

# Methods for Assessing Biological Integrity of Surface Waters in Kentucky



**Commonwealth of Kentucky  
Natural Resources and Environmental Protection Cabinet  
Division of Water  
Water Quality Branch  
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**METHODS FOR ASSESSING BIOLOGICAL INTEGRITY  
OF SURFACE WATERS**

**Kentucky Department for Environmental Protection**

**Division of Water**

**Ecological Support Section**

**Frankfort, Kentucky**

**July 2002**

**This report has been approved for release:**

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**Jeffrey W. Pratt, Director**

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**Date**



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# 1 - INTRODUCTION

This manual has been developed by the Ecological Support Section of the Water Quality Branch (WQB) as guidance for the uniform collection, analysis, interpretation and quality assurance/quality control of biological samples from surface waters. The procedures defined herein are followed for all biological monitoring and assessment studies conducted by WQB and are recommended for use in assessments required by other Kentucky Division of Water (KDOW) programs. Compliance with these procedures is critical to basing sound assessments on waterbodies throughout the Commonwealth. Specific uses for these data are: (1) to determine legitimate waterbody uses as defined under Kentucky Water Quality Standards (KSWS) 401 KAR 5:031, (2) to determine effects of known point or nonpoint sources of pollution on the aquatic biota of the waterbody and (3) to determine background conditions within particular drainages or ecological regions.

*Biological integrity* is defined as "the condition of the aquatic community occurring in natural habitats of unimpaired surface waters as measured by community structure and function." *Biological assessments*, or bioassessments, are performed to evaluate the biological condition of surface water using biological surveys and other direct measurements of resident biota. The United States Environmental Protection Agency (U.S. EPA) document, "Biological Criteria: National program guidance for surface waters" (1990), has defined these and several other terms relating to biological assessments, and their definitions have been adopted by KDOW. Definitions for chapter-specific terms are provided in each chapter, where applicable.

The WQB integrates the collection and analysis of algal, macroinvertebrate, fish, habitat, bacteriological, and water chemistry data to arrive at conclusions on the health of Kentucky's surface waters. Because environmental stressors (e.g., excessive nutrients, organic wastes, industrial toxins and habitat alterations) reveal their impacts on the resident biota, biological communities leave detectable response signatures related to the various types of pollution. Therefore, environmental impacts and pollution abatement success can be directly measured with the standard ecological methods described in this manual.

As with any type of field sampling, safety precautions must be followed. Sampling during unsafe weather conditions (e.g., high water, electrical storms, extreme cold) should be avoided at all times. Field crews must consist of two or more people and all permanent employee crewmembers must be trained in First Aid and CPR. When using any field equipment (e.g. electrofishers, dredges, etc.), manufacturers' safety guidelines must be followed. Electrofishing units require extreme caution and all samplers must be trained in their use. To prevent injuries in the laboratory, safety procedures must be followed according to OSHA Standards.

If you have any questions or comments concerning this manual, please contact the Ecological Support Section at the following address:

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## **2 - STUDY PLANS AND STATION SELECTION**

### **I. STUDY PLAN**

The purpose of the study plan is to clearly state the objectives of the ecological survey, describe the study area, list known impacts to and uses of the waterbody, and list the types of data to be collected and measured. All available historical data are reviewed during design of the survey. The following sections are included in the study plan:

#### **A. Waterbody Data**

The following descriptive information is listed at the beginning of the study plan:

Stream name

Major river basin

Stream order (at mouth)

County or counties included in the survey

USGS 7.5-min. quadrangle names

#### **B. Survey Dates**

Tentative starting dates for the survey is listed on the study plan. For routine watershed monitoring (e.g., waterbody use-support classifications) appropriate sample index periods are stated in the algal, macroinvertebrate and fish chapters. These dates may not correspond to special situations (e.g., permitting activities and emergency spill or impact studies). Weather, streamflows or workload may affect actual starting dates. Surveys that are to be repeated seasonally are identified as such in the study plan.

#### **C. Objectives**

Biological surveys will have one or more stated objectives. Examples of objectives for biological surveys include, but are not limited to:

1. Determining legitimate waterbody uses as defined under Kentucky Surface Water Standards (KSWS) 401 KAR 5:031.
2. Determining effects of known point or nonpoint sources of pollution on the aquatic biota of the waterbody.
3. Determining background conditions within the drainage.

#### **D. Study Area Description**

The study area is described in detail and includes the following information:

1. Physical description of the study area, including topography, geology, physiographic region and ecoregion;
2. Stream length;

3. Drainage basin area;
4. Major tributaries;
5. Flow characteristics based on existing or extrapolated data;
6. Land use (agriculture, mining, silviculture, urban, etc.);
7. Location of known point or nonpoint sources of pollution.

#### **E. Parameter Coverage**

Parameter coverage varies depending on the objective(s) of the survey. Full coverage includes collection of habitat, biological, physicochemical and sediment samples. Biological samples include algal, macroinvertebrate and fish collections. Sampling of more than one taxonomic group encompasses more than one trophic level (primary producers and secondary and tertiary consumers) and provides a more realistic evaluation of the aquatic ecosystem. Full coverage may also include fish/shellfish contaminant analysis, bacteriological analysis and/or toxicity testing. Analytical chemistry parameters are selected based on the objectives of the study. At a minimum, hand-held multi-parameter meters that measure temperature, dissolved oxygen, pH and specific conductance should be employed on each sampling occasion. Habitat assessment (Chapter 6) is based on conditions observed at a site and the immediate area around the site at the time of the survey.

#### **F. Quality Assurance/Quality Control (QA/QC) Statement**

The QA/QC statement describes the procedures used to ensure the completeness and accuracy of all data. Procedures for field and laboratory QA/QC are described in detail in the study plan if they differ from the routine QA/QC procedures outlined in this manual. Quality Assurance/Quality Control procedures will be discussed in each chapter of this document, where applicable.

## **II. STATION SELECTION**

Stream sites are initially selected from 7.5 min. USGS topographical maps and/or Arcview GIS software. A reconnaissance visit, if necessary, is made to finalize the site selections. A sampling site should include habitats that are typical for the stream reach under study. Locally modified sites with channelized areas, impounded sections, etc., are avoided unless they are the focus of the study. In addition, sampling at or near the mouths of tributaries should be avoided if possible.

The Probabilistic (random) Sampling Program selects stations using a different method. The random survey approach is used to assess aquatic life use support for streams in each watershed management unit. For each basin management unit U.S. EPA in Corvallis, Oregon, is contacted to provide the population of streams to be assessed. This population consists of wadeable streams, orders 1<sup>st</sup>-4<sup>th</sup>. Fifty to 75 streams are typically assessed in each basin-year. All streams in the defined population are then randomly selected, along with the latitude and longitude of the exact location where the assessment is to be conducted. The stream population is weighted so the less numerous, higher order (3<sup>rd</sup> and 4<sup>th</sup>) streams will have an equivalent chance of being selected, based on percentages of each stream order in a given basin. Often these sample sites have no public access and landowner permission to gain access must be obtained.

## **A. Number and Location of Sampling Sites**

The number of sites selected for a survey often depends on the size of the drainage basin and the severity and number of impacts to the stream. However, available personnel and workload, funding or timeline may also limit the magnitude and scope of the study. All point sources, nonpoint sources and inputs from major tributaries are bracketed with an upstream site, a site at the mixing zone of each pollution source and intermediate sites. Control and reference sites (see below) and downstream recovery sites are also established. Downstream sites are selected between the point source and the next major tributary to avoid dilution effects. The number of downstream sites, and distance between these sites, depends on the objectives of the survey, the number of pollution sources, the number of major tributaries and the types of pollutants entering the stream. When the rate of movement of a contaminant or effluent plume is required, several samples are collected along a longitudinal gradient at regular intervals based on flow and travel time of the stream.

## **B. Control/Reference Sites**

Whenever possible, at least two control sites are sampled for comparative purposes. Control sites are located either upstream of the source(s) of pollution or, when this is not possible, on a nearby, unaffected tributary. Streams within the same drainage basin and ecoregion and with similar physical characteristics and habitats should be used. Control sites from different drainage basins should be used only if no suitable site can be located within the basin of the stream being surveyed. Fixed ecoregional reference sites (streams that are considered the most natural and undisturbed for the particular ecoregion) should be used where possible.

## **C. Tributary Sites**

When applicable, sites may be selected on each major tributary. Upstream and downstream sites to determine tributary and dilution effects should bracket the tributary.

## **D. Recovery Sites**

At least one site should be located far enough downstream of the polluted area to detect recovery of the aquatic biota (i.e., when the biota begins to show a similarity to the control/reference site). Location of this site depends on the magnitude and downstream extent of the pollution.

## **E. Site Numbering**

Stations are numbered consecutively from the mouth of the stream to the headwaters in a hierarchical method. The station nearest the mouth is assigned the lowest number not previously used. Sites are then numbered consecutively from mouth to headwaters. Next, tributary sites are assigned numbers, starting with the tributary closest to the mouth of the stream being surveyed. All stations on that tributary are numbered consecutively upstream from the confluence. Then, the next tributary is numbered until all sites have been assigned numbers. If additional sites are added after the initial study, they must be assigned numbers that have not been previously used, following the above outlined convention as closely as possible.

## 3 - ECOREGIONS AND THE REFERENCE CONDITION

### I. INTRODUCTION TO ECOREGIONS

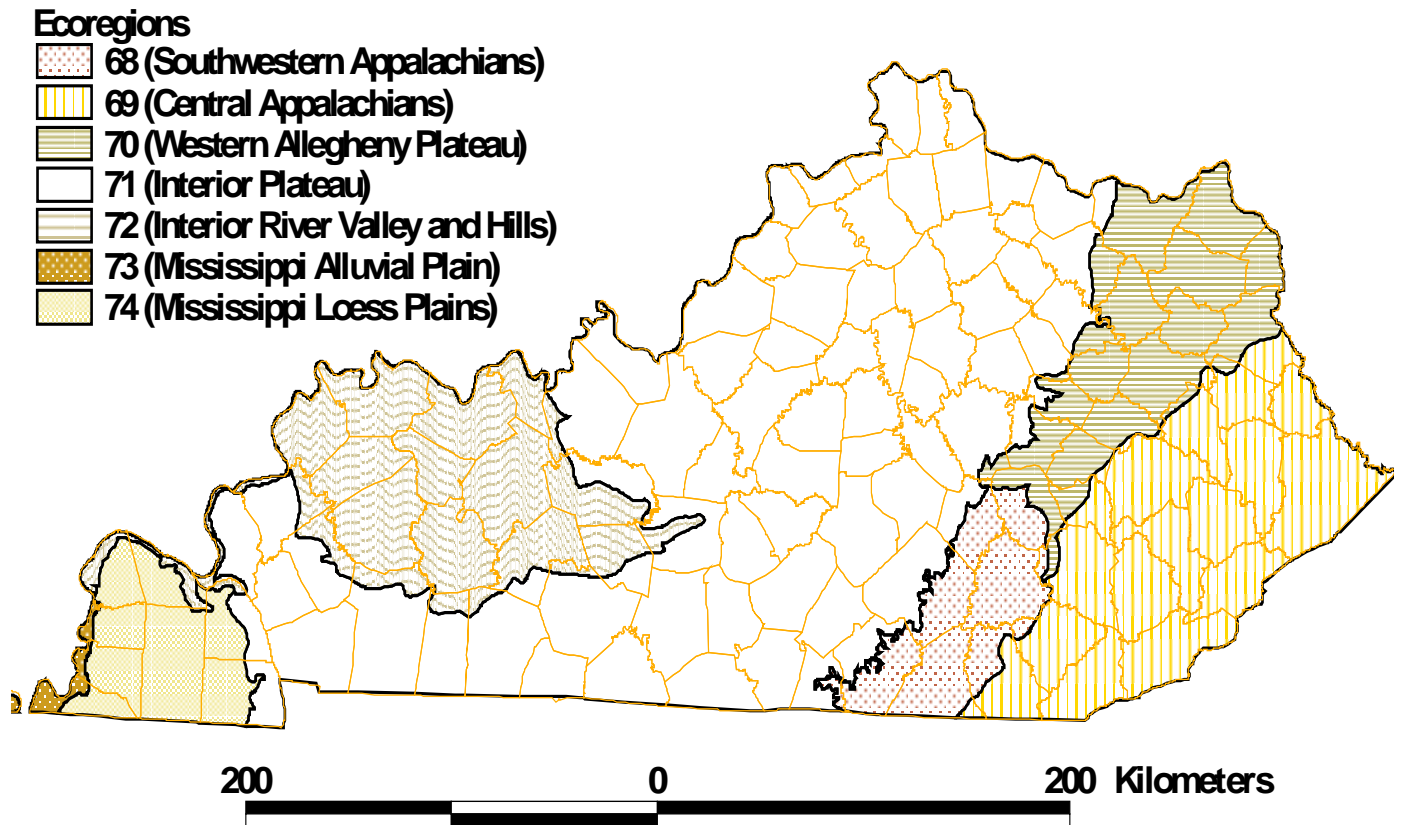
The concept of ecological regions, or ecoregions, within physiographic regions is a recent postulation that has received considerable attention. The conceptual framework of aquatic ecoregions is based on definable similarities among streams in a region that are related to the terrestrial characteristics of that region. In effect, streams acquire their characteristics from their watersheds (Likens and Bormann 1974, Hynes 1975). Streams draining watersheds of the same size, with comparable land uses and habitats, and in the same region are more likely to contain similar aquatic communities than those draining watersheds of a different size or in a different region (Hughes et al. 1986). This regional framework facilitates water quality resource management and the development of region-specific biological and chemical criteria.

Omernik (1987) mapped the Level III terrestrial ecoregions of the conterminous United States based on physiography, soil types, potential natural vegetation, geology and land-use types. A revised version of the Level III ecoregions in Kentucky is shown in Figure 3-1. Analysis of the data revealed regional homogeneities, which allowed for the delineation of boundaries between ecoregions and the location of the most typical areas within each ecoregion. Each of the delineated Level III ecoregions is descriptively compared below to adjacent ecoregions (modified after Woods et al. 2002). Level IV subecoregions have been delineated and a map and descriptions are available in Woods et al. (2002).

#### **Ecoregion 68: SOUTHWESTERN APPALACHIANS**

This region has a mixture of open, low mountains containing a mosaic of forest and woodland with some cropland and pasture. A deeply incised escarpment occurs in the west near the boundary with the Interior Plateau's Eastern Highland Rim (Ecoregion 71). The landscape is underlain by Pennsylvanian and Mississippian rock strata. Ultisols and Inceptisols (soil types) are common and contrast with the Alfisols that dominate the lowlands of the Interior Plateau to the west. Mixed mesophytic forest is generally restricted to the deeper ravines and escarpment slopes, and mixed oaks with shortleaf pine dominate the upland forests. Resource extraction (oil, gas and coal), agriculture and silviculture are the major land uses. Moderate- to high-gradient streams are common and generally have cobble- or boulder-dominated substrates; a few low-gradient streams occur and have gravelly or sandy bottoms. Nutrient and alkalinity levels are typically low compared to adjacent Ecoregion 71. Fish and mussel distributions in Cumberland River tributaries are distinct from the tributaries of the Kentucky River. They are also distinct from areas above Cumberland Falls in Ecoregion 69.

# Figure 3-1. Level III Ecoregions of Kentucky



## **Ecoregion 69: CENTRAL APPLACHIANS**

Ecoregion 69 is a high, dissected and rugged plateau made up of sandstone, shale, conglomerate and coal of Pennsylvanian age. The plateau is locally punctuated by a few anticlinal ridges. Its rugged terrain, cool climate and nutrient-poor soils sharply limit agricultural potential and result in a mostly forested land cover. The high hills and low mountains are mostly covered by mixed mesophytic forest. Bituminous coal mines are common and have caused the siltation and acidification of streams. Soils have developed from residuum and are mostly Ultisols and Inceptisols; they contrast with the Alfisols that dominate the lowlands of the Interior Plateau (Ecoregion 71) to the west. Its western boundary with Ecoregion 70 occurs at the elevation and forest density break; the more densely forested Ecoregion 69 is higher, cooler, and steeper than the Western Allegheny Plateau (Ecoregion 70) and is underlain by more resistant rock. The region is drained by relatively high gradient streams that make up the headwaters of the Kentucky, Cumberland, Licking and Big Sandy river basins. These streams are typically cool, with substrates composed mainly of cobbles and boulders.

## **Ecoregion 70: WESTERN ALLEGHENY PLATEAU**

The hilly and wooded terrain of Ecoregion 70 was not muted by glaciation. It is more rugged than the agricultural till plains of Ecoregions 55 and 61 in Ohio, but is less rugged than the Central Appalachians (Ecoregion 69). Extensive mixed mesophytic forests originally grew in Ecoregion 70, and contrast with the oak-hickory forest that is found farther west in the lower Interior Plateau (Ecoregion 71). Today, most of the rounded hills of Ecoregion 70 remain in forest; dairy, livestock and general farms, as well as residential developments, are concentrated in the valleys. Resource extraction (oil, gas and coal), silviculture and agriculture remain the major land uses within this ecoregion. Horizontally bedded, Pennsylvanian sedimentary rock containing sandstone, siltstone, shales and coal underlies the region. Some areas have eroded down to limestone and may have localized karst development. Moderate to high-gradient headwater streams of the Kentucky, Licking, Little Sandy and Ohio river basins originate within the Western Allegheny Ecoregion. Upland streams are generally cool with cobble-boulder substrates similar to Ecoregion 69. Nutrient and alkalinity levels are generally higher than the Central Appalachians (Ecoregion 69) but lower than the Interior Plateau (Ecoregion 71).

## **Ecoregion 71: Interior Plateau**

The Interior Plateau Ecoregion is the largest ecoregion in area encompassing most of the Highland Rim and Blue Grass Sections of the Interior Low Plateau Physiographic Province. Ecoregion 71 is composed of irregular plains, open hills, knobs and large areas of karst topography. Its landforms and soils are the result of the differential weathering of sedimentary strata, stream erosion and deposition, and the solvent action of subterranean water on carbonates. Ecoregion 71 is underlain by Mississippian-through Ordovician-age limestone, chert, sandstone, siltstone and shale. Lithologic and related physiographic boundaries tend to determine the natural subdivisions of the Interior Plateau (71). In areas underlain by cavernous limestone, drainage is primarily underground and only a few entrenched master streams and rivers occur. Rock types are distinct from the unconsolidated coastal plain sands that underlie the Mississippi Valley Loess Plains (74) in western Kentucky and the Pennsylvanian coal measures found in Ecoregions 69, 70 and 72. Maximum elevations and local relief are lower than in the Appalachian and Cumberland Plateau ecoregions to the east (68, 69 and 70) but are greater than in the Interior River Valley and Hills (72). The soils of Ecoregion 71 are highly varied and developed from the underlying sandstone, siltstone, shale and limestone and are not from glacial till like those of the Eastern Corn Belt Plains (55) to the north. Lowlands are generally dominated by Alfisols and not Ultisols or Inceptisols that are more characteristic of Ecoregions 68, 69 and 70. The natural vegetation is primarily oak-hickory forest, but large areas of bluestem prairie originally occurred in the western karst areas. The potential natural vegetation is distinct from the mixed mesophytic forests of higher, cooler, wetter Ecoregions 68, 69 and 70. Rolling pastures, crop fields and woodlots are common, and agriculture, urban expansion and construction are major land uses. Most of the streams in the Interior Plateau Ecoregion are heavily influenced by human activities. Parts of the Licking, Kentucky, Cumberland, Salt, Green, Tradewater and Ohio river basins are located within the ecoregion. Stream morphology is highly variable across this large ecoregion. Both moderate-to high-gradient streams with boulder-cobble substrates and low-gradient streams with sand-gravel bottoms occur. Cool groundwater-fed streams punctuate those areas underlain by extensive and well-developed karst geology. Stream nutrient, alkalinity and hardness levels are higher in most parts of Ecoregion 71 than in Ecoregions 68, 69 and 70.

## **Ecoregion 72. Interior River Valleys and Hills**

This broad, undulating lowland was formed in nonresistant, noncalcareous sedimentary rock of Pennsylvanian age. Often referred to as the "Western Coalfields," it is rich in coal reserves. Large upland areas are veneered by windblown material. Many wide, flat-bottomed, terraced valleys occur and are filled with alluvium, loess and lacustrine deposits. Bottomland hardwood forests and swamp forests once grew on poorly drained, nearly level sites, whereas the upland areas had oak-hickory forests. Patterns of land use are more varied than in the neighboring ecoregions, and large areas have been strip mined for coal. Drained alluvial soils are farmed for feed grains and soybeans. Undrained valleys are used for forage crops, pasture or woodlots; uplands are used for mixed farming and livestock. Extensive strip mining, as well as crop and livestock production, have impacted stream water quality (acidification, sedimentation, nutrients) and stream habitat (channelization, sedimentation); sheet erosion can be severe on cultivated slopes. Streams feed the Tradewater, Green and Ohio river basins and have relatively low (<5 ft/mile) gradients. Unchannelized upland streams often have gravel bottoms, while sand, silt and mud dominate most lowland channels. Streams typically have lower nutrient, alkalinity and hardness levels than those in Ecoregion 71 do. Wetlands were once common in this region, but many have since been drained for agricultural uses.

## **Ecoregion 73: Mississippi Alluvial Plain**

The Mississippi Alluvial Plain is the smallest ecoregion found in Kentucky, occupying the areas immediately adjacent to the Mississippi River. Rock stratum is almost exclusively composed of alluvial deposits. Mostly flat, broad floodplains with river terraces and levees provide the main elements of relief. Abandoned channels, oxbow lakes, bayous, backswamps and point bars are found in the ecoregion. Fine-grained, poorly drained soils are common, but better drained loamy and sandy soils also occur. Winters are milder and summers are hotter than other Kentucky ecoregions. Bottomland deciduous forest vegetation covered the region before clearance for cultivation. All of the streams within the area drain into the Mississippi River and have been extensively channelized for agriculture. Streams are very low-gradient and have sandy to muddy substrates and often contain extensive wetland vegetation.

## **Ecoregion 74: Mississippi Valley Loess Plains**

Ecoregion 74 in far western Kentucky consists of irregular plains, gently rolling hills and, near the Mississippi River, bluffs. It is mostly covered by thick loess and alluvium and underlain by Cretaceous and Tertiary coastal plain sediments. Rock types are distinctly different from the limestone, chert, sandstone, siltstone and shale of the Interior Plateau (71). Ecoregion 74 has less relief than ecoregions farther to the east in Kentucky, and elevations are much lower than in the Appalachian ecoregions. The potential natural vegetation is oak-hickory forest (Kuchler 1966), but agriculture is the dominant land use in Ecoregion 74 in Kentucky. Typically, streams have moderate to low gradients and gravelly to sandy bottoms. Wetlands are common throughout the area. In natural streams, nutrient and alkalinity levels are generally low compared to those of Ecoregion 71.

## II. DEFINING ECOREGIONAL REFERENCE CONDITIONS

To address levels of impact on any given stream, a firm understanding of the inherent biological variability and potential of natural streams in a collective region is necessary. This is accomplished using a regional reference approach (Hughes et al. 1986) which is based on the range of conditions found in a population of sites or streams with similar physical characteristics and minimal human impact. Reference sites are tightly associated with ecoregions and an effort is made to obtain good coverage of sites within all regions. Collectively, the reference condition refers to the range of quantifiable ecological elements (chemistry, habitat and biology) that are found in natural environments. In many regions of Kentucky, finding reference streams can be a difficult task, as few regions are without areas of human disturbance. Therefore, reference reaches are more appropriately deemed "least-disturbed." The application of the reference condition involves its comparison to a stream exposed to environmental stress using defined sampling methodology and assessment criteria. Impairment of the test site would be detected if indicator measurements (e.g., species richness, habitat rating, nutrients) fell outside the range of threshold criteria established by the reference condition.

### 1. Selection of Candidate Reference Reach Waterbodies

Ecoregional reference reach site selection and evaluation analysis is an ongoing process supported by intensive map and ground reconnaissance. The U.S. Environmental Protection Agency's (EPA) Biocriteria Program suggests that the selection process for candidate reference reach waterbodies should be well documented so that the data defining the reference condition will be scientifically defensible.

In order to comply with U.S. EPA guidelines, the Reference Reach Program follows a step-by-step process for the selection of candidate reference reach waterbodies. This process involves the analysis of topographic maps and aerial photography, cross-referencing with other available data sources and field reconnaissance of the candidate watersheds. Because of the diverse land use, topography and physiography across the state, fixed statewide reference criteria are not used. Therefore, reference reach candidates are qualified in a regional context in an effort to find "least-disturbed" conditions among the various ecoregions.

U.S. Geological Survey (USGS) 7.5-min. topographic maps and aerial Digital Orthogonal Quadrangles (DOQs) lying within the boundaries of the Commonwealth of Kentucky are analyzed as part of the initial step of the selection process. Each stream is then evaluated based upon the presence or absence of the following:

- 1) High proportion of forestland and riparian zone quality;
- 2) Towns or communities along the streambank;
- 3) Resource extraction in the watershed (e.g., coal mines, oil wells, gas wells);
- 4) Hydrologic modification in the watershed (e.g., impoundment, channelization);
- 5) Major sewage treatment/industrial discharges.

If an adequate riparian zone exists along most of the stream length and minimal land-use activities occur within the watershed, then the stream is recorded as an initial candidate. An initial candidates' list is then compiled for each county and Level III ecoregion. Once the initial



candidates' list is created, it is cross-referenced with other available data sources including the following:

- 1) Kentucky Rivers Assessment (KDOW and NPS 1992);
- 2) Kentucky Nonpoint Source Assessment Report (KDOW 1999);
- 3) Kentucky Nature Preserves Commission's Fish Collection Catalogue (Warren et al. 1983);
- 4) Kentucky Division of Water's Fish Collection Catalogue (Mills 1988);
- 5) Aquatic Biota and Water Quality Survey of the Appalachian Province, Vol. 1-3 (Harker et al. 1979);
- 6) Aquatic Biota and Water Quality Survey of the Upper Cumberland River Basin, Vol. 1-2 (Harker et al. 1980);
- 7) Aquatic Biota and Water Quality Survey of the Western Kentucky Coal Field, Vol. 1-2 (Harker et al. 1981);
- 8) Recommendations for Kentucky's Outstanding Resource Water Classifications with Water Quality Criteria for Protection (KNPC 1982);
- 9) Kentucky Division of Water Intensive Survey Reports (KDOW); and
- 10) Personal communication with Kentucky Division of Water, Kentucky Nature Preserves Commission, Kentucky Department of Fish and Wildlife Resources, and Daniel Boone National Forest personnel, where applicable.

Cross-referencing allows for amendment of the initial candidates' list resulting in a smaller, secondary list. Upon completion of cross-referencing, field reconnaissance of the watersheds is conducted. Each site is evaluated on categories and criteria shown in Table 3-1.

**Table 3-1. Summary of physical criteria used in the Reference Reach selection process.**

Category	Criterion
1) riparian zone condition	well-developed providing some canopy over the stream; presence of adequate aquatic habitats in the form of root mats, coarse woody debris and other allochthonous material
2) bank stability	at least moderately stable with only a few erodible areas within the sampling station
3) degree of sedimentation	the substrate is 25 percent or less embedded by fine sediment
4) suspended material	the water is relatively free from suspended solids during normal weather conditions
5) evidence of nutrient enrichment	the substrate is relatively free from extensive algal mats that could choke riffle habitats
6) conductivity	conductivity is not highly elevated above what naturally occurs (region-specific)
7) aquatic habitat availability	there is a 50 percent or greater mix of rubble, gravel, boulders, submerged logs, root mats, aquatic vegetation or other stable habitats available for aquatic organisms
8) the presence or absence of trash in the stream	solid waste within the stream and on the streambank is at a minimum
9) evidence of new land-use activities in the watershed	the land-use conditions remain constant from what is depicted on the most recent USGS topographic or DOQ maps
10) accessibility of the site for collection	Accessible

Note: the reference criteria listed above may vary somewhat among ecoregions.

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# 4 - SITE CHARACTERIZATION

## I. INTRODUCTION

Assessing the quality of an area to be sampled is an integral part of any aquatic survey. In intensive impact surveys, every attempt is made to sample waterbodies, such as streams and wetlands, with comparable habitat types. Qualitative documentation of habitat quality is recorded on field data sheets. A more quantitative approach to determine the habitat conditions of a sampling site is the Habitat Assessment procedures found in Chapter 6.

## II. FIELD DATA SHEET

Field observations of site conditions and the habitat assessment are recorded on a field data sheet (FDS), (Appendices A-1 and A-2, depending on high/moderate or low-gradient stream classification). This type of habitat analysis allows the investigator to quickly check off or record observed habitat conditions. Procedures for scoring habitat features with the EPA Rapid Bioassessment Protocol (RBP) Habitat Assessment are presented in Chapter 6.

### A. General Information

Some of the information included on the FDS is gathered in advance, using data from maps and prior reconnaissance visits.

#### 1. Map Information

The following FDS information is gathered from maps, primarily using USGS 7.5 minute topographical maps and county maps: basin, stream name, location (e.g. highway no., bridge, nearby town), county, latitude and longitude, map name or KDOW map number.

#### 2. On-Site Information

The following general information is recorded on the FDS onsite, prior to biological sampling: weather (e.g. hot, cold, rainy, dry, sunny, etc.), time (beginning) and sample type (e.g., biological, bacteriological, physicochemical, sediment, tissue).

##### a. Water Quality Observations

A portable, multi-parameter water quality meter that measures, at a minimum, pH, dissolved oxygen, conductivity and water temperature should be used on each sampling occasion and recorded on the FDS. In addition, note any water odors, water surface oils and water clarity/turbidity observations.

### B. Stream Substrate Quality and Composition

#### 1. Substrate Composition

In general, variations in particle size and type are reflected in flowing bodies of water by gradation of habitat types from stream headwaters to mouth. Each longitudinal gradation in substrate type harbors a characteristic biotic community. The absence of characteristic

community members in the presence of a favorable substrate type can be a useful indication of stream disturbance. For visual estimates of substrate size, a transect is surveyed in a pool (mid-pool) and riffle (mid-riffle) to estimate the substrate by percent particle size and type of material. Results are expressed as percent of total. Additionally, a Wohlman Pebble Count (or equivalent) can be conducted following procedures found in (Harrelson et al. 1994, Wolman 1965). Sample particles are measured against the particle size chart (Table 4-1) to provide the investigator with a fixed concept of category size. In deep waters, particle size may be determined from dredge grab samples. Results are recorded on the FDS. In addition, record the estimated percent riffle, run, and pool within the sampling reach. References: (Cummins 1962, Platts et al. 1983, Wentworth 1922, Winger 1981)

<b>Table 4-1: Substrate Particle Size Chart</b>	
<b>Categories</b>	<b>Size (mm)</b>
Boulders	>256 (= 10 in)
Cobble	64 - 256 (= 2.5 - 10 in)
Pebble	16 - 64 (= .63 - 2.5 in)
Gravel	2 - 16 (= .08 - .63 in)
Fines	<2 (= .08 in)
Exposed Bedrock	--
Hardpan Clay	--
Detritus	--

### C. Stream Physical Features

#### 1. Stream Flow Condition

The condition of the stream is evaluated, and one of the following assessments is recorded on the FDS: perennial, intermittent or interrupted. In addition, record the gradient of the stream as either high, medium or low.

#### 2. Stream Discharge/Velocity

The stage of the stream is estimated, and one of the following assessments is recorded on the FDS: dry, no flow (pooled), low, normal, high or flooded. Velocities are measured with an acceptable flow meter or with neutral-buoyant objects (e.g., oranges, small sticks, small sponge rubber balls) in areas of laminar flow and along a uniform transect of the channel. Reference: (Lazorchak, Klemm, and Peck 1998)

#### 3. Channel Morphology

##### a. Stream-Depth Range

The stream-depth range is estimated for the entire reach and recorded on the FDS.

##### b. Stream-Width Range

The stream-width range is estimated for the entire reach and recorded on the FDS.

#### **4. Canopy**

An exposed stream often experiences increased water temperatures that may be directly or indirectly limiting to some organisms and may be favorable for nuisance algal blooms and decreased dissolved oxygen. Light intensity may be limiting to some organisms and favorable to others. A partially shaded stream generally achieves the greatest diversity. In wadeable streams, sufficient shade to maintain temperatures and habitats that will support indigenous organisms is generally created by a 50% to 75% tree canopy. Natural headwater streams should generally have 75% to 100% tree canopy.

The percent canopy shading the stream is recorded on the FDS using these categories.

Fully Exposed	(0 - 25%)
Partially Exposed	(25% - 50%)
Partially Shaded	(50% - 75%)
Fully Shaded	(75% - 100%)

References: (Hawkins et al. 1982, U.S. EPA 1982, Pfankuch 1978, Karr and Schlosser 1977, Winger 1981, Platts et al. 1983)

#### **5. Channel Alterations**

Many activities that alter the stream channel require water quality certification by KDOW and Section 404 permits from the U.S. Army Corps of Engineers. Some of these activities include: dredging, channelization, clear and snag, bridge construction and artificial bank stabilization. The occurrence of any channel-altering activities at the site is recorded in the FDS.

References: (Simpson et al. 1982, Johnson and McCormick 1978, Hewlett 1978, Newbold et al. 1980, Burns and Hewlett 1983, Platts 1978, 1981a, 1981b and 1982, Platts et al. 1983).

#### **6. Hydraulic Structures**

Hydraulic structures include any natural obstructions or human-made devices that impede or deflect the course of water from its original pathway. The presence and type of hydraulic structures are noted on the FDS. Some typical examples are dams, bridge abutments, islands, gravel and mud bars and any others to be listed individually.

#### **7. Watershed Features/Land Use**

All land uses occurring within the vicinity of the sampling site are recorded on the FDS. Examples of land use include silviculture, agriculture, oil exploration, construction, mining, urbanization, onsite wastewater treatment (e.g. "straight pipes"), etc. In addition, note the intensity of localized watershed erosion as heavy, moderate or none.

#### **8. Pollution Types**

The presence and proximity of point source discharges, such as municipal, private or industrial wastewater treatment plants (WWTP), should be noted on the FDS. In addition, record any localized nonpoint source impacts near or upstream of the sampling location.

## D. Riparian Vegetation

Indicate the dominant trees, shrubs and herbaceous plants in the riparian zone. Because of its stabilizing effects and its ability to influence water temperatures, a riparian zone of 18 meters or more is preferred. The width of the riparian zone is scored in the RBP Habitat Assessment (Chapter 6). In addition, count the number of canopy strata present in the riparian zone as an indication of riparian age and quality (e.g., overstory, understory, herb layer).

References: (Hawkins et al. 1982, Schlosser and Karr 1981a and 1981b, Hewlett 1978, Karr and Schlosser 1977 and 1978, Johnson and McCormick 1978, Karr et al. 1981, Platts et al. 1983)

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# 5 - BACTERIOLOGICAL EVALUATION

## I. INTRODUCTION

Bacteriological analysis includes, but is not necessarily limited to, the collection and analysis of *E. coli* and fecal coliform bacteria; i.e., bacteria that are commonly found in the intestinal tracts of warm-blooded animals.

## II. BACTERIOLOGICAL SAMPLE COLLECTION

- A. Samples are collected from directly below the water surface in sterile 250 ml Nalgene bottles, 125 ml borosilicate glass bottles containing sodium thiosulfate to counteract the presence of chlorine, or Corning sterile disposable 120 ml coliform water sample containers (1700-100), labeled, placed on wet ice and analyzed within eight hours of collection, preferably within six hours.
- B. Stream samples are collected by using the surface grab technique and should be free of debris and bottom sediment. Grasp the bottle at the base with one hand and plunge the bottle, mouth down, into the water to avoid introducing surface scum. Position the mouth of the bottle upstream toward current. Collect the sample from a depth of 6-12 inches. Allow an air space for mixing in each sample (approximately 25%). In a 250 ml capacity sample bottle, collect approximately 200 ml. If more than one sample is taken, identify each sample with a number (i.e., 1, 2, 3) in chronological order of collection. This order should be repeated on the chain-of-custody sheet. If direct stream sampling is not practical, an alternative sampling device may be used (i.e., rod and reel, as long as sterile sampling bottles are used at each sample station).
- C. A grab sample is obtained using a sterile sample bottle of 250 ml capacity if more than one bacterial parameter is to be analyzed. Identify the sampling site on a chain-of-custody tag and/or sheet and on a field log sheet, including location, time, date and the name of the actual sample collector.
- D. Transport samples on wet ice in a cooler capable of holding a temperature of 1°–4°C.
- E. Holding times for bacteriological samples should not be exceeded.
- F. Note: Because of the reproduction potential of the bacteria, samples should be iced and analyzed as soon as possible, thus reducing the degree of error in the final result. Sewage samples and organically rich wastes are particularly susceptible to rapid increases or die-away; therefore, they should be held for the shortest time possible to minimize change.
  - 1. If iced, samples must be delivered to the laboratory within six hours.
  - 2. If not iced, samples must be delivered within one hour of collection, and processing must be initiated immediately upon delivery to avoid unpredictable changes. The time of collection and time of delivery to the laboratory must be documented, and proper chain-of-custody procedures followed.



Samples collected from a chlorinated source are dechlorinated by adding 0.1 ml of a 10% sodium thiosulfate solution per 125 ml of sample. This concentration will neutralize approximately 15 mg/l of residual chlorine.

### III. SAMPLE HOLDING TIMES

- A. A maximum time of eight hours may elapse from time of collection of the sample to completion of sample processing. Notify the laboratory at least 24 hours prior to sample collection, unless an emergency condition exists.
  
- B. Once the samples have been processed, they must be resuscitated for two hours at 35 degrees Centigrade and incubated for 22 hours for *E. coli* (analyses by MTEC media procedure) or 24 ( $\pm 2$ ) hours at 44.5 degrees Centigrade for fecal coliform analyses. IDEXX procedure may take 48 hours. This means that samples delivered to the laboratory on Thursday or Friday require members of the staff to work Saturday or Sunday. Therefore, no samples will be accepted on Friday except in an emergency. No samples will be analyzed after normal working hours without prior approval. The laboratory must have time to prepare media, glassware and buffer solution for unexpected samples. The laboratory should be notified as soon as possible so that preparation can begin and conflicts can be avoided.

