprinid species in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 67.



TEN5 - RGV. Number of native round-bodied sucker species in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 67.



TEN6a - RGV. Total number of species ranked as sensitive at headwater sites (<15 square miles drainage basin area) in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed valued of the drainage basin area (square miles). Total samples equal 50.



TEN6b - RGV. Number of species ranked as intolerant in the Ridge and Valley ecoregion of the Tennessee drainage basin plotted against the log (base 10) transformed value of the drainage basin area (square miles). Total samples equal 17.

Fish List for the Ridge and Valley Ecoregion of Georgia. (Updated May 11, 2005)

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Petromyzontidae				
Ohio Lamprey *Ichthyomyzon bdellium*		PR		TEN
Chestnut Lamprey *Ichthyomyzon castaneus*		PR		COO, TEN
Southern Brook Lamprey *Ichthyomyzon gagei*		HB		COO
Mountain Brook Lamprey *Ichthyomyzon greeleyi*		HB		TEN
Least Brook Lamprey *Lampetra aepyptera*		HB		COO
American Brook Lamprey *Lampetra appendix*	HWI	HB		TEN
Acipenseridae				
Lake Sturgeon *Acipenser fulvescens*		IN		COO
Lepisosteidae				
Spotted Gar *Lepisosteus oculatus*		CR		COO, TEN
Longnose Gar *Lepisosteus osseus*		CR		COO, TEN
Hiodontidae				
Mooneye *Hiodon tergisus*		IN		COO
Clupeidae				
Skipjack Herring *Alosa chrysochloris*		CR		TEN
Gizzard Shad *Dorosoma cepedianum*		GE		COO, TEN
Threadfin Shad *Dorosoma petenense*		HB		COO, TEN
Cyprinidae				
Largescale Stoneroller *Campostoma oligolepis*		HB		COO, TEN
Goldfish *Carassius auratus*		GE		EXOTIC

Fish List for the Ridge and Valley Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Grass Carp *Ctenopharyngodon idella*		HB		EXOTIC
Blue Shiner *Cyprinella caerulea*		IC	SMM	COO
Alabama Shiner *Cyprinella callistia*	INT	IC	SMM	COO
Whitetail Shiner *Cyprinella galactura*	INT	IC	SMM	TEN
Red Shiner *Cyprinella lutrensis*		GE		EXOTIC
Spotfin Shiner *Cyprinella spiloptera*		IC		TEN
Tricolor Shiner *Cyprinella trichroistia*	INT	IC		COO
Blacktail Shiner *Cyprinella venusta*		IC		COO
Common Carp *Cyprinus carpio*		GE		EXOTIC
Flame Chub *Hemitremia flammea*		IC		TEN
Bigeye Chub *Hybopsis amblops*		IC	SMM	TEN
Lined Chub *Hybopsis lineapunctata*		IC	SMM	COO
Striped Shiner *Luxilus chrysocephalus*		IC		COO, TEN
Warpaint Shiner *Luxilus coccogenis*		IC		TEN
Bandfin Shiner *Luxilus zonistius*		IC		COO**
Rosefin Shiner *Lythrurus fasciolaris*	HWI	IC		TEN
Mountain Shiner *Lythrurus lirus*	INT	IC		COO, TEN

Fish List for the Ridge and Valley Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Speckled Chub *Macrhybopsis aestivalis*		IC	SMM	COO
Silver Chub *Macrhybopsis storeriana*		IC	SMM	COO
River Chub *Nocomis micropogon*		IC	SMM	COO**, TEN
Golden Shiner *Notemigonus crysoleucas*		GE		COO, TEN
Popeye Shiner *Notropis ariommus*		IC		TEN
Burrhead Shiner *Notropis asperifrons*	INT	IC	SMM	COO
Emerald Shiner *Notropis atherinoides*		IC		TEN
Rainbow Shiner *Notropis chrosomus*	HWI	IC		COO
Tennessee Shiner *Notropis leuciodus*		IC		TEN
Silver Shiner *Notropis photogenis*		IC		TEN
Silverstripe Shiner *Notropis stilbius*	INT	IC		COO
Telescope Shiner *Notropis telescopus*	INT	IC		TEN
Mimic Shiner *Notropis volucellus*	INT	IC	SMM	COO, TEN
Coosa Shiner *Notropis xaenocephalus*		IC		COO
Riffle Minnow *Phenacobius catostomus*		IC	SMM	COO
Stargazing Minnow *Phenacobius uranops*	INT	IC	SMM	TEN
Tennessee Dace *Phoxinus tennesseensis*		HB		TEN