### IV. GENERAL LABORATORY PRACTICE

The following analyses are performed in the bacteriological laboratory:

Total coliform analysis

*E. coli* analysis

Fecal coliform analysis

1. Total coliform analysis will be performed using IDEXX techniques and instrumentation (IDEXX Laboratories Inc., Westbrook, ME.)
  
2. *E. coli* procedures using membrane filter analysis will be performed as described in the latest edition of Standard Methods for the Examination of Water and Wastewater (Standard Methods).
  
3. Fecal coliform procedures using membrane filter analysis will be performed as described in the latest edition of Standard Methods.
  
4. Fecal coliform analyses for chlorinated effluents by membrane filter will be performed as described in the latest edition of Standard Methods.
  
5. For the purpose of resolving any controversy over results of membrane filter analysis, fecal coliform analyses will be performed, as described by the latest edition of Standard Methods, by the Most-Probable-Number Method.
  
6. A laboratory bench sheet provided in Appendix B-1 should be used to record findings.

References: APHA (1992), Weber (1973), U.S. EPA (1973)

## V. INTERPRETATION OF BACTERIOLOGICAL ANALYSES

### A. Total Coliform Analysis

This analysis will be performed during *E. coli* analysis with IDEXX (i.e., Quanti-Tray®/2000). It will provide information on water quality, but will not be used as a standard indicator. Total coliform analysis will be most beneficial in well water testing.

### B. *E. coli* Analysis

Instream *E. coli* levels to determine water quality for primary contact recreational uses will be compared to maximum allowable limits established by U.S. EPA. Currently a single sample maximum of 235 per 100 ml is considered acceptable. *E. coli* testing by standard methodology (i.e., MTEC procedure) or IDEXX will be acceptable.

*E. coli* levels are used for:

- 1) primary contact recreation use determination
- 2) well water testing

### C. Fecal Coliform Analysis

Instream fecal coliform levels will be compared to maximum allowable limits established in 401 KAR 5:031, Surface Water Standards.

Fecal coliform levels are used for:

- 1) primary contact recreation use determination
- 2) secondary contact recreation use determination
- 3) domestic water supply use determination
- 4) point/nonpoint assessment

## VI. QUALITY ASSURANCE/QUALITY CONTROL

**A. Duplication.** Each laboratory must establish quality control over the microbiological analysis in use. Run duplicate analyses on one of every ten samples analyzed (10%) and a minimum of one duplicate analyses per month. The duplicates may be run as split samples by more than one analyst. At least once per month, two or more analysts should count the colonies on the same membranes containing 20 – 60 colonies.

**B. Verification.** Five percent of the samples measured should be verified as fecal coliform bacteria. Verify ten blue colonies as fecal coliform bacteria on one of every twenty samples. Lauryl tryptose broth and EC broth in 10 mL test tubes with gas fermentation vials made of borosilicate glass should be used for the analyses. Each analyst should verify ten colonies as fecal coliform bacteria a minimum of once per month.

**C. Water Supply.** Once per year a sample supply should be delivered and analyzed by the Division of Environmental Services for the presence of total organic carbon, ammonia nitrogen, cadmium, chromium, copper, lead, nickel, and zinc. Once per year test 100 ml of the water supply for the presence of bacteria.

**D. Performance samples.** Laboratories should analyze at least one unknown performance sample per year when available, for the parameters measured.

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# 6 - HABITAT ASSESSMENT

## I. INTRODUCTION

A habitat assessment should be conducted at every biological sampling reach. Such an assessment will allow investigators to evaluate the quality of instream and riparian habitat. The availability of quality habitat directly influences the biological integrity of the stream reach. Information obtained from the habitat assessment can be used to supplement biological and physicochemical data when determining the overall health of the stream reach and stream-use designation. Additionally, habitat assessments can be used to document physical changes that occur at a sampling reach over time. In multi-agency monitoring projects (such as watershed monitoring), habitat assessments provide continuity and consistency between all entities involved in the monitoring effort. Habitat assessment procedures follow those outlined in *Rapid Bioassessment Protocols for Use in Wadeable Streams and Rivers* (Barbour et al. 1999).

## II. ASSESSMENT PROCEDURES

Investigators should conduct a visual-based habitat evaluation of the stream reach by filling out the appropriate Habitat Assessment Field Data Sheet (Barbour et al. 1999). In streams where riffles should naturally be present (e.g., most stream reaches of the Central Appalachian, Western Allegheny, Southwestern Appalachian and Interior Plateau ecoregions would qualify), the High-Gradient Habitat Assessment Field Data Sheet should be used (Appendix A-1). In low-gradient streams where rocky riffles are not naturally present (e.g., most stream reaches in the Mississippi Valley Loess Plain and the Interior River Lowland ecoregions would qualify), the Low-Gradient Habitat Assessment Field Data Sheet should be used (Appendix A-2).

The visual-based habitat evaluation consists of ten parameters that rate instream habitat, channel morphology, bank stability and riparian vegetation for each sampling reach. A numerical scale of 0 (lowest) to 20 (highest) is used to rate each parameter (Barbour et al. 1999). For each parameter, the investigators will determine which of the following conditions exist at the sampling reach: Optimal, Suboptimal, Marginal or Poor. A parameter score will then be given within the condition category chosen above: Optimal (20-16), Suboptimal (15-11), Marginal (10-6) or Poor (5-0). The investigators will total all of the parameter ratings to obtain a final habitat ranking (Barbour et al. 1999).

## III. PARAMETERS FOR HABITAT ASSESSMENT

- i. These parameters should be evaluated within the sampling reach. All of the areas within the reach should be evaluated together as a composite.**

### **Parameter #1**

**Epifaunal Substrate/Available Cover (Both High and Low Gradient Sheets)**- this metric measures the relative quantity and the variety of structures: cobble, boulders, fallen trees, logs, branches, root mats, undercut banks, aquatic vegetation, etc., that provide refugia, feeding opportunities and sites for spawning and nursery functions.

1. Optimal (High Gradient): >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 not new fall and not transient) (20-16)

Optimal (Low Gradient): >50% of substrate favorable for epifaunal colonization and fish cover, mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at a stage to allow full colonization potential (i.e., logs/snags that are not new fall and not transient) (20-16)

2. Suboptimal (High Gradient): 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 new fall, but not yet prepared for colonization (may rate at the high end of the scale) (15-11)

Suboptimal (Low Gradient): 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 new fall, but not yet prepared for colonization (may rate at high end of the scale) (15-11)

3. Marginal (High Gradient): 20%-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed (10-6)

Marginal (Low Gradient): 10%-30% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed (10-6)

4. Poor (High Gradient): <20% stable habitat; lack of habitat is obvious; substrate unstable or lacking (5-0)

Poor (Low Gradient): <10% stable habitat; lack of habitat is obvious; substrate unstable or lacking (5-0)

## **Parameter #2**

1. **Embeddedness - (High Gradient Sheet)** - the extent to which rocks and snags are covered or sunken into the silt, sand, mud or biofilms (algal, fungal or bacterial mats) of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish (for shelter, spawning and egg incubation) is decreased; assess in the upstream or central portions of riffles.

- a. Optimal: Rocks are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space (20-16)

- b. Suboptimal: Rocks are 25%-50% surrounded by fine sediment (15-11)

- c. Marginal: Rocks are 50%-75% surrounded by fine sediment (10-6)

- d. Poor: Rocks are >75% surrounded by fine sediment (5-0)

2. **Pool Substrate Characterization** - (**Low Gradient Sheet**) - evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (e.g., gravel and sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.

- a. Optimal: Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common (20-16)
- b. Suboptimal: Mixture of soft sand, mud or clay; mud may be dominant; some root mats and submerged vegetation present (15-11)
- c. Marginal: All mud or clay or sand bottom; little or no root mat; no submerged vegetation (10-6)
- d. Poor: Hard-pan clay or bedrock; no root mat or vegetation (5-0)

### Parameter #3

1. **Velocity/Depth Regime** - (**High Gradient Sheet**) - the best streams in most high-gradient regions will have all of the following patterns of velocity and depth: 1) slow-deep, 2) slow-shallow, 3) fast-deep, and 4) fast-shallow; the occurrence of these four patterns relates to the stream's ability to provide and maintain a stable aquatic environment. Investigators may have to scale deep and shallow depending upon the stream size; a general guideline is 0.5 m between shallow and deep.

- a. Optimal: All 4 regimes present (20-16)
- b. Suboptimal: Only 3 of the 4 regimes present; if fast-shallow is missing, score lower than if missing other regimes (15-11)
- c. Marginal: Only 2 of the 4 regimes present; if fast-shallow or slow-shallow are missing, score low (10-6)
- d. Poor: Dominated by 1 regime (usually slow-deep) (5-0)

2. **Pool Variability** - (**Low Gradient Sheet**) - rates the overall mixture of pool types found in streams, according to size and depth. The four basic types of pools are large-shallow, large-deep, small-shallow and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. General guidelines are any pool dimension (i.e., length, width, oblique)

greater than half the cross-section of the stream for separating large from small and 1 m depth separating shallow and deep.

- a. Optimal: Even mix of large-shallow, large-deep, small-shallow and small-deep pools present (20-16)
- b. Suboptimal: Majority of pools large-deep; very few shallow (15-11)
- c. Marginal: Shallow pools much more prevalent than deep pools (10-6)
- d. Poor: Majority of pools small-shallow or pools absent (5-0)

#### **Parameter #4**

**Sediment Deposition (Both Sheets)** - measures the amount of sediment that has accumulated in pools and changes that have occurred to the stream bottom as a result of deposition. This 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) or shoals or result in the filling of runs and pools. Sediment is often found in areas that are obstructed and areas where the stream flow decreases, such as bends. Deposition is a symptom of an unstable and continually changing environment that becomes unsuitable for many organisms.

1. Optimal (High Gradient): Little or no enlargement of islands or point bars and less than 5% of the bottom affected by sediment deposition (20-16)

Optimal (Low Gradient): Little or no enlargement of islands or point bars and less than 20% of the bottom affected by sediment deposition (20-16)

2. Suboptimal (High Gradient): Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5%-30% of the bottom affected; slight deposition in pools (15-11)

Suboptimal (Low Gradient): Some new increase in bar formation, mostly from gravel, sand or fine sediment; 20%-50% of the bottom affected; slight deposition in pools (15-11)

3. Marginal (High Gradient): Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30%-50% of the bottom affected; moderate sediment deposits apparent at most obstructions and slow areas, bends and pools (10-6)

Marginal (Low Gradient): Moderate deposition of new gravel, sand or fine sediment on old and new bars; 50%-80% of the bottom affected; sediment deposits at obstruction, constrictions and bends; moderate deposition of pools prevalent (10-6)

4. Poor (High Gradient): Heavy deposits of fine material, increased bar development; more than 50% of the bottom changing frequently; pools almost absent due to substantial sediment deposition (5-0)

Poor (Low Gradient): Heavy deposits of fine material, increased bar development; more than 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition (5-0)

#### **Parameter #5**

**Channel Flow Status (Both Sheets)** - the degree to which the channel is filled with water; will change with seasons.

1. Optimal: Water reaches base of both lower banks; minimal amount of channel substrate exposed (20-16)
2. Suboptimal: Water fills >75% of the available channel; or <25% of channel substrate exposed (15-11)
3. Marginal: Water fills 25%-75% of the available channel; riffle substrates are mostly exposed (10-6)
4. Poor: Very little water in channel; mostly present in pools (5-0)

**ii. The next 5 parameters should evaluate an area from approx. 100-m upstream of the sampling reach through the sampling reach. This whole area should be evaluated as a composite. When determining left and right bank, look downstream.**

#### **Parameter #6**

**Channel Alteration (Both Sheets)** - measures the large-scale changes in the shape of the stream channel; channel alteration is present when 1) artificial embankments, rip-rap and other forms of bank stabilization or structures are present, 2) the stream is very straight for significant distances, 3) dams and bridges are present and 4) other such changes have occurred.

1. Optimal: Channelization or dredging absent or minimal; stream with normal pattern (20-16)
2. Suboptimal: Some channelization present, usually in areas of bridge abutments; evidence of past channelization (dredging, etc., >20 past years) may be present, but recent channelization not present (15-11)
3. Marginal: Channelization may be extensive; embankments or shoring structures present on both banks; and 40%-80% of the stream reach channelized and disrupted (10-6)
4. Poor: Banks shored with gabion or cement; >80% of the stream disrupted; instream habitat greatly altered or removed entirely (5-0)



## Parameter #7

1. **Frequency of Riffles (or Bends) - (High Gradient Sheet)** - measures the sequence of riffles and thus the heterogeneity occurring in a stream.
  - a. **Optimal**: Occurrence of riffles relatively frequent; ratio of distance between riffles divided by the width of the stream  $<7:1$  (generally 5 to 7); variety of habitat is key; in streams where riffles are continuous, placement of boulders or other large, natural obstruction is important (20-16)
  - b. **Suboptimal**: Occurrence of riffles infrequent; distance between riffles divided by the width of the stream is between 7 and 15 (15-11)
  - c. **Marginal**: Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 and 25 (10-6)
  - d. **Poor**: Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is  $>25$  (5-0)
  
2. **Channel Sinuosity - (Low Gradient Sheet)** - 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 water levels in the stream fluctuate as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in low-gradient streams, a longer segment or reach than that designated for sampling may be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In "oxbow" streams of coastal areas and deltas, meanders are highly exaggerated and transient. Natural conditions in these streams are shifting channels and bends, and alteration is usually in the form of flow regulation and diversion. A stable channel is one that does not exhibit progressive changes in slope, shape or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).
  - a. **Optimal**: 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.) (20-16)
  - b. **Suboptimal**: The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line (15-11)
  - c. **Marginal**: The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line (10-6)

- d. Poor: Channel straight; waterway has been channelized for a long distance (5-0)

### **Parameter #8**

**Bank Stability (Both Sheets)** - measures whether the stream banks are eroded or have the potential to erode. Each bank is scored independently from 10-0.

1. Optimal: Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems; <5% of bank affected (10-9)
2. Suboptimal: Moderately stable; infrequent, small areas of erosion mostly healed over; 5%-30% of the bank affected (8-6)
3. Marginal: Moderately unstable; 30%-60% of bank in reach has areas of erosion; high erosion potential during floods (5-3)
4. Poor: Unstable; many raw, eroded areas; obvious bank sloughing; >60% of bank has erosional scars (2-0)

### **Parameter #9**

**Bank Vegetative Protection (Both Sheets)** - measures the amount of vegetative protection afforded to the stream and the near-stream portion of the riparian zone. Each bank is scored independently from 10-0.

1. Optimal: >90% of the streambank surfaces and immediate riparian zones covered by natural vegetation, including trees, understory shrubs, herbs and nonwoody macrophytes; vegetation disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally (10-9)
2. Suboptimal: 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 (8-6)
3. Marginal: 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 (5-3)
4. Poor: <50% of the streambank surfaces covered by vegetation; disruption is very high; vegetation has been removed to 5 cm or less in average stubble height (2-0)

## Parameter #10

**Riparian Vegetative Zone Width (Both Sheets)** - measures the width of the natural vegetation from the edge of the streambank through the riparian zone. The presence of old fields, paths, walkways, etc., in otherwise undisturbed riparian zones may be judged to be inconsequential to destruction of the riparian zone. Each bank is scored independently from 10-0.

1. Optimal: Width of riparian zone >18 m; human activities (parking lots, roadbeds, clear-cuts, lawns, pastures or crops) have not impacted the zone (10-9)
2. Suboptimal: Width of riparian zone 13-18 m; human activities have impacted the zone only minimally (8-6)
3. Marginal: Width of riparian zone 6-12 m; human activities have impacted the zone a great deal (5-3)
4. Poor: Width of riparian zone <6 m; little or no riparian zone due to human activities (2-0) (Barbour et al. 1999)

## IV. TENTATIVE HABITAT CRITERIA

Habitat evaluations were conducted at biological sampling sites in the Kentucky River Basin for 1998, the Licking and Salt River Basins for 1999, the Cumberland, Tennessee and Mississippi River Basins for 2000, and the Green and Tradewater River Basins for 2001 using the new Rapid Bioassessment Protocols format. Historical and current reference reach habitat data were used to produce tentative habitat criteria for certain areas of Kentucky. The scores for all reference reach stations were ranked and divided into percentiles. The lower quartile (25<sup>th</sup> percentile) may be considered the dividing line between those habitats fully supporting biotic integrity and those supporting biotic integrity, but threatened. Those scores falling within the 25<sup>th</sup> and 10<sup>th</sup> percentile may be considered supporting, but threatened. Habitats scoring in the last 10<sup>th</sup> percentile may be considered partially supporting biotic integrity. Habitats may be considered poor if they fall below the lowest reference condition score for that area.

Mountainous ecoregions of the Commonwealth provided similar habitat opportunities for aquatic community colonization and use. Habitat scores from reference sites in the Central Appalachian, Southwestern Appalachian and Western Allegheny Ecoregions reflected these similarities. Therefore, habitat data from all of these ecoregions were combined to develop habitat criteria for the Mountains.

For the Interior Plateau Ecoregion, reference sites were divided into wadeable and headwater based upon drainage area. Those above six square miles were considered wadeable and below six square miles headwater. Headwater streams in this large ecoregion have a tendency to score higher on certain metrics (i.e. frequency of riffles, bank stability, riparian zone width) than wadeable streams as a result of increased land use. This bias was reflected in final habitat scores. Therefore, separate habitat criteria were developed for wadeable and headwater streams.

Streams sampled within the Mississippi Valley Loess Plains and Interior River Valleys and Hills Ecoregions were generally low-gradient streams with very few riffles, if any. Low-gradient habitat

assessment sheets were used in these ecoregions to rate the available habitat. Additionally, in low-gradient sections of Interior Plateau and mountainous streams, low-gradient habitat assessment sheets were used. Most low-gradient streams provide similar habitats for biotic colonization and use; therefore, all low-gradient reference were analyzed together. Habitat criteria for low-gradient streams were developed from this entire data set.

The following tentative habitat criteria can be used to determine whether the sampling reach is fully supporting, supporting, but threatened, partially supporting or not supporting its designated use:

**1. Central Appalachian, Southwestern Appalachian and Western Allegheny Ecoregions (High-Gradient Assessments)**

Fully Supporting:	165 and above
Supporting, But Threatened:	164 - 156
Partially Supporting:	155 - 145
Not Supporting:	144 and below

**2. Interior Plateau Ecoregion (High-Gradient Assessments) - Wadeable Streams (>6.0 mi<sup>2</sup>)**

Fully Supporting:	145 and above
Supporting, But Threatened:	144 - 131
Partially Supporting:	130 - 105
Not Supporting:	104 and below

**3. Interior Plateau Ecoregion (High-Gradient Assessments) - Headwater Streams (<6.0 mi<sup>2</sup>)**

Fully Supporting:	155 and above
Supporting, But Threatened:	154 - 145
Partially Supporting:	144 - 138
Not Supporting:	137 and below

**4. Mississippi Valley Loess Plain and Interior River Hills and Valleys Ecoregions (Low-Gradient Assessments)**

Fully Supporting:	132 and above
Supporting, But Threatened:	131 - 119
Partially Supporting:	118 - 110
Not Supporting:	109 and below

Criteria development is an ongoing process. Updates of the criteria will occur as more reference data is collected.

## V. QUALITY ASSURANCE/QUALITY CONTROL

- A. Habitat assessment is subjective; however, annual training can help to standardize the assessment.
- B. It is best to do the habitat assessment after the biological sampling. This allows the investigator to become more familiar with the types of substrate, degree of embeddedness, the conditions of the banks, etc.
- C. If at all possible, have more than one person conduct a habitat assessment. The sampling crew may want to come together and discuss each parameter to attain a final score.
- D. If only one person is assessing the habitat, make sure that person continues to conduct the habitat assessments at other sites.
- E. Be consistent with your scoring, paying close attention to narrative statements made for each parameter.
- F. Repetition will allow you to differentiate between suboptimal and marginal, for instance.
- G. Photos of the site could help document habitat conditions and changes.

### LITERATURE CITED

- Barbour, M.T., J. Gerritsen, B.D. Snyder, and J.B. Stribling. 1999. Rapid bioassessment protocols for use in wadeable streams and rivers: periphyton, benthic macroinvertebrates, and fish (2<sup>nd</sup> edition). The United States Environmental Protection Agency, Washington, D.C. EPA 841-B-99-002.
- Gordon, N.D., T.A. McMahon, and B.L. Finlayson. 1992. Stream hydrology: an introduction for ecologists. John Wiley and Sons, Inc. West Sussex, England.

# 7 - ALGAE

## *BENTHIC ALGAE*

### I. INTRODUCTION

Benthic (attached) algae are sensitive indicators of change in lotic waters, as well as being the primary producers within the stream ecosystem. The benthic algae are usually the dominant component of the periphyton. Because it is attached to the substrate, the benthic algae community integrates physical and chemical disturbances to a stream. Another advantage of using benthic algae in water quality assessments is that the benthic algae community contains a naturally high number of species, making data useful for statistical and numerical applications to assess water quality. Response time of benthic algae is rapid, as is recovery, with recolonization after a disturbance often more rapid than for other organisms. Diatoms, in particular, are useful indicators of biological integrity because they are ubiquitous; at least a few can be found under almost any conditions. In addition, most can be identified to species by experienced biologists, and tolerances or sensitivities to specific changes in environmental conditions are known for many species (Dixit, et al. 1992, Rott 1991). By using benthic algal data in association with macroinvertebrate and fish data, the biological integrity of the entire ecosystem can be ascertained.

### II. SAMPLING PROCEDURES

#### A. Benthic Algae Field Data Sheet (Appendix C-1)

Conduct a cursory, visual assessment of the reach and record the results on the benthic algae field data sheet. Record the following station information in the appropriate spaces at the top of the sheet: KDOW station identification number, stream name, location of the station, collection date, time, county, river basin, KDOW program and name of investigator(s). Most of this information can be completed in the office prior to sampling, which will facilitate faster sample collection. After sampling has been conducted, assess the following conditions and record these assessments in the proper spaces: macrohabitats sampled (e.g., riffle, run, pool), microhabitats sampled (Table 7-1), the collection method used, macroalgae present (e.g., *Cladophora*, *Batrachospermum*, *Tetraspora*, etc.), an estimation of benthic algae coverage (e.g., absent, sparse, moderate, heavy), any comments (e.g., whether scour has recently occurred, the flow status of the stream, etc.) and a qualitative ranking of the algal community (See Section I.4.A for details). Use this information to supplement more detailed benthic algal community assessments.

#### B. Sample Collection

##### 1. Natural Substrates (Headwater and Wadeable streams)

Collect benthic algae from all available microhabitats in wadeable streams (Table 7-1). Collect a composite, qualitative sample from microhabitats in roughly the proportion that they occur at the site. Sample both riffles and pools, or select one major habitat type (usually riffle) if it occurs at all sites to be compared in the study. During low-flow periods, pools may be the only habitat available. Collect samples during stable flow conditions. After extremes of flooding or drought,

allow at least a two-week recolonization period before sampling. Collect samples using the following methods:

- a. Use a knife blade, microspatula, toothbrush or similar device to scrape algae from rocks and other hard substrates making every effort to remove all algae from the scraped area. Rinse with distilled water if necessary. Five replicate samples are collected from rocks of similar size whenever possible. Samples from individual rocks can be composited or analyzed separately. Quantitative data can be obtained by measuring the area of substrate sampled.
- b. Use a suction device to collect algae from bedrock substrates. Press a section of PVC pipe equipped with a neoprene rubber gasket against the bedrock substrate so that the benthic algae within the enclosed area can be dislodged with a stiff-bristled brush. Suction dislodged material into a filter flask using a hand-operated pump. This method can be used to obtain quantitative data from bedrock (modified from Douglas 1958).
- c. Gently lift algal mats from depositional areas with forceps or by suctioning with a disposable pipette.

<b>Epipellic</b>	Silt and sediment habitats usually in depositional areas with slow current. Algae may form a thin mat that loosely adheres to the surface of the epipelon. To collect epipellic algae, suction material from the mud-water interface using an eye-dropper bulb and a disposable pasteur pipette or gently lift the algal mat from the surface of the sediment using a knife.
<b>Episammic</b>	Sand habitats. Collection is made in the same manner as above.
<b>Epilithic</b>	Rock or other hard surface habitats including dams, bridge abutments, boat ramps, etc. To sample epilithic algae, scrape or hand pick material from epilithon in riffles, pools and runs.
<b>Epidendric</b>	Woody habitats. To collect epidendric algae, scrape or hand pick material from submerged logs, tree roots, drifts, etc.
<b>Epiphytic</b>	Plant habitats usually associated with aquatic mosses, macrophytes and filamentous algae. To sample epiphytic algae, scrape, wring out, hand pick or collect the entire substrate.
<b>Epizooic</b>	Animal habitats including turtle shells, snail shells and other macroinvertebrates. To sample epizooic algae, scrape, hand pick or collect the entire substrate.

## 2. Artificial Substrate (Non-wadeable Streams)

Algal data from artificial substrates can be used as a tool to assess water quality, even if the natural assemblage is not exactly duplicated (Patrick 1973, Stevenson and Lowe 1986). In non-wadeable streams, rivers with no riffle areas, wetlands or littoral zones of lakes, surface (floating) or benthic (bottom) periphytometers equipped with glass slides are used for collecting algae. Clay tiles, plexiglass plates or other substrates may be substituted for glass slides. Pre-clean glass slides and

rinse with acetone before placing them in the periphytometer. Place artificial substrates in the stream for three to four weeks to allow sufficient time for colonization (Aloi 1990, Weber and Raschke 1970, Weber 1973). To minimize difficulty or errors in analysis of benthic algae from artificial substrates, set a minimum of three periphytometers at each site, from which individual or composite samples can be obtained. Attach periphytometers to trees, snags, bridge pilings or other sturdy anchor sites and away from frequently visited areas (swimming and fishing "holes") to avoid vandalism, theft or other disturbances. Reset periphytometers if flooding or desiccation occur during the exposure period. Allow streams to return to pre-scouring or pre-drought conditions before resetting periphytometers. Samples from replicate periphytometers can be analyzed separately or composited, depending on the goals of the study.

### 3. Biomass Samples

For biomass samples, scrape natural substrates in the field using distilled water to rinse the substrates. Collect all material and rinse water from the scraped area into an opaque jar and return it, on ice, to the laboratory for filtration of subsamples. Filtration may be performed in the field and filters transported on ice to the laboratory. Do not add preservatives to chlorophyll *a* samples. For natural substrates, measure the area sampled by tracing its outline on paper and determining size with a digital planimeter. Artificial substrates are wrapped in aluminum foil and transported on ice to the laboratory for sample preparation and analysis.

## III. SAMPLE PRESERVATION AND LABELING

Collect samples in small, watertight glass vials or screw-top bottles of at least 60 ml in volume. Preserve taxonomic samples with 3% to 4% buffered formalin, 2% glutaraldehyde, Lugol's solution or other preservatives listed in the latest edition of Standard Methods (APHA 1992). Chlorophyll *a* and ash-free dry-weight samples should be placed on ice only. Affix a label permanently to the sample container, recording the following information on the label using a pencil or waterproof, indelible ink: waterbody name, location, sample number, date and name of collector. Record this information in a laboratory sample logbook (Appendix C-2). Refrigerate samples until taxonomic evaluation is completed. After taxonomy is completed, archive the samples in a cool, dry place. Permanent mounts of diatom samples are retained in wooden or plastic slide boxes.

## IV. SAMPLE ANALYSIS

### A. Field Assessment

While in the field, qualitatively rank the benthic algal community at each station. A score of 1 (lowest quality) to 5 (highest quality) is possible. Record scores and a description of the benthic algal community on the benthic algae field data sheet (Appendix C-1). While somewhat subjective, this information can be used later to support a detailed assessment of the benthic algal community. The algae are carefully judged using the following ranking criteria:

- 1) **Excellent Quality (5):** The benthic algal community appears diverse with several divisions represented, including chrysophytes, chlorophytes, cyanophytes and rhodophytes. Phytoplankton sub-community are not apparent. Floating algal mats are not present. The algal community is similar to that of reference stations within the same ecoregion.



- 2) **Fair - Good Quality (2-4)**: Benthic algae are present in moderate amounts. The benthic algal community may be dominated by one type of growth, such as long filaments of *Cladophora*. Diversity is low to moderate, and a phytoplankton sub-community is not apparent. Floating algal mats may be present, but are not extensive. Clean water benthic algal taxa (e.g., red algae, *Chaetophora*, etc.) present in reference reach stations may not be present.
- 3) **Poor Quality (1)**: In cases of toxic pollution (acid mine drainage, toxic discharges, etc.), substrates and water column may appear sterile, bleached or rust-colored. Little or no algae are observed. With organic pollution (sewage discharges, etc.), substrates may be covered with thick white, black or gray mats of filamentous bacteria, thick algal mats of cyanophytes (blue-green algae) and/or chlorophytes (green algae). The water column may have a "pea green" appearance as a result of high abundances of euglenophytes, or large floating mats of algae may be present, especially in pools and slow-moving streams. Look for extremes of either characteristic. Diversity is very low. Very few, if any, clean water taxa are present.

## **B. Laboratory Analysis**

### **1. Non-Diatom Algae Slide Preparation and Analysis**

#### Equipment:

Microscope with 20X and 40X objectives  
Glass microscope slides  
Glass coverslips  
Watchglass  
Disposable pipettes

#### Procedure:

In this document, the term "non-diatom algae" refers to all taxa that do not belong to the Class Bacillariophyceae. Thoroughly shake the sample container to dislodge epiphytes from filamentous taxa and randomly mix all algal organisms. Pour the contents into a shallow watchglass so that all filamentous and mat-forming taxa can be separated. Using dissecting probes or needle-nosed forceps, place representative filamentous taxa on a pre-cleaned microscope slide or in a settling chamber for inverted microscope use. Cover the filaments with approximately 0.5 ml of sample liquid. Gently place a coverslip over the subsample, completing the wet mount. First, examine each slide at 200x, then at 400x to ensure that smaller organisms are not overlooked. Identify all non-diatom algae to the lowest taxonomic level using current taxonomic references. Scan each slide until no new organisms are seen. Examine a minimum of three slides for each sample. Record observed taxa on the non-diatom bench sheet (Appendix C-3) along with taxonomic division, estimated relative abundance (abundant, common, rare) and any known autecological information. In some instances, counts of the non-diatom community may be needed in order to obtain specific autecological information. Homogenize the sample with a blender. Pipette a subsample into a Palmer counting cell. Identify and count 300 algal non-diatom units to the lowest taxonomic level at 400X. Non-diatom algal units are considered instead of cells. Since colonial, coenobial or filamentous cells normally do not occur singly in a natural environment, counting each cell does not accurately portray relative abundance for that specific taxon. For example, although *Pediastrum*

*duplex* is composed of several cells, the colony as a whole is counted as one unit. Likewise, coenobia and unicells are each considered one unit. Filaments may be counted either as one unit or counted in units of 10 um lengths (A 100-um filament is 10 units.) Record numbers of non-diatom algal units on the non-diatom bench sheet. Enter the data into an appropriate database, such as EDAS (Ecological Data Application System). Taxonomic references are listed at the end of this chapter.

## 2. Diatom Slide Preparation and Analysis

### A. Cleaning Methods for Diatoms

After the non-diatom algae have been identified, clear diatom frustules of organic and intercellular material using one of the following oxidation methods (APHA 1992, van der Werff 1955):

#### 1. Nitric Acid Oxidation

Equipment:

2000 ml Erlenmeyer flask  
1000 ml graduated cylinder  
20-30 ml of algae sample  
50 ml of HNO<sub>3</sub>

Procedure:

Shake the sample vigorously and immediately pour a portion (about 20-30 ml) of the sample into a large 2000 ml Erlenmeyer flask. Under a flame hood, add 50 ml of concentrated HNO<sub>3</sub>. Allow the sample to oxidize overnight, then fill the flask with distilled water. Let the sample resettle overnight, siphon off the supernatant, and pour the remaining diatom solution into a 1000 ml graduated cylinder. Fill with distilled water, settle and siphon supernatant at least twice more, until the yellow color changes to clear.

#### 2. Hydrogen Peroxide/Potassium Dichromate Oxidation

Equipment:

2000 ml Erlenmeyer flask  
1000 ml graduated cylinder  
20-30 ml algae sample  
50 ml of 50% H<sub>2</sub>O<sub>2</sub>  
Microspatula  
Dash of K<sub>2</sub>Cr<sub>2</sub>O<sub>7</sub>

Procedure:

Prepare sample as in nitric acid method, but use 50 ml of 50% H<sub>2</sub>O<sub>2</sub> instead of HNO<sub>3</sub>. Allow to oxidize overnight; then add a microspatula of K<sub>2</sub>Cr<sub>2</sub>O<sub>7</sub>. This will cause a violent exothermic reaction, so be careful to perform this method only under a fume hood. When

the sample color changes from purple to yellow and boiling stops, fill the flask with distilled water. Repeat rinsing steps as outlined for the HNO<sub>3</sub> method until the yellow color is gone.

## **B. Diatom Slide Preparation**

### Equipment:

Pre-cleaned microscope slide  
Glass coverslip  
Naphrax mounting medium  
Hot plate

### Procedure:

Using a disposable pipette, drop a small amount of well-mixed sample onto a coverslip that has been placed on a hot plate. Dry at low heat until the moisture is removed. Place a large drop of Naphrax mounting medium onto a pre-cleaned microscope slide. Invert the coverslip onto the Naphrax. Place the slide on a hot plate on high heat. Allow the toluene to boil out of the Naphrax. Make sure that this procedure is conducted in or near a laboratory hood where ventilation is good. After bubbles of toluene have stopped forming, remove the slide from the hot plate. Allow the slide to cool and harden. Remove all excess Naphrax from the slide with a razor blade. Affix a slide label to the slide and place in a slide box for storage.

## **C. Diatom Identification and Enumeration**

### Equipment:

Microscope with 100X oil immersion objective  
Immersion oil

### Procedure:

Identify diatoms at 1000X to the lowest possible taxonomic level, preferably to the species or variety level, using current taxonomic references. Record all taxa encountered on the diatom bench sheet (Appendix C-4) creating a species list prior to enumeration. Scan the slide until several minutes pass without producing any new taxa. For quantitative data, count a minimum of 500 valves recording taxa and number counted on the diatom bench sheet. Data assessment is based on the completed species list. Enter data into an appropriate database, such as EDAS.

## **D. Biomass**

### **A. Chlorophyll *a***

Chlorophyll *a* analyses are performed as described for phytoplankton (Chapter 3, Part II), using U.S. EPA Method 445.0 (USEPA 1992).

### **B. Ash-Free Dry-Weight (AFDW)**

Replicate samples are analyzed in accordance with Standard Methods (APHA 1992).

## V. DATA ANALYSIS

An assessment of biological integrity can be made based on the benthic algal data. The goal is to categorize water quality as excellent, good, fair or poor and to determine the degree and cause of aquatic life use impairments in fair or poor streams. A multiple metric index called the Diatom Bioassessment Index (DBI) is used to assess the benthic algal community.

### A. Diatom Bioassessment Index

Biological indices represent mathematical models of community changes (Perkins 1983). Changes in water quality will affect resident biota, and indices that reflect these changes in a particular community are useful biological indicators of water quality. The benthic algal community, especially diatoms, is a useful biological indicator because: 1) they are attached to the substrate and, therefore, subjected to any immediate or prolonged disturbances; 2) diatoms are ubiquitous, with at least a few individuals found under almost any aquatic conditions; 3) total number of taxa at any given site is usually high enough for use in calculating various metrics; 4) diatoms, especially the most abundant species, are identifiable to species by trained professionals; 5) tolerance of or sensitivity to changes (autecological requirements) is known or suspected for many species or assemblages of diatoms; and 6) benthic algal communities, especially diatoms, have a rapid response and recovery time because of their relatively short lifecycle (as compared to fish or macroinvertebrates) and their ability to quickly recolonize formerly disturbed (impacted) sites (Dixit et al. 1992).

Several metrics have been used to assess water quality conditions using benthic algae. Some have the diagnostic ability to indicate the type of impact (nutrient enrichment, toxicity, acidity, salinity, sewage (organic) pollution and siltation). However, each metric alone could not accurately describe the overall water quality at a site. In the mid-1980s, phycologists at the KDOW developed a multi-metric index using four benthic diatom metrics. This multi-metric approach was modeled after successfully used indices such as Karr (1981) with fish and Plafkin, et al. (1989) with macroinvertebrates. The metrics chosen for the original Diatom Bioassessment Index (DBI) were as follows: total number of diatom taxa (TNDT), Shannon diversity ( $H'$ ), pollution tolerance index (PTI) and % sensitive species (%SS). Recently, phycologists at KDOW have refined the DBI using box and whisker plots to test metric sensitivity and the Pearson's correlation coefficient index to test for metric redundancy. A new DBI was developed using three of the original metrics (TNDT,  $H'$ , and PTI) and the following three new metrics: *Cymbella* group richness (CGR), *Fragilaria* group richness (FGR), and % *Navicula*, *Nitzschia* and *Surirella* (%NNS). The new DBI provides water resource managers with a very sensitive, community structure-based tool for assessing water quality.

#### 1. Diatom Bioassessment Index Metrics

##### a. Total Number of Diatom Taxa (TNDT)

Total number of diatom taxa (TNDT) is an estimate of diatom species richness. High species richness is assumed to be the case in an unimpacted site, and species richness is expected to decrease with increasing pollution. Slight levels of nutrient enrichment, however, may increase species richness in naturally unproductive, nutrient-poor streams

(Bahls 1992). Low-order, pristine streams in the Central Appalachian or Western Allegheny ecoregion of eastern Kentucky may fall into this category.

- i. TNDT = total number of diatom taxa identified
- ii. TNDT/500 = total number of diatom taxa encountered in a count of 500 individuals.

**b. Shannon Diversity**

The mean Shannon diversity index is used in diatom assessments. It was chosen primarily because it is commonly used by many aquatic biologists, so values will be more readily interpreted and compared with other literature values. Using this index,  $H' = 0$  when only one species is present in the collection, and  $H'$  is at a maximum when all individuals are evenly distributed among the  $S$  species.

$$H' = - \sum \frac{n_i}{N} \log_{10} \frac{n_i}{N}$$

where:

- $n_i$  = number of individuals of species  $i$   
 $N$  = total number of individuals

$H'$  can also be expressed using natural logarithms ( $\ln$ ); however,  $\log_{10}$  is used for historical comparison with other data collected across the state (Harker et al. 1979). Conversion factors for  $\ln$  and  $\log_2$  are available in Weber (1973).

i. Disadvantages

Diversity is affected by both the number of species in a sample and the distribution of individuals among those species (Klemm et al. 1992). Because species richness and evenness may vary independently, under certain conditions diversity values can be misleading. For example, streams with poor water quality due to toxic discharges may have very low taxa richness, but the individuals present may be very evenly distributed among those few taxa. This often results in high diversity values under stressed water quality conditions (KDOW unpublished data, Pontasch and Brusven 1988, Pontasch et al. 1989). Archibald (1972) also warns of the dangers of using diatom diversity alone as an indicator of water quality because low diversity values may indicate either heavily polluted water or clean water. Hurlbert (1971) goes so far as to state that species diversity is an ecologically meaningless concept and suggests its use be abandoned; and according to Perkins (1983), the assumption that individuals in more polluted environments should be less evenly distributed is speculative. It can be argued that the evenness assumption of the diversity index may be ecologically unsound and that in natural communities individuals are not evenly distributed among species. This is supported by the work of Patrick et al. (1954) on the structure of natural diatom communities.

ii. Advantages

Species diversity, despite the controversy surrounding it, has historically been used with success as an indicator of organic (sewage) pollution (Wilhm and Dorris 1966, Weber 1973, Cooper and Wilhm 1975). Bahls (1992) uses Shannon diversity because of its sensitivity to water quality changes, and Stevenson (1984) suggests that changes in species diversity, rather than the diversity value, may be useful indicators of changes in water quality. It is obvious that successful use of the diversity index depends upon careful application and interpretation. As a metric used in the DBI, it is only one of several metrics and will not be used alone as a water quality indicator.

c. **Pollution Tolerance Index (PTI)**

Several recent water quality indices based on pollution tolerance (or sensitivity) of diatom species have been proposed, including the saprobity index of Sladeczek (1973), and diatom pollution tolerance indices developed by Lange-Bertalot (1979), Descy (1979), Leclercq and Maquet (1987) and Watanabe et al. (1988). What these indices have in common is that species are differentiated into groups and assigned values relating to pollution tolerance. Calculations involve estimates of the dominance of species and a "subjective assessment of the value of each species as an indicator" (Round 1991). Other state agencies in the U.S. using some type of diatom pollution tolerance index include the Montana Water Quality Bureau (Bahls 1992) and Oklahoma Conservation Commission (Bob Lynch, pers. comm.). Ideally, a consensus will be reached on which diatom pollution tolerance index should be used, and a common list of pollution tolerance values will be developed. However, because a species may react differently across different ecoregions and may have variable sensitivities to different types of pollution (e.g., nutrients, metals, pH, salinity), universal values may not be appropriate.

The pollution tolerance index (PTI) used by the Kentucky Division of Water is most similar to that of Lange-Bertalot (1979) and resembles the Hilsenhoff Biotic Index for macroinvertebrates (Hilsenhoff, 1987). Lange-Bertalot distinguished three categories of diatoms according to their tolerance to increased pollution, with species assigned a value of 1 for most tolerant taxa (e.g., *Nitzschia palea* or *Gomphonema parvulum*) to 3 for relatively sensitive species. For the PTI, Lange-Bertalot's list has been adapted to four categories to differentiate a large moderately tolerant group of species (similar to his splitting of category 2 diatoms into 2a and 2b); the KDOW diatom pollution tolerance values range from one (most tolerant) to four (most sensitive). Tolerance values for Kentucky diatoms were generated from a multitude of literature, including Lowe (1974), Patrick and Reimer (1966 and 1975), Patrick (1977), Lange-Bertalot (1979), Descy (1979), Sabater, et al. (1988), Bahls (1992), Mississippi Department of Environmental Quality (Stanley Rogers, pers. comm.) and Oklahoma Conservation Commission (Bob Lynch, pers. comm.). The extensive KDOW diatom database collected from 1977 to the present and data collections by the Kentucky Nature Preserves Commission (1979 - 1986) were also instrumental in the determination of tolerance values. The list of tolerance values presently in use (Appendix C-5) will be revised and updated as new autecological data is discovered; however, the tolerances of most common species are fairly well understood. Because the index is based on relative abundances, rare species will have little effect on the final index value. If no autecological data is known, the species is given a PTI value of 0 and is not used in PTI index calculation.

The formula used to calculate PTI is:

$$PTI = \frac{\sum n_i \times t_i}{N}$$

where  $n_i$  = number of individuals in species  $i$

$t_i$  = tolerance value of species  $i$

$N$  = total number of individuals

**d. Siltation Index (%NNS)**

The sum of the relative abundances of all *Navicula* (including *Aneumastus*, *Cavinula*, *Chamaepinnularia*, *Cosmioneis*, *Craticula*, *Diadesmis*, *Fallacia*, *Fistulifera*, *Geissleria*, *Hippodonta*, *Kobarasia*, *Luticola*, *Lyrella*, *Mayamaia*, *Muellaria*, *Placoneis* and *Sellaphora*), *Nitzschia* (including *Psammodictyon* and *Tryblionella*) and *Surirella* taxa reflects the degree of sedimentation at a reach. These three genera are motile, using their raphes to slide through sediment if they become covered. Their abundance expresses the frequency and severity of sedimentation. As sedimentation increases, the %NNS is expected to increase (Bahls et al. 1992).

%NNS = the sum of the relative abundances of all *Navicula*+*Nitzschia*+*Surirella* taxa

**e. Fragilaria Group Richness (FGR)**

The total number taxa represented in the sample from the genera *Ctenophora*, *Fragilaria*, *Fragilariforma*, *Pseudostaurosira*, *Punctastriata*, *Stauroforma*, *Staurosira*, *Staurosirella*, *Tabularia* and *Synedra* reflects high water quality. As water pollution increases, the FGR is expected to decrease.

FGR=*Ctenophora*+*Fragilaria*+*Fragilariforma*+*Pseudostaurosira*+*Punctastriata*+*Stauroforma*+*Staurosira*+*Staurosirella*+*Synedra*+*Tabularia*

**f. Cymbella Group Richness (CGR)**

The total number of taxa represented in the sample from the genera *Cymbella*, *Cymbopleura*, *Encyonema*, *Encyonemopsis*, *Navicella*, *Pseudoencyonema* and *Reimeria* reflects high water quality. As water pollution increases, the CGR is expected to decrease.

CGR=*Cymbella*+*Cymbopleura*+*Encyonema*+*Encyonemopsis*+*Navicella*+*Pseudoencyonema*+*Reimeria*

**2. Future Diatom Bioassessment Index Metrics**

Other metrics may be included in the new DBI as their usefulness is evaluated and more data are collected.

**a. Total Number of All Algal Genera (TNG)**

Total number of all algal genera (TNG) may provide a better estimate of diversity than taxa richness. As water pollution increases, TNG is expected to decrease (Barbour et al. 1999).

TNG = total number of benthic algal genera identified (non-diatom + diatom)

**b. Total Number of Divisions Represented (TDiv)**

Representatives from several divisions of algae are common from sites with good water quality. The number of divisions represented is reported as an indicator of diversity.

**3. Calculating the Diatom Bioassessment Index**

Each metric is given a calculated score (range 0-100) based on the percent of the standard metric value (i.e., the 95<sup>th</sup> percentile or 5<sup>th</sup> percentile) of the entire database (impaired and reference). These percentile thresholds are used to eliminate outliers. The formulae for calculating DBI scores are shown in Table 7-2.

<b>Table 7-2: Metric Scoring Formulae for the Diatom Bioassessment Index</b>	
<b>Metric</b>	<b>Formula</b>
TNDT	$(TNDT/95^{th}\%ile) \times 100$
H'	$(H'/95^{th}\%ile) \times 100$
PTI	$(PTI/95^{th}\%ile) \times 100$
FGR	$(FGR/95^{th}\%ile) \times 100$
CGR	$(CGR/95^{th}\%ile) \times 100$
%NNS	$(100 - \%NNS)/(100 - 5^{th}\%ile) \times 100$

Metric scores with values greater than 100 receive a score of 100.0. The mean of the six DBI metrics is the final DBI score on a 0-100 scale.

Final DBI scoring criteria are currently being developed for each ecoregion in Kentucky using an extensive KDOW database collected from 1986 through 2000. Refinement of these criteria will continue as more data are collected.

**B. Other Data Evaluation Methods**

Data assessment and interpretation is by no means restricted to the above metrics. Statistical analyses, when appropriate, may render useful insights into benthic algal data. Often, however, biological data are not appropriate for parametric statistical analyses because the basic assumption of normal distribution is not always met. In addition, statistically significant differences are not always ecologically significant (APHA 1992, Elliott 1983). Multivariate analyses are becoming popular in environmental assessments (Ter Braak 1988, Gauch 1982) and as user-friendly software packages become available, these methods will certainly be explored in the future. Such methods are especially useful in determining the tolerances of benthic algal species to specific environmental parameters (Agbeti 1991, del Giorgio et al. 1991, Dixit et al. 1992 and Sabater et al. 1988).

**1. Chlorophyll *a***

Benthic chlorophyll *a* values are used as an estimate of algal biomass. Chlorophyll *a* values can be extremely variable because of the patchiness of benthic algal distribution; therefore, assessments are



based on a mean of three or more replicate samples. These values are used to compare biomass accrual at the same station over time or between stations during the same sampling period. High chlorophyll *a* values may indicate nutrient enrichment, while low values may either indicate low nutrient availability, toxicity or low-light availability because of shading, sedimentation or high turbidity. Chlorophyll *a* values are used only in support of other analyses.

## 2. Ash-free Dry-weight (AFDW)

Benthic AFDW values are used as an estimate of total organic material accumulated on the artificial substrate. This organic material includes all living organisms (algae, bacteria, fungi, protozoa and macroinvertebrates) as well as non-living detritus. Ash-free dry-weight values have been used in conjunction with chlorophyll *a* as a means of determining the trophic status (autotrophic vs. heterotrophic) of streams. The Autotrophic Index (AI) is calculated as follows:

$$AI = \frac{AFDW (mg/m^2)}{Chlorophyll\ a (mg/m^2)}$$

High AI values (>200) indicate the community is dominated by heterotrophic organisms, and extremely high values indicate poor water quality (Weber 1973, Weitzel 1979, Matthews et al. 1980). This index should be used with discretion, as non-living organic detritus can artificially inflate the AFDW value.

## VI. QUALITY ASSURANCE AND QUALITY CONTROL

### A. Quality Assurance and Quality Control in the field (Barbour et al. 1999)

1. Make sure that sample labels are accurate and complete with all pertinent information that were previously discussed.
2. Before leaving a reach, all sampling equipment will be checked for residual algal material, rubbed clean and thoroughly rinsed with distilled water.
3. One duplicate sample from 10% of the reaches sampled within a sampling season will be collected and analyzed to ensure precision and repeatability of the sampling technique.
4. Phycologists are trained in the sampling techniques on a regular basis.

### B. Quality Assurance and Quality Control in the laboratory (Barbour et al. 1999)

1. Record all samples in the laboratory sample logbook (Appendix C-2).
1. A voucher collection will be maintained of all samples and diatom slides. Label information must be accurate and complete.
2. Phycologists should discuss any problem identifications with other phycologists. Any problem taxa should be sent to an outside taxonomist to assist in identification.

3. Duplicate samples will be processed and analyzed in the same procedures as regular samples.
4. Another in-house phycologist should identify and enumerate the duplicate samples. If there is not another phycologist in the lab, an outside phycologist should be utilized to periodically check identification of samples.
5. Duplicate samples should exceed 75% similarity (Whittaker 1952) of the replicate sample.
6. The most recent taxonomic references will be used for the identification of algae.
7. Phycologists will participate in algal identification workshops, seminars and training sessions when opportunities arise.

## ***PHYTOPLANKTON***

### **I. INTRODUCTION**

*Phytoplankton* are important primary producers in lakes, impoundments, ponds, wetlands, low-gradient streams and backwater areas. *Lotic* phytoplankton samples may be collected by the KDOW from biological monitoring stations and from selected intensive survey sites. In addition, phytoplankton samples are collected from streams and reservoirs used as drinking water supplies if taste and odor problems occur. Samples are analyzed for chlorophyll *a*, phytoplankton density (cells/ml) and/or phytoplankton community structure, depending upon the nature of the survey. *Lentic* phytoplankton samples may also be collected to assess water quality and recreation uses.

### **II. SAMPLING PROCEDURES**

#### **A. Phytoplankton Field Data Sheet (Appendix D-6)**

Conduct a cursory, visual assessment of the reach and record the results on the phytoplankton field data sheet. Record the following station information in the appropriate spaces at the top of the sheet: KDOW station identification number, waterbody name, location of the station, collection date, time, county, river basin, purpose of the study (e.g., chlorophyll *a*, taste and odor, etc.) and name of investigator(s). Most of this information can be completed in the office prior to sampling, which will facilitate faster sample collection. After sampling has been conducted, record the collection method used, analyses to be conducted (e.g., chlorophyll *a*, ash-free dry-mass, phytoplankton count), bloom characteristics (e.g., color of surface scum, any odor present, any surface sheen present, thickness of the bloom, etc.) and the surface bloom coverage (See Section II.4.A) in the appropriate spaces on the phytoplankton field data sheet.

#### **B. Sample Collection**

Collect samples in one-liter glass or plastic containers. Samples from wadeable streams are collected near mid-stream, approximately 0.5 m beneath the surface. Composite, euphotic zone samples are collected from unwadeable streams, wetlands, lakes and impoundments using a Kemmerer or Van Dorn sampler. Skim surface scums formed by algal blooms for additional algal identification when present.

### C. Chlorophyll *a* Samples

For chlorophyll *a* analysis, filter 50 to 100 ml aliquots of the phytoplankton sample through 2.4 cm Whatman GF/C glass microfibre filters. Fold filters in half, with algal cells folded to the inside. If filtering is performed in the field, place filters in light-excluding containers, such as empty 35mm film containers, or wrap filters in aluminum foil and place them in a zip-lock bag. Preserve samples on ice in a cooler until they can be returned to the laboratory. As an alternative to filtering in the field, whole-water samples (1 L or greater) may be preserved on ice and filtered upon return to the laboratory. DO NOT add chemical preservatives to samples to be analyzed for chlorophyll *a*. Samples should be filtered within 24 hours of collection and immediately frozen.

## III. SAMPLE PRESERVATION

Samples for identification and enumeration are preserved with 3 to 4% buffered formalin, Lugols solution, 2% glutaraldehyde, M<sup>3</sup> fixative or 6-3-1 preservative. Samples should be labeled with waterbody name, location, site number, date and name of collector. Labels should be written in pencil or indelible ink and attached firmly to the container.

## IV. SAMPLE ANALYSIS

### A. Field Assessment

A *qualitative* field determination of surface algal bloom coverage is recorded on the field data sheet using the following criteria:

Total - 100% (bank to bank)

Dense - 60% to 99%

Moderate - 30% to 59%

Sparse – 1% to 29%

Absent - 0%

### B. Primary Productivity

If primary productivity studies take place, the procedures followed will be those described in the latest edition of Standard Methods (APHA 1992).

### C. Chlorophyll *a* Analysis

#### 1. Filtration, Extraction and Determination

Replicate samples will be analyzed using a Turner Designs Model 10 fluorometer, using U.S. EPA Method 445.0 (U.S. EPA 1992). The fluorometric method is more sensitive than the spectrophotometric method, and smaller sample volumes can be analyzed.

**a. Filtration**

Equipment:

Hand-operated or electric pump at a vacuum pressure of less than 6 in of mercury.

500 ml filter flask  
50 ml filter funnel  
2.4 cm Whatman GF/C glass microfibre filters

Procedure:

Phytoplankton - filter 50 to 100 ml of sample through a 2.4 cm Whatman GF/C filter.

Periphyton - scrape a glass microscope slide from periphytometer into a watch glass, rinsing the slide with distilled water. Filter the resulting solution, rinsing the watch glass with distilled water. Remove the filter from the funnel, fold it in half, and place it in a light-excluding container (e.g., 35 mm film container). Repeat the process for each replicate sample. For natural substrate samples, scrape a known area of substrate into a jar and record the volume of sample. Filter a subsample of 1 to 10 ml, depending upon the amount of algae present in the sample. Mechanical blending may be necessary to homogenize the sample if large mats or filaments are present. For storage, freeze filters at 4°C for no longer than 30 days.

**b. Extraction**

Reagents:

90% aqueous acetone (mix 900 ml reagent grade acetone with 100 ml distilled water)

Equipment:

Tissue grinder with teflon pestle  
Glass-grinding vessel with a round bottom that matches the teflon pestle  
Plastic centrifuge tubes, graduated

Procedure:

Place one filter in the grinding vessel and add 5 ml of acetone solution and macerate the filter. Pour the resulting solution into a centrifuge tube. Rinse the grinder with another 5 ml of the acetone solution and transfer to the centrifuge tube. Steep samples in a dark refrigerator for at least 2 hours, but no more than 24 hours. Centrifuge at 1000 g for 5 to 20 minutes to clarify the extract.

**c. Analysis**

Reagents:

90% aqueous acetone  
2 N HCl (for acid-corrected method)

Equipment:

Turner Designs model 10 Fluorometer equipped with chlorophyll kit filters and bulb (as recommended by the manufacturer)

Glass cuvette

Procedure:

Calibrate the fluorometer at each sensitivity level annually, or whenever filters or lamps are changed, using directions and samples obtained from the U.S. EPA Quality Assurance Lab in Cincinnati, Ohio. If necessary, prepare 0.1 or 0.01 dilutions of the sample if the chlorophyll *a* extract sample is too concentrated. Usually, phytoplankton samples from streams need no dilution. Phytoplankton from enriched streams or eutrophic lakes may need dilutions of 0.1, and periphyton samples may need to be diluted by a factor of 0.01.

**d. Acidification method for corrected chlorophyll *a*:**

Fill cuvette about 2/3 full with chlorophyll extract. Record the initial reading as R<sub>b</sub> (reading before acidification). Record the instrument sensitivity level and keep readings between 20 and 80 by either changing the sensitivity or diluting the sample. Remove the cuvette and add 3 drops of 2N HCl. Gently agitate, allowing approximately 60 seconds for mixing and degradation of chlorophyll *a* to phaeophytin, then record, from the same scale as before, R<sub>a</sub> (reading after acidification).

The formula used for calculating corrected chlorophyll *a* is:

$$\text{Chl } a = F_s \frac{r}{r - 1} (R_b - R_a)$$

where:

F<sub>s</sub> = calibration factor for each sensitivity level

R<sub>b</sub> = reading before acidification

R<sub>a</sub> = reading after acidification.

r = R<sub>b</sub>/R<sub>a</sub> (when the instrument is calibrated using pure chlorophyll standards of known concentration)

Record the results on a chlorophyll *a* bench sheet (Appendix D-7).

**e. Uncorrected chlorophyll *a***

To calculate chlorophyll *a* using narrow band filters, use the formula:

$$\text{Chl } a = \frac{F_s r R_b}{r - 1}$$

Acidification of the sample is not necessary if this formula is used.

Record the results on a chlorophyll *a* bench sheet (Appendix D-7).

## D. Identification and Enumeration

Phytoplankton is concentrated by settling, identified to the lowest possible taxon using appropriate taxonomic references, and if *quantitative* data are required, enumerated as follows:

1. Place an aliquot of concentrated sample into a calibrated counting chamber such as a Sedgewick-Rafter cell or a sedimentation cylinder with a standardized bottom area (inverted microscope technique).
2. Allow the sample to settle for an appropriate amount of time. For 1 ml of sample, allow a sedimentation time of one hour; for greater volumes of sample, increase the sedimentation time to ensure settling of all phytoplankton.
3. Identify and enumerate phytoplankton with a binocular compound microscope or an inverted phase-contrast microscope equipped with a standard Whipple grid. Examine sedimentation chambers at 400X on an inverted microscope. Examine Sedgewick-Rafter counting chambers at 200X on a compound microscope. A list of taxonomic references follows the text.
4. Count a minimum of 200 algal units for each sample. Report results for each taxon as a relative percentage of the total count (relative abundance). Algal density, biovolume, taxa richness, diversity, percent similarity or other metrics will be calculated when needed.

## V. INTERPRETATION OF PHYTOPLANKTON DATA

### A. Chlorophyll *a*

Chlorophyll *a* data are compared with other sites in the same drainage, reference reach sites, or intensive survey control sites. Chlorophyll *a* data will also be used for biological trend monitoring.

### B. Community Structure

Phytoplankton abundance will be used to determine the degree and extent of algal blooms. Metrics such as taxa richness, relative abundance, community similarity, diversity, or other appropriate measures of phytoplankton community health will be evaluated and compared to other sites in the drainage, reference or control sites, available historical data and appropriate scientific literature.

### C. Use Impairments

Algal blooms will be evaluated to determine use impairment for Aquatic Life, Recreational and Domestic Water Supply Uses, as well as the minimum criteria applicable to all surface waters. These uses are defined in Kentucky's surface water quality standards (401 KAR 5:031).

1. **Aquatic Life** - the autecology of dominant and abundant taxa and associations will be determined when information is available (Anonymous 1967, Bennet 1969, Collins and Weber 1978, Lowe 1974, Palmer 1977, Patrick 1977, Patrick et al. 1975, VanLandingham 1982, Whitford and Schumacher 1963). Presence of the following indicator groups will be recorded:

heterotrophic taxa - indicative of organically enriched conditions.

halophilic taxa - indicative of high salinities

acidophilic taxa - indicative of low pH

2. **Domestic Water Supply** - taste and odor algae are identified and enumerated when appropriate. Potentially toxic species will be identified and enumerated.
3. **Recreational Use** - algae will be identified and enumerated. Algal blooms and bloom potential will be evaluated using chlorophyll *a*, cell density or both types of data.

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# 8 - MACROINVERTEBRATES

## I. INTRODUCTION

### A. Macroinvertebrates as Environmental Indicators

Since the early 1900s, aquatic organisms have been used extensively in water quality monitoring and impact assessment (Cairns and Pratt 1993), and macroinvertebrate assemblages have proven to be very useful in detecting even the subtlest changes in habitat and water quality. Today, macroinvertebrates are used throughout the world in water quality assessment, as environmental indicators of biological integrity, to describe water quality conditions or health of the aquatic ecosystem, and to identify causes of impairment. The KDOW relies on the analyses of macroinvertebrate communities in use-attainment designations for sections 305b and 303d of the Clean Water Act (CWA) reporting, assessing specific effects of pollutants on water quality and for listing Exceptional Water designations to streams and rivers throughout the Commonwealth. KDOW defines benthic macroinvertebrates as organisms large enough to be seen by the unaided eye, can be retained by a U.S. Standard No. 30 sieve (28 mesh/inch, 600  $\mu\text{m}$  openings) and live at least part of their life cycle within or upon available substrates of a waterbody.

### B. Type of Collections

The WQB typically collects macroinvertebrates either to assess baseline water quality or to make a bioassessment of areas impacted by anthropogenic activities. In addition, a random survey approach is used to assess aquatic life use-support for streams in each watershed management unit (see Section 2.E.). Standardized, semi-quantitative collections are made at all sampling locations; however, precise quantitative data may be collected on a case-by-case basis or if biological monitoring requires more rigorous statistical analyses. In general, collection methods used by KDOW are similar to those discussed in Lenat (1988), Plafkin et al. (1989), Klemm et al. (1990), Eaton and Lenat (1991), USEPA (1997b) and Barbour et al. (1999).

### C. When to Sample

Adherence to sample index periods is very important for accurate bioassessments using macroinvertebrates since biocriteria are calibrated for seasonality. For *wadeable* and *non-wadeable* streams, sampling must occur from May through September. For *headwater* streams (<5  $\text{mi}^2$  in drainage area), sampling must take place from February through May. In some cases, sampling outside of these index periods is necessary to assess immediate impacts (e.g., chemical spills) or to adhere to specific guidelines set forth by the U.S. Fish and Wildlife Service and KDOW Standards and Specifications Section for trend monitoring and bioassessments in streams containing Federally Listed species. For routine bioassessments or baseline data collection, samples collected outside of these index periods may be considered unacceptable. Also, biological samples should not be collected during periods of excessively high or low flows or within two weeks of a scouring flow event.

## II. SAMPLING METHODS

The methods described in this section are modifications of the single and multi-habitat approaches outlined by Barbour et al. (1999). The methods vary somewhat among stream sizes and stream gradients because of differences in habitat types present. The WQB of KDOW must approve any deviation from the methods described below. For multiple-site intensive surveys of known impacts, and for comparison to regional reference conditions, the 1-m<sup>2</sup> kicknet samples should be kept separate from other habitat collections to facilitate data interpretation, provide for better diagnosis of impairment and minimize the influence of uncontrolled variables.

### A. Wadeable-Moderate/High Gradient

For macroinvertebrates, KDOW generally considers wadeable streams as 3<sup>rd</sup> order or larger and usually greater than 5 square miles (13 km<sup>2</sup>) in drainage area. A summary of collection techniques required for these types of streams is shown in Table 8-1. Wadeable low-gradient streams, high-gradient headwater streams and large non-wadeable rivers require different methods and are discussed in Sections B, C and D, respectively. For routine and intensive surveys, it is important that both riffle/run samples and multi-habitat sampling are conducted (see below). As a quick screening tool, a single habitat (riffle/run) sample may be taken to detect obvious impairment from reference conditions. This simplified method may be employed for identifying impacted areas for TMDL development.

#### 1. Riffle Sample

This technique samples the most important subhabitat (i.e., riffle) found in moderate to high-gradient streams. It requires using a 600 µm mesh, one meter wide net in moderate to fast current in areas with gravel to cobble substrate. Four (4) 0.25 m<sup>2</sup> samples are taken from mid-riffle or the thalweg (path of deepest thread of water), dislodging benthos by vigorously disturbing 0.25 m<sup>2</sup> (20 x 20 in.) in front of the net. Large rocks should be hand washed into the net. The contents of the net are then washed and all four samples are composited into a 600 µm mesh wash bucket. ***This sample must be kept separate from all other subhabitat collections.***

#### 2. Mutli-Habitat Sample

a) **Sweep Sample** - This sample involves sampling a variety of non-riffle habitats with the aid of an 800 x 900 µm mesh triangular or D-frame dipnet. Each habitat is sampled in at least three (3) replicates, where possible.

1) *Undercut banks/root mats* - sampled by placing a large rootwad into the triangular or D-frame dipnet and shaken vigorously. The contents are removed from the dipnet and placed into a mesh wash bucket. Note: if undercut banks are present in both run and pool areas, each is sampled separately with three replicates.

2) *Marginal emergent vegetation* (exclusive of *Justicia americana* beds) - sampled by thrusting (i.e., “jabbing”) the dipnet into the vegetation for ca. 1 m, and then sweeping through the area to collect dislodged organisms. Material is then rinsed in the wash bucket and any sticks, leaves and vegetation are thoroughly washed and inspected before discarding.

- 3) *Bedrock or slab-rock habitats* - sampled by placing the edge of the dipnet flush on the substrate, disturbing approximately 0.1 m<sup>2</sup> of area to dislodge attached organisms. Material is emptied into a wash bucket.
- 4) *Justicia americana (water willow) beds* - sampled by working the net through a 1 m section in a jabbing motion. The material is then emptied into a wash-bucket and any *J. americana* stems are thoroughly washed, inspected and discarded.
- 5) *Leaf Packs* - preferably “conditioned” (i.e., not new-fall material) where possible; samples are taken from a variety of locations (i.e., riffles, runs and pools) and placed into the wash-bucket. The material is thoroughly rinsed to dislodge organisms and then inspected and discarded.

**b) Silt, sand, and fine gravel**

- 1) *Sieving* - a U.S. No. 10 sieve is used to sort out larger invertebrates (e.g., mussels, burrowing mayflies, dragonfly larvae) from silt, sand and fine gravel by scooping the substrate to a depth of ca. 5 cm. A variety of collection sites are sampled in order to obtain 3 replicates in each substrate type (silt, sand and fine gravel).
- 2) *Netting* (optional) - a fine-mesh Surber sampler or a fine-mesh bag (300 µm) is used to collect sand and silt depositional areas by placing the net on the substrate and vigorously stirring the sediments in front of the net. An area of 0.1 m<sup>2</sup> is sampled for each replicate making sure, where possible, that replicates are taken from different depositional areas. The material is elutriated and sieved in a 300 µm nitex sampler constructed of PVC or other fine-meshed net.

**c) Aufwuchs sample** - small invertebrates associated with this habitat are obtained by washing a small amount of rocks, sticks, leaves, filamentous algae and moss into a medium-sized bucket half filled with water. The material is then elutriated and sieved with the nitex sampler.

**d) Rock Picking** - invertebrates are picked from 15 rocks (large cobble-small boulder size; 5 each from riffle, run and pool). Selected rocks are washed in a bucket half filled with water, then carefully inspected to remove invertebrates with fine-tipped forceps.

**e) Wood Sample** - pieces of submerged wood, adding ranging from roughly 3 to 6 meters (10 to 20 linear feet) and ranging from 5–15 cm (2–6 inches) in diameter, are individually rinsed into the wash-bucket. Pieces of wood are inspected for burrowers and crevice dwellers. Large diameter, well-aged logs should be inspected and hand-picked with fine-tipped forceps.

**3. Alternative Methods**

The following are methods that may be used for special studies, as approved by the WQB of KDOW.

**a) Modified Traveling-Kick Method (TKM)** - this is an adaptation of Hornig and Pollard's (1978) method. Three TKMs are taken on a transect across the stream at mid-riffle in the thalweg. In the event that the TKMs cannot be taken on a transect,

they will be collected diagonally in an upstream direction. A triangular or D-frame dipnet is placed on small cobble sizes or smaller sized substrate and moved in an upstream direction disturbing an area in front of the dipnet of 30 cm (1 ft). The distance covered in the Interior Plateau portion of the state is 1.5 m (5 ft) in 30 seconds, while the distance in the Eastern and Western Coal Fields and Jackson Purchase area is 3 m (10 ft) in one minute.

- b) *Surber Sampler*** - this sampler should be employed only when the riffles are shallow (20 cm or less) and the current is moderate. Three to four Surbers are taken on a transect across the stream at mid-riffle in the thalweg by methods outlined in Needham and Needham (1962) and Klemm et al. (1990). In the event that the Surbers cannot be taken on a transect, they will be collected diagonally in an upstream direction.
- c) *Modified Hester-Dendy Multiplate Sampler*** - three multiplates, as described by Fullner (1971), are attached by wire or cord to a flotation device and allowed to float 25–30 cm (1 ft) below the surface. The multiplates are collected at the end of three weeks during the summer in the Interior Plateau or six weeks in the spring or fall in the Interior Plateau and for six weeks in the rest of the state regardless of season.
- d) *Basket Sampler*** - these samplers, described by Mason et al. (1967), may be used in lieu of multiplates. The basket samplers should be filled with limestone rock of approximately 7.5 cm (3 in) in diameter or porcelain spheres of approximately the same diameter. Residence time is the same as the multiplates.
- e) *Drift Nets*** - these should be used in streams that have a current velocity of more than 0.5 m/sec (1.5 ft/sec). A minimum of two drift nets per station should be used. One net should be set 25 cm (10 in) from the surface of the river and one set 10 cm (4 in) below the surface of the water. The collection period should be from one to three hours. Data should be reported in number of organisms per 100 m<sup>3</sup> of flow. Additional information can be found in Klemm et al. (1990).
- f) *Dredges*** - these may be used in lentic-type environments in water depths greater than 1 m and collected on a transect. All dredge samples are to be washed in a 600 µm mesh bucket.
- g) *Nylon rope ladder*** - for freshwater mussel monitoring. This method was developed specifically to observe mussel populations or individual mussels in wadeable streams. It is used for long-term monitoring purposes. A rope ladder is constructed to form successive square-meter blocks, using ½ inch nylon rope and ¼ inch metal bars as rungs. Concrete rebar (½ X 36 in.) stakes driven downward into the stream banks serve as a permanent reference and attachment site for the ladder. The square meter blocks are numbered from left to right facing upstream. Mussel(s) located within each block are charted on paper to represent their location in the streambed.

**Table 8-1. Summary of sampling methods for wadeable, moderate/high-gradient streams.**

Technique	Sampling Device	Habitat	Replicates (composited)
1m <sup>2</sup> Kicknet*	Kick Seine/Mesh Bucket	Riffle	4- 0.25m <sup>2</sup>
Sweep Sample	Dipnet/Mesh Bucket	All Applicable	
Undercut Banks/Roots	"	"	3
Emergent Vegetation	"	"	3
Bedrock/Slabrock	"	"	3
<i>Justicia</i> beds	"	"	3
Leaf Packs	Dipnet/Mesh Bucket	Riffle-Run-Pool	3
Silt,Sand, Fine Gravel		Margins	
Coarse Seive	US No. 10 Seive		3
Fine Mesh	300 µm nitex net		3
Aufwuchs Sample	300 µm nitex net	Riffle-Run-Pool	3
Rock Pick	Forceps	Riffle-Run-Pool	15 rocks (5-5-5)
Wood Sample	Mesh Bucket	Riffle-Run-Pool	3-6 linear m

\*Sample contents kept separate from other habitats

## B. Wadeable Low-Gradient

Low-gradient streams are defined as streams that have velocities less than 0.013 m/sec (0.5 ft/sec) and naturally lack riffle habitat. The most productive habitats of these streams are typically woody snags, undercut banks and aquatic vegetation. A proportional sampling technique is used in most streams of the western parts of the state, particularly in ecoregions 72, 73 and 74. The method follows, in part, the Mid-Atlantic Coastal Plain Streams Workgroup (MACS) protocol (US EPA 1997a), which is also described in Barbour et al. (1999). Essentially, the technique is considered "proportional sampling," in which some predetermined number of sample units (20 in this case) is allocated among distinct and productive mesohabitats in relation to their proportion found within a 100 m stream reach.

A sample unit is called a "jab" in which a D- or A-frame net is thrust into the targeted habitat in a jabbing motion for approximately 0.5 m and then swept with the net 2 or 3 times to collect the dislodged organisms. For example, in a 100 m stream reach, if woody snags make up roughly 50% of the reach, submerged root mats 25%, and submerged macrophytes 25%, then 10 jabs are to be allocated to the snags, 5 jabs allocated to the root mats, and the last 5 jabs would be allocated to the macrophytes. If a jab becomes heavily clogged with debris and sediment, discard and repeat the jab. ***All material is composited*** into a wash bucket for further processing. Large leaves and twigs can be washed, inspected, and discarded to reduce the volume of the debris in the sample. Sand and sediment can be elutriated using a bucket and 600 µm sieve.

## C. Headwater High-Gradient

In these small streams, riffle habitat predominates and is the primary targeted habitat. Benthic invertebrates should be collected in the spring index period (mid-February to early June) as this period offers the highest potential for macroinvertebrate diversity and abundance in these headwater streams. A collection consists of a composited semi-quantitative riffle sample and a composited



multi-habitat sample. *These two sample types must be kept separate for a more effective diagnosis of impairment.* A summary of these collection techniques is shown in Table 8-2.

**1. Riffle Sample**

For the semi-quantitative sample, invertebrates are collected from four 0.25 m<sup>2</sup> quadrat kicknet samples stratified within the thalweg of cobble-boulder riffle habitat. This habitat is targeted to ensure the highest species richness and abundance of macroinvertebrates. The thalweg of a riffle also guarantees the most flow permanence and substrate stability in these often intermittent streams. Two kicknet samples are allocated to each of two distinct riffles that are separated by at least one pool or run. This is done to help reduce between-riffle variability. The four samples are composited into a 600 µm mesh wash bucket to yield a 1 m<sup>2</sup> semi-quantitative sample. The composited sample is partially field processed using a U.S. #30 sieve (600 µm) and wash bucket. Large stones, leaves and sticks are individually rinsed and inspected for organisms and then discarded. Small stones and sediment are removed by elutriation using the wash bucket and U.S. #30 sieve.

**2. Multi-Habitat Sample**

The multi-habitat, qualitative sample consists of a composite of 3 leafpacks, 3 jabs in sticks/wood, 3 jabs in soft sediments, 3 jabs into undercut banks/submerged roots, handpicking of 5 small pool boulders and approximately 2 linear feet of large woody debris. These techniques were described above in the wadeable Moderate/High-Gradient section.

<b>Technique</b>	<b>Sampling Device</b>	<b>Habitat</b>	<b>Replicates (composited)</b>
1m <sup>2</sup> Kicknet*	Kick Seine/Mesh Bucket	Riffle	4-0.25m <sup>2</sup>
Sweep Sample	Dipnet/Mesh Bucket	All Applicable	
Undercut Banks/Roots	Dipnet/Mesh Bucket		3
Sticks/Wood			3
Leaf Packs	Dipnet/Mesh Bucket	Riffle-Run-Pool	3
Silt,Sand, Fine Gravel	Dipnet/Mesh Bucket	Margins	3
Rock Pick	Forceps	Pool	5 sm. boulders
Wood Sample	Forceps/Mesh Bucket	Riffle-Run-Pool	2 linear m

\* Sample contents kept separate from other habitats

**D. Non-wadeable Streams**

Samples taken from non-wadeable (boatable) streams will utilize methods similar to those outlined for wadeable low-gradient streams (section 2.B.). A site will consist of a 300-meter stream reach. The proportional sampling technique, utilizing the 20 "jab" method discussed in Section 2B will be augmented with dredge samples, rock picking and a wood sample. Three dredge samples, taken with a petite ponar grab sampler, are taken from each of three transects; one dredge sample is taken from

mid-stream and one each from the right and left bank. The sample material is washed thoroughly in a 600 µm mesh wash bucket, then the macroinvertebrates are removed and stored in 70% ethanol. Where available, pick 15 rocks (large cobble-small boulder) and wash and pick 6 m of wood (5–15 cm in diameter).