Fish List for the Ridge and Valley Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Bluntnose Minnow *Pimephales notatus*		GE		TEN
Fathead Minnow *Pimephales promelas*		GE		EXOTIC
Bullhead Minnow *Pimephales vigilax*		GE		COO, TEN
Blacknose Dace *Rhinichthys atratulus*		IC	SMM	COO, TEN
Creek Chub *Semotilus atromaculatus*		GE		COO, TEN
Catostomidae				
River Carpsucker *Carpiodes carpio*		GE		TEN
Quillback *Carpiodes cyprinus*		GE		TEN
White Sucker *Catostomus commersoni*		IN	RBS	TEN
Alabama Hogsucker *Hypentelium etowanum*		IN	RBS	COO
Northern Hogsucker *Hypentelium nigricans*		IN	RBS	TEN
Smallmouth Buffalo *Ictiobus bubalus*		GE		COO, TEN
Spotted Sucker *Minytrema melanops*		IN	RBS	COO, TEN
Silver Redhorse *Moxostoma anisurum*		IN	RBS	TEN
River Redhorse *Moxostoma carinatum*		IN	RBS	COO, TEN
Black Redhorse *Moxostoma duquesnei*		IN	RBS	COO, TEN
Golden Redhorse *Moxostoma erythrurum*		IN	RBS	COO, TEN
Blacktail Redhorse *Moxostoma poecilurum*		IN	RBS	COO

Fish List for the Ridge and Valley Ecoregion of Georgia.

Species	Tolerance Ranking	Feeding Guild	Species Category	Drainage Basin
Ictaluridae				
Snail Bullhhead *Ameiurus brunneus*		GE		COO, TEN**
Black Bullhead *Ameiurus melas*		GE		COO, TEN
Yellow Bullhead *Ameiurus natalis*		GE		COO, TEN
Brown Bullhead *Ameiurus nebulosus*		GE		COO, TEN
Blue Catfish *Ictalurus furcatus*		CR		COO, TEN
Channel Catfish *Ictalurus punctatus*		GE		COO, TEN
Speckled Madtom *Noturus leptacanthus*	HWI	IN	BI	COO
Mountain Madtom *Noturus eleutherus*		IN	BI	TEN
Yellowfin Madtom *Noturus flavipinnis*		IN	BI	TEN
Frecklebelly Madtom *Noturus munitus*		IN	BI	COO
Flathead Catfish *Pylodictis olivaris*		CR		COO, TEN
Esocidae				
Redfin Pickerel *Esox americanus*		CR		COO
Chain Pickerel *Esox niger*		CR		COO
Salmonidae				
Rainbow Trout *Oncorhynchus mykiss*		CR		EXOTIC
Brown Trout *Salmo trutta*		CR		EXOTIC
Brook Trout *Salvelinus fontinalis*		CR		COO**, TEN

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Fundulidae				
Northern Studfish *Fundulus catenatus*	INT	IN		TEN
Blackspotted Topminnow *Fundulus olivaceus*		IN		COO, TEN
Southern Studfish *Fundulus stellifer*	HWI	IN		COO
Poeciliidae				
Mosquitofish *Gambusia* sp.		GE		COO, TEN
Atherinidae				
Brook Silversides *Labidesthes sicculus*		IN		TEN
Cottidae				
Mottled Sculpin *Cottus bairdi*		IN	BI	COO, TEN
Banded Sculpin *Cottus carolinae*		IN	BI	COO, TEN
Percichthyidae				
White Bass *Morone chrysops*		CR		COO**, TEN
Yellow Bass *Morone mississippiensis*		CR		TEN
Striped Bass *Morone saxatalis*		CR		COO
Centrarchidae				
Shadow Bass *Ambloplites ariommus*	INT	CR	SF	COO
Rock Bass *Ambloplites rupestris*	HWI	CR	SF	TEN
Redbreast Sunfish *Lepomis auritus*		IN	SF	COO**, TEN**
Green Sunfish *Lepomis cyanellus*		IN	SF	COO, TEN
Warmouth *Lepomis gulosus*		CR	SF	COO, TEN