#### **E. Probabilistic (Random) Bioassessments**

Probabilistic bioassessments incorporate most of the sampling protocols for macroinvertebrates described above. The stream reach to be assessed is defined as 40Xs the width of the stream. In sampling each stream, the random design restricts the investigator to available habitat offered at each reach. If the stream is a high-to moderate-gradient stream and the particular reach contains no riffle-run habitat (a rare occurrence), the macroinvertebrates will be collected using the 20-jab proportional sampling technique (see Section 2.B.). This method is employed routinely in coastal and low-gradient streams where riffle-run habitat does not occur. Otherwise, a kicknet is used to collect macroinvertebrates from the riffle habitat and a D- or A-frame dipnet is employed to collect macroinvertebrates from other habitats that may be available, such as roots/undercut banks, woody debris, aquatic vegetation and leaf packs. Collected benthos are partially field processed and preserved in 95% ethyl alcohol. Those organisms collected from the riffle habitat are kept separate from those collected in other habitats.

### **III. SAMPLE PROCESSING**

#### **A. Sorting**

In the field, large sticks and leaves are washed, inspected for organisms and discarded. Rocks should be elutriated and hand washed with a bucket and 600 µm sieve. This process is repeated until a manageable amount of debris and organisms (relative to size of sample container) can be preserved for laboratory sorting. Samples may be partially field picked in the field using a white pan and fine-tipped forceps. Club soda may be used to retard the movement of aquatic organisms when field sorting. Approximately a tablespoon of material to be sorted is placed in a white pan with a small amount of water. The material is dispersed evenly throughout the pan, and all macroinvertebrates are removed and placed in 70% ethanol.

Sorting in the laboratory is done with the aid of a circline lamp or dissecting scope against a white background. Staining invertebrates with either rose bengal or phloxine B at a concentration of 100 g/L of ethanol may be done to aid in sorting operations. Sugar floatation techniques may also be employed to facilitate sorting. While entire picks are routinely done at KDOW, unmanageable amounts of organisms and debris may be proportionally subsampled by dividing all of the material into 1/4<sup>th</sup>s, randomly choose one of the quarters and remove all organisms. If at least 300 organisms are not obtained from the subsample, pick additional quarters completely until the target number is reached. After subsampling, scan the remaining sample under low magnification for rare or large organisms not found in the original subsample. In addition, certain taxa (e.g., chironomids, hydropsyhid caddisflies) may be subsampled (10%, 20% or 25%) using gridded pans and a random numbers table.

#### **B. Preservation**

Samples are initially preserved in the field in 95% ethanol. Upon returning to the laboratory, all sorted samples are transferred to a fresh 70% ethanol solution.

#### **C. Labeling**

While at the sampling location, all macroinvertebrate samples will receive a label. The label may be placed in the sample jar or written directly on some portion of the jar. The label will include the site number, if known, stream name, location, county, date sampled and the collector's initials. A permanent label will be placed in the collection jar either when the sample is returned to the laboratory, or when it is identified. This label will include the site number, stream name, county, date sampled, latitude, longitude, mile point and collector names.

#### **D. Taxonomy**

All taxonomic identifications are made to the lowest practical level, using the most current taxonomic references available. A partial list of taxonomic references used by KDOW is found at the end of this chapter. A current master taxa list is found in Appendix D-1.

## IV. DATA ANALYSIS

The use of multiple community attributes to assess instream biological impairment has become widely accepted. This approach was first developed by Karr (1981) for midwest fish communities and now has been refined regionally and used throughout the United States. Karr's methods involved using a variety of community attributes, referred to as "metrics," to assess the condition of biological communities. Each metric is expected to contribute pertinent ecological information about the community under study. Examples of the application of the metric approach to aquatic macroinvertebrate communities can be found in Nuzzo (1986), Ohio Environmental Protection Agency (1987), Bode (1988), Shackelford (1988), Plafkin et al. (1989) and Barbour et al. (1999).

### A. Core Metrics

The KDOW's metric selection process uses statistical properties of redundancy and sensitivity to evaluate the power of metrics that can discriminate between impaired and unimpaired sites. Metric scoring criteria are established using percentiles of the reference and non-reference data distribution. The following is a list and explanation of each metric commonly used by KDOW. Note that metric combinations and scoring criteria may vary among ecoregions and stream sizes. **[Many of these metrics (e.g., mHBI, % composition metrics) require quantitative or semi-quantitative sampling (i.e., calculated from riffle kicknet sample, or 20-jab technique.)]**

- 1. Taxa Richness.** This refers to the total number of distinct taxa present in the composited sample (both semi-quantitative and qualitative samples combined). In general, increasing taxa richness reflects increasing water quality, habitat diversity and/or habitat suitability.
- 2. Ephemeroptera, Plecoptera, Trichoptera Richness (EPT).** This is the total number of distinct taxa (both semi-quantitative and qualitative samples combined) within the generally pollution-sensitive insect orders of Ephemeroptera, Plecoptera and Trichoptera found in the composited sample. This index value will usually increase with increasing water quality, habitat diversity and/or habitat suitability.
- 3. Modified Hilsenhoff Biotic Index (mHBI).** The HBI was developed to summarize the overall pollution tolerance of a benthic arthropod community with a single value (Klemm et al. 1990). Hilsenhoff (1977), using a range of 0-5, originally developed the index for Wisconsin riffle/run streams experiencing organic pollution. Hilsenhoff (1982, 1987) later refined the index, expanding the scale to range from 0 to 10. Plafkin et al. (1989) modified the index to include non-arthropod benthic macroinvertebrates. Hilsenhoff (1987) developed tolerance values for a variety of macroinvertebrates from Wisconsin, and Plafkin et al. (1989) added additional tolerance values. However, KDOW uses tolerance values developed by North Carolina Division of Environmental Management (NCDEM) (Lenat 1993) as well as values developed from KDOW data. These HBI values have been regionally modified for streams of the southeastern United States. Both Hilsenhoff (1988) and NCDEM have developed seasonal correction factors for the HBI. Several states, including Kentucky, have used the mHBI to assess impacts other than organic enrichment and found the mHBI to be a valuable metric. An increasing mHBI value indicates decreasing water quality. The formula for the HBI is as follows:

$$m\ HBI = \frac{\sum n_i \times a_i}{N}$$

where:

$n_i$  = number of individuals within a species (**maximum of 25**),

$a_i$  = tolerance value of the species,

$N$  = total number of organisms in the sample (**adjusted for  $n_i \geq 25$** ).

4. **Modified Percent EPT Abundance (m%EPT).** This metric measures the abundance of the generally pollution-sensitive insect orders of Ephemeroptera, Plecoptera and Trichoptera. The relatively tolerant and ubiquitous caddisfly *Cheumatopsyche* is excluded from the calculation. Increasing values indicate increasing water quality and/or habitat conditions.
5. **Percent Ephemeroptera (%Ephem).** The relative abundance of mayflies is calculated to show impacts of metals and high conductivity associated with mining and oil well impacts. Ephemeroptera abundance normally declines in the presence of brine and metal contamination.
6. **Percent Chironomidae+Oligochaeta (%Chir+%Olig).** This metric measures the relative abundance of these generally pollution tolerant organisms. Increasing abundance of these groups suggests decreasing water quality conditions.
7. **Percent Primary Clingers (%Clingers).** This habit metric measures the relative abundance of those organisms that need hard, silt-free substrates to "cling" to. Merritt and Cummins (1996) and Barbour et al. (1999) list habits for most insect genera. Habit information for non-insect taxa can be determined from Pennak (1989), Thorp and Covich (1991) and Barbour et al. (1999).

## B. Supplemental Metrics

These metrics may be used in special situations such as intensive surveys and nonpoint source studies that evaluate a particular impact. Functional feeding group abundances may also be calculated to provide insight into food/energy relationships among sites.

1. **Jaccard Coefficient of Community Similarity.** This similarity index measures the degree of taxonomic similarity between two stations in terms of taxon presence or absence. Coefficient values, ranging from 0 to 1.0, increase as the similarity with the reference station increases. The formula is as follows:

$$Jaccard\ Coefficient = \frac{c}{a + b + c}$$

where:

a = number of taxa in sample A but not B

b = number of taxa in sample B but not A

c = number of taxa common to both samples

Sample A = reference station

Sample B = station of comparison

2. **Percent Community Similarity (PSc).** The PSc index, discussed by Whittaker (1952), was used by Whittaker and Fairbanks (1958) to compare planktonic copepod communities. It is a good index in bioassessments because it shows community similarities based on relative abundance, thus, giving more weight to dominant taxa than rare ones. Percent community similarity values range from 0 (no similarity) to 100 percent. The formula for calculating PSc is:

$$PSc = 100 - 0.5 \sum(a-b) = \sum \min(a,b)$$

Where:

a = percentage of taxa a in sample a

b = percentage of taxa b in sample b

3. **Percent Contribution of Dominant Taxa (PCD<sub>5</sub>).** This simple measure of redundancy and evenness adds the relative abundance percentages of the five dominant taxa. Highly redundant communities (i.e., communities highly dominated by a few taxa) may reflect a degraded condition.
4. **EPT/Chironomidae (Ratio of EPT to Chironomidae Abundances).** This metric uses the ratio of these indicator groups as a measure of community balance. Communities with a good biotic condition would be expected to have a substantial representation of EPT taxa. Skewed populations having a disproportionate number of generally tolerant chironomids relative to the more sensitive insect groups may indicate environmental stress (Ferrington 1987).
5. **Dominants in Common, Five (DIC<sub>5</sub>).** This metric, developed by Shackelford (1988), measures the similarity of a reference station and a station of comparison based on the five most abundant taxa at each station. The DIC<sub>5</sub> provides a measurement of substitution between the reference community and the downstream station.
6. **Dominants in Common, Ten (DIC<sub>10</sub>).** This metric, also developed by Shackelford (1988), is the same as the DIC<sub>5</sub> metric, but is based on the ten most abundant taxa at each station.
7. **Percentage of *Cricotopus*+*Chironomus* Abundance to Total Chironomidae (Cr+Ch /Chironomidae).** This measures the abundance of the pollution-tolerant genera *Cricotopus* and *Chironomus* to the total abundance of the family Chironomidae.

Additional information on various metrics may be found in Plafkin et al. (1989), Klemm et al. (1990), Resh and Jackson (1993) and Barbour et al. (1992, 1999). The functional feeding group concept is discussed by Cummins (1973), and genus-level functional feeding group designations for aquatic insects are provided by Merritt and Cummins (1996).

### C. Macroinvertebrate Bioassessment Index

Macroinvertebrate data analysis for wadeable or headwater streams is accomplished by using the multimetric approach. Metric criteria are based on reference reach data and may vary from region to region and with stream size (i.e., wadeable or headwater). Core metrics in wadeable streams are

Taxa Richness, modified Hilsenhoff Biotic Index, EPT Richness, modified %EPT Abundance and %Chironomidae+Oligochaeta Abundance and % Clinger Abundance. Additionally, supplemental metrics are used depending on type of impact, stream size, subcoregion, etc. Each metric is given a calculated score (range 0–100) based on the percent of the standard metric value (i.e., the 95<sup>th</sup> percentile or 5<sup>th</sup> percentile). These percentile thresholds are used to eliminate outliers. The formulae for calculating MBI scores are shown in Table 8–3. The individual metric scores are summed and then averaged to produce a Macroinvertebrate Bioassessment Index (MBI) value. Thresholds are established to assign narrative water-quality ranking of Excellent, Good, Fair, Poor and Very Poor. The KDOW is in the process of calibrating, or refining, metric scoring criteria for ecoregions and stream sizes. A separate document detailing the metric selection and evaluation process and bioassessment interpretation is currently in preparation. Contact the Ecological Support Section of the WQB for current 95<sup>th</sup> %ile and 5<sup>th</sup> %ile values.

<b>Table 8-3. Examples of metric scoring formulae for the Macroinvertebrate Bioassessment Index.</b>	
<b>Metric</b>	<b>Formula</b>
<b>TR</b>	$\frac{TR}{95th\%ile} \times 100$
<b>EPT</b>	$\frac{EPT}{95th\%ile} \times 100$
<b>MHBI</b>	$\frac{10 - mHBI}{10 - 5th\%ile} \times 100$
<b>m% EPT</b>	$\frac{m\% EPT}{95th\%ile} \times 100$
<b>%Clingers</b>	$\frac{\%Clingers}{95th\%ile} \times 100$
<b>% Chir+Olig</b>	$\frac{100 - \%Chir + Olig}{100 - 5th\%ile} \times 100$

In probabilistic (random) sampling, the MBI may not be representative in cases when atypical habitat is sampled. For example, in streams where riffle-run habitat typically occurs but by chance this habitat did not occur in the randomly chosen stream reach, the sampling techniques and the habitat assessment must be considered in making the final aquatic life use determination. Otherwise, the evaluation is made solely using the MBI.

#### **D. Quality Assurance/Quality Control (QA/QC)**

The following is a list of QA/QC procedures that will be followed for the collection, sorting and identification of macroinvertebrate samples. All personnel involved in the collection and identification of macroinvertebrates will have prior educational qualifications as well as appropriate in-house training.

1. **Sample Collection.** Five percent of samples collected by KDOW will be duplicated to evaluate precision and repeatability of the technique and/or the sampling crew. The field crew will select a stream reach that has sufficient habitat to accommodate a duplicate sample. After the duplicate sample has been taken, with the appropriate sampling methods for that stream reach, it will be labeled as the duplicate and processed with the rest of the samples. After any sampling has been completed, all sampling gear (nets, sieves, buckets, etc.) will be thoroughly cleaned to remove all macroinvertebrates so that specimens are not carried over to the next site. The equipment should be examined just prior to sampling the next site to insure that no macroinvertebrates are carried over.
2. **Laboratory Sorting.** Ten percent of the pans sorted will be examined by a second qualified biologist to insure that all organisms are being removed from the sorting pan or grid. If less than 10 organisms are found in the pan or grid, the sample is considered passed. If more than 10 organisms are found, then the sample fails and another successive pan will be checked. This will continue until the sorter passes the procedure. The sorting pans will be thoroughly rinsed after each pan is picked.
3. **Identification.** Five percent of samples collected by KDOW will be re-identified by a second taxonomist to evaluate precision and accuracy. A target of 90% similarity in taxonomic composition is acceptable. A third taxonomist will reconcile differences in identifications between the two taxonomists, if necessary. A voucher collection of macroinvertebrates will be maintained by the WQB of KDOW. If possible, three to five specimens of the vouchered taxon will be placed in the vial. These specimens will be properly labeled, preserved and stored in the WQB laboratory for future reference. Rare and/or difficult taxa will be verified by other WQB biologists or noted taxonomic experts.
  - a) A macroinvertebrate bench sheet (Appendix D-2) shall be produced during the sample identification process. After the sample has been identified, the data will be entered into the WQB database.
  - b) When available, KDOW biologists will take part in suitable macroinvertebrate identification training (offered by U.S. EPA, state universities and other groups) as this will help ensure consistent and accurate macroinvertebrate identification. In addition, the WQB will maintain a library and list of taxonomic references.

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# 9 - FISH

## *FISH COMMUNITY STRUCTURE*

### I. INTRODUCTION

The evaluation of fish community structure is an important component of biological monitoring, intensive surveys, Exceptional Water determinations and reference reach surveys, all of which provide reliable assessments for the Clean Water Act (CWA) Section 305b. The primary goal of evaluating fish community structure to ensure accurate assessments for the CWA is to perform a fish Index of Biotic Integrity (IBI) on the sampling event. Advantages of using fish as biological indicators include their widespread distribution from small streams to all but the most polluted waters, their utilization of a variety of trophic levels, their stable populations during summer months and the availability of extensive life history information (Karr et al. 1986). The methods used for collecting, analyzing and reporting of fish community structure data are outlined in this section.

### II. SAMPLING METHODS

Standardization of methods is essential since it will allow streams to be assessed and compared, therefore, establishing trends and long-term monitoring (Bonar and Hubert 2001). To ensure collection of standardized fish community data, stream size (i.e., drainage area) has been used to designate streams to two classes, headwater and wadeable, and a set methodology is outlined for each classification. For fish assessments, headwater streams are streams with a drainage area less than 12 mi.<sup>2</sup> (30.1 km<sup>2</sup>). Wadeable streams are streams with a drainage area greater than 12 mi.<sup>2</sup> (30.1 km<sup>2</sup>). However, streams between the drainages of 8-12 mi.<sup>2</sup> (20.7-30.1 km<sup>2</sup>) fall within a “gray” area of stream classification and best professional judgment should be used to determine if the stream is headwater or wadeable.

The sampling index period is **mid-March through October**. Prior to sampling, the field crew should research the distributional records and familiarize themselves with the possible ichthyofauna for a particular basin and compile a species list for that basin. Special awareness and precautions should be made for the possibility of any Federal Threatened/Endangered Species encountered.

#### A. Headwaters

The sample reach must range between 100-125 meters in length, which should consist of riffles, runs and pools if they are present. Also, the reach must not be associated within the immediate area (<150 meters) of major tributary confluences or human structural influences, such as bridges, road crossings (fords), low head dams or any other similar structure, unless the purpose of obtaining the fish community data is related to these influences. Sampling in headwater streams must consist of using a backpack electrofisher unit working in an upstream manner (**collectors must be familiar with and follow all safety procedures as suggested by the manufacturer**). The electrofishing duration within the sample reach should be a minimum of 600 “shocking” seconds and a maximum of 1000 “shocking” seconds.

The lower and upper ends of the reach should be associated with a natural instream barrier such as a riffle. The sampling crew should consist of a minimum of two members with at least one member having prior electrofishing experience. The crew should work upstream shocking in a side-to-side/bank-to-bank sweeping technique. The crew collects the stunned fish with dip nets and places them into a livewell/bucket for preservation or identification after the reach has been sampled. One pass of the reach is sampled from the downstream end to the upstream end with all recognizable habitats thoroughly sampled, following the sweeping technique (Barbour et al. 1999). In streams with larger pool habitat, a seine should be used to sample the area more efficiently. The experience of the crew and their ability to see and net the fish improves the effectiveness of sampling the reach. Polarized sunglasses are optional, since they will cut down on the glare of the water. Also, physical features such as water clarity, flow and depth and time of day need to be considered to obtain optimal success in sampling. If proper conditions are not present sampling needs to be postponed until an efficient sampling effort can be made.

## **B. Wadeable**

Wadeable streams are larger bodies of water and provide more variability of habitat than headwater streams, so the reach sampled will be determined by a greater time designation within the given length designation. Because of that variability, a combination of seining and electrofishing is used, since both methods have advantages and disadvantages in habitat types and species groups. The combination of seining and electrofishing yields better results than with one technique independently (Onorato et al. 1998; Yoder and Smith 1999). The collectors should be aware of the advantages and shortcomings of each technique. KDOW has observed electrofishing to be more effective in streams that may have numerous boulders and/or undercut banks and/or woody debris. KDOW also has observed that electrofishing tends to be biased toward catostomid and centrarchid members while not fully representing the schools of cyprinids in large pools. However, cyprinids can be effectively sampled with a seine in large pool habitats to yield a better representation of their presence in the community (Onorato et al. 1998).

The reach length should be a minimum of 100 meters and should not exceed 200 meters (documentation of the sample reach should be noted). The sample reach should consist of at least two riffles, runs and pools each. In cases where two riffles, runs and pools cannot be sampled, either one riffle, run and pool is sampled or the recommended reach length of the stream is sampled. Sampling must be done using a seine (3.4 × 1.8 m with 0.3 cm mesh or any other suitable size) and an electrofisher (any suitable generator or battery powered electrofisher may be used). A seine is used for approximately 30-60 minutes from start to finish. The electrofisher (backpack, tote barge or similar units) is typically used in areas not efficiently sampled with the seine (e.g., root masses, undercut banks, rock slabs, boulder/cobble substrates, fallen trees, etc). The electrofisher should be used for at least 600 “shocking” seconds to a maximum of 1800 “shocking” seconds of effort. Also, as in headwater streams, the electrofishing method should be done in a sweeping manner, and the reach must not be associated within the immediate area (<150 meters) of major tributary confluences or human structural influences. Best professional judgment by the collector is used to determine the appropriate gear and effort for a particular habitat, but a combination of the two methods (seine and electrofisher) must be used. Typically a reach is thoroughly sampled when no new species are being collected and all habitats have been sampled.



Documentation of methodology is required to help provide insight into the results of the sampling effort, particularly if only one technique is used. Again the efficiency of sampling depends on the experience of the crew and their ability to see and net the fish. Polarized sunglasses are optional. The water clarity, flow and depth and the time of day need to be considered to obtain optimal success in sampling. If these conditions are not adequate and/or practical, sampling needs to be postponed until an efficient sampling effort can be made.

### **C. Preservation**

Fish collections are preserved in the field with a 10%-15% buffered formalin solution. Large specimens are to be identified in the field, recorded and released, unless the specimen(s) represent a significant ichthyological find (e.g., state or drainage record), then they are to be preserved as voucher specimens. Easily identified fish that are collected in large numbers (i.e., *Campostoma* spp.) are also recorded in the field and released. Photographs of large specimens and/or vouchers of all released specimens need to be made for verification. If possible, at least five specimens of each species released should be kept as vouchers from the sample event. If a species or genus is viewed but not collected and if positively identified, these records should be noted (i.e., *Hypentelium nigricans*, *Micropterus* spp. or *Lepomis* spp.).

## **III. LABORATORY PROCESSING**

Fix fish in formalin for at least 2 weeks (larger fish should be fixed for at least 3–4 weeks unless injected with a formalin solution in the field). Samples are then rinsed and soaked for 1–2 days in tapwater. Transfer fish to 70% ethanol solution for long-term preservation and storage. Identify and count fish using all available taxonomic keys and distribution records. In a given sampling season, 10% of the samples collected should be re-identified by another qualified/trained fish taxonomist for the purpose of quality assurance/quality control. Independent taxonomic verifications of selected species are obtained, when needed, from recognized experts (e.g., Ecological Support Section KDOW or Kentucky State Nature Preserves Commission).

Species and number of individuals collected are recorded on data sheets (Appendix E-1) and used for evaluating species composition, relative abundance, species richness and presence/absence of indicator species. The occurrence of species that are listed as rare, endangered or threatened by federal or state agencies is also documented, and the appropriate agencies are notified with the Threatened/Endangered Species Report Form (Appendix E-2). Other characteristics of fish communities that are evaluated, as needed, include the presence of hybrids, size/age distribution of populations, incidence of disease and occurrence of parasites. These and other non-routine assessments of fish communities are included in specific study plans.

## **IV. QUALITY ASSURANCE AND QUALITY CONTROL**

### **A. Quality Assurance and Quality Control in the field (Barbour et al. 1999):**

A field crew will consist of at least one trained ichthyologist. The ichthyologist is to make sure voucher representatives (5 specimens, if possible) of all fish are taken, and all specimens which are in question of identification are preserved correctly for laboratory examination. All released

specimens will be noted. Collection labels are to be accurate and complete with all pertinent information that was previously discussed.

In a sampling season 5% of the samples will be duplicated/replicated by a field crew. The duplicate/replicate samples will be taken either at the exact sample site within a two-week period or taken on the same day within the sample reach by either the same field crew or an alternate crew to ensure precision and repeatability of the sampling technique.

## **B. Quality Assurance and Quality Control in the laboratory (Barbour et al. 1999)**

All samples will be preserved correctly to ensure optimal condition of specimens. Label information must be accurate and complete. The ichthyologist will be trained and up to date with the current identification and nomenclature of Kentucky fishes. The use of all pertinent keys and distributional records (see taxonomic references) is required and final identification will be cross-referenced with the KDOW voucher collection. Ichthyologists should discuss any problem identifications with other in-house ichthyologists. Recognized taxonomic experts will assist in difficult specimens and confirmation of specimens. Ichthyologists will participate in fish identification workshops, seminars and training sessions when opportunities arise.

In a sampling season 10% of the collections will be re-identified and enumerated by another ichthyologist. The re-identification and enumeration of a collection should exceed 90% accuracy. Duplicate samples should exceed 75% similarity (Whittaker 1952). Duplicate/replicate samples will be processed and analyzed in the same procedures as regular samples.

## **V. DATA ANALYSIS**

### **A. Index of Biotic Integrity**

The Index of Biotic Integrity (IBI) as described by Karr (1981) was used to assess the fish community structure and biotic integrity of midwestern streams. This IBI was composed of 12 equally weighted metrics that were grouped into three general categories: Species Richness and Composition (Category I), Trophic Composition (Category II), and Fish Abundance and Condition (Category III). Each metric was assigned a 5, 3 or 1 value depending upon whether the obtained value strongly approximates the expected value (5), somewhat approximates the expected value (3) or does not approximate the expected value (1). The individual metric scores were summed and a total IBI score ranging from 12–60 was achieved. Metrics in Category I often vary with region and stream size, while less variation was usually found among those in Categories II and III (Karr et al. 1986). Five classifications based on total IBI scores were assigned by Karr (1981) to describe the quality of the fish community at each site. Scores that fall between categories were evaluated based on professional experience.

While the theory of using a multimetric fish index to assess stream health has sustained and prospered over time, numerous modifications and testing of the original IBI have been made to provide more accuracy and precision within a particular region and/or water body type (Ohio 1987, Minns et al. 1994; Barbour et al. 1999, Hughes and Oberdorff 1999, Smogor and Angermeier 2001). In the early 1990s KDOW started a Reference Reach Program to document

natural patterns in the biota and to develop biocriteria among the Commonwealth's lotic systems (KDOW 1997). However, gaps exist in the state database; therefore, further collection and data analysis is needed. Given the existing data and following recent IBI framework approaches (Barbour et al. 1999, Simon [ed.] 1999, McCormick et al. 2001, Smogor and Angermeier 2001), the IBI is being modified from the KDOW (1997) version to provide statewide coverage, easier application and better precision among users.

## B. Metrics

Fish assemblages in the state have shown strong correlation with ecoregions, basins, physiographic regions and stream size. Development of criteria for an IBI must be region- and stream size-specific to correspond with the differences within the ecoregion/basin framework (Fausch et al. 1984, Angermeier et al. 2000). Therefore, several candidate metrics were tested and validated for their sensitivity and variation among each region and stream size following methodology found in Smogor and Angermeier (1999) and McCormick et al. (2001). Metrics were selected to demonstrate attributes of a fish community that show responsiveness to disturbances, predictability and uniformity throughout the state. A list of candidate metrics is provided in Table 9-1. The metric selection process uses statistical properties of redundancy and sensitivity to evaluate the discriminating power of a metric between impaired and relatively unimpaired sites (reference sites). After testing the candidate metrics, eight metrics were selected to structure the multimetric IBI. The following list is an explanation of each metric used in the Kentucky IBI. A master list of Kentucky fishes with their trophic and tolerance classifications is provided in Appendix E-3. The ecological classifications in the master taxa list has been compiled using scientific literature, historic data, consultation with fisheries biologists, and professional experience (Ohio EPA 1987, Etnier and Starnes 1993, KDOW 1997, Goldstein and Simon 1999, McCormick et al. 2001). Any necessary modifications to the list will be made as additional information becomes available

- 1. Native Species Richness (NS).** This is the total number of native species present in a sample. Native richness is expected to decline with impairment. Non-native species are excluded since they tend to invade with impairment. This metric will usually increase with increasing water quality, habitat diversity and/or habitat suitability.
- 2. Darter, Madtom and Sculpin Richness (DMS).** This is the total number of the species in a sample within the tribe Etheostomatini (darters), the genus *Noturus* (madtoms) and the genus *Cottus* (sculpins). These groups are generally sensitive to pollution and are expected to decline with impairment. This metric will usually increase with increasing water quality, habitat diversity and/or habitat suitability.
- 3. Intolerant Species Richness (INT).** This metric represents the total number of intolerant species collected in a sample. Intolerant species are expected to decline with impairment. This metric will usually increase with increasing water quality, habitat diversity and/or habitat suitability.
- 4. Water Column Species Richness (WC).** This metric is a combination of four metrics. It is the total number of species, excluding tolerant members, from the family Cyprinidae along with the

Sunfish, Top Carnivore and Sucker metrics used in KDOW (1997). This metric is expected to decline with impairment. This metric will usually increase with increasing water quality, habitat diversity and/or habitat suitability.

5. **Simple Lithophilic Spawning Species Richness (SL).** This metric is the total number of simple lithophilic spawning species. This metric represents species that require relatively clean gravel and exhibit simple spawning behavior (Ohio 1987, Simon 1991). The metric is considered a habitat metric and is expected to decline with impairment and be particularly sensitive to siltation (Berkman and Rabeni 1987).
6. **Proportion of Insectivorous Individuals (% INST).** This metric is the relative abundance of insectivorous individuals excluding tolerant individuals. This metric represents a proportion of individuals that are fairly sensitive and feed primarily on insects. The metric is expected to decline with impairment. This metric will usually increase with increasing water quality, habitat diversity and/or habitat suitability.
7. **Proportion of Omnivorous Individuals (% OMNI).** This metric is the relative abundance of omnivorous individuals. This metric represents a proportion of individuals that are considered generalist in their feeding behavior. This metric is expected to increase with impairment. This metric will usually increase with decreasing water quality, habitat diversity and/or habitat suitability.
8. **Proportion of Tolerant Individuals (% TOL).** This metric is the relative abundance of tolerant individuals. This metric is to represent a proportion of individuals that are pollution tolerant and invade with impairment. Therefore, this metric is expected to increase with impairment. This metric will usually increase with decreasing water quality, habitat diversity and/or habitat suitability.

**Table 9-1: Candidate Metrics for the Index of Biotic Integrity (IBI)<sup>1</sup>**

<b>Metrics</b>	<b>Description</b>	<b>Response</b>
<b>Total Number of Species</b>	Measures the number of species	Decreases
Total Number of Native Species	Measures the number of native species	Decreases
Number of DMS Species	Measures the number of Darter, Madtom and Sculpin Species	Decreases
<b>Number of Darter Species</b>	Measures the number of Darter Species	Decreases
<b>Number of Sunfish Species</b>	Measures the number of Sunfish Species	Decreases
<b>Number of Intolerant Species</b>	Measures the number of Intolerant Species	Decreases
Number of Cyprinid Species	Measures the number of Minnow Species	Decreases
<b>Number of Sucker Species</b>	Measures the number of Sucker Species	Decreases
Number of Simple Lithophilic Spawning Species	Measures the number of Simple Lithophilic Spawning Species	Decreases
Number of Top Carnivores	Measures the number of Top Carnivores Species	Decreases
Percent Tolerant Species	Relative abundance of tolerant individuals	Increase

**Table 9-1: Candidate Metrics for the Index of Biotic Integrity (IBI)<sup>1</sup>**

<b>Metrics</b>	<b>Description</b>	<b>Response</b>
<b>Percent Omnivorous Species</b>	Relative abundance of individuals as generalist feeders	Decreases
Percent Insectivorous Species	Relative abundance of individuals as insect dominant Feeders	Decreases
Percent Darter Individuals	Relative abundance of darter individuals	Decreases
Number of Tolerant Species	Measures the number of Tolerant Species	Increases
<b>Percent Green Sunfish</b>	Relative abundance of Green Sunfish individuals	Increases
<b>Total Number of Individuals</b>	Measures the number of individuals collected (catch/effort)	Decreases
<b>Percent Top carnivores</b>	Relative abundance of individuals as top carnivores	Decreases
Percent Dace Individuals	Relative abundance of <i>Clinostomus</i> , <i>Phoxinus</i> and <i>Rhinichthys</i>	Decreases
Number of Darter and Sculpin Species	Measures the number of Darter and Sculpin Species	Decreases
Number of Darter and Madtom Species	Measures the number of Darter and Madtom Species	Decreases
Number of Headwater Species	Measures the number of Headwater Species	Decreases
Percent Headwater Species	Relative abundance of Headwater Species	Decreases
<b>Percent Insectivorous Cyprinids</b>	Relative abundance of Insectivorous Cyprinid Individuals	Decreases
Percent Pioneering Species	Relative abundance of Pioneering individuals	Increases
Percent Simple Lithophilic Spawning Species	Relative abundance of Simple Lithophilic spawning Individuals	Decreases
<b>Percent Hybrids</b>	Relative abundance of hybrid individuals	Increases
Percent Intolerant Species	Relative abundance of intolerant individuals	Decreases
<b>Percent Diseased Individuals</b>	Relative abundance of individuals with diseases, deformities, tumors and/or fin and skeletal anomalies	Increases
Percent Stoneroller Individuals	Relative abundance of <i>Campostoma</i> individuals	Increases
Percent Suckers	Relative abundance of Round-bodied Sucker individuals	Decreases
Percent DMS Species	Relative abundance of Darter, Madtom and Sculpin individuals of the community	Decreases
Number of Water Column Species	Measures the number of cyprinids, sunfish, top carnivores and catostomids, excluding tolerant members	Decreases
Number of Pool Species	Measures the number of Cyprinidae, Catostomidae and Centrarchidae Species, excluding tolerant members	Decreases

<sup>1</sup> **Bolded Metrics from Karr (1981)**

**Table 9-2: Metrics Used in the Kentucky IBI**

<b>SPECIES RICHNESS AND COMPOSITION</b>		<b>Response to Disturbances</b>
1.	Richness of native fish species (NS)	Negative
2.	Richness of darter, madtom, sculpin species (DMS)	Negative
3.	Richness of intolerant species (INT)	Negative
4.	Richness of water column species (WC)	Negative
<b>TROPHIC COMPOSITION</b>		
5.	Proportion of individuals as insectivores (% INST)	Positive
6.	Proportion of individuals as omnivores (% OMNI)	Negative
<b>FISH ABUNDANCE AND CONDITION</b>		
7.	Richness of simple lithophilic spawning species (SL)	Negative
8.	Proportion of individuals as tolerant species (% TOL)	Positive

### **C. IBI Criteria**

Currently, criteria are being developed, calibrated for drainage area, updated and tested for the various regions in the state using the 8 metrics. Each metric is given a calculated score (range 0–100) based on the percent of the standard metric value (i.e., the 95<sup>th</sup> percentile or 5<sup>th</sup> percentile predicted from the reference database). These percentile thresholds are used to eliminate outliers. The individual metric scores are summed and then averaged to produce an Index of Biotic Integrity (IBI) score. Thresholds are established to assign narrative water-quality ranking of Excellent, Good, Fair, Poor and Very Poor. The KDOW is in the process of calibrating, or refining metric scoring criteria for ecoregions and stream sizes. A separate document detailing the metric selection, the evaluation process and the bioassessment interpretation is currently in preparation. When available, the criteria for the state and/or a particular region can be obtained from the Ecological Support Section upon request.

## **VI. REPORTING FISH DATA**

### **A. Community Structure Data**

The condition of the fish community and the related stream health is determined by calculating an IBI for the site. Relative abundance, species composition, richness, the evaluation of species tolerances to environmental perturbations and the condition of fishes are all attributes that are factored into the IBI score in the form of metrics. Shifts in these respective attributes within the community should indicate a change in the stream condition, positive or negative. In addition, the presence or absence of indicator species and the presence of rare or endangered species should provide insight into the stream condition and the causes for respective changes or condition. Correlations with habitat, landuse, water quality and any other pertinent information should be made to interpret the IBI score and to provide intuitive information to the stream ecosystem function and condition. The age and size class distribution of species populations should be noted and assessed in relation to recruitment potential; spawning and nursery area availability should also be considered. Any limiting factors, either natural or human-induced, are reported.

## **B. Distribution Data**

Monitoring of the fish community structure is useful for stream health and fish populations as a whole, but may be too broad for the monitoring of a particular species. Therefore, reliable records from collections can provide useful status information (e.g., rare, common, etc.) for a given species in the state. Considerable data are available on fish distribution in Kentucky (Burr and Warren 1986, KDOW data) and environmental requirements of many of the more common species. Distribution is affected by many natural factors, including evolution of drainage patterns, stream order, substrate, temperature, cover availability, gradient, current velocity, seasonal flow variability and presence and abundance of food organisms (Maret 1999 and Strange 1999). Fish have inherent ranges of tolerances for many of these natural factors. A variation outside these ranges of tolerance may be the result of detrimental conditions that are reflected in the presence, structure and relative abundance of fish populations. Therefore, a compilation of all existing ichthyological data (both historic and present fish population data and field survey data) is necessary to assist in the management of a species or species groups. Also, physical and chemical characteristics of the stream are assessed in order to determine habitat availability and suitability for a species. All of these aspects help provide insight into the status and condition of a given species population.

## **C. Conclusions**

Conclusions based on all existing data for a site are drawn, and recommendations or subsequent corrective actions are made, as applicable. Conclusions should take into account the relations of the surrounding land use, in-stream habitat (micro and macro), water chemistry, collecting conditions, watershed history and other suitable factors. Fish collection data may be compiled and published as a separate technical report as necessary.

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# 10 - FISH AND MACROINVERTEBRATE CONTAMINANT ANALYSIS

## *FISH CONTAMINANT ANALYSIS*

### I. INTRODUCTION

Kentucky's watersheds, which include streams, rivers and lakes/reservoirs, have the potential to become polluted. Pollutants enter the watershed through permitted point source discharges (e.g., industrial and municipal facilities) and nonpoint sources (e.g., agricultural practices, urban runoff and atmospheric deposition). The pollutants may bioaccumulate in organisms living in these waters. These organisms provide a good method to monitor for potential pollution problems in the system. Contaminant analyses are performed on fish in these watersheds to provide background information on contaminant concentrations and to indicate potentially harmful concentrations. When harmful concentrations of contaminants are found, the data are compared to the respective consumption concentration guidelines for issuing risk-based advisories. In the United States, states have responsibilities for issuing fish consumption advisories for the purpose of protecting citizens from the harmful effects of eating fish contaminated with dangerous levels of chemicals introduced by environmental pollution. The following information includes the standardized methods for obtaining fish samples that can be used for issuing the advisories.

### II. FIELD SAMPLING PROCEDURES

Fish specimens for contaminant analysis are collected at various sites within Kentucky's watersheds including streams, rivers and lakes/reservoirs. An attempt is made to collect composite samples of target species at each sampling site to facilitate comparisons between sites.

#### 1. Target Species

For all biological sampling events, an initial list of target species may be developed on the basis of prior sampling, types of fish known to occur in the area, stream size, type of collecting equipment required and the purpose of the study. Although the actual species collected varies, two trophic groups are preferentially sought: predators and bottom feeders. The following fish are used for contaminant analysis whenever possible:

- a) Bottom feeders:
  - Carp - *Cyprinus carpio*
  - Channel Catfish - *Ictalurus punctatus* or other ictalurids
  - Redhorse sucker - *Moxostoma* sp.
  - or other sucker species - *Ictiobus* sp.
  - Carpiodes* sp., *Catostomus* sp., *Minytrema* sp.
  
- b) Predators:
  - Black Bass - *Micropterus* sp.
  - Rock Bass - *Ambloplites rupestris*

Crappie - *Pomoxis* sp.  
Sunfish - *Lepomis* sp.  
Sauger - *Stizostedion canadense*  
Walleye - *Stizostedion vitreum*  
Bowfin - *Amia calva*

In the event that target species are not collected at the site, other species that are consumed locally and are of harvestable size may be collected. If no harvestable sized fish are collected, a large sample of small fish of the same species may be collected for a whole body composite sample to determine if more intensive sampling needs to be performed.

## 2. Sampling

Typically, samples should be collected from late summer to early fall (August - October) Phillips, 1980). This is when the lipid content that contains many organic pollutants is highest. However, there are many exceptions coinciding with target species such as spawning period, budget constraints or when temporary help is available in summer months.

Various sampling techniques are used to collect fish. The most common methods of collection use:

- a) Active gear:
  - Electrofishing units
  - Seines
  
- b) Passive gear:
  - Gill nets
  - Trammel nets
  - Hoop nets
  - Rotenone (lock chambers only)

The size difference of individuals making up a composite sample shall not have a size difference (largest to smallest) greater than 75% (GLSFATF, 1993). Composite samples should contain three to ten individuals of the same species. Replicate composite samples are collected approximately 10% of the time. Sometimes both fillet and whole-body fish tissue samples are needed. When possible, the right fillet is taken from an individual and the left fillet and body is retained for a whole-body sample.

Fish are identified, weighed and measured in the field to the nearest ounce and 0.1 inch total length and recorded on the fish tissue field data sheet (Appendix F-1). The total body length is determined by measuring from the anterior most part of the fish to the tip of the longest caudle fin ray (when the lobes of the caudle fin are compressed dorsoventrally). (Anderson and Gutreuter, 1983). Any additional information, such as abnormalities (fin erosion, skin ulcers, skeletal deformities, tumors), should also be noted.

### **3. Fillet Procedure**

Fillets are the primary sample to be analyzed for contaminants. Fish samples are usually filleted in the field. However, if this cannot be done, they should be stored on ice immediately and returned to the lab within 24 hours for processing. Periodic wipe tests should be conducted in the work area to monitor for significant levels of metal and organic concentrations (U.S. EPA, 1999). The work surface for filleting is covered with clean (10% nitric acid then acetone rinsed) aluminum foil or teflon. The fish should be scaled, except for scaleless fish such as catfish and bullheads where the skin is removed. The fish are then filleted so as to include all flesh and fatty deposits from the back of the head to the tail and from the top of the back down to and including the belly flap area of the fish. Carefully remove the fillet from the body cavity to avoid puncturing it and internal organs; the rupture of internal organs will contaminate edible tissue samples (Stober, 1991; U.S. EPA 1986). If the rupture of internal organs contaminates the fillet tissue, rinse it in contaminant-free water. Remove all fins, the tail, head, viscera and major bones. Only fillets, typically removed from the right side of each fish, are used for the composite edible portion sample for consumption advisories, not whole bodies (Federal Register 1979). If only a small sample could be obtained, right and left fillets may be used and are included in the advisory. The remainder is reserved for the whole-body composite sample (right fillet + whole body right fillet was taken from) if needed. When sampling and filleting, caution should be taken to not contaminate the samples (e.g., handling gasoline containers then the fish samples or exposing fish samples to engine exhaust, etc.). The fillet knife is cleaned and rinsed with 10% nitric acid then acetone after each sample and the aluminum foil is replaced.

### **4. Sample packaging**

Once cleaned, the filleting utensils should be wrapped in aluminum foil to prevent contamination. Fillet samples are rinsed with ambient water, wrapped in extra heavy-duty aluminum foil, and placed in a waterproof plastic bag. Composite samples of the same species can be wrapped together in aluminum foil and placed in one bag. If the remaining carcass is to be used for whole-body analysis (fish with one fillet removed), it is also rinsed with ambient water, wrapped and labeled. If more than one species is collected from a site, these packaged composite samples should be kept together in one large waterproof plastic bag if possible. Once packaged, the samples are stored in ice for transport to the laboratory freezer. If samples are not to be transported to the laboratory that day, they should be frozen, preferably with dry ice.

### **5. Labeling and Chain-of-Custody**

Labels for each sample will contain the following information: stream name and sampling location, date, county, latitude and longitude, collectors, collection method (be specific as to type of electrofisher, net, etc.), type of fish, individual lengths (inches) and weights (ounces), and type of fillet. Types of fillets include whole body (WB), left fillet (LF), right fillet (RF), or left plus right fillet (BF). These variables are needed to enter the data into the EDAS database. Other information that is needed can be obtained at a later date. All samples should be properly labeled and returned with a fish tissue data sheet (Appendix F-1). Proper chain of custody procedures should be followed (KDOW, 1986).

### III. LABORATORY PROCEDURES

Tissue samples are frozen prior to processing. The frozen samples are cut into small pieces with a meat saw, blended with dry ice and homogenized in a stainless steel industrial blender or a meat grinder, depending on the size of the fish being processed. Equipment for processing fish tissue samples will be cleaned between samples as follows:

- a) Wash with mild detergent
- b) Rinse with hot tap water
- c) Rinse with distilled water
- d) Rinse with 10% nitric acid
- e) Rinse with acetone

#### 1. Composite Samples

Approximately one pint (500 ml) of ground, homogenized composite fillet tissue is placed in a pre-cleaned glass jar with a teflon-lined lid. The sample is then labeled and kept frozen until analysis by the DES analytical laboratory. The remainder of the fillet sample is combined with the whole-body fish tissue and ground together to produce the final composite whole-body sample. Approximately one pint (500 ml) of the whole-body sample will be placed in a pre-cleaned glass jar with a teflon-lined lid, labeled and kept frozen until analysis.

#### 2. Individual Fish Samples

When individual fish are processed, procedures similar to those outlined above are followed for both fillet and whole-body samples if at least one-half pint (250 ml) of fillet tissue can be obtained from the sample.

### IV. INTERPRETATION OF FISH TISSUE DATA

Fish tissue data can often be difficult to interpret accurately. But as we become more aware of harmful side effects of contaminants and the guidelines are adjusted accordingly, the advisories provide the most up-to-date guidelines for interpreting and comparing the data obtained from the tissue samples. For example, advances in technology and research have allowed us to better determine the effects of specific contaminants at specific concentrations. This allows us to issue levels of contaminant advisories for individuals based on the amount of risk it poses. The Commonwealth of Kentucky is currently using risk-based advisories. These advisories help to establish a guideline for comparing the data obtained from the tissue samples. These consumption guidelines are based on the edible portion (e.g., fillet) of the fish which is checked for an array of contaminants (Table 10-1). When the sample contaminant concentrations are obtained, they are compared to consumption concentration guidelines that range from unrestricted consumption to no consumption.

Consumption guidelines for children, pregnant and nursing women and potential childbearing women are one group higher than for the general population (GLSFATF, 1993). This separate guideline was provided because of concern for developmental effects in children and fetuses because to reduce body weight causes sensitivity to and builds up contaminant concentrations. An example of risk-based protocols with associated contaminant concentration guidelines for mercury and PCBs are presented at the end of this section (Tables 10-2 and 10-3, respectively). Based on the results, an advisory is

implemented until future studies indicate a change in advisory levels. Once an advisory is issued for a specific waterbody, residual levels of the contaminant must fall below the state criterion for at least 2 years before the waterbody is removed from the advisory list. If the contaminant does not yet have an associated risk-based protocol, one should be implemented on a need basis (FDA action levels could be used as a threshold concentration to determine that there is a need for researching and implementing a risk-based protocol for those substances). Whole-body fish tissue data cannot be used for consumption advisories. Whole bodies can be used, however, as an indicator of areas where more extensive sampling needs to be performed as a result of inflated contaminant concentrations.

## **V. QUALITY ASSURANCE/QUALITY CONTROL (QA/QC)**

To assure that samples are being processed and analyzed properly 10 percent of the ground tissue samples will be submitted as duplicate samples for comparison. Duplicate sample results within 20% relative standard deviation (RSD) will be accepted as accurate data. Duplicate sample results that fall outside 20% RSD will be considered suspect and resampling will be considered.

### ***MACROINVERTEBRATE TISSUE ANALYSIS***

#### **I. INTRODUCTION**

Macroinvertebrate tissue samples can be used to assess the bioaccumulation of pollutants in aquatic food chains and to provide background, control or reference tissue data for streams and rivers. Collection and analysis of these types of samples are performed in biological surveys on a case-by-case basis.

#### **II. TARGET SPECIES - SAMPLE PREPARATION - LABORATORY PROCEDURES**

Composite macroinvertebrate samples consist of individuals of the same or similar species. Target macroinvertebrate species are crayfish, mussels and helgrammites. Species and sample size should be recorded for all samples. Additionally, length/weight and age measurements should be recorded for mussels. Only the mussel body, not the shell, is used for analysis.

All samples are either wrapped in clean aluminum foil or placed in pre-cleaned glass jars with teflon lined lids and held on ice until return to the laboratory. The samples are frozen until processing. Samples are labeled, stored and processed in a manner similar to that used for fish tissue preparation.

At least 5 grams of homogenized macroinvertebrate tissue must be obtained for analysis by the DES laboratory. The chemical analyses performed are the same as for fish tissue analysis (Table 10-1).

#### **III. QUALITY ASSURANCE/QUALITY CONTROL (QA/QC)**

To assure that samples are being processed and analyzed properly 10 percent of the ground tissue samples will be submitted as duplicate samples for comparison. Duplicate sample results within 20% relative standard deviation (RSD) will be accepted as accurate data. Duplicate sample results that fall outside 20% RSD will be considered suspect and resampling will be considered.

**Table 10-1. PARAMETERS FOR TISSUE ANALYSIS**

% Lipids	O,P' -DDT, Total
Aluminum, Total	P,P' -DDT, Total
Arsenic, Total	Endosulfan I, alpha
Beryllium, Total	Endosulfan II, beta
Cadmium, Total	Endosulfan sulfate
Chromium, Total	Endrin
Copper, Total	Endrin aldehyde
Lead, Total	Endrin ketone
Manganese, Total	Heptachlor
Mercury, Total	Heptachlor epoxide
Nickel, Total	Hexachlorobenzene
Zinc, Total	Hexachlorocyclohexane
Aldrin	alpha-BHC
Aroclor	beta-BHC
cis-Chlordane	gamma-BHC
trans-Chlordane	delta-BHC
Oxychlordane	1,2,3,4,5,5-Hexachloro-1,3-cyclopentadiene
Technical Chlordane	Methoxychlor
Chlordane	<u>Mirex</u>
Chlorpyrifos	cis-Nonachlor
Dieldrin	trans-Nonachlor
O,P' -DDD, Total	Pentachlorophenol
P,P' -DDD, Total	Tetrachlorophenol (2,3,4,5)
O,P' -DDE, Total	Tetrachlorophenol (2,3,4,6)
P,P' -DDE, Total	Toxaphene
DDT, Total	

<b>Table 10-2. MONTHLY RISK-BASED FISH CONSUMPTION LIMITS FOR CONTAMINANTS</b>	
<b>Fish Meals/Month</b>	<b>Methylmercury EPA Fish Tissue Concentrations</b>
16	> 0.03–0.06
12	> 0.06–0.08
8	> 0.08–0.12
4	> 0.12–0.24
3	> 0.24–0.32
2	> 0.32–0.48
1	> 0.48–0.97
0.5	> 0.97–1.9
No consumption	> 1.9

<b>Table 10-3. MONTHLY RISK-BASED FISH CONSUMPTION LIMITS FOR CONTAMINANTS</b>	
<b>Fish Meals/Month</b>	<b>PCBs GLP Fish Tissue Concentrations* (ppm, wet weight)</b>
<b>Unrestricted</b>	<b>0.00-0.05</b>
<b>4</b>	<b>&gt; 0.05-0.20</b>
<b>1</b>	<b>&gt; 0.20-1.00</b>
<b>0.5</b>	<b>&gt; 1.00-1.90</b>
<b>No consumption</b>	<b>&gt;1.90</b>

**\*Pregnant and nursing women, potential childbearing women and children would be one group higher**



<b>Table 10-4. Short List of Action Levels for Poisonous or Deleterious Substances in Human Food and Animal Feed<sup>1</sup> <a href="http://vm.cfsan.fda.gov/~lrd/fdaact.html">http://vm.cfsan.fda.gov/~lrd/fdaact.html</a></b>		
<b>Substance</b>	<b>Level Established in:</b>	<b>Action Level<sup>2</sup></b>
Alfatoxin	Food & Feeds	20 ug/kg
Aldrin, Dieldrin	Fish & Seafoods	0.3 ppm
BHC	Frog Legs	0.5 ppm
Chlordane	Fish	0.3 ppm
DDT, DDE, TDE	Fish	5.0 ppm
Dioxin	Fish	50 ppt / 25 ppt <sup>3</sup>
Endrin	Fish & Shellfish Fishmeal, Fish soluble, Fish oil	0.3 ppm
Heptachlor, Heptachlor Epoxide	Fish & Shellfish	0.3 ppm
Chlordecone (formerly Kepone)	Fish	0.3 ppm
Mercury	Fish, Shellfish, Crustaceans, Aquatic Animals (Edible portions only)	1.0 ppm
Mirex	Fish	0.1 ppm
Paralytic Shellfish Toxin	Clam, Mussels, Oysters	80 ug/100g meat
Toxaphene	Fish	5.0 ppm
PCB	Fish	2 ppm

<sup>1</sup> Action Levels are established and revised according to criteria specified in Title 21 Code of Federal Regulations, parts 109 & 509 and are revoked when a regulation established a tolerance for the same substance and use becomes effective.

<sup>2</sup> Represent limits at or above which FDA will take legal action to remove adulterated products from the market.

<sup>3</sup> Represents limit at which FDA issues advisory to limit consumption.

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For more in depth discussion on risk-based fish consumption advisories see:

- U.S. EPA. 1999. Guidance for assessing chemical contaminant data for use in fish advisories: Fish sampling and analysis, vol. 1. EPA 823-R-99-007.
- U.S. EPA. 1999. Guidance for assessing chemical contaminant data for use in fish advisories: Risk assessment and fish consumption limits, vol. 2. EPA 823-R-99-008.
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# 11 - NONPOINT SOURCE MONITORING

## I. INTRODUCTION

Under §319 of the Clean Water Act (CWA) amendments of 1987 (P.L. 100-4), each state must develop and submit a management plan (KDOW 2000) to the U.S. Environmental Protection Agency (U.S. EPA) for approval to control nonpoint source pollution (NPS) impacting navigable waters within that state. Essentially, NPS pollution is that which does not exit a discrete discharge point (i.e., pipe) and thus is not regulated under the Kentucky Pollutant Discharge Elimination System. Examples of NPS pollution include runoff from animal feedlots, crop fields, logging operations, construction sites, abandoned mine lands, urban areas and residential landscapes. One of the central means of addressing NPS pollution is through the implementation of watershed projects. The goal of watershed projects is to improve or protect water quality through the voluntary incorporation of best management practices (BMPs), educational programs, technical assistance and other activities. Examples of BMPs include manure lagoons for animal waste management systems, silt fences, vegetative buffer strips and straw bales for reducing sedimentation. The type of activity being controlled governs the particular BMPs selected. Additional information concerning this topic can also be obtained from specific BMP manuals that are available for agriculture (KDOC 1996), mining (KDOW 1996), construction (U.S. EPA 1992 and KDOC 1994), onsite wastewater (902 KAR 10:081-902 KAR 10:085) and the Kentucky NPS Management Program document (KDOW 2000) for agriculture and forestry. In order to determine the effectiveness of watershed projects, water quality monitoring is required to detect changes in water quality.

### A. Project Coordination

State and federal agencies, and other organizations, can be involved with NPS water quality monitoring projects. Agencies/organizations that have participated in Kentucky NPS monitoring projects include U.S. Department of Agriculture, U.S. Army Corps of Engineers, Kentucky Division of Conservation, U.S. Geological Survey, Kentucky Department of Fish and Wildlife Resources, National Park Service and state universities. Communication and cooperation are essential for project success.

### B. Strategy for NPS Monitoring

The monitoring plan design will dictate station selection. However, site-specific factors will also need to be considered. When selecting monitoring stations for nonpoint source projects, having 1) distinct monitoring objectives, 2) an understanding of the project area and 3) a knowledge of the location and movement of local point and nonpoint pollution sources (USEPA 1997b) are all important. Specific factors that need to be considered for selecting monitoring stations include:

- a) project objectives;
- b) proximity to the BMP implementation locations;
- c) station accessibility;
- d) land-use/land treatment of surrounding area;
- e) location of point source pollution sources;
- f) USGS gauging station locations;
- e) monitoring locations of existing or past studies/surveys;
- f) the drainage area;

- g) severity of the pollution problem under investigation;
- h) the type of monitoring approach being utilized
- i) land owner cooperation;
- j) project funding (may limit the number of stations);
- k) other site-specific features (e.g., presence of wetlands, sinks).

Arrangement of monitoring stations for NPS projects can usually be handled through one of three basic approaches (USEPA 1997b and NRCS 1996). These approaches include upstream-downstream, paired-subwatershed and single-downstream designs. Whenever feasible, the upstream-downstream or paired-subwatershed approach should be selected since both spatial and temporal data comparisons can be made, and greater level of comparability is provided than with the single-downstream design. This greater level of comparability will allow better documentation of water quality changes (USEPA 1997a and 1997b, Grabow et al. 1998, 1999a and 1999b).

Targeting smaller sub-watersheds for BMP implementation and monitoring may be advantageous or even necessary when project areas encompass large drainage basins. Smaller watersheds are more responsive to water quality changes than large watersheds. Further, certain portions of a watershed may be more degraded from NPS pollution, thus warranting more attention in the way of remediation. Targeting would affect monitoring station locations since they would tend to be clustered in certain areas rather than spread out within the study area. However, as resources allow, establishing an additional station(s) some distance downstream of the NPS concern to determine if water quality changes can be observed on a broader scale may be desirable.

Control stations are established for comparative purposes (upstream, downstream and paired-subwatershed monitoring). A station being used as a measure of the ecological integrity of a stream or watershed is considered a test site. On the other hand, a station used to monitor influences outside of the specific focus of the project, such as severe weather conditions, is referred to as a control site.

Additional factors need to be addressed for biological sampling. When biological data from multiple stations will be compared, as with upstream-downstream and paired-subwatershed projects, the stations need to be as similar as possible to each other with respect to habitat, stream order, length and width, riffle and pool depths, riffle-pool ratio, etc. Specific habitat elements that should be similar when biological data is being compared include stream substrate composition, area soils and geology, aquatic vegetation, canopy cover, climate, gradient and flow. Compared stations should be contained within the same ecoregion. Furthermore, atypical reaches of stream, such as channelized or impounded sections, should be avoided unless they are the focus of the study (e.g., habitat restoration projects or reservoir studies) (USEPA 1997b).

Most NPS monitoring projects involve the collection of water quality data prior to and after BMP implementation. Collecting an adequate amount of data is crucial to making meaningful comparisons. Therefore, the frequency and duration of sample collection become important considerations. At a minimum, a full annual hydrologic cycle should be monitored prior to and after BMP implementation. However, it is preferable to obtain up to 3 years of pre-BMP and post-BMP data (Lambardo et al. 2000). The greater the variability among parameter values being examined, the greater the number of samples needed to accurately depict site conditions. Because of natural variations, biological monitoring conducted over subsequent years should be collected

during the same seasonal regime for a given project. For example, biological data should not be collected during spring and fall one year then during summer and winter the next. Likewise, only data sets representing the same approximate time of year should be compared (e.g., spring to spring, fall to fall, etc.).

Project planners should consult U.S. EPA (1997b and 2000) and NRCS (1996) for general NPS monitoring strategies and guidance. U.S. EPA has also published guidance for specific NPS categories, including forestry (U.S. EPA 1997d), agriculture (U.S. EPA 1997c) and urban (U.S. EPA 2001).

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**APPENDIX A-1**  
**WATER QUALITY BRANCH FIELD DATA SHEET (HIGH GRADIENT)**

**APPENDIX A-2**  
**WATER QUALITY BRANCH FIELD DATA SHEET (LOW GRADIENT)**

### Appendix A-1 High Gradient Stream Data Sheet

STREAM NAME:		LOCATION:			
STATION #:		MILE:		BASIN/WATERSHED:	
LAT.:		LONG.:		COUNTY:	
DATE:		TIME: <input type="checkbox"/> AM <input type="checkbox"/> PM		USGS 7.5 TOPO:	
TYPE SAMPLE: <input type="checkbox"/> P-CHEM <input type="checkbox"/> Macroinvertebrate <input type="checkbox"/> FISH <input type="checkbox"/> BACT.					
<b>WEATHER:</b> Now Past 24 hours Has there been a heavy rain in the last 7 days? <input type="checkbox"/> Heavy rain <input type="checkbox"/> Yes <input type="checkbox"/> No <input type="checkbox"/> Steady rain Air Temperature _____ °C. Inches rainfall in past 24 hours _____ in. <input type="checkbox"/> Intermittent showers _____ % Cloud Cover <input type="checkbox"/> Clear/sunny					
P-Chem: Temp(°C)_____ D.O. (mg/l)_____ % Saturation_____ pH(S.U.)_____ Cond._____ <input type="checkbox"/> Grab					
<b>INSTREAM WATERSHED FEATURES:</b> Stream Width _____ ft Range of Depth _____ ft Average Velocity _____ ft/s Discharge _____ cfs Est. Reach Length _____			<b>LOCAL WATERSHED FEATURES:</b> <u>Predominant Surrounding Land Use:</u> <input type="checkbox"/> Surface Mining <input type="checkbox"/> Construction <input type="checkbox"/> Forest <input type="checkbox"/> Deep Mining <input type="checkbox"/> Commercial <input type="checkbox"/> Pasture/Grazing <input type="checkbox"/> Oil Wells <input type="checkbox"/> Industrial <input type="checkbox"/> Silviculture <input type="checkbox"/> Land Disposal <input type="checkbox"/> Row Crops <input type="checkbox"/> Urban Runoff/Storm Sewers		
<u>Hydraulic Structures:</u> <input type="checkbox"/> Dams <input type="checkbox"/> Bridge Abutments <input type="checkbox"/> Island <input type="checkbox"/> Waterfalls <input type="checkbox"/> Other _____		<u>Stream Flow:</u> <input type="checkbox"/> Dry <input type="checkbox"/> Pooled <input type="checkbox"/> Low <input type="checkbox"/> Normal <input type="checkbox"/> High <input type="checkbox"/> Very Rapid or Torrential		<u>Stream Type:</u> <input type="checkbox"/> Perennial <input type="checkbox"/> Intermittent <input type="checkbox"/> Ephemeral <input type="checkbox"/> Seep	
Riparian Vegetation: Dom. Tree/Shrub Taxa Dominate Type: <input type="checkbox"/> Trees <input type="checkbox"/> Shrubs <input type="checkbox"/> Grasses <input type="checkbox"/> Herbaceous Number of strata _____		<u>Canopy Cover:</u> <input type="checkbox"/> Fully Exposed (0-25%) <input type="checkbox"/> Partially Exposed (25-50%) <input type="checkbox"/> Partially Shaded (50-75%) <input type="checkbox"/> Fully Shaded (75-100%)		<u>Channel Alterations:</u> <input type="checkbox"/> Dredging <input type="checkbox"/> Channelization <input type="checkbox"/> Full <input type="checkbox"/> Partial	
Substrate <input type="checkbox"/> Est. <input type="checkbox"/> P.C.		Riffle _____ %		Run _____ %	
Silt/Clay (<0.06 mm)					
Sand (0.06 – 2 mm)					
Gravel (2-64 mm)					
Cobble (64 – 256 mm)					
Boulders (>256 mm)					
Bedrock					
<b>Habitat</b>		<b>Condition Category</b>			
<b>Parameter</b>	<b>Optimal</b>	<b>Suboptimal</b>	<b>Marginal</b>	<b>Poor</b>	
<b>1. Epifaunal Substrate/Available Cover</b>	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.	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	
<b>2. Embeddedness</b>	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.	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	
<b>3. Velocity/Depth Regime</b>	All four velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (Sow 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).	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	



<b>4. Sediment Deposition</b>	Little or no enlargement of islands or point bars and less than 5% (<20% for low-gradient streams) of the bottom affected by sediment deposition.	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5-30% (20-50% for low-gradient) of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% (50-80% for low-gradient) 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% (80% for low-gradient) of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>5. Channel Flow Status</b>	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.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>6. Channel Alteration</b>	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.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>7. Frequency of Riffles (or bends)</b>	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream <7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important.	Occurrence of riffles infrequent; distance between riffles divided by the width of the stream is between 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is a ratio of >25.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>8. Bank Stability (score each bank)</b>	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.
Note: determine left or right side by facing downstream.				
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0
<b>9. Vegetative Protection (score each bank)</b>	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.
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0
<b>10. Riparian Vegetative Zone Width (score each bank riparian zone)</b>	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.
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0

Total Score

NOTES/COMMENTS:

### Appendix A-2 Low Gradient Stream Data Sheet

STREAM NAME:		LOCATION:			
STATION #:		MILE:		BASIN/WATERSHED:	
LAT.:		LONG.:		COUNTY:	
DATE:		TIME: <input type="checkbox"/> AM <input type="checkbox"/> PM		USGS 7.5 TOPO:	
INVESTIGATORS:					
TYPE SAMPLE: <input type="checkbox"/> P-CHEM <input type="checkbox"/> Macroinvertebrate <input type="checkbox"/> FISH <input type="checkbox"/> BACT.					
<b>WEATHER:</b> Now Past 24 hours Has there been a heavy rain in the last 7 days? <input type="checkbox"/> Heavy rain <input type="checkbox"/> Yes <input type="checkbox"/> No <input type="checkbox"/> Steady rain Air Temperature _____ °C. Inches rainfall in past 24 hours _____ in. <input type="checkbox"/> Intermittent showers _____ % Cloud Cover <input type="checkbox"/> Clear/sunny					
P-Chem: Temp(°C)_____ D.O. (mg/l)_____ % Saturation_____ pH(S.U.)_____ Cond._____ <input type="checkbox"/> Grab					
<b>INSTREAM WATERSHED FEATURES:</b> Stream Width _____ ft Range of Depth _____ ft Average Velocity _____ ft/s Discharge _____ cfs Est. Reach Length _____			<b>LOCAL WATERSHED FEATURES:</b> <u>Predominant Surrounding Land Use:</u> <input type="checkbox"/> Surface Mining <input type="checkbox"/> Construction <input type="checkbox"/> Forest <input type="checkbox"/> Deep Mining <input type="checkbox"/> Commercial <input type="checkbox"/> Pasture/Grazing <input type="checkbox"/> Oil Wells <input type="checkbox"/> Industrial <input type="checkbox"/> Silviculture <input type="checkbox"/> Land Disposal <input type="checkbox"/> Row Crops <input type="checkbox"/> Urban Runoff/Storm Sewers		
<u>Hydraulic Structures:</u> <input type="checkbox"/> Dams <input type="checkbox"/> Bridge Abutments <input type="checkbox"/> Island <input type="checkbox"/> Waterfalls <input type="checkbox"/> Other _____		<u>Stream Flow:</u> <input type="checkbox"/> Dry <input type="checkbox"/> Pooled <input type="checkbox"/> Low <input type="checkbox"/> Normal <input type="checkbox"/> High <input type="checkbox"/> Very Rapid or Torrential		<u>Stream Type:</u> <input type="checkbox"/> Perennial <input type="checkbox"/> Intermittent <input type="checkbox"/> Ephemeral <input type="checkbox"/> Seep	
Riparian Vegetation: Dom. Tree/Shrub Taxa Dominate Type: <input type="checkbox"/> Trees <input type="checkbox"/> Shrubs <input type="checkbox"/> Grasses <input type="checkbox"/> Herbaceous Number of strata _____		<u>Canopy Cover:</u> <input type="checkbox"/> Fully Exposed (0-25%) <input type="checkbox"/> Partially Exposed (25-50%) <input type="checkbox"/> Partially Shaded (50-75%) <input type="checkbox"/> Fully Shaded (75-100%)		<u>Channel Alterations:</u> <input type="checkbox"/> Dredging <input type="checkbox"/> Channelization ( <input type="checkbox"/> Full <input type="checkbox"/> Partial)	
Substrate <input type="checkbox"/> Est. <input type="checkbox"/> P.C.	<b>Riffle _____ %</b>		<b>Run _____ %</b>		<b>Pool _____ %</b>
Silt/Clay (<0.06 mm)					
Sand (0.06 – 2 mm)					
Gravel (2-64 mm)					
Cobble (64 – 256 mm)					
Boulders (>256 mm)					
Bedrock					
<b>Habitat</b>	<b>Condition Category</b>				
<b>Parameter</b>	<b>Optimal</b>	<b>Suboptimal</b>	<b>Marginal</b>	<b>Poor</b>	
<b>1. Epifaunal Substrate/ Available Cover</b>	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.	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	
<b>2. Pool Substrate Characterization</b>	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.	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	
<b>3. Pool Variability</b>	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.	
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0	

<b>4. Sediment Deposition</b>	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; 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>5. Channel Flow Status</b>	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.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>6. Channel Alteration</b>	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.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>7. Channel Sinuosity</b>	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 2 to 1 times longer than if it was in a straight line.	Channel straight; waterway has been channelized for a long distance.
<b>SCORE</b>	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
<b>8. Bank Stability (score each bank)</b>  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.
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0
<b>9. Vegetative Protection (score each bank)</b>	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.
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0
<b>10. Riparian Vegetative Zone Width (score each bank riparian zone)</b>	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.
<b>SCORE (LB)</b>	Left Bank 10 9	8 7 6	5 4 3	2 1 0
<b>SCORE (RB)</b>	Right Bank 10 9	8 7 6	5 4 3	2 1 0

**Total Score**

**NOTES/COMMENTS:**

## **B-1 BACTERIOLOGICAL BENCH SHEET**



## **APPENDIX C**

**C-1 PERIPHYTON FIELD DATA SHEET**

**C-2 SAMPLE ALGAE LABORATORY LOGBOOK**

**C-3 NON-DIATOM BENCH SHEET**

**C-4 DIATOM BENCH SHEET**

**C-5 MASTER TAXA LISTS**

**C-6 PHYTOPLANKTON FIELD DATA SHEET**

**C-7 CHLOROPHYLL *a* AND ASH-FREE DRY MASS BENCH SHEETS**

**C-8 PHYTOPLANKTON BENCH SHEET**

## C-1 BENTHIC ALGAE FIELD DATA SHEET

<b>DOW Station ID Number:</b>	<b>Stream Name:</b>	<b>Location:</b>
<b>Collection Date:</b>	<b>Time:</b>	<b>County:</b>
<b>River Basin:</b>	<b>DOW Program:</b>	<b>Name of Investigator(s):</b>
<b>Macrohabitats Sampled:</b>	Riffle ____ Run ____ Pool ____	
<b>Microhabitats Sampled:</b>	Epipellic ____ Episammic ____ Epilithic ____ Epidendric ____ Epiphytic ____ Epizooic ____ Artificial Substrate ____	
<b>Collection Method Used:</b>	Periphytometer ____ Natural Scraping ____ Suction Device ____	
<b>Macroalgae Present:</b>	<i>Cladophora</i> ____ <i>Tetraspora</i> ____ <i>Hydrodictyon</i> ____ <i>Draparnaldia</i> ____ <i>Batrachospermum</i> ____ <i>Lemanea/Paralemanea</i> ____ <i>Thorea</i> ____ <i>Boldia</i> ____ <i>Vaucheria</i> ____ <i>Chara</i> ____ <i>Nitella</i> ____ Filamentous Green Filaments ____ Blue-Green Mats ____ Diatom Mats ____	
<b>Periphyton Coverage:</b>	Dense (>75%) ____ Moderate (25-75%) ____ Sparse (10-25%) ____ Absent (<10%) ____	

**Field Algal Community Assessment Score** \_\_\_\_

<b>Excellent (5)</b>	Phytobenthos appears diverse with several algal divisions represented, including chrysophytes, chlorophytes, cyanophytes, and rhodophytes. Phytoplankton sub-community not apparent. Floating algal mats are not present. The algal community is similar to that of reference stations within the same ecoregion.
<b>Fair - Good (2 - 4)</b>	Phytobenthic algae are present in moderate amount. The algal community may be dominated by one type of algal growth, such as long filaments of <i>Cladophora</i> . Diversity is low to moderate, and a phytoplankton sub-community is not apparent. Floating algal mats may be present, but are not extensive. Clean water algal taxa (e.g. red algae, <i>Chaetophora</i> , etc.) present in reference reach stations may not be present.
<b>Poor (1)</b>	In cases of toxic pollution (acid mine drainage, toxic discharges, etc.), substrates and water column may appear sterile, bleached, or rust-colored. Little or no algae are observed. With organic pollution (sewage discharges, etc.), substrates may be covered with thick white, black, or gray mats of filamentous bacteria, thick algal mats of cyanophytes (blue-green algae), and/or chlorophytes (green algae). The water column may have a "pea green" appearance as a result of high abundances of euglenophytes, or large floating mats of algae may be present, especially in pools and slow-moving streams. Look for extremes of either characteristic. Diversity is very low. Very few, if any, clean water taxa are present.