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Bluegill *Lepomis macrochirus*		IN	SF	COO, TEN
Longear Sunfish *Lepomis megalotis*		IN	SF	COO, TEN
Redear Sunfish *Lepomis microlophus*		IN	SF	COO, TEN
Spotted Sunfish *Lepomis punctatus*		IN	SF	COO, TAL
Redeye Bass *Micropterus coosae*	HWI	CR	CENT	COO, TEN**
Smallmouth Bass *Micropterus dolomieu*	INT	CR	CENT	TEN
Spotted Bass *Micropterus punctulatus*		CR	CENT	COO, TEN
Largemouth Bass *Micropterus salmoides*		CR	CENT	COO, TEN
White Crappie *Pomoxis annularis*		CR	CENT	COO, TEN
Black Crappie *Pomoxis nigromaculatus*		CR	CENT	COO, TEN
Percidae				
Greenside Darter *Etheostoma blennioides*	HWI	IN	BI	TEN
Holiday Darter *Etheostoma brevirostrum*		IN	BI	COO
Rainbow Darter *Etheostoma caeruleum*	HWI	IN	BI	TEN
Coosa Darter *Etheostoma coosae*		IN	BI	COO
Coldwater Darter *Etheostoma ditrema*		IN	BI	COO
Black Darter *Etheostoma duryi*		IN	BI	TEN
Blueside Darter *Etheostoma jessiae*	INT	IN	BI	TEN

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Greenbreast Darter *Etheostoma jordani*	INT	IN	BI	COO
Stripetail Darter *Etheostoma kennicotti*		IN	BI	TEN
Redline Darter *Etheostoma rufilineatum*		IN	BI	TEN
Rock Darter *Etheostoma rupestre*		IN	BI	COO
Tennessee Snubnose Darter *Etheostoma simoterum*		IN	BI	TEN
Speckled Darter *Etheostoma stigmaeum*	INT	IN	BI	COO
Trispot Darter *Etheostoma trisella*		IN	BI	COO
Banded Darter *Etheostoma zonale*	INT	IN	BI	TEN
Amber Darter *Percina antesella*		IN	BI	COO
Goldline Darter *Percina aurolineata*		IN	BI	COO
Logperch *Percina caprodes*	INT	IN	BI	TEN
Conasauga Logperch *Percina jenkinsi*		IN	BI	COO
Freckled Darter *Percina lenticula*		IN	BI	COO
Mobile Logperch *Percina kathae*	INT	IN	BI	TAL
Dusky Darter *Percina sciera*		IN	BI	TEN
River Darter *Percina shumardi*		IN	BI	COO, TEN
Blackbanded Darter *Percina nigrofasciata*		IN	BI	COO

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Bronze Darter *Percina palmaris*	HWI	IN	BI	COO
Snail Darter *Percina tanasi*		IN	BI	TEN
Upland Bridled Darter *Percina* sp.		IN	BI	COO
Yellow perch *Perca flavescens*		CR		EXOTIC
Sauger *Stizostedion canadense*		CR		TEN
Walleye *Stizostedion vitreum*		CR		COO, TEN
Sciaenidae				
Freshwater Drum *Aplodinotus grunniens*		CR		COO, TEN

Water Quality Tolerance: **HWI** = headwater intolerant; **INT** = intolerant

Feeding Guild: **CR** = top carnivore; **GE** = generalist; **HB** = herbivore; **IC** = insectivorous cyprinid; **IN** = insectivore/invertivore; **PR** = parasitic

Species Category: **BI** = benthic insectivore species; **CENT** = centrarchid species; **RBS** = round-bodied sucker species; **SF** = sunfish species; **SMM** = subterminal mouth minnow species;

Drainage Basin: **COO** = Coosa; **TEN** = Tennessee

EXOTIC = species introduced to Georgia

****** = species introduced to that drainage basin