### C-3 Non-DIATOM BENCH SHEET (CONT'D)

<b>DOW Station ID Number:</b>	<b>Stream Name:</b>	<b>Location:</b>		
<b>Collection Date:</b>	<b>County:</b>	<b>Name of Collector(s):</b>		
<b>River Basin:</b>	<b>DOW Program:</b>	<b>Name of Taxonomist(s):</b>		
<b>Taxa Name</b>	<b>Algal Division</b>	<b>Tally (If Counting)</b>	<b># of Algal Units (If Counting)</b>	<b>Estimated Abundance</b>





## C-5 DIATOM MASTER TAXA LIST

Taxa	New Taxa Name	PTI Value
<i>Achnanthes childanos</i>		0
<i>Achnanthes clevei</i>	<i>Karayevia clevei</i>	4
<i>Achnanthes clevei</i> var. <i>rostrata</i>	<i>Karayevia clevei</i> var. <i>rostrata</i>	4
<i>Achnanthes coarctata</i>	<i>Achnanthidium coarctatum</i>	0
<i>Achnanthes deflexa</i>		4
<i>Achnanthes deflexa</i> var. <i>alpestris</i>		4
<i>Achnanthes detha</i>	<i>Achnanthes subatomoides</i>	0
<i>Achnanthes exigua</i>		4
<i>Achnanthes hauckiana</i>	<i>Achnanthidium delicatulum</i>	0
<i>Achnanthes hungarica</i>	<i>Lemnicola hungarica</i>	4
<i>Achnanthes hustedtii</i>		4
<i>Achnanthes inflata</i>		0
<i>Achnanthes lanceolata</i>	<i>Planothidium lanceolata</i>	3
<i>Achnanthes lanceolata</i> var. <i>apiculata</i>	<i>Planothidium lanceolata</i> var. <i>apiculata</i>	3
<i>Achnanthes lanceolata</i> var. <i>dubia</i>	<i>Planothidium lanceolata</i> var. <i>dubia</i>	3
<i>Achnanthes lapponica</i> var. <i>ninckei</i>	<i>Achnanthes laevis</i>	3
<i>Achnanthes linearis</i>	<i>Rossithidium linearis</i>	3
<i>Achnanthes linearis</i> f. <i>curta</i>	<i>Rossithidium linearis</i> f. <i>curta</i>	3
<i>Achnanthes linearis</i> var. <i>pusilla</i>	<i>Rossithidium linearis</i> var. <i>pusilla</i>	3
<i>Achnanthes microcephala</i>	<i>Achnanthidium microcephalum</i>	3
<i>Achnanthes minutissima</i>	<i>Achnanthidium minutissimum</i>	3
<i>Achnanthes pinnata</i>	<i>Achnanthes conspicua</i>	3
<i>Achnanthes</i> sp.		3
<i>Achnanthes</i> sp. 1		2
<i>Achnanthes</i> spp.		2
<i>Achnanthes stewartii</i>		4
<i>Achnanthes wellsiae</i>	<i>Achnanthes solea</i>	0
<i>Actinocyclus normanii</i>		0
<i>Amphipleura pellucida</i>		3
<i>Amphora bullatoides</i>		0
<i>Amphora ovalis</i>		3
<i>Amphora ovalis</i> var. <i>affinis</i>	<i>Amphora libyca</i>	3
<i>Amphora ovalis</i> var. <i>pediculus</i>	<i>Amphora libyca</i>	3
<i>Amphora perpusilla</i>	<i>Amphora pediculus</i>	3
<i>Amphora</i> sp.		3
<i>Amphora submontana</i>	<i>Amphora montana</i>	3
<i>Amphora veneta</i>		1
<i>Anomoeoneis serians</i> var. <i>brachysira</i>	<i>Brachysira serians</i>	0
<i>Anomoeoneis vitrea</i>	<i>Brachysira vitrea</i>	2
<i>Asterionella formosa</i>		3
<i>Aulacoseira alpigena</i>		3
<i>Aulacoseira distans</i>		3
<i>Aulacoseira granulata</i>		3
<i>Aulacoseira italica</i>		3
<i>Bacillaria paradoxa</i>		2
<i>Biddulphia laevis</i>	<i>Pleurosira laevis</i>	3
<i>Caloneis amphisbaena</i>	<i>Pinnularia amphisbaena</i>	0
<i>Caloneis bacillaris</i> var. <i>thermalis</i>	<i>Pinnularia thermalis</i>	0
<i>Caloneis bacillum</i>	<i>Pinnularia bacillum</i>	3
<i>Caloneis branderi</i>	<i>Pinnularia branderi</i>	0
<i>Caloneis budensis</i>	<i>Pinnularia budensis</i>	0
<i>Caloneis hyalina</i>	<i>Pinnularia hyalina</i>	0
<i>Caloneis lewisii</i>	<i>Pinnularia schumanniana</i> var. <i>biconstricta</i>	2
<i>Caloneis lewisii</i> var. <i>inflata</i>	<i>Pinnularia schumanniana</i> var. <i>biconstricta</i>	2
<i>Caloneis limosa</i>	<i>Pinnularia schumanniana</i>	0
<i>Caloneis</i> sp.	<i>Pinnularia</i> sp.	2
<i>Caloneis undulata</i>	<i>Pinnularia undulata</i>	2
<i>Caloneis ventricosa</i>	<i>Pinnularia silicula</i>	3
<i>Caloneis ventricosa</i> var. <i>alpina</i>	<i>Pinnularia silicula</i> var. <i>alpina</i>	3
<i>Caloneis ventricosa</i> var. <i>minuta</i>	<i>Pinnularia silicula</i> var. <i>minuta</i>	3
<i>Caloneis ventricosa</i> var. <i>subundulata</i>	<i>Pinnularia silicula</i> var. <i>subundulata</i>	3
<i>Caloneis ventricosa</i> var. <i>truncatula</i>	<i>Pinnularia silicula</i> var. <i>truncatula</i>	3
<i>Campylodiscus hibernicus</i>		0
<i>Capartogramma crucicula</i>		2

## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Cocconeis pediculus</i>		3
<i>Cocconeis placentula</i>		3
<i>Cocconeis placentula</i> var. <i>euglypta</i>		3
<i>Cocconeis placentula</i> var. <i>lineata</i>		3
<i>Cyclotella atomus</i>		2
<i>Cyclotella meneghiniana</i>	<i>Stephanocyclus meneghiniana</i>	1
<i>Cyclotella pseudostelligera</i>		2
<i>Cyclotella</i> sp.		2
<i>Cyclotella stelligera</i>		3
<i>Cyclotella striata</i>		4
<i>Cyclotella striata</i> var. <i>ambigua</i>		2
<i>Cylindrotheca gracilis</i>		3
<i>Cymatopleura solea</i>		3
<i>Cymbella affinis</i>		4
<i>Cymbella amphicephala</i>	<i>Cymbopleura amphicephala</i>	4
<i>Cymbella aspera</i>		4
<i>Cymbella cesatii</i>	<i>Encyonemopsis cesatii</i>	4
<i>Cymbella cistula</i>		4
<i>Cymbella cuspidata</i>		4
<i>Cymbella cymbiformis</i>		4
<i>Cymbella delicatula</i>		4
<i>Cymbella hauckii</i>		0
<i>Cymbella hebridica</i>		4
<i>Cymbella hustedtii</i>		0
<i>Cymbella laevis</i>		0
<i>Cymbella lanceolata</i>		4
<i>Cymbella leptoceros</i>		4
<i>Cymbella lunata</i>	<i>Encyonema lunatum</i>	4
<i>Cymbella mexicana</i>		0
<i>Cymbella microcephala</i>		4
<i>Cymbella minuta</i>	<i>Encyonema minutum</i>	3
<i>Cymbella minuta</i> var. <i>pseudogracilis</i>	<i>Encyonema mesianum</i>	3
<i>Cymbella muelleri</i>	<i>Encyonema muelleri</i>	4
<i>Cymbella naviculiformis</i>		4
<i>Cymbella prostrata</i>	<i>Encyonema prostratum</i>	4
<i>Cymbella prostrata</i> var. <i>auerswaldii</i>	<i>Encyonema caespitosum</i>	4
<i>Cymbella pusilla</i>	<i>Navicella pusilla</i>	0
<i>Cymbella silesiaca</i>	<i>Encyonema silesiacum</i>	4
<i>Cymbella sinuata</i>	<i>Reimeria sinuata</i>	4
<i>Cymbella</i> sp.		4
<i>Cymbella</i> sp. ( <i>K</i> )		4
<i>Cymbella subaequalis</i>		0
<i>Cymbella subcuspidata</i>		4
<i>Cymbella triangulum</i>	<i>Encyonema triangulum</i>	4
<i>Cymbella tumida</i>		4
<i>Cymbella turgidula</i>		4
<i>Denticula elegans</i>		3
<i>Denticula kuetzingii</i>		3
<i>Denticula</i> sp.		3
<i>Diatoma hiemale</i>	<i>Diatoma hyemalis</i>	1
<i>Diatoma tenue</i>	<i>Diatoma tenue</i>	0
<i>Diatoma vulgare</i>	<i>Diatoma vulgare</i>	3
<i>Diploneis elliptica</i>		3
<i>Diploneis finnica</i>		0
<i>Diploneis oblonella</i>		3
<i>Diploneis puella</i>		0
<i>Diploneis smithii</i> var. <i>dilatata</i>		0
<i>Diploneis</i> sp.		3
<i>Diploneis subovalis</i>		0
<i>Entomoneis alata</i>		1
<i>Entomoneis ornata</i>		1
<i>Epithemia adnata</i>		2
<i>Epithemia adnata</i> var. <i>saxonica</i>		0
<i>Epithemia argus</i>		1

## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Epithemia argus</i> var. <i>protracta</i>		1
<i>Epithemia sorex</i>		3
<i>Epithemia</i> sp.		2
<i>Epithemia turgida</i>		3
<i>Epithemia turgida</i> var. <i>granulata</i>		3
<i>Eunotia arcus</i>		2
<i>Eunotia curvata</i>	<i>Eunotia bilunaris</i>	3
<i>Eunotia exigua</i>		2
<i>Eunotia formica</i>		0
<i>Eunotia incisa</i>		0
<i>Eunotia maior</i>		3
<i>Eunotia naegeli</i>		0
<i>Eunotia pectinalis</i>		3
<i>Eunotia pectinalis</i> var. <i>minor</i>		3
<i>Eunotia pectinalis</i> var. <i>undulata</i>		3
<i>Eunotia perpusilla</i>	<i>Eunotia muscicola</i> var. <i>tridentula</i>	3
<i>Eunotia praerupta</i>		0
<i>Eunotia quaternaria</i>	<i>Eunotia muscicola</i> var. <i>tridentula</i>	0
<i>Eunotia rhomboidea</i>		4
<i>Eunotia septentrionalis</i>		0
<i>Eunotia serra</i> var. <i>diadema</i>		3
<i>Eunotia</i> sp.		3
<i>Eunotia tenella</i>		3
<i>Eunotia triodon</i>		0
<i>Eunotia vanheurckii</i> var. <i>intermedia</i>		0
<i>Fragilaria brevistriata</i>	<i>Pseudostaurosira brevistriata</i>	0
<i>Fragilaria capucina</i> var. <i>mesolepta</i>		2
<i>Fragilaria construens</i>	<i>Staurosira construens</i>	0
<i>Fragilaria construens</i> var. <i>binodis</i>	<i>Pseudostaurosira binodis</i>	0
<i>Fragilaria construens</i> var. <i>pumila</i>	<i>Staurosira construens</i> var. <i>subsalina</i>	0
<i>Fragilaria construens</i> var. <i>venter</i>	<i>Staurosira construens</i> f. <i>venter</i>	0
<i>Fragilaria crotonensis</i>		0
<i>Fragilaria lapponica</i>	<i>Staurosirella lapponica</i>	0
<i>Fragilaria leptostauron</i>	<i>Staurosirella leptostauron</i>	3
<i>Fragilaria nanana</i>		3
<i>Fragilaria pinnata</i>	<i>Punctastriata pinnata</i>	3
<i>Fragilaria</i> sp.		3
<i>Fragilaria vaucheriae</i>	<i>Fragilaria capucina</i> var. <i>vaucheriae</i>	2
<i>Fragilaria virescens</i>		2
<i>Frustulia assymetrica</i>		0
<i>Frustulia rhomboides</i>		3
<i>Frustulia rhomboides</i> var. <i>amphipleuroides</i>		3
<i>Frustulia rhomboides</i> var. <i>capitata</i>		3
<i>Frustulia rhomboides</i> var. <i>crassinervia</i>		4
<i>Frustulia rhomboides</i> var. <i>saxonica</i>		3
<i>Frustulia</i> sp.		3
<i>Frustulia vulgaris</i>		3
<i>Frustulia weinholdii</i>		3
<i>Gomphoneis herculeana</i>	<i>Gomphoneis minutum</i>	0
<i>Gomphoneis herculeana</i> var. <i>robusta</i>	<i>Gomphoneis minutum</i>	0
<i>Gomphonema abbreviatum</i>	<i>Rhoicosphenia abbreviata</i>	3
<i>Gomphonema acuminatum</i>		4
<i>Gomphonema acuminatum</i> var. <i>elongatum</i>		4
<i>Gomphonema affine</i>		3
<i>Gomphonema angustatum</i>		2
<i>Gomphonema angustatum</i> var. <i>productum</i>		2
<i>Gomphonema apuncto</i>		2
<i>Gomphonema augur</i>		2
<i>Gomphonema brasiliense</i>	<i>Gomphonema grovei</i> var. <i>lingulatum</i>	4
<i>Gomphonema clevei</i>		3
<i>Gomphonema dichotomum</i>	<i>Gomphonema angustum</i>	1
<i>Gomphonema gracile</i>		3
<i>Gomphonema instabilis</i>	<i>Gomphonema angustatum</i>	3

## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Gomphonema intricatum</i>	<i>Gomphonema angustum</i>	3
<i>Gomphonema intricatum</i> var. <i>pulvinatum</i>	<i>Gomphonema angustum</i>	3
<i>Gomphonema manubrium</i>		4
<i>Gomphonema mehleri</i>		0
<i>Gomphonema olivaceoides</i>	<i>Gomphonema olivaceum</i> var. <i>minutissimum</i>	0
<i>Gomphonema olivaceum</i>		2
<i>Gomphonema parvulum</i>		1
<i>Gomphonema puiggarianum</i> var. <i>aequatorialis</i>		2
<i>Gomphonema rhombicum</i>	<i>Gomphonema clevei</i>	4
<i>Gomphonema</i> sp.		3
<i>Gomphonema sparsistriatum</i> f. <i>maculatum</i>		4
<i>Gomphonema sphaerophorum</i>	<i>Gomphonema hebridense</i> var. <i>sphaerophorum</i>	4
<i>Gomphonema subclavatum</i>	<i>Gomphonema clavatum</i>	2
<i>Gomphonema subclavatum</i>	<i>Gomphonema clavatum</i> var. <i>mexicanum</i>	3
<i>Gomphonema tenellum</i>	<i>Gomphonema minutum</i>	2
<i>Gomphonema tergestinum</i>		0
<i>Gomphonema truncatum</i>		4
<i>Gomphonema truncatum</i> var. <i>capitatum</i>		4
<i>Gomphonema truncatum</i> var. <i>turgidulum</i>		4
<i>Gyrosigma acuminatum</i>		3
<i>Gyrosigma attenuatum</i>		3
<i>Gyrosigma distortum</i>	<i>Gyrosigma parkerii</i>	2
<i>Gyrosigma nodiferum</i>		4
<i>Gyrosigma obscurum</i>		0
<i>Gyrosigma obtusatum</i>		0
<i>Gyrosigma scalproides</i>		3
<i>Gyrosigma sciotense</i>		0
<i>Gyrosigma</i> sp.		3
<i>Gyrosigma spencerii</i>		3
<i>Gyrosigma spencerii</i> var. <i>curvula</i>		3
<i>Hantzschia amphioxys</i>		3
<i>Hantzschia elongata</i>		0
<i>Mastogloia smithii</i>		0
<i>Melosira distans</i> var. <i>alpigena</i>	<i>Auloseira alpigena</i>	3
<i>Melosira granulata</i>	<i>Auloseira granulata</i>	3
<i>Melosira granulata</i> var. <i>angustissima</i>	<i>Auloseira granulata</i> var. <i>angustissima</i>	3
<i>Melosira italica</i>	<i>Auloseira italica</i>	3
<i>Melosira varians</i>		2
<i>Merideon circulare</i>		3
<i>Merideon circulare</i> var. <i>constrictum</i>		3
<i>Navicula accomoda</i>	<i>Craticula accomoda</i>	1
<i>Navicula agrestis</i>		0
<i>Navicula anglica</i> var. <i>subsalsa</i>	<i>Navicula pseudanglica</i> var. <i>signata</i>	0
<i>Navicula angusta</i>		0
<i>Navicula arvensis</i>		3
<i>Navicula atomus</i>	<i>Mayamaia atomus</i>	1
<i>Navicula auriculata</i>	<i>Fallacia auriculata</i>	3
<i>Navicula bacillum</i>	<i>Sellaphora bacillum</i>	4
<i>Navicula capitata</i>	<i>Hippodonta capitata</i>	3
<i>Navicula capitata</i> var. <i>luneburgensis</i>	<i>Hippodonta luneburgensis</i>	3
<i>Navicula cari</i>		1
<i>Navicula clementioides</i>		0
<i>Navicula clementis</i>		0
<i>Navicula cocconeiformis</i>	<i>Cavinula cocconeiformis</i>	0
<i>Navicula confervacea</i>	<i>Diadensis confervacea</i>	2
<i>Navicula confervacea</i> var. <i>peregrina</i>	<i>Diadensis confervacea</i>	2
<i>Navicula contenta</i>	<i>Diadensis contenta</i>	2
<i>Navicula contenta</i> var. <i>biceps</i>	<i>Diadensis contenta</i>	2
<i>Navicula cryptocephala</i>		3
<i>Navicula cryptocephala</i> var. <i>exilis</i>	<i>Navicula cryptocephala</i>	4
<i>Navicula cryptocephala</i> var. <i>veneta</i>	<i>Navicula veneta</i>	1
<i>Navicula cuspidata</i>	<i>Craticula cuspidata</i>	2
<i>Navicula decussis</i>		3
<i>Navicula dibola</i>		0



## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Navicula elginensis</i>		3
<i>Navicula elginensis</i> var. <i>neglecta</i>		3
<i>Navicula elginensis</i> var. <i>rostrata</i>		3
<i>Navicula exigua</i>		3
<i>Navicula exigua</i> var. <i>capitata</i>		3
<i>Navicula exigua</i> var. <i>signata</i>		3
<i>Navicula gastrum</i>	<i>Placoneis gastrum</i>	0
<i>Navicula gibbosa</i>		0
<i>Navicula gottlandica</i>		2
<i>Navicula graciloides</i>	<i>Navicula cari</i>	1
<i>Navicula gregaria</i>		2
<i>Navicula gysingensis</i>		0
<i>Navicula halophila</i>	<i>Craticula halophila</i>	2
<i>Navicula halophila</i> var. <i>tenuirostris</i>	<i>Craticula halophila</i>	2
<i>Navicula hasta</i>		2
<i>Navicula heufleri</i> var. <i>leptocephala</i>	<i>Navicula erifuga</i>	2
<i>Navicula hustedtii</i>		3
<i>Navicula ingenua</i>		0
<i>Navicula integra</i>		0
<i>Navicula lacustris</i>	<i>Cavinula lacustris</i>	0
<i>Navicula laevisissima</i>	<i>Sellaphora laevisissima</i>	0
<i>Navicula lanceolata</i>		2
<i>Navicula lateropunctata</i>		0
<i>Navicula laterorostrata</i>		1
<i>Navicula luzonensis</i>	<i>Navicula subminuscula</i>	1
<i>Navicula menisculus</i>		2
<i>Navicula menisculus</i> var. <i>upsaliensis</i>		2
<i>Navicula minima</i>		1
<i>Navicula miniscula</i>		0
<i>Navicula mutica</i>	<i>Luticola mutica</i>	2
<i>Navicula mutica</i> var. <i>binodis</i>	<i>Luticola mutica</i>	2
<i>Navicula mutica</i> var. <i>cohnii</i>	<i>Luticola cohnii</i>	1
<i>Navicula mutica</i> var. <i>nivalis</i>	<i>Luticola nivalis</i>	2
<i>Navicula mutica</i> var. <i>stigma</i>	<i>Luticola mutica</i>	2
<i>Navicula mutica</i> var. <i>undulata</i>	<i>Luticola mutica</i>	2
<i>Navicula mutica</i> var. <i>ventricosa</i>	<i>Luticola mutica</i>	2
<i>Navicula notha</i>		3
<i>Navicula paucivittata</i>		0
<i>Navicula pelliculosa</i>	<i>Fistulifera pelliculosa</i>	2
<i>Navicula placenta</i>		0
<i>Navicula placentula</i>		0
<i>Navicula pseudarvensis</i>		0
<i>Navicula pseudolanceolata</i>		0
<i>Navicula pupula</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> f. <i>rostrata</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> var. <i>capitata</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> var. <i>elliptica</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> var. <i>mutata</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> var. <i>rectangularis</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pupula</i> var. <i>subcapitata</i>	<i>Sellaphora pupula</i>	3
<i>Navicula pygmaea</i>	<i>Fallacia pygmaea</i>	3
<i>Navicula radiosa</i>		3
<i>Navicula radiosa</i> var. <i>parva</i>		3
<i>Navicula radiosa</i> var. <i>tenella</i>	<i>Navicula cryptotenella</i>	2
<i>Navicula rhynchocephala</i>		3
<i>Navicula rhynchocephala</i> var. <i>germanii</i>	<i>Navicula viridula</i> var. <i>rostellata</i>	2
<i>Navicula salinarum</i>		2
<i>Navicula salinarum</i> var. <i>intermedia</i>	<i>Navicula capitatoradiata</i>	2
<i>Navicula savannahiana</i>		0
<i>Navicula saxophila</i>	<i>Luticola saxophila</i>	0
<i>Navicula schroeteri</i> var. <i>escambia</i>	<i>Navicula schroeteri</i>	3
<i>Navicula scutelloides</i>		0
<i>Navicula secreta</i> var. <i>apiculata</i>	<i>Navicula expecta</i>	2
<i>Navicula seminulum</i>	<i>Sellaphora seminulum</i>	1

## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Navicula seminulum</i> var. <i>hustedtii</i>	<i>Sellaphora seminulum</i>	1
<i>Navicula</i> sp.		2
<i>Navicula subhamulata</i>	<i>Fallacia subhamulata</i>	3
<i>Navicula subhamulata</i> var. <i>undulata</i>	<i>Fallacia helensis</i>	3
<i>Navicula subminuscula</i>		1
<i>Navicula symmetrica</i>	<i>Navicula schroeteri</i>	2
<i>Navicula tantula</i>	<i>Navicula minima</i>	1
<i>Navicula tenelloides</i>		3
<i>Navicula tenera</i>	<i>Fallacia tenera</i>	2
<i>Navicula tridentula</i>		0
<i>Navicula tripunctata</i>		3
<i>Navicula tripunctata</i> var. <i>schizonemoides</i>	<i>Navicula recens</i>	2
<i>Navicula trivialis</i>		2
<i>Navicula tuscula</i>	<i>Aneumastus tusculus</i>	0
<i>Navicula vanheurckii</i>		0
<i>Navicula viridula</i>		2
<i>Navicula viridula</i> var. <i>avenacea</i>	<i>Navicula lanceolata</i>	2
<i>Navicula viridula</i> var. <i>linearis</i>		2
<i>Navicula viridula</i> var. <i>rostellata</i>		2
<i>Navicula zanoni</i>		2
<i>Neidium affine</i>		3
<i>Neidium affine</i> var. <i>amphirhynchus</i>	<i>Neidium affine</i>	3
<i>Neidium affine</i> var. <i>longiceps</i>		3
<i>Neidium affine</i> var. <i>undulatum</i>	<i>Neidium affine</i>	3
<i>Neidium apiculatum</i>		0
<i>Neidium binodis</i>		0
<i>Neidium bisulcatum</i>		0
<i>Neidium bisulcatum</i> var. <i>baicalense</i>		0
<i>Neidium dubium</i>		0
<i>Neidium dubium</i> f. <i>constrictum</i>		0
<i>Neidium iridis</i> var. <i>amphigomphus</i>	<i>Neidium iridis</i>	0
<i>Neidium ladogense</i> var. <i>densestriatum</i>		0
<i>Neidium</i> sp.		3
<i>Nitzschia accomoda</i>		0
<i>Nitzschia acicularioides</i>		2
<i>Nitzschia acicularis</i>		2
<i>Nitzschia acicula</i>		0
<i>Nitzschia adapta</i>		2
<i>Nitzschia agnita</i>		3
<i>Nitzschia amphibia</i>		1
<i>Nitzschia angustata</i> var. <i>acuta</i>		2
<i>Nitzschia angustula</i>		2
<i>Nitzschia apiculata</i>	<i>Tryblionella apiculata</i>	1
<i>Nitzschia brevissima</i>		0
<i>Nitzschia capitellata</i>		1
<i>Nitzschia chasei</i>	<i>Simonsenia delognei</i>	3
<i>Nitzschia clausii</i>		2
<i>Nitzschia coarctata</i>	<i>Tryblionella coarctata</i>	3
<i>Nitzschia communis</i>		1
<i>Nitzschia compressa</i>		0
<i>Nitzschia constricta</i>	<i>Psammodictyon constrictum</i>	3
<i>Nitzschia debilis</i>		0
<i>Nitzschia denticula</i>		3
<i>Nitzschia dissipata</i>		3
<i>Nitzschia dissipata</i> var. <i>media</i>		3
<i>Nitzschia dubia</i>		2
<i>Nitzschia elegantula</i>		0
<i>Nitzschia filiformis</i>		1
<i>Nitzschia fonticola</i>		2
<i>Nitzschia frustulum</i>		1
<i>Nitzschia frustulum</i> var. <i>perpusilla</i>		1
<i>Nitzschia gandarsheimiensis</i>		0
<i>Nitzschia gracilis</i>		2
<i>Nitzschia hantzschiana</i>		0

## C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Nitzschia heufferiana</i>		3
<i>Nitzschia hungarica</i>	<i>Tryblionella hungarica</i>	2
<i>Nitzschia inconspicua</i>		2
<i>Nitzschia intermedia</i>		2
<i>Nitzschia kutzingiana</i>	<i>Nitzschia pusilla</i>	0
<i>Nitzschia levidensis</i>	<i>Tryblionella levidensis</i>	3
<i>Nitzschia linearis</i>		3
<i>Nitzschia linearis</i> var. <i>tenuis</i>		3
<i>Nitzschia littoralis</i>	<i>Tryblionella littoralis</i>	3
<i>Nitzschia lorenziana</i> var. <i>subtilis</i>	<i>Nitzschia lorenziana</i>	3
<i>Nitzschia microcephala</i>		1
<i>Nitzschia nana</i>		3
<i>Nitzschia obtusa</i>		0
<i>Nitzschia palea</i>		1
<i>Nitzschia palea</i> var. <i>debilis</i>		1
<i>Nitzschia palea</i> var. <i>tenuirostris</i>		1
<i>Nitzschia paleacea</i>		2
<i>Nitzschia parvula</i>		0
<i>Nitzschia pellucida</i>		0
<i>Nitzschia perminuta</i>		2
<i>Nitzschia pumila</i>		2
<i>Nitzschia pusilla</i>		1
<i>Nitzschia radicola</i>		0
<i>Nitzschia rautenbachiae</i>		3
<i>Nitzschia recta</i>		3
<i>Nitzschia reversa</i>		2
<i>Nitzschia romana</i>	<i>Nitzschia fonticola</i>	3
<i>Nitzschia rostellata</i>		0
<i>Nitzschia sigma</i>		1
<i>Nitzschia sigmoidea</i>		3
<i>Nitzschia sinuata</i> var. <i>tabellaria</i>		3
<i>Nitzschia sociabilis</i>		2
<i>Nitzschia</i> sp.		2
<i>Nitzschia stagnorum</i>	<i>Nitzschia umbonata</i>	0
<i>Nitzschia stricta</i>		0
<i>Nitzschia subacicularis</i>		0
<i>Nitzschia sublinearis</i>		2
<i>Nitzschia termalis</i>	<i>Nitzschia umbonata</i>	0
<i>Nitzschia tropica</i>		2
<i>Nitzschia tryblionella</i>	<i>Tryblionella gracilis</i>	3
<i>Nitzschia tryblionella</i> var. <i>levidensis</i>	<i>Tryblionella levidensis</i>	3
<i>Nitzschia tryblionella</i> var. <i>victoriae</i>	<i>Tryblionella victoriae</i>	3
<i>Nitzschia umbonata</i>		0
<i>Nitzschia vermicularis</i>		2
<i>Nitzschia vitrea</i>		0
<i>Orthoseira roseana</i>		0
<i>Pinnularia abaujensis</i>	<i>Pinnularia gibba</i>	3
<i>Pinnularia abaujensis</i> var. <i>rostrata</i>	<i>Pinnularia gibba</i> var. <i>rostrata</i>	3
<i>Pinnularia abaujensis</i> var. <i>subundulata</i>	<i>Pinnularia gibba</i> f. <i>subundulata</i>	3
<i>Pinnularia acrosphaeria</i> var. <i>turgidula</i>	<i>Pinnularia acrosphaeria</i>	0
<i>Pinnularia appendiculata</i>		2
<i>Pinnularia biceps</i>	<i>Pinnularia interrupta</i>	3
<i>Pinnularia borealis</i>		2
<i>Pinnularia borealis</i> var. <i>rectangularis</i>		2
<i>Pinnularia braunii</i> var. <i>amphicephala</i>		3
<i>Pinnularia legumen</i>		3
<i>Pinnularia maior</i>		0
<i>Pinnularia mesogonglyta</i>	<i>Pinnularia gibba</i> var. <i>mesogonglyta</i>	3
<i>Pinnularia mesolepta</i>	<i>Pinnularia interrupta</i>	3
<i>Pinnularia microstauron</i>		0
<i>Pinnularia nodosa</i>		0
<i>Pinnularia obscura</i>		3
<i>Pinnularia</i> sp.		3
<i>Pinnularia stomatophora</i>		0

**C-5 DIATOM MASTER TAXA LIST (CONT'D)**

Taxa	New Taxa Name	PTI Value
<i>Pinnularia subcapitata</i>		3
<i>Pinnularia subcapitata</i> var. <i>paucistriata</i>		3
<i>Pinnularia terminata</i>		0
<i>Pinnularia viridis</i>		0
<i>Pinnularia viridis</i> var. <i>minor</i>	<i>Pinnularia streptoraphe</i>	0
<i>Plagiotropis lepidoptera</i> var. <i>proboscidea</i>		3
<i>Pleurosigma delicatulum</i>		0
<i>Rhoicosphenia curvata</i>	<i>Rhoicosphenia abbreviata</i>	3
<i>Rhopalodia gibba</i>		3
<i>Rhopalodia gibba</i> var. <i>ventricosa</i>	<i>Rhopalodia gibba</i>	4
<i>Rhopalodia gibberula</i> var. <i>vanheurckii</i>		0
<i>Skeletonema potomos</i>		3
<i>Stauroneis anceps</i>		4
<i>Stauroneis anceps</i> var. <i>americana</i>		4
<i>Stauroneis anceps</i> var. <i>gracilis</i>		4
<i>Stauroneis anceps</i> var. <i>linearis</i>		4
<i>Stauroneis kriegeri</i>		0
<i>Stauroneis legumen</i>		0
<i>Stauroneis nana</i>		0
<i>Stauroneis nobilis</i> f. <i>alabamae</i>	<i>Stauroneis nobilis</i>	0
<i>Stauroneis obtusa</i>		0
<i>Stauroneis phoenicenteron</i>		0
<i>Stauroneis phoenicenteron</i> f. <i>gracilis</i>		0
<i>Stauroneis</i> sp.		4
<i>Stauroneis smithii</i>		4
<i>Stauroneis smithii</i> var. <i>incisa</i>		4
<i>Stauroneis smithii</i> var. <i>sagitta</i>		4
<i>Stenopterobia delicatissima</i>		4
<i>Stephanodiscus alpinus</i>		0
<i>Stephanodiscus dubius</i>	<i>Cyclostephanos dubius</i>	3
<i>Stephanodiscus hantzschii</i>		3
<i>Stephanodiscus invisitatus</i>	<i>Cyclostephanos invisitatus</i>	3
<i>Stephanodiscus minutulus</i>		3
<i>Stephanodiscus niagarae</i>		0
<i>Stephanodiscus</i> sp.		3
<i>Stephanodiscus subtilis</i>		0
<i>Stephanodiscus tenuis</i>	<i>Stephanodiscus hantzschii</i>	3
<i>Surirella agmatilis</i>		3
<i>Surirella angustata</i>		2
<i>Surirella brebissonii</i>		0
<i>Surirella elegans</i>		4
<i>Surirella gracilis</i>		0
<i>Surirella linearis</i>		2
<i>Surirella linearis</i> var. <i>helvetica</i>		2
<i>Surirella ovalis</i>		3
<i>Surirella ovata</i>	<i>Surirella minuta</i>	2
<i>Surirella ovata</i> var. <i>africana</i>	<i>Surirella minuta</i>	2
<i>Surirella ovata</i> var. <i>pinnata</i>	<i>Surirella minuta</i>	3
<i>Surirella robusta</i>		0
<i>Surirella robusta</i> f. <i>lata</i>		0
<i>Surirella robusta</i> var. <i>splendida</i>	<i>Surirella splendida</i>	0
<i>Surirella</i> sp.		2
<i>Surirella splendida</i>		0
<i>Surirella tenera</i>		3
<i>Surirella tenera</i> var. <i>nervosa</i>		3
<i>Synedra acus</i>	<i>Fragilaria ulna</i>	3
<i>Synedra capitata</i>	<i>Fragilaria dilatata</i>	0
<i>Synedra delicatissima</i>	<i>Fragilaria delicatissima</i>	4
<i>Synedra famelica</i>	<i>Fragilaria capucina</i>	4
<i>Synedra fasciculata</i>	<i>Fragilaria fasciculata</i>	1
<i>Synedra fasciculata</i> var. <i>truncata</i>	<i>Fragilaria fasciculata</i>	1
<i>Synedra filiformis</i> var. <i>exilis</i>		4
<i>Synedra nana</i>	<i>Fragilaria nanana</i>	4
<i>Synedra parasitica</i>	<i>Fragilaria parasitica</i>	4

### C-5 DIATOM MASTER TAXA LIST (CONT'D)

Taxa	New Taxa Name	PTI Value
<i>Synedra parasitica</i> var. <i>subconstricta</i>	<i>Fragilaria parasitica</i>	4
<i>Synedra pulchella</i>	<i>Ctenophora pulchella</i>	1
<i>Synedra pulchella</i> var. <i>lacerata</i>	<i>Ctenophora pulchella</i>	1
<i>Synedra radians</i>	<i>Fragilaria capucina</i>	1
<i>Synedra rumpens</i>	<i>Fragilaria capucina</i> var. <i>rumpens</i>	4
<i>Synedra rumpens</i> var. <i>familiaris</i>	<i>Fragilaria capucina</i> var. <i>gracilis</i>	4
<i>Synedra rumpens</i> var. <i>fragilarioides</i>	<i>Fragilaria capucina</i> var. <i>vaucheriae</i>	4
<i>Synedra rumpens</i> var. <i>scotica</i>	<i>Fragilaria capucina</i> var. <i>gracilis</i>	4
<i>Synedra</i> sp.		3
<i>Synedra ulna</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>amphirhynchus</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>contracta</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>danica</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>oxyrhynchus</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>oxyrhynchus</i> f. <i>mediocontracta</i>	<i>Fragilaria ulna</i>	3
<i>Synedra ulna</i> var. <i>ramesi</i>	<i>Fragilaria ulna</i>	3
<i>Tabellaria fenestrata</i>		4
<i>Tabellaria flocculosa</i>		4
<i>Tetracyclus glans</i>		0
<i>Tetracyclus rupestris</i>		0
<i>Thalassiosira weissflogii</i>		2

## C-6 PHYTOPLANKTON FIELD DATA SHEET

<b>DOW Station ID Number:</b>	<b>Stream Name:</b>	<b>Location:</b>
<b>Collection Date:</b>	<b>Time:</b>	<b>County:</b>
<b>River Basin:</b>	<b>Purpose of Study:</b>	<b>Name of Investigator(s):</b>

<b>Collection Method Used:</b>	
<b>Analyses to be Completed:</b>	Chlorophyll <i>a</i> ____ Ash-Free Dry-Mass ____ Phytoplankton Count ____
<b>Bloom Characteristics:</b>	Is there an odor? Is there an oily sheen? What is the thickness of the bloom? What is the blooms color? What organism might be the problem?

### Surface Bloom Coverage Assessment (Check One)

<b>Total (100% Coverage; Bank to Bank)</b>	
<b>Dense (60 - 99% Coverage)</b>	
<b>Moderate (30 - 59% Coverage)</b>	
<b>Sparse (1 - 29% Coverage)</b>	
<b>Absent</b>	

## C-7 CHLOROPHYLL a (mg/m<sup>2</sup>) BENCH SHEET

Stream or Lake: \_\_\_\_\_ Station #: \_\_\_\_\_

Location: \_\_\_\_\_

Date Collected: \_\_\_\_\_ Date Analyzed: \_\_\_\_\_

Replicates: \_\_\_\_\_ Vial #: \_\_\_\_\_

	A	B	C
Area:	_____	_____	_____
Volume:	_____	_____	_____
Subsample:	_____	_____	_____
Extract ml:	_____	_____	_____
Dilution:	_____	_____	_____
Sensitivity:	_____	_____	_____
Fluorometer:	_____	_____	_____

Chlorophyll a: \_\_\_\_\_

Mean: \_\_\_\_\_  
Standard Deviation: \_\_\_\_\_  
c.v.%: \_\_\_\_\_

Sensitivity:

MS = 1  
3.16 = 2  
10 = 3  
31.6 = 4

## C-7 ASH-FREE DRY MASS (g/m<sup>2</sup>) BENCH SHEET

Stream or Lake: \_\_\_\_\_ Station #: \_\_\_\_\_

Location: \_\_\_\_\_

Date Collected: \_\_\_\_\_ Date Analyzed: \_\_\_\_\_

Replicates: \_\_\_\_\_

	A	B	C
Crucible Number:	_____	_____	_____
Area:	_____	_____	_____
Volume:	_____	_____	_____
Subsample Volume:	_____	_____	_____
Crucible Weight:	_____	_____	_____
Dried Weight:	_____	_____	_____
Ashed Weight:	_____	_____	_____
Dry Weight:	_____	_____	_____
AFDM:	_____	_____	_____

### DRY WEIGHT

Mean: \_\_\_\_\_  
Standard Deviation: \_\_\_\_\_  
c.v.%: \_\_\_\_\_

### ASH-FREE DRY MASS

Mean: \_\_\_\_\_  
Standard Deviation: \_\_\_\_\_  
c.v.%: \_\_\_\_\_



# C-8 PHYTOPLANKTON BENCH SHEET

<b>DOW Station ID Number:</b>		<b>Stream Name:</b>		<b>Location:</b>	
<b>Collection Date:</b>		<b>County:</b>		<b>Name of Collector(s):</b>	
<b>River Basin:</b>		<b>DOW Program:</b>		<b>Name of Taxonomist(s):</b>	
Taxa Name	Algal Division	Tally (If Counting)	# of Algal Units (If Counting)	Estimated Abundance	

Total Number of Taxa \_\_\_\_\_

Total Number of Divisions \_\_\_\_\_

**C-8 PHYTOPLANKTON BENCH SHEET (CONT'D)**

<b>DOW Station ID Number:</b>		<b>Stream Name:</b>		<b>Location:</b>	
<b>Collection Date:</b>		<b>County:</b>		<b>Name of Collector(s):</b>	
<b>River Basin:</b>		<b>DOW Program:</b>		<b>Name of Taxonomist(s):</b>	
<b>Taxa Name</b>	<b>Algal Division</b>	<b>Tally (If Counting)</b>	<b># of Algal Units (If Counting)</b>	<b>Estimated Abundance</b>	

**APPENDIX D-1 MASTER MACROINVERTEBRATE TAXA LIST**

**APPENDIX D-2 MACROINVERTEBRATE LABORATORY BENCH SHEET**

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Haplosclerina	Spongillidae	Spongilla sp	3.0	3	CF	
Haplosclerina	Spongillidae	Unidentified Spongillid	3.0	3	CF	
Trachylina	Petasidae	Craspedacusta sowerbyi	8.4	8	CG	
Hydroida	Hydriidae	Hydra sp	5.0	5	CG	
Tricladida	Planariidae	Dugesia sp	5.0	5	CG	
Tricladida	Planariidae	Phagocata sp	5.0	5	CG	
Tricladida	Planariidae	Planaria sp	5.0	5	CG	
Tricladida	Planariidae	Unidentified Planariid	5.0	5	CG	
Gordioidea	Gordiidae	Gordius sp	5.0	5		
Gordioidea	Parachordodidae	Paragordius sp	5.9	6		
Mesogastropoda	Valvatidae	Valvata bicarinata	5.0	5	SC	
Mesogastropoda	Valvatidae	Valvata tricarinata	5.0	5	SC	
Mesogastropoda	Viviparidae	Campeloma crassulum	5.0	5	SH	
Mesogastropoda	Viviparidae	Campeloma decisum	6.5	5	SH	
Mesogastropoda	Viviparidae	Campeloma integrumd	5.0	5	SH	
Mesogastropoda	Viviparidae	Campeloma sp	5.0	5	SH	
Mesogastropoda	Viviparidae	Cipangopaludina chinensis malleata	5.0	5	SC	
Mesogastropoda	Viviparidae	Lioplax subcarinata	5.0	5	SC	
Mesogastropoda	Viviparidae	Lioplax sulculosa	5.0	5	SC	
Mesogastropoda	Viviparidae	Viviparus georgianus	5.0	5	SC	
Mesogastropoda	Viviparidae	Viviparus sp	5.0	5	SC	
Mesogastropoda	Hydrobiidae	Amnicola limosa limosa	4.8	6	SC	
Mesogastropoda	Hydrobiidae	Amnicola limosa parva	4.8	6	SC	
Mesogastropoda	Hydrobiidae	Amnicola sp	5.2	6	SC	
Mesogastropoda	Hydrobiidae	Cincinnatiata integra	5.7	6	SC	
Mesogastropoda	Hydrobiidae	Probythinella lacustris	5.7	6	SC	
Mesogastropoda	Hydrobiidae	Somatogyrus integra	6.5	6	SC	
Mesogastropoda	Hydrobiidae	Somatogyrus sp	6.4	6	SC	
Mesogastropoda	Hydrobiidae	Somatogyrus trothis	6.5	6	SC	
Mesogastropoda	Pomatiopsidae	Pomatiopsis cincinnatiensis	5.0	5	SC	
Mesogastropoda	Bithyniidae	Bithynia tentaculata	5.0	5	SC	
Lymnophila	Ancylidae	Ferrissia fragilis	6.9	7	SC	
Lymnophila	Ancylidae	Ferrissia rivularis	6.9	7	SC	
Lymnophila	Ancylidae	Ferrissia sp	6.6	7	SC	
Lymnophila	Ancylidae	Laevapex diaphanus	7.0	7	SC	
Lymnophila	Ancylidae	Laevapex fuscus	7.5	7	SC	
Lymnophila	Ancylidae	Laevapex sp	7.5	7	SC	
Lymnophila	Ancylidae	Unidentified Ancylidae	5.0	7	SC	
Lymnophila	Lymnaeidae	Fossaria sp	7.0	8	SC	
Lymnophila	Lymnaeidae	Lymnaea sp	7.0	8	SC	
Lymnophila	Lymnaeidae	Lymnaea stagnalis	7.0	8	SC	
Lymnophila	Lymnaeidae	Pseudosuccinea columella	7.7	8	SC	
Lymnophila	Lymnaeidae	Stagnicola sp	8.2	8	SC	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Lymnophila	Lymnaeidae	Unidentified Lymnaeid	7.0	8	SC	
Basommatophora	Physidae	Physella globosa	8.8	9	SC	
Basommatophora	Physidae	Physella gyrina	8.8	9	SC	
Basommatophora	Physidae	Physella heterostropha	8.8	9	SC	
Basommatophora	Physidae	Physella heterostropha pomila	8.8	9	SC	
Basommatophora	Physidae	Physella integra	8.8	9	SC	
Basommatophora	Physidae	Physella integra brevispira	8.8	9	SC	
Basommatophora	Physidae	Physella sp	8.8	9	SC	
Mesogastropoda	Pleuroceridae	Elimia costifera	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia curreyana	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia ebenum	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia laqueata costulata	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia laqueata laqueata	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia livescens	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia plicatastriata	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia semicarinata	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia sp	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Elimia sp #1	2.5	3	SC	
Mesogastropoda	Pleuroceridae	Leptoxis praerosa	1.6	3	SC	
Mesogastropoda	Pleuroceridae	Leptoxis sp	1.8	3	SC	
Mesogastropoda	Pleuroceridae	Leptoxis trilineata	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia armigera	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia geniculata	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia obovata	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia salebroza	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia sp	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Lithasia verrucosa	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera acuta	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera alveare	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera canaliculata	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera canaliculata undulatum	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera sp	3.0	3	SC	
Mesogastropoda	Pleuroceridae	Pleurocera sp 1	3.0	3	SC	
Lymnophila	Planorbidae	Gyraulus parvus	7.5	7	SC	
Lymnophila	Planorbidae	Helisoma anceps anceps	6.2	7	SC	
Lymnophila	Planorbidae	Helisoma sp	6.5	7	SC	
Lymnophila	Planorbidae	Menetus dilatatus	8.2	7	SC	
Lymnophila	Planorbidae	Planorbella sp	6.8	7	SC	
Mesogastropoda	Planorbidae	Planorbella trivolvis	6.5	7	SC	
Lymnophila	Planorbidae	Planorbula sp	6.8	7	SC	
Lymnophila	Planorbidae	Promenetus exacuus	7.5	7	SC	
Lymnophila	Planorbidae	Promenetus sp	7.5	7	SC	
Lymnophila	Planorbidae	Unidentified Planorbid	7.0	7	SC	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Heterodonta	Sphaeriidae	Eupera cubensis	5.7	7	CF	
Heterodonta	Sphaeriidae	Musculium lacustre	7.7	7	CF	
Heterodonta	Sphaeriidae	Musculium partumeium	7.7	7	CF	
Heterodonta	Sphaeriidae	Musculium securis	7.7	7	CF	
Heterodonta	Sphaeriidae	Musculium sp	7.7	7	CF	
Heterodonta	Sphaeriidae	Musculium transversum	7.7	7	CF	
Heterodonta	Sphaeriidae	Pisidium adamsi	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium amnicum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium casertanum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium compressum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium conventus	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium cruciatum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium dubium	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium equilaterale	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium fallax	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium ferrugineum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium idahoense	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium lilljeborgi	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium nitidum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium sp	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium subtruncatum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium variabile	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium ventricosum	6.5	7	CF	
Heterodonta	Sphaeriidae	Pisidium walkeri	6.5	7	CF	
Heterodonta	Sphaeriidae	Sphaerium corneum	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium fabale	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium nitidum	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium occidentale	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium patella	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium rhomboideum	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium sexmaculatus	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium simile	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium sp	7.6	7	CF	
Heterodonta	Sphaeriidae	Sphaerium striatinum	7.6	7	CF	
Heterodonta	Sphaeriidae	Unidentified Sphaeriid		7	CF	
Pelecypoda	Unionidae	Actinonaias ligamentina	5.0	4	CF	
Pelecypoda	Unionidae	Actinonaias pectorosa	1.0	4	CF	
Pelecypoda	Unionidae	Alasmidonta atropurpurea	1.0	4	CF	
Pelecypoda	Unionidae	Alasmidonta marginata	3.0	4	CF	
Pelecypoda	Unionidae	Alasmidonta viridis	3.0	4	CF	
Pelecypoda	Unionidae	Amblema plicata	5.0	4	CF	
Pelecypoda	Unionidae	Anodonta suborbiculata	5.0	4	CF	
Pelecypoda	Unionidae	Anodontoides ferussacianus	3.0	4	CF	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Pelecypoda	Unionidae	Arcidens confragosus	3.0	4	CF	
Pelecypoda	Unionidae	Cumberlandia monodonta	1.0	4	CF	
Pelecypoda	Unionidae	Cyclonaias tuberculata	5.0	4	CF	
Pelecypoda	Unionidae	Cyrogenia stegaria	1.0	4	CF	
Pelecypoda	Unionidae	Dromus dromas	1.0	4	CF	
Pelecypoda	Unionidae	Ellipsaria lineolata	3.0	4	CF	
Pelecypoda	Unionidae	Elliptio crassidens	5.0	4	CF	
Pelecypoda	Unionidae	Elliptio dilatata	5.0	4	CF	
Pelecypoda	Unionidae	Epioblasma arcaeformis	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma biemarginata	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma brevidens	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma capsaeformis	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma flexuosa	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma florentina florentina	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma florentina walkeri	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma haysiana	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma lewisii	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma obliquata obliquata	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma obliquata perobliqua	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma personata	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma propinqua	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma sampsonii	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma stewardsonii	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma torulosa rangiana	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma torulosa torulosa	1.0	4	CF	
Pelecypoda	Unionidae	Epioblasma triquetra	1.0	4	CF	
Pelecypoda	Unionidae	Fusconaia ebena	5.0	4	CF	
Pelecypoda	Unionidae	Fusconaia flava	5.0	4	CF	
Pelecypoda	Unionidae	Fusconaia subrotunda	1.0	4	CF	
Pelecypoda	Unionidae	Glebulia rotundata	3.0	4	CF	
Pelecypoda	Unionidae	Hemistena lata	1.0	4	CF	
Pelecypoda	Unionidae	Lampsilis abrupta	1.0	4	CF	
Pelecypoda	Unionidae	Lampsilis cardium	3.0	4	CF	
Pelecypoda	Unionidae	Lampsilis fasciola	3.0	4	CF	
Pelecypoda	Unionidae	Lampsilis ovata	1.0	4	CF	
Pelecypoda	Unionidae	Lampsilis siliquoidea	5.0	4	CF	
Pelecypoda	Unionidae	Lampsilis teres	3.0	4	CF	
Pelecypoda	Unionidae	Lasmigona complanata complanata	3.0	4	CF	
Pelecypoda	Unionidae	Lasmigona compressa	3.0	4	CF	
Pelecypoda	Unionidae	Lasmigona costata	5.0	4	CF	
Pelecypoda	Unionidae	Lasmigona subviridis	3.0	4	CF	
Pelecypoda	Unionidae	Leptodea fragilis	5.0	4	CF	
Pelecypoda	Unionidae	Leptodea leptodon	1.0	4	CF	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Pelecypoda	Unionidae	Lexingtonia dolabelloides	1.0	4	CF	
Pelecypoda	Unionidae	Ligumia recta	3.0	4	CF	
Pelecypoda	Unionidae	Ligumia subrostrata	3.0	4	CF	
Pelecypoda	Unionidae	Medionidus conradicus	1.0	4	CF	
Pelecypoda	Unionidae	Megaloniaias nervosa	5.0	4	CF	
Pelecypoda	Unionidae	Obliquaria reflexa	1.0	4	CF	
Pelecypoda	Unionidae	Obovaria olivaria	1.0	4	CF	
Pelecypoda	Unionidae	Obovaria retusa	1.0	4	CF	
Pelecypoda	Unionidae	Obovaria subrotunda	1.0	4	CF	
Pelecypoda	Unionidae	Pegias fabula	1.0	4	CF	
Pelecypoda	Unionidae	Plectomerus dombeyanus	1.0	4	CF	
Pelecypoda	Unionidae	Plethobasus cicatricosus	1.0	4	CF	
Pelecypoda	Unionidae	Plethobasus cooperianus	1.0	4	CF	
Pelecypoda	Unionidae	Plethobasus cyphus	3.0	4	CF	
Pelecypoda	Unionidae	Pleurobema clava	1.0	4	CF	
Pelecypoda	Unionidae	Pleurobema coccineum	1.0	4	CF	
Pelecypoda	Unionidae	Pleurobema cordatum	5.0	4	CF	
Pelecypoda	Unionidae	Pleurobema oviforme	1.0	4	CF	
Pelecypoda	Unionidae	Pleurobema plenum	1.0	4	CF	
Pelecypoda	Unionidae	Pleurobema rubrum	1.0	4	CF	
Pelecypoda	Unionidae	Pleurobema sintoxia	1.0	4	CF	
Pelecypoda	Unionidae	Potamilus alatus	5.0	4	CF	
Pelecypoda	Unionidae	Potamilus capax	1.0	4	CF	
Pelecypoda	Unionidae	Potamilus ohioensis	3.0	4	CF	
Pelecypoda	Unionidae	Potamilus purpuratus	3.0	4	CF	
Pelecypoda	Unionidae	Ptychobranhus fasciolaris	5.0	4	CF	
Pelecypoda	Unionidae	Ptychobranhus subtentum	1.0	4	CF	
Pelecypoda	Unionidae	Pyganodon grandis	5.0	4	CF	
Pelecypoda	Unionidae	Quadrula apiculata	1.0	4	CF	
Pelecypoda	Unionidae	Quadrula cylindrica	1.0	4	CF	
Pelecypoda	Unionidae	Quadrula fragosa	1.0	4	CF	
Pelecypoda	Unionidae	Quadrula metanevra	3.0	4	CF	
Pelecypoda	Unionidae	Quadrula nodulata	3.0	4	CF	
Pelecypoda	Unionidae	Quadrula pustulosa	5.0	4	CF	
Pelecypoda	Unionidae	Quadrula quadrula	5.0	4	CF	
Pelecypoda	Unionidae	Quadrula sparsa	1.0	4	CF	
Pelecypoda	Unionidae	Simpsonaias ambigua	1.0	4	CF	
Pelecypoda	Unionidae	Strophitus undulatus	3.0	4	CF	
Pelecypoda	Unionidae	Toxolasma lividus	3.0	4	CF	
Pelecypoda	Unionidae	Toxolasma parvus	5.0	4	CF	
Pelecypoda	Unionidae	Toxolasma texasiensis	5.0	4	CF	
Pelecypoda	Unionidae	Tritogonia verrucosa	5.0	4	CF	
Pelecypoda	Unionidae	Truncilla donaciformis	3.0	4	CF	



**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Pelecypoda	Unionidae	Truncilla truncata	3.0	4	CF	
Pelecypoda	Unionidae	Uniomerus tetralasmus	5.0	4	CF	
Pelecypoda	Unionidae	Utterbackia imbecillis	5.0	4	CF	
Pelecypoda	Unionidae	Venustaconcha ellipsiformis	3.0	4	CF	
Pelecypoda	Unionidae	Villosa fabalis	1.0	4	CF	
Pelecypoda	Unionidae	Villosa iris	3.0	4	CF	
Pelecypoda	Unionidae	Villosa lienosa	3.0	4	CF	
Pelecypoda	Unionidae	Villosa ortmanni	3.0	4	CF	
Pelecypoda	Unionidae	Villosa taeniata	5.0	4	CF	
Pelecypoda	Unionidae	Villosa trabalis	1.0	4	CF	
Pelecypoda	Unionidae	Villosa vanuxemensis	3.0	4	CF	
Pelecypoda	Corbiculidae	Corbicula fluminea	6.1	6	CF	
Pelecypoda	Dreissenidae	Dreissena polymorpha	5.0	5	CF	
Haplotaxida	Enchytraeidae	Enchytraeus sp	10.0	10	CG	
Lumbriculida	Lumbriculidae	Eclipidrilus sp	7.3	7	CG	
Lumbriculida	Lumbriculidae	Lumbriculus inconstans	7.3	7	CG	
Lumbriculida	Lumbriculidae	Lumbriculus sp	7.3	7	CG	
Lumbriculida	Lumbriculidae	Lumbriculus variegatus	7.3	7	CG	
Lumbriculida	Lumbriculidae	Unidentified Lumbriculid	7.3	7	CG	
Haplotaxida	Naididae	Arcteonais lomondi	8.0	9	CG	
Haplotaxida	Naididae	Bratislavia unidentata	9.0	9	CG	
Haplotaxida	Naididae	Chaetogaster limnaei	6.0	9	CG	
Haplotaxida	Naididae	Chaetogaster sp	6.0	9	CG	
Haplotaxida	Naididae	Dero digitata	10.0	9	CG	
Haplotaxida	Naididae	Dero furcata	9.0	9	CG	
Haplotaxida	Naididae	Dero nivea	10.0	9	CG	
Haplotaxida	Naididae	Dero sp	9.0	9	CG	
Haplotaxida	Naididae	Dero trifida	8.0	9	CG	
Haplotaxida	Naididae	Haemonais waldvogeli	9.5	9	CG	
Haplotaxida	Naididae	Nais barbata	5.0	9	CG	
Haplotaxida	Naididae	Nais bretscheri	8.0	9	CG	
Haplotaxida	Naididae	Nais communis	8.8	9	CG	
Haplotaxida	Naididae	Nais simplex	8.8	9	CG	
Haplotaxida	Naididae	Nais sp	8.9	9	CG	
Haplotaxida	Naididae	Nais variabilis	8.9	9	CG	
Haplotaxida	Naididae	Ophidonais serpentina	7.5	9	CG	
Haplotaxida	Naididae	Pristina aquiseta	9.0	9	CG	
Haplotaxida	Naididae	Pristina jenhinae		9	CG	
Haplotaxida	Naididae	Pristina leidyi	9.0	9	CG	
Haplotaxida	Naididae	Pristina longeseta		9	CG	
Haplotaxida	Naididae	Pristina sp	9.6	9	CG	
Haplotaxida	Naididae	Pristina synclites	8.0	9	CG	
Haplotaxida	Naididae	Pristinella jenkiniae	8.0	9	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Haplotaxida	Naididae	Pristinella longidentata	8.0	9	CG	
Haplotaxida	Naididae	Pristinella osborni	8.0	9	CG	
Haplotaxidae	Naididae	Pristinella sp	8.0	9	CG	
Haplotaxida	Naididae	Slavina appendiculata	7.1	9	CG	
Haplotaxida	Naididae	Stephensoniana trivandrana	9.1	9	GG	
Haplotaxida	Naididae	Stylaria fossularis	8.5	9	CG	
Haplotaxida	Naididae	Stylaria lacustris	8.5	9	CG	
Haplotaxida	Naididae	Stylaria sp	8.5	9	CG	
Haplotaxida	Naididae	Unidentified Naidid	9.1	9	CG	
Haplotaxida	Tubificidae	Aulodrilus pigueti	5.5	9	CG	
Haploplaxida	Tubificidae	Aulodrilus pleuriseteta	5.0	9	CG	
Haplotaxida	Tubificidae	Branchiura sowerbyi	8.3	9	CG	
Haplotaxida	Tubificidae	Ilyodrilus templetoni	9.3	9	CG	
Haplotaxida	Tubificidae	Limnodrilus cervix	9.0	9	CG	
Haplotaxida	Tubificidae	Limnodrilus claparedeianus	9.6	9	CG	
Haplotaxida	Tubificidae	Limnodrilus hoffmeisteri	9.5	9	CG	
Haplotaxida	Tubificidae	Limnodrilus maumeensis	9.0	9	CG	
Haplotaxida	Tubificidae	Limnodrilus sp	9.5	9	CG	
Haplotaxida	Tubificidae	Limnodrilus ukedemianus	9.5	9	CG	
Haplotaxida	Tubificidae	Limnodrilus/tubifix	9.0	9	CG	
Haplotaxida	Tubificidae	Pelosclex multisetosus	8.8	9	CG	
Haplotaxida	Tubificidae	Spirosperma ferox	7.7	9	CG	
Haplotaxida	Tubificidae	Tubifex sp	10.0	9	CG	
Haplotaxida	Tubificidae	Tubifex tubifex	10.0	9	CG	
Haplotaxida	Tubificidae	UIW/OCS sp	9.0	9	CG	
Haplotaxida	Tubificidae	UIWCS sp	9.0	9	CG	
Haplotaxida	Tubificidae	Unidentified Tubificidae	9.0	9	CG	
Haplotaxida	Lumbricidae	Haplotaxis sp	9.0	9	CG	
Haplotaxida	Lumbricidae	Unidentified Lumbricid	5.0	9	CG	
Haplotaxida	Branchiobdellidae	Branchiobdella americana	5.0	6	SC	
Haplotaxida	Branchiobdellidae	Cambarincola elevata	6.2	6	SC	
Haplotaxida	Aeolosomatidae	Aeolosoma sp	5.0	5	CG	
Rhynchobdellida	Piscicolidae	Cystobranchnus mammillatus	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Cystobranchnus virginicus	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Myzobdella lugubris	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Piscicola geometra	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Piscicola punctata	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Piscicolaria reducta	8.2	8	PC	
Rhynchobdellida	Piscicolidae	Piscicolaria sp	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Actinobdella annectens	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Actinobdella inequiannulata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Alboglossiphonia heteroclita	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Batracobdella cryptobranchii	7.6	8	PC	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Rhynchobdellida	Glossiphoniidae	Batracobdella michiganensis	7.6	8	PC	
Rhynchobdellida	Glossiphoniidae	Batracobdella paludosa	7.6	8	PC	
Rhynchobdellida	Glossiphoniidae	Batracobdella phalera	7.6	8	PC	
Rhynchobdellida	Glossiphoniidae	Batracobdella picta	7.6	8	PC	
Rhynchobdellida	Glossiphoniidae	Boreobdella verrucata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Glossiphonia complanata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella elongata	9.5	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella fusca	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella papillata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella sp	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella stagnalis	8.6	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella transversa	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Helobdella triserialis	9.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Oligobdella biannulata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella montifera	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella multilineata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella ornata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella papillifera	9.0	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella parasitica	8.7	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella pediculata	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Placobdella sp	9.0	8	PC	
Rhynchobdellida	Glossiphoniidae	Theromyzon biannulatum	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Theromyzon rude	8.2	8	PC	
Rhynchobdellida	Glossiphoniidae	Unidentified Glossiphoniid	8.2	8	PC	
Pharyngobdellida	Erpobdellidae	Dina anoculata	8.2	8	PC	
Pharyngobdellida	Erpobdellidae	Dina dubia	8.2	8	PC	
Pharyngobdellida	Erpobdellidae	Dina parva	8.2	8	CG	
Pharyngobdellida	Erpobdellidae	Erpobdella punctata	7.8	8	CG	
Pharyngobdellida	Erpobdellidae	Mooreobdella bucera	7.8	8	CG	
Pharyngobdellida	Erpobdellidae	Mooreobdella fervida	7.8	8	CG	
Pharyngobdellidae	Erpobdellidae	Mooreobdella melanostoma	7.8	8	CG	
Pharyngobdellida	Erpobdellidae	Mooreobdella microstoma	7.8	8	CG	
Pharyngobdellida	Erpobdellidae	Mooreobdella sp	7.8	8	CG	
Pharyngobdellida	Erpobdellidae	Unidentified Erpobdellid	8.2	8	CG	
Gnathobdellida	Hirudinidae	Haemopsis grandis	8.2	8	PC	
Gnathobdellida	Hirudinidae	Haemopsis lateromaculata	8.2	8	PC	
Gnathobdellida	Hirudinidae	Haemopsis marmorata	8.2	8	PC	
Gnathobdellida	Hirudinidae	Haemopsis sp	8.2	8	PC	
Gnathobdellida	Hirudinidae	Haemopsis terrestris	8.2	8	PC	
Gnathobdellida	Hirudinidae	Hirudo medicinalis	8.2	8	PC	
Gnathobdellida	Hirudinidae	Macrobdella decora	8.2	8	PC	
Gnathobdellida	Hirudinidae	Macrobdella diploptertia	8.2	8	PC	
Gnathobdellida	Hirudinidae	Macrobdella ditetra	8.2	8	PC	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Gnathobdellida	Hirudinidae	Macrobdella sp	8.2	8	PC	
Gnathobdellida	Hirudinidae	Philobdella floridana	8.2	8	PC	
Gnathobdellida	Hirudinidae	Philobdella gracilis	8.2	8	PC	
Gnathobdellida	Hirudinidae	Philobdella sp	8.2	8	PC	
Gnathobdellida	Hirudinidae	Unidentified Hirudinea	8.2	8	PC	
Bryozoa	Pectinatellidae	Pectinatella magnifica	3.0	3	CG	
Bryozoa	Plumatellidae	Hyalinella punctata	5.0	5	CF	
Bryozoa	Plumatellidae	Plumatella emarginata	5.0	5	CF	
Bryozoa	Plumatellidae	Plumatella repens	5.0	5	CF	
Bryozoa	Plumatellidae	Plumatella sp	5.0	5	CF	
Collembola	Isotomidae	Folsomia sp	5.0	5	CG	
Collembola	Isotomidae	Isotoma sp	5.0	5	CG	
Collembola	Isotomidae	Isotomurus sp	5.0	5	CG	
Ephemeroptera	Polymitarcyidae	Ephoron album	2.0	2	CG	
Ephemeroptera	Polymitarcyidae	Ephoron leukon	1.3	2	CG	
Ephemeroptera	Leptophlebiidae	Choroterpes basalis	2.3	3	SC	
Ephemeroptera	Leptophlebiidae	Choroterpes sp	2.3	3	SC	X
Ephemeroptera	Leptophlebiidae	Habrophlebia vibrans	0.5	3	CG	
Ephemeroptera	Leptophlebiidae	Habrophlebiodes americana	2.3	3	CG	
Ephemeroptera	Leptophlebiidae	Habrophlebiodes sp	2.3	3	SC	
Ephemeroptera	Leptophlebiidae	Leptophlebia austrina	6.2	3	CG	
Ephemeroptera	Leptophlebiidae	Leptophlebia grandis	6.2	3	SC	
Ephemeroptera	Leptophlebiidae	Leptophlebia intermedia	6.2	3	CG	
Ephemeroptera	Leptophlebiidae	Leptophlebia sp	6.2	3	CG	
Ephemeroptera	Leptophlebiidae	Paraleptophlebia assimilis	0.9	3	CG	
Ephemeroptera	Leptophlebiidae	Paraleptophlebia debilis	0.9	3	CG	
Ephemeroptera	Leptophlebiidae	Paraleptophlebia guttata	0.9	3	CG	
Ephemeroptera	Leptophlebiidae	Paraleptophlebia sp	0.9	3	CG	
Ephemeroptera	Leptophlebiidae	Unidentified Leptophlebiid	3.3	3	CG	
Ephemeroptera	Isonychiidae	Isonychia sp	3.5	4	CF	
Ephemeroptera	Heptageniidae	Cinygmula subequalis	0.0	3	SC	X
Ephemeroptera	Heptageniidae	Epeorus dispar	1.0	3	SC	X
Ephemeroptera	Heptageniidae	Epeorus rubidus/subpallidus	1.2	3	SC	X
Ephemeroptera	Heptageniidae	Epeorus sp	1.3	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia flavescens	2.8	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia julia	0.0	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia marginalis	2.3	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia pulla	1.9	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia sp	2.6	3	SC	X
Ephemeroptera	Heptageniidae	Heptagenia spinosa	2.8	3	SC	X
Ephemeroptera	Heptageniidae	Leucrocuta aphrodite	2.4	3	SC	X
Ephemeroptera	Heptageniidae	Leucrocuta hebe	2.8	3	SC	X
Ephemeroptera	Heptageniidae	Leucrocuta junio	2.8	3	SC	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Ephemeroptera	Heptageniidae	Leucrocuta maculipennis	2.8	3	SC	X
Ephemeroptera	Heptageniidae	Leucrocuta sp	2.4	3	SC	X
Emphemeroptera	Heptageniidae	Nixe sp		3	CG	X
Ephemeroptera	Heptageniidae	Rhithrogena amica	0.3	3	SC	X
Ephemeroptera	Heptageniidae	Stenacron candidum	4.0	3	CG	X
Ephemeroptera	Heptageniidae	Stenacron carolina	1.1	3	CG	X
Ephemeroptera	Heptageniidae	Stenacron gildersleevi	2.5	3	SC	X
Ephemeroptera	Heptageniidae	Stenacron interpunctatum	6.9	3	CG	X
Ephemeroptera	Heptageniidae	Stenacron minnetonka	4.0	3	CG	X
Ephemeroptera	Heptageniidae	Stenacron pallidum	2.7	3	SC	X
Ephemeroptera	Heptageniidae	Stenacron sp	4.0	3	CG	X
Ephemeroptera	Heptageniidae	Stenonema bednariki	5.0	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema exiguum	3.8	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema femoratum	7.2	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema integrum	5.8	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema ithaca	3.6	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema mediopunctatum	3.8	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema meririvulanum	0.1	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema modestum	5.5	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema pudicum	2.0	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema pulchellum	4.1	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema sp	4.1	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema terminatum	4.1	3	SC	X
Ephemeroptera	Heptageniidae	Stenonema vicarium	1.3	3	SC	X
Ephemeroptera	Heptageniidae	Unidentified Heptageniid	3.2	3	SC	X
Ephemeroptera	Potamanthidae	Anthopotamus distinctus	1.6	2	CG	
Ephemeroptera	Potamanthidae	Anthopotamus myops	1.6	2	CG	
Ephemeroptera	Potamanthidae	Anthopotamus sp	1.6	2	CG	
Ephemeroptera	Potamanthidae	Anthopotamus verticis	1.6	2	CG	
Ephemeroptera	Siphonuridae	Siphonurus mirus	2.6	4	CG	
Ephemeroptera	Siphonuridae	Siphonurus sp	5.8	4	CG	
Ephemeroptera	Ameletidae	Ameletus lineatus	2.4	2	SC	
Ephemeroptera	Ameletidae	Ameletus sp	2.4	2	SC	
Ephemeroptera	Palingeniidae	Pentagenia robusta	5.0	5	CG	
Ephemeroptera	Palingeniidae	Pentagenia sp	5.0	5	CG	
Ephemeroptera	Palingeniidae	Pentagenia vittigera	5.0	5	CG	
Ephemeroptera	Tricorythidae	Leptohyphes sp	1.4	5	CG	X
Ephemeroptera	Tricorythidae	Tricorythodes albilineatus	5.4	5	CG	
Ephemeroptera	Tricorythidae	Tricorythodes sp	5.1	5	CG	
Ephemeroptera	Tricorythidae	Tricorythodes sp #1	5.1	5	CG	
Ephemeroptera	Ephemeridae	Ephemera guttulata	0.0	4	CG	
Ephemeroptera	Ephemeridae	Ephemera simulans	2.2	4	CG	
Ephemeroptera	Ephemeridae	Ephemera sp	2.2	4	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Ephemeroptera	Ephemeridae	Ephemera varia	2.2	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia atrocaudata	4.9	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia bilineata	4.9	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia limbata	4.9	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia munda	4.9	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia rigida	4.9	4	CG	
Ephemeroptera	Ephemeridae	Hexagenia sp	4.9	4	CG	
Ephemeroptera	Ephemerellidae	Attenella attenuata	1.6	2	CG	X
Ephemeroptera	Ephemerellidae	Attenella sp	1.6	2	CG	X
Ephemeroptera	Ephemerellidae	Drunella allegheniensis	0.8	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella cornutella	0.0	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella lata	0.0	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella longicornis	0.7	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella sp	0.7	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella tuberculata	0.0	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella walkeri	1.0	2	SC	X
Ephemeroptera	Ephemerellidae	Drunella wayah	0.0	2	SC	X
Ephemeroptera	Ephemerellidae	Ephemerella argo	1.7	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella catawba	4.4	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella crenula	1.7	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella dorothea	1.7	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella hispida	0.8	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella inconstans	1.7	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella invaria gr	2.4	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella needhami	0.0	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella rossi	0.0	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella rotunda	2.6	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella septentrionalis	2.0	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella sp	2.0	2	CG	X
Ephemeroptera	Ephemerellidae	Ephemerella subvaria	0.0	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella aestiva	1.5	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella bicolor	4.9	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella enoensis	4.0	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella funeralis	2.1	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella macdunnoughi	1.5	2	SC	X
Ephemeroptera	Ephemerellidae	Eurylophella minimella	3.0	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella sp	4.3	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella temporalis	4.3	2	CG	X
Ephemeroptera	Ephemerellidae	Eurylophella verisimilis	0.3	2	CG	X
Ephemeroptera	Ephemerellidae	Serratella deficiens	2.8	2	CG	X
Ephemeroptera	Ephemerellidae	Serratella serrata	1.9	2	CG	X
Ephemeroptera	Ephemerellidae	Serratella sordida	1.7	2	CG	X
Ephemeroptera	Ephemerellidae	Serratella sp	2.7	2	CG	X
Ephemeroptera	Ephemerellidae	Serratella spiculosa	2.7	2	CG	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Ephemeroptera	Ephemerellidae	Timpanoga cornutella	2.0	2	CG	X
Ephemeroptera	Ephemerellidae	Timpanoga lita	0.0	2	CG	X
Ephemeroptera	Ephemerellidae	Timpanoga simplex	3.9	2	CG	X
Ephemeroptera	Ephemerellidae	Timpanoga sp	2.0	2	CG	X
Ephemeroptera	Ephemerellidae	Unidentified Ephemerellid	1.0	2		X
Ephemeroptera	Neophemeridae	Neoemphemera purpurea	1.6	2	CG	
Ephemeroptera	Caenidae	Brachycercus sp	3.0	8	CG	
Ephemeroptera	Caenidae	Caenis amica	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis anceps	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis diminuta	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis hilaris	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis latipennis	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis macafferti	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis punctata	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis sp	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis sp #1	7.4	8	CG	
Ephemeroptera	Caenidae	Caenis tardata	7.4	8	CG	
Ephemeroptera	Caenidae	Cercobrachys sp	1.0	8	CG	
Ephemeroptera	Caenidae	Unidentified Caenid	7.6	8	CG	
Ephemeroptera	Baetidae	Acentrella ampla	3.6	5	CG	
Ephemeroptera	Baetidae	Acentrella sp	4.0	5	CG	
Ephemeroptera	Baetidae	Acentrella turbida	3.6	5	CG	
Ephemeroptera	Baetidae	Acerpenna harti	3.7	5	CG	
Ephemeroptera	Baetidae	Acerpenna macdunnoughi	5.4	5	CG	
Ephemeroptera	Baetidae	Acerpenna pygmaea	3.9	5	CG	
Ephemeroptera	Baetidae	Acerpenna sp	5.0	5	CG	
Ephemeroptera	Baetidae	Baetis anachris	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis armillatus	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis bicaudatus	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis brunneicolor	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis cinctus	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis flavistriga	6.6	5	CG	
Ephemeroptera	Baetidae	Baetis fuscatus gr	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis intercalaris	5.0	5	CG	
Ephemeroptera	Baetidae	Baetis pluto	4.3	5	CG	
Ephemeroptera	Baetidae	Baetis posticatus	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis sp	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis sp #1	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis sp #3	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis spinosus	5.4	5	CG	
Ephemeroptera	Baetidae	Baetis tricaudatus	1.6	5	CG	
Ephemeroptera	Baetidae	Callibaetis pretiosus	9.8	5	CG	
Ephemeroptera	Baetidae	Callibaetis sp	9.8	5	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Ephemeroptera	Baetidae	Centroptilum alamanace	6.3	5	CG	
Ephemeroptera	Baetidae	Centroptilum similie	6.3	5	CG	
Ephemeroptera	Baetidae	Centroptilum sp	6.6	5	CG	
Ephemeroptera	Baetidae	Centroptilum vividocularis	6.6	5	CG	
Ephemeroptera	Baetidae	Cloeon sp	7.4	5	CG	
Ephemeroptera	Baetidae	Dipheter hageni	1.6	5	CG	
Ephemeroptera	Baetidae	Fallceon sp	5.4	5	CG	
Ephemeroptera	Baetidae	Heterocloeon curiosum	3.5	5	CG	
Ephemeroptera	Baetidae	Heterocloeon frivolus	3.6	5	CG	
Ephemeroptera	Baetidae	Paracloeodes minutus	8.3	5	SC	
Ephemeroptera	Baetidae	Paracloeodes sp	8.3	5	SC	
Ephemeroptera	Baetidae	Plauditus dubius	5.4	5	CG	
Ephemeroptera	Baetidae	Plauditus sp	5.4	5	CG	
Ephemeroptera	Baetidae	Procloeon bellum	5.0	5	CG	
Ephemeroptera	Baetidae	Procloeon fragile	5.4	5	CG	
Ephemeroptera	Baetidae	Procloeon sp	5.0	5	CG	
Ephemeroptera	Baetidae	Procloeon sp1	3.6	5	CG	
Ephemeroptera	Baetidae	Procloeon sp2	3.6	5	CG	
Ephemeroptera	Baetidae	Pseudocentroptiloides sp	5.0	5	CG	
Ephemeroptera	Baetidae	Pseudocloeon ephippiatus	3.7	5	CG	
Ephemeroptera	Baetidae	Pseudocloeon frondalis	7.4	5	CG	
Ephemeroptera	Baetidae	Pseudocloeon longipalpus	5.6	5	CG	
Ephemeroptera	Baetidae	Pseudocloeon propinquus	5.7	5	CG	
Ephemeroptera	Baetidae	Pseudocloeon sp	4.0	5	CG	
Ephemeroptera	Baetidae	Unidentified Baetid	5.0	5	CG	
Ephemeroptera	Baetiscidae	Baetisca berneri	2.0	2	SC	
Ephemeroptera	Baetiscidae	Baetisca carolina	3.5	2	SC	
Ephemeroptera	Baetiscidae	Baetisca gibbera	1.4	2	SC	
Ephemeroptera	Baetiscidae	Baetisca lacustris	1.0	2	SC	
Ephemeroptera	Baetiscidae	Baetisca obesa	1.9	2	SC	
Ephemeroptera	Baetiscidae	Baetisca rogersi	1.9	2	SC	
Ephemeroptera	Baetiscidae	Baetisca sp	2.1	2	SC	
Odonata	Lestidae	Archilestes grandis	8.0	9	PR	
Odonata	Lestidae	Lestes disjunctus	9.4	9	PR	
Odonata	Lestidae	Lestes eurinus	9.4	9	PR	
Odonata	Lestidae	Lestes forcipatus	9.4	9	PR	
Odonata	Lestidae	Lestes sp	9.4	9	PR	
Odonata	Lestidae	Lestes vidua	9.4	9	PR	
Odonata	Coenagrionidae	Amphiagrion saucium	9.0	9	PR	
Odonata	Coenagrionidae	Anomalagrion hastatum	9.0	9	PR	
Odonata	Coenagrionidae	Argia apicalis	8.5	9	PR	
Odonata	Coenagrionidae	Argia bipunctulata	8.5	9	PR	
Odonata	Coenagrionidae	Argia fumipennis	8.5	9	PR	



**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Odonata	Coenagrionidae	Argia moesta	8.5	9	PR	
Odonata	Coenagrionidae	Argia sedula	8.5	9	PR	
Odonata	Coenagrionidae	Argia sp	8.2	9	PR	
Odonata	Coenagrionidae	Argia tibialis	8.5	9	PR	
Odonata	Coenagrionidae	Argia translata	8.5	9	PR	
Odonata	Coenagrionidae	Chromagrion conditum	9.0	9	PR	
Odonata	Coenagrionidae	Enallagma aspersum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma basidens	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma civile	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma daeckii	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma divagans	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma dubium	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma exsulans	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma geminatum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma hageni	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma laterale	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma minusculum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma pallidum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma sexsulans	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma signatum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma sp	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma sulcatum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma traviatum	8.9	9	PR	
Odonata	Coenagrionidae	Enallagma vesperum	8.9	9	PR	
Odonata	Coenagrionidae	Ischnura posita	9.5	9	PR	
Odonata	Coenagrionidae	Ischnura prognatha	9.5	9	PR	
Odonata	Coenagrionidae	Ischnura ramburi	9.5	9	PR	
Odonata	Coenagrionidae	Ischnura sp	9.5	9	PR	
Odonata	Coenagrionidae	Ischnura verticalis	9.5	9	PR	
Odonata	Calopterygidae	Calopteryx amata	7.8	7	PR	
Odonata	Calopterygidae	Calopteryx angustipennis	7.8	7	PR	
Odonata	Calopterygidae	Calopteryx dimidiata	7.8	7	PR	
Odonata	Calopterygidae	Calopteryx maculata	7.8	7	PR	
Odonata	Calopterygidae	Calopteryx sp	7.8	7	PR	
Odonata	Calopterygidae	Hetaerina americana	6.2	7	PR	
Odonata	Calopterygidae	Hetaerina sp	5.6	7	PR	
Odonata	Calopterygidae	Hetaerina titia	6.2	7	PR	
Odonata	Aeshnidae	Aeshna sp	7.1	7	PR	
Odonata	Aeshnidae	Aeshna umbrosa	7.1	7	PR	
Odonata	Aeshnidae	Aeshna verticalis	7.1	7	PR	
Odonata	Aeshnidae	Anax junius	7.1	7	PR	
Odonata	Aeshnidae	Anax longipes	7.1	7	PR	
Odonata	Aeshnidae	Anax sp	7.1	7	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Odonata	Aeshnidae	Basiaeschna janata	7.4	7	PR	
Odonata	Aeshnidae	Basiaeschna pentacantha	7.7	7	PR	
Odonata	Aeshnidae	Basiaeschna sp	7.7	7	PR	
Odonata	Aeshnidae	Boyeria grafiana	6.1	7	PR	
Odonata	Aeshnidae	Boyeria sp	6.0	7	PR	
Odonata	Aeshnidae	Boyeria vinosa	5.9	7	PR	
Odonata	Aeshnidae	Epiaeschna sp	8.0	7	PR	
Odonata	Aeshnidae	Nasiaeschna pentacantha	8.1	7	PR	
Odonata	Aeshnidae	Nasiaeschna sp	8.0	7	PR	
Odonata	Gomphidae	Dromogomphus armatus	5.9	6	PR	
Odonata	Gomphidae	Dromogomphus sp	5.9	6	PR	
Odonata	Gomphidae	Dromogomphus spinosus	5.9	6	PR	
Odonata	Gomphidae	Dromogomphus spoliatus	5.9	6	PR	
Odonata	Gomphidae	Erpetogomphus designatus	6.3	6	PR	
Odonata	Gomphidae	Gomphus abbreviatus	5.8	6	PR	
Odonata	Gomphidae	Gomphus amnicola	5.8	6	PR	
Odonata	Gomphidae	Gomphus apomyius/brevis	5.8	6	PR	
Odonata	Gomphidae	Gomphus australis	5.8	6	PR	
Odonata	Gomphidae	Gomphus borealis	5.8	6	PR	
Odonata	Gomphidae	Gomphus cavillaris	5.8	6	PR	
Odonata	Gomphidae	Gomphus descriptus	5.8	6	PR	
Odonata	Gomphidae	Gomphus dilatatus	5.8	6	PR	
Odonata	Gomphidae	Gomphus diminutus	5.8	6	PR	
Odonata	Gomphidae	Gomphus lividus	5.8	6	PR	
Odonata	Gomphidae	Gomphus minutus	5.8	6	PR	
Odonata	Gomphidae	Gomphus notatus	5.8	6	PR	
Odonata	Gomphidae	Gomphus pallidus	5.8	6	PR	
Odonata	Gomphidae	Gomphus rogersi	5.8	6	PR	
Odonata	Gomphidae	Gomphus sp	5.8	6	PR	
Odonata	Gomphidae	Gomphus spiniceps	5.8	6	PR	
Odonata	Gomphidae	Gomphus vastus	5.8	6	PR	
Odonata	Gomphidae	Gomphus viridifrons	5.8	6	PR	
Odonata	Gomphidae	Hagenius brevistylus	4.0	6	PR	
Odonata	Gomphidae	Hagenius sp	4.0	6	PR	
Odonata	Gomphidae	Lanthus parvulus	1.8	6	PR	
Odonata	Gomphidae	Lanthus sp	1.8	6	PR	
Odonata	Gomphidae	Lanthus vernalis	1.8	6	PR	
Odonata	Gomphidae	Ophiogomphus carolinus	5.5	6	PR	
Odonata	Gomphidae	Ophiogomphus howei	5.5	6	PR	
Odonata	Gomphidae	Ophiogomphus mainensis	5.5	6	PR	
Odonata	Gomphidae	Ophiogomphus sp	5.5	6	PR	
Odonata	Gomphidae	Progomphus obscurus	8.2	6	PR	
Odonata	Gomphidae	Progomphus sp	8.7	6	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Odonata	Gomphidae	Stylogomphus albistylus	4.7	6	PR	
Odonata	Gomphidae	Stylurus sp	6.2	6	PR	
Odonata	Gomphidae	Unidentified Gomphid	6.0	6	PR	
Odonata	Libellulidae	Celithemis amanda	9.0	9	PR	
Odonata	Libellulidae	Celithemis elisa	9.0	9	PR	
Odonata	Libellulidae	Celithemis ornata	9.0	9	PR	
Odonata	Corduliidae	Didymops sp	5.9	7	PR	
Odonata	Corduliidae	Didymops transversa	5.9	7	PR	
Odonata	Libellulidae	Dythemis sp	9.0	9	PR	
Odonata	Libellulidae	Dythemis velox	9.0	9	PR	
Odonata	Corduliidae	Epicordulia princeps	5.6	7	PR	
Odonata	Corduliidae	Epitheca (Epicordulia) sp	5.6	7	PR	
Odonata	Corduliidae	Epitheca (Tetragoneuria) sp	8.5	7	PR	
Odonata	Corduliidae	Epitheca (Tetragoneuria/Epicordulia) sp	7.0	7	PR	
Odonata	Corduliidae	Epitheca costalis	8.5	7	PR	
Odonata	Corduliidae	Epitheca regina	5.6	7	PR	
Odonata	Corduliidae	Epitheca spinosa	8.5	7	PR	
Odonata	Libellulidae	Erythemis simplicicollis	9.7	9	PR	
Odonata	Libellulidae	Erythrodiplax berenice	9.0	9	PR	
Odonata	Libellulidae	Erythrodiplax sp	9.0	9	PR	
Odonata	Corduliidae	Helocordulia selysii	5.9	7	PR	
Odonata	Corduliidae	Helocordulia sp	4.8	7	PR	
Odonata	Corduliidae	Helocordulia uhleri	4.9	7	PR	
Odonata	Libellulidae	Ladona exusta	9.0	9	PR	
Odonata	Libellulidae	Libellula auripennis	9.6	9	PR	
Odonata	Libellulidae	Libellula insecta	9.6	9	PR	
Odonata	Libellulidae	Libellula luctuosa	9.6	9	PR	
Odonata	Libellulidae	Libellula needhami	9.6	9	PR	
Odonata	Libellulidae	Libellula pulchella	9.6	9	PR	
Odonata	Libellulidae	Libellula sp	9.6	9	PR	
Odonata	Libellulidae	Libellula vibrans	9.6	9	PR	
Odonata	Corduliidae	Macrodiplax balteata	9.0	7	PR	
Odonata	Corduliidae	Macromia alleghaniensis	6.2	7	PR	
Odonata	Corduliidae	Macromia georgina	6.2	7	PR	
Odonata	Corduliidae	Macromia illinoiensis	6.2	7	PR	
Odonata	Corduliidae	Macromia illinoiensis/georgina	6.2	7	PR	
Odonata	Corduliidae	Macromia margarita	6.2	7	PR	
Odonata	Corduliidae	Macromia sp	6.2	7	PR	
Odonata	Corduliidae	Macromia taeniolata	6.2	7	PR	
Odonata	Libellulidae	Nannothemis bella	9.0	9	PR	
Odonata	Corduliidae	Neurocordulia alabamensis	5.4	7	PR	
Odonata	Corduliidae	Neurocordulia molesta	1.8	7	PR	
Odonata	Corduliidae	Neurocordulia obsoleta	5.2	7	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Odonata	Corduliidae	Neurocordulia sp	5.0	7	PR	
Odonata	Corduliidae	Neurocordulia virginiensis	2.1	7	PR	
Odonata	Corduliidae	Neurocordulia yamaskanensis		7	PR	
Odonata	Libellulidae	Pachydiplax longipennis	9.9	9	PR	
Odonata	Libellulidae	Perithemis sp	9.9	9	PR	
Odonata	Libellulidae	Perithemis tenera	9.9	9	PR	
Odonata	Libellulidae	Plathemis lydia	10.0	9	PR	
Odonata	Corduliidae	Somatochlora provocans	9.2	7	PR	
Odonata	Corduliidae	Somatochlora sp	9.2	7	PR	
Odonata	Corduliidae	Somatochlora tenebrosa	9.2	7	PR	
Odonata	Libellulidae	Sympetrum ambiguum	7.3	9	PR	
Odonata	Libellulidae	Sympetrum sp	7.3	9	PR	
Odonata	Libellulidae	Sympetrum vicinum	7.3	9	PR	
Odonata	Libellulidae	Tramea carolina	9.8	9	PR	
Odonata	Corduliidae	Unidentified Corduliid	6.6	7	PR	
Odonata	Libellulidae	Unidentified Libellulid	9.1	9	PR	
Odonata	Cordulegastridae	Cordulegaster erronea	5.7	6	PR	
Odonata	Cordulegastridae	Cordulegaster fasciata	5.7	6	PR	
Odonata	Cordulegastridae	Cordulegaster maculata	5.7	6	PR	
Odonata	Cordulegastridae	Cordulegaster obliqua	5.7	6	PR	
Odonata	Cordulegastridae	Cordulegaster sp	5.7	6	PR	
Plecoptera	Pteronarcyidae	Pteronarcys comstocki	1.7	2	SH	X
Plecoptera	Pteronarcyidae	Pteronarcys biloba	1.7	2	SH	X
Plecoptera	Pteronarcyidae	Pteronarcys dorsata	1.8	2	SH	X
Plecoptera	Pteronarcyidae	Pteronarcys proteus	1.7	2	SH	X
Plecoptera	Pteronarcyidae	Pteronarcys sp	1.7	2	SH	X
Plecoptera	Perlodidae	Clioperla clio	4.7	2	PR	X
Plecoptera	Perlodidae	Cultus decisus	1.6	2	PR	X
Plecoptera	Perlodidae	Diploperla duplicata	2.7	2	PR	X
Plecoptera	Perlodidae	Diploperla robusta	2.7	2	PR	X
Plecoptera	Perlodidae	Hydroperla crosbyi	2.0	2	PR	X
Plecoptera	Perlodidae	Isogenoides doratus	2.0	2	PR	X
Plecoptera	Perlodidae	Isogenoides hansonii	0.5	2	PR	X
Plecoptera	Perlodidae	Isogenoides varians	2.0	2	PR	X
Plecoptera	Perlodidae	Isoperla bellona	1.8	2	PR	X
Plecoptera	Perlodidae	Isoperla bilineata	5.4	2	PR	X
Plecoptera	Perlodidae	Isoperla cotta	2.2	2	SC	X
Plecoptera	Perlodidae	Isoperla holochlora	0.0	2	PR	X
Plecoptera	Perlodidae	Isoperla lata	0.0	2	PR	X
Plecoptera	Perlodidae	Isoperla nana	1.8	2	PR	X
Plecoptera	Perlodidae	Isoperla similis	0.2	2	PR	X
Plecoptera	Perlodidae	Isoperla sp	1.8	2	PR	X
Plecoptera	Perlodidae	Malirekus hastatus	1.2	2	PR	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Plecoptera	Perlodidae	Malirekus sp	1.2	2	PR	X
Plecoptera	Perlodidae	Remenus bilobatus	0.3	2	PR	X
Plecoptera	Perlodidae	Unidentified Perlodid	2.0	2	PR	X
Plecoptera	Perlodidae	Yugus sp	0.0	2	PR	X
Plecoptera	Capniidae	Allocapnia aurora	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia forbesi	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia fumosa	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia granulata	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia mystica	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia rickeri	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia sp	2.5	3	SH	
Plecoptera	Capniidae	Allocapnia vivipara	2.5	3	SH	
Plecoptera	Capniidae	Neocapnia carolina	1.5	3	SH	
Plecoptera	Capniidae	Paracapnia angulata	0.1	3	SH	
Plecoptera	Capniidae	Paracapnia sp	0.1	3	SH	
Plecoptera	Capniidae	Unidentified Capniid	2.8	3	SH	
Plecoptera	Peltoperlidae	Peltoperla arcuata	1.0	2	SH	X
Plecoptera	Peltoperlidae	Peltoperla sp	1.0	2	SH	X
Plecoptera	Peltoperlidae	Tallaperla sp	1.2	2	SH	X
Plecoptera	Nemouridae	Amphinemura delosa	3.3	4	SH	
Plecoptera	Nemouridae	Amphinemura nigritta	3.3	4	SH	
Plecoptera	Nemouridae	Amphinemura sp	3.3	4	SH	
Plecoptera	Nemouridae	Amphinemura wui	3.3	4	SH	
Plecoptera	Nemouridae	Ostrocerca sp	2.5	4	SH	
Plecoptera	Nemouridae	Paranemura perfecta	2.0	4	SH	
Plecoptera	Nemouridae	Prostoia completa	6.1	4	SH	
Plecoptera	Nemouridae	Prostoia similis	6.1	4	SH	
Plecoptera	Nemouridae	Prostoia sp	5.8	4	SH	
Plecoptera	Nemouridae	Soyedina vallicularia		4	SH	
Plecoptera	Nemouridae	Unidentified Nemourid	4.5	4	SH	
Plecoptera	Leuctridae	Leuctra ferruginea	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra moha	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra rickeri	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra sibleyi	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra sp	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra tenuis	0.6	1	SH	
Plecoptera	Leuctridae	Leuctra triloba	0.7	1	SH	
Plecoptera	Leuctridae	Leuctra variabilis	0.7	1	SH	
Plecoptera	Leuctridae	Megaleuctra sp	1.0	1	SH	
Plecoptera	Leuctridae	Paraleuctra sp	2.8	1	SH	
Plecoptera	Leuctridae	Unidentified Leuctrid	1.0	1	SH	
Plecoptera	Leuctridae	Zealeuctra claasseni	1.0	1	SH	
Plecoptera	Taeniopterygidae	Strophopteryx appalachia	2.5	5	SC	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Plecoptera	Taeniopterygidae	Strophopteryx fasciata	2.5	5	SC	
Plecoptera	Taeniopterygidae	Strophopteryx sp	2.7	5	SC	
Plecoptera	Taeniopterygidae	Taenionema atlanticum	5.0	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx burksi	6.1	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx lita	6.3	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx maura	6.3	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx metequi	1.4	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx parvula	6.3	5	SH	
Plecoptera	Taeniopterygidae	Taeniopteryx sp	5.4	5	SH	
Plecoptera	Taeniopterygidae	Unidentified Taeniopterygid	4.6	5	SC	
Plecoptera	Perlidae	Acroneuria abnormis	2.1	2	PR	X
Plecoptera	Perlidae	Acroneuria arida	1.4	2	PR	X
Plecoptera	Perlidae	Acroneuria carolinensis	0.0	2	PR	X
Plecoptera	Perlidae	Acroneuria evoluta	1.4	2	PR	X
Plecoptera	Perlidae	Acroneuria frisoni	4.0	2	PR	X
Plecoptera	Perlidae	Acroneuria internata	1.4	2	PR	X
Plecoptera	Perlidae	Acroneuria lycorias	2.1	2	PR	X
Plecoptera	Perlidae	Acroneuria mela	0.9	2	PR	X
Plecoptera	Perlidae	Acroneuria sp	1.4	2	PR	X
Plecoptera	Perlidae	Agentina sp	0.0	2	PR	X
Plecoptera	Perlidae	Agnetina capitata	0.0	2	PR	X
Plecoptera	Perlidae	Agnetina flavescens	3.0	2	PR	X
Plecoptera	Perlidae	Agnetina sp	3.0	2	PR	
Plecoptera	Perlidae	Attaneuria ruralis	3.0	2	PR	X
Plecoptera	Perlidae	Beloneuria sp	0.0	2	PR	X
Plecoptera	Perlidae	Beloneuria stewarti	0.0	2	PR	X
Plecoptera	Perlidae	Eccoptura xanthenes	3.7	2	PR	X
Plecoptera	Perlidae	Neoperla clymene	5.0	2	PR	X
Plecoptera	Perlidae	Neoperla freytagi	5.0	2	PR	X
Plecoptera	Perlidae	Neoperla sp	5.0	2	PR	X
Plecoptera	Perlidae	Paragnetina fumosa	3.4	2	PR	X
Plecoptera	Perlidae	Paragnetina immarginata	1.4	2	PR	X
Plecoptera	Perlidae	Paragnetina kansensis	2.0	2	PR	X
Plecoptera	Perlidae	Paragnetina media	1.8	2	PR	X
Plecoptera	Perlidae	Paragnetina sp	1.5	2	PR	X
Plecoptera	Perlidae	Perlesta frisoni	4.7	2	PR	X
Plecoptera	Perlidae	Perlesta placida	4.7	2	PR	X
Plecoptera	Perlidae	Perlesta sp	4.7	2	PR	X
Plecoptera	Perlidae	Perlinella drymo	1.3	2	PR	X
Plecoptera	Perlidae	Perlinella ephyre	1.3	2	PR	X
Plecoptera	Perlidae	Perlinella sp	1.3	2	PR	X
Plecoptera	Perlidae	Unidentified Perlid	3.0	2	PR	X
Plecoptera	Chloroperlidae	Alloperla atlantica	1.4	1	PR	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Plecoptera	Chloroperlidae	Alloperla caudata	1.4	1	PR	X
Plecoptera	Chloroperlidae	Alloperla imbecilla	1.4	1	PR	X
Plecoptera	Chloroperlidae	Alloperla neglecta	1.4	1	PR	X
Plecoptera	Chloroperlidae	Alloperla sp	1.2	1	PR	X
Plecoptera	Chloroperlidae	Haploperla brevis	1.0	1	PR	X
Plecoptera	Chloroperlidae	Rasvena terna	0.0	1	PR	X
Plecoptera	Chloroperlidae	Sweltsa lateralis	0.0	1	PR	X
Plecoptera	Chloroperlidae	Sweltsa mediana	0.0	1	PR	X
Plecoptera	Chloroperlidae	Sweltsa nanina	0.0	1	PR	X
Plecoptera	Chloroperlidae	Sweltsa sp	0.0	1	PR	X
Plecoptera	Chloroperlidae	Unidentified Chloroperlid	0.8	1	PR	X
Plecoptera	Chloroperlidae	Utaperla gaspesiana	0.9	1	CG	X
Hemiptera	Corixidae	Corixini sp	9.0	9	PH	
Hemiptera	Corixidae	Hesperocorixa brimleyi	9.0	9	PH	
Hemiptera	Corixidae	Hesperocorixa sp	9.0	9	PH	
Hemiptera	Corixidae	Palmacorixa sp	9.0	9	PH	
Hemiptera	Corixidae	Sagara sp	9.0	9	PH	
Hemiptera	Corixidae	Sigara modesta	9.0	9	PH	
Hemiptera	Corixidae	Sigara signata	9.0	9	PH	
Hemiptera	Corixidae	Sigara sp	9.1	9	PH	
Hemiptera	Corixidae	Sigara variabilis	9.0	9	PH	
Hemiptera	Corixidae	Trichocorixa sp	9.0	9	PR	
Hemiptera	Corixidae	Unidentified Corixid	9.0	9	PH	
Hemiptera	Veliidae	Microvelia americana	9.0	9	PR	
Hemiptera	Veliidae	Microvelia paludicola	9.0	9	PR	
Hemiptera	Veliidae	Microvelia sp	9.0	9	PR	
Hemiptera	Veliidae	Rhagovelia obesa	9.0	9	PR	
Hemiptera	Veliidae	Rhagovelia sp	9.0	9	PR	
Hemiptera	Veliidae	Unidentified Veliid	9.0	9	PR	
Hemiptera	Hebridae	Merragata sp	10.0	10	PR	
Hemiptera	Saldidae	Micracanthia sp	9.0	9	PR	
Hemiptera	Saldidae	Saldula sp	9.0	9	PR	
Hemiptera	Gelastocoridae	Gelastocoris oculatus	9.0	9	PR	
Hemiptera	Notonectidae	Buenoa sp	9.0	9	PR	
Hemiptera	Notonectidae	Notonecta irrorata	9.0	9	PR	
Hemiptera	Notonectidae	Notonecta rahleighi	9.0	9	PR	
Hemiptera	Notonectidae	Notonecta sp	8.7	9	PR	
Hemiptera	Nepidae	Nepa apiculata	9.0	8	PR	
Hemiptera	Nepidae	Nepa sp	9.0	8		
Hemiptera	Nepidae	Ranatra australis	7.5	8	PR	
Hemiptera	Nepidae	Ranatra buenoi	7.5	8	PR	
Hemiptera	Nepidae	Ranatra fusca	7.5	8	PR	
Hemiptera	Nepidae	Ranatra kirkaldyi	7.5	8	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Hemiptera	Nepidae	Ranatra nigra	7.5	8	PR	
Hemiptera	Nepidae	Ranatra sp	7.8	8	PR	
Hemiptera	Mesoveliidae	Mesovelia amoena	9.8	10	PR	
Hemiptera	Mesoveliidae	Mesovelia cryptophila	9.8	10	PR	
Hemiptera	Mesoveliidae	Mesovelia mulsanti	9.8	10	PR	
Hemiptera	Mesoveliidae	Mesovelia sp	9.8	10	PR	
Hemiptera	Pleidae	Neoplea sp	9.0	9	PR	
Hemiptera	Pleidae	Neoplea striola	9.0	9	PR	
Hemiptera	Pleidae	Paraplea sp	9.0	9	PR	
Hemiptera	Belostomatidae	Belostoma flumineum	9.8	10	PR	
Hemiptera	Belostomatidae	Belostoma lutarium	9.8	10	PR	
Hemiptera	Belostomatidae	Belostoma sp	9.8	10	PR	
Hemiptera	Belostomatidae	Lethocerus americanus	9.0	10	PR	
Hemiptera	Hydrometridae	Hydrometra australis	9.0	9	PR	
Hemiptera	Hydrometridae	Hydrometra hungerfordi	9.0	9	PR	
Hemiptera	Hydrometridae	Hydrometra martini	9.0	9	PR	
Hemiptera	Hydrometridae	Hydrometra sp	9.0	9	PR	
Hemiptera	Hydrometridae	Hydrometra wileyae	9.0	9	PR	
Hemiptera	Gerridae	Aquarius remigeris		9	PR	
Hemiptera	Gerridae	Gerris conformis	9.0	9	PR	
Hemiptera	Gerridae	Gerris marginatus	9.0	9	PR	
Hemiptera	Gerridae	Gerris nebularis	9.0	9	PR	
Hemiptera	Gerridae	Gerris remigis	9.0	9	PR	
Hemiptera	Gerridae	Gerris sp	9.0	9	PR	
Hemiptera	Gerridae	Halobates micans	9.0	9	PR	
Hemiptera	Gerridae	Limnopus canaliculatus	9.0	9	PR	
Hemiptera	Gerridae	Limnopus sp	9.0	9	PR	
Hemiptera	Gerridae	Metrobates hesperius	9.0	9	PR	
Hemiptera	Gerridae	Metrobates sp	9.0	9	PR	
Hemiptera	Gerridae	Neogerris sp	9.0	9	PR	
Hemiptera	Gerridae	Rheumatobates hungerfordi	9.0	9	PR	
Hemiptera	Gerridae	Rheumatobates rileyi	9.0	9	PR	
Hemiptera	Gerridae	Rheumatobates sp	9.0	9	PR	
Hemiptera	Gerridae	Rheumatobates tenuipes	9.0	9	PR	
Hemiptera	Gerridae	Trepobates imermis	9.0	9	PR	
Hemiptera	Gerridae	Trepobates pictus	9.0	9	PR	
Hemiptera	Gerridae	Trepobates sp	9.0	9	PR	
Hemiptera	Gerridae	Trepobates subnitidus	9.0	9	PR	
Hemiptera	Gerridae	Unidentified Gerrid	9.0	9	PR	
Megaloptera	Corydalidae	Chauloides pecticornis	9.6	6	PR	X
Megaloptera	Corydalidae	Chauloides pectinicornis	9.6	6	PR	X
Megaloptera	Corydalidae	Chauloides rastricornis	8.4	6	PR	X
Megaloptera	Corydalidae	Corydalus cornutus	5.2	6	PR	X



**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Megaloptera	Corydalidae	Neohermes angusticollis	5.8	6	PR	X
Megaloptera	Corydalidae	Neohermes concolor	5.8	6	PR	X
Megaloptera	Corydalidae	Nigronia fasciatus	5.6	6	PR	X
Megaloptera	Corydalidae	Nigronia serricornis	5.0	6	PR	X
Megaloptera	Corydalidae	Nigronia sp	5.3	6	PR	X
Megaloptera	Sialidae	Sialis aequalis	7.2	8	PR	
Megaloptera	Sialidae	Sialis americana	7.2	8	PR	
Megaloptera	Sialidae	Sialis infumata	7.2	8	PR	
Megaloptera	Sialidae	Sialis iola	7.2	8	PR	
Megaloptera	Sialidae	Sialis itasca	7.2	8	PR	
Megaloptera	Sialidae	Sialis joppa	7.2	8	PR	
Megaloptera	Sialidae	Sialis mohri	7.2	8	PR	
Megaloptera	Sialidae	Sialis sp	7.2	8	PR	
Megaloptera	Sialidae	Sialis vagans	7.2	8	PR	
Megaloptera	Sialidae	Sialis velata	7.2	8	PR	
Neuroptera	Sisyridae	Climacia areolaris	8.4	7	PR	
Neuroptera	Sisyridae	Climacia sp	8.4	7	PR	
Neuroptera	Sisyridae	Sisyra sp	5.0	7	PR	
Trichoptera	Limnephilidae	Goera calcarata	0.3	3	SC	
Trichoptera	Limnephilidae	Goera sp	0.1	3	SC	
Trichoptera	Limnephilidae	Goera stylata	0.3	3	SC	
Trichoptera	Limnephilidae	Goerita betteni	0.5	3	SC	
Trichoptera	Limnephilidae	Hydatophylax argus	2.2	3	SH	
Trichoptera	Limnephilidae	Ironoquia punctatissima	7.8	3	SH	
Trichoptera	Limnephilidae	Ironoquia sp	7.7	3	SH	
Trichoptera	Limnephilidae	Platycentropus radiatus	2.0	3	SH	
Trichoptera	Limnephilidae	Pseudostenophylax sp	2.0	3	SC	
Trichoptera	Limnephilidae	Pseudostenophylax sparsus	2.0	3	SH	
Trichoptera	Limnephilidae	Pseudostenophylax uniformis	2.0	3	SH	
Trichoptera	Limnephilidae	PyXopsyche divergens	2.5	3	SH	
Trichoptera	Limnephilidae	PyXopsyche gentilis	0.6	3	SH	
Trichoptera	Limnephilidae	PyXopsyche guttifer	2.6	3	SH	
Trichoptera	Limnephilidae	PyXopsyche lepida	2.7	3	SH	
Trichoptera	Limnephilidae	PyXopsyche lepida/subfasciata gr	2.3	3	SH	
Trichoptera	Limnephilidae	PyXopsyche luculenta/sonso	2.5	3	SH	
Trichoptera	Limnephilidae	PyXopsyche scrabripennis	2.5	3	SH	
Trichoptera	Limnephilidae	PyXopsyche sp	2.5	3	SH	
Trichoptera	Limnephilidae	PyXopsyche sp1	2.3	3	SH	
Trichoptera	Limnephilidae	PyXopsyche sp2	2.3	3	SH	
Trichoptera	Molannidae	Molanna blenda	2.0	2	SC	
Trichoptera	Molannidae	Molanna sp	2.0	2	SC	
Trichoptera	Hydroptilidae	Agraylea multipunctata	5.9	3	PH	X
Trichoptera	Hydroptilidae	Agraylea sp	5.9	3	PH	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Hydroptilidae	Dibusa angata	3.0	3	SH	X
Trichoptera	Hydroptilidae	Hydroptila ajax	6.2	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila amoena	6.2	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila armata	6.2	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila consimilis	6.2	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila delineata	3.0	3	SC	X
Trichoptera	Hydroptilidae	Hydroptila grandiosa	3.0	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila hamata	4.0	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila howelli	0.5	3	Sc	X
Trichoptera	Hydroptilidae	Hydroptila kuehnei	0.5	3	Sc	X
Trichoptera	Hydroptilidae	Hydroptila perdita	4.0	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila sp	6.2	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila sp B	6.2	3	SC	X
Trichoptera	Hydroptilidae	Hydroptila spatulata	5.0	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila talladaga	1.0	3	SC	X
Trichoptera	Hydroptilidae	Hydroptila waskesia	2.5	3	PH	X
Trichoptera	Hydroptilidae	Hydroptila waubesiana	5.0	3	PH	X
Trichoptera	Hydroptilidae	Ithytrichia mazon	3.0	3	SC	X
Trichoptera	Hydroptilidae	Ithytrichia sp	3.0	3	SC	X
Trichoptera	Hydroptilidae	Leucotrichia pictipes	4.1	3	SC	X
Trichoptera	Hydroptilidae	Mayatrichia ayama	1.0	3	SC	X
Trichoptera	Hydroptilidae	Neotrichia minutisimella	2.0	3	SC	X
Trichoptera	Hydroptilidae	Neotrichia okopa	2.0	3	SC	X
Trichoptera	Hydroptilidae	Neotrichia sp	2.0	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia anisca	5.9	3	CG	X
Trichoptera	Hydroptilidae	Ochrotrichia arva	1.5	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia confusa	5.9	3	CG	X
Trichoptera	Hydroptilidae	Ochrotrichia eliaga	1.0	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia reisi	1.0	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia shawnee	1.0	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia sp	4.0	3	CG	X
Trichoptera	Hydroptilidae	Ochrotrichia spinosa	4.0	3	CG	X
Trichoptera	Hydroptilidae	Ochrotrichia tarsalis	3.5	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia unio	3.0	3	SC	X
Trichoptera	Hydroptilidae	Ochrotrichia xena	3.0	3	SC	X
Trichoptera	Hydroptilidae	Orthotrichia aegerfasciella	5.0	3	CG	X
Trichoptera	Hydroptilidae	Orthotrichia cristata	5.0	3	CG	X
Trichoptera	Hydroptilidae	Orthotrichia sp	5.0	3	CG	X
Trichoptera	Hydroptilidae	Oxyethira pallida	2.0	3	CG	X
Trichoptera	Hydroptilidae	Oxyethira sp	2.2	3	CG	X
Trichoptera	Hydroptilidae	Palaeagapetus celsus	1.5	3	CG	X
Trichoptera	Hydroptilidae	Stactobiella delira	1.5	3	SC	X
Trichoptera	Hydroptilidae	Stactobiella martynovi	1.3	3	SC	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Hydroptilidae	Stactobiella palmata	1.5	3	CG	X
Trichoptera	Hydroptilidae	Stactobiella sp	1.3	3	CG	X
Trichoptera	Hydroptilidae	Unidentified Hydroptilid	3.0	3		X
Trichoptera	Lepidostomatidae	Lepidostoma sp	0.9	1	SH	
Trichoptera	Lepidostomatidae	Lepidostoma togatum	1.0	1	SH	
Trichoptera	Lepidostomatidae	Theliopsyche melas	1.0	1	SC	X
Trichoptera	Sericostomatidae	Agarodes sp	0.7	1	SH	
Trichoptera	Leptoceridae	Ceraclea ancylus	2.3	4	PR	
Trichoptera	Leptoceridae	Ceraclea cancellata	2.5	4	PR	
Trichoptera	Leptoceridae	Ceraclea diluta	2.3	4	PR	
Trichoptera	Leptoceridae	Ceraclea flava	1.0	4	PR	
Trichoptera	Leptoceridae	Ceraclea maculata	6.5	4	PR	
Trichoptera	Leptoceridae	Ceraclea neffi	1.0	4	PR	
Trichoptera	Leptoceridae	Ceraclea ophioderus	2.4	4	PR	
Trichoptera	Leptoceridae	Ceraclea punctata	5.0	4	SC	
Trichoptera	Leptoceridae	Ceraclea resurgens	2.9	4	PR	
Trichoptera	Leptoceridae	Ceraclea sp	2.0	4	PR	X
Trichoptera	Leptoceridae	Ceraclea tarsipunctata	3.0	4	PR	
Trichoptera	Leptoceridae	Ceraclea transversa	2.5	4	PR	
Trichoptera	Leptoceridae	Mystacides sepulchralis	2.7	4	PR	
Trichoptera	Leptoceridae	Mystacides sp	2.5	4	PR	
Trichoptera	Leptoceridae	Nectopsyche albida	3.5	4	PR	
Trichoptera	Leptoceridae	Nectopsyche candida	5.5	4	PR	
Trichoptera	Leptoceridae	Nectopsyche exquisita	4.1	4	PR	
Trichoptera	Leptoceridae	Nectopsyche pavida	4.1	4	PR	
Trichoptera	Leptoceridae	Nectopsyche sp	2.9	4	PR	
Trichoptera	Leptoceridae	Oecetis avara	5.7	4	PR	
Trichoptera	Leptoceridae	Oecetis cinerascens	5.7	4	PR	
Trichoptera	Leptoceridae	Oecetis ditissa	5.7	4	PR	
Trichoptera	Leptoceridae	Oecetis inconspicua	1.9	4	PR	
Trichoptera	Leptoceridae	Oecetis nocturna	4.1	4	PR	
Trichoptera	Leptoceridae	Oecetis parva	5.7	4	PR	
Trichoptera	Leptoceridae	Oecetis persimilis	4.7	4	PR	
Trichoptera	Leptoceridae	Oecetis scala	5.0	4	PR	
Trichoptera	Leptoceridae	Oecetis sp	4.7	4	PR	
Trichoptera	Leptoceridae	Setodes sp	0.0	4	SC	
Trichoptera	Leptoceridae	Triaenodes abus	4.1	4	PR	
Trichoptera	Leptoceridae	Triaenodes cumberlandensis	3.5	4	SC	
Trichoptera	Leptoceridae	Triaenodes flavescens	3.0	4	PR	
Trichoptera	Leptoceridae	Triaenodes helo		4	PR	
Trichoptera	Leptoceridae	Triaenodes ignitus	4.6	4	PR	
Trichoptera	Leptoceridae	Triaenodes injustus	2.5	4	PR	
Trichoptera	Leptoceridae	Triaenodes melacus		4	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Leptoceridae	Trienodes nox		4	PR	
Trichoptera	Leptoceridae	Trienodes perna	4.1	4	PR	
Trichoptera	Leptoceridae	Trienodes sp	4.5	4	PR	
Trichoptera	Leptoceridae	Trienodes tardus	4.6	4	PR	
Trichoptera	Leptoceridae	Unidentified Leptocerid	3.6	4	PR	
Trichoptera	Calamoceratidae	Anisocentropus pyraloides	0.9	1	SH	
Trichoptera	Calamoceratidae	Heteroplectron americanum	3.2	1	SH	
Trichoptera	Rhyacophilidae	Rhyacophila carolina	1.0	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila fuscula	1.9	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila glaberrima	0.8	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila invaria gp	0.0	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila ledra/fenestra	3.9	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila lobifera	2.5	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila minor	0.0	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila nigrita	0.0	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila sp	0.8	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila torva	1.6	1	PR	X
Trichoptera	Rhyacophilidae	Rhyacophila vibox	0.8	1	PR	X
Trichoptera	Helicopsychidae	Helicopsyche borealis	5.0	5	SC	X
Trichoptera	Helicopsychidae	Helicopsyche sp	5.0	5	SC	X
Trichoptera	Uenoidae	Neophylax autumnus	1.6	2	SC	X
Trichoptera	Uenoidae	Neophylax ayanus	1.6	2	SC	X
Trichoptera	Uenoidae	Neophylax consimilis	1.5	2	SC	X
Trichoptera	Uenoidae	Neophylax fuscus	0.0	2	SC	X
Trichoptera	Uenoidae	Neophylax mitchelli	0.0	2	SC	X
Trichoptera	Uenoidae	Neophylax oligius	2.2	2	SC	X
Trichoptera	Uenoidae	Neophylax sp	2.2	2	SC	X
Trichoptera	Brachycentridae	Brachycentrus lateralis	0.6	1	CG	X
Trichoptera	Brachycentridae	Brachycentrus nigrosoma	2.3	1	CF	X
Trichoptera	Brachycentridae	Brachycentrus numerosus	1.7	1	CG	X
Trichoptera	Brachycentridae	Brachycentrus sp	2.1	1	CG	X
Trichoptera	Brachycentridae	Micrasema bennetti	0.0	1	SH	X
Trichoptera	Brachycentridae	Micrasema charonis	0.8	1	SH	X
Trichoptera	Brachycentridae	Micrasema rusticum	0.0	1	SH	X
Trichoptera	Brachycentridae	Micrasema sp	1.0	1	SH	X
Trichoptera	Brachycentridae	Micrasema wataga	2.6	1	SH	X
Trichoptera	Glossosomatidae	Agapetus fuscipes	0.0	1	SC	X
Trichoptera	Glossosomatidae	Agapetus hessi	0.0	1	SC	X
Trichoptera	Glossosomatidae	Agapetus illini	3.0	1	SC	X
Trichoptera	Glossosomatidae	Agapetus rossi	0.0	1	SC	X
Trichoptera	Glossosomatidae	Agapetus sp	0.0	1	SC	X
Trichoptera	Glossosomatidae	Agapetus tomus	3.0	1	SC	X
Trichoptera	Glossosomatidae	Glossosoma intermedium	1.5	1	SC	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Glossosomatidae	Glossosoma nigrior	1.5	1	SC	X
Trichoptera	Glossosomatidae	Glossosoma sp	1.6	1	SC	X
Trichoptera	Glossosomatidae	Matrioptila jeanae	0.0	1	SC	X
Trichoptera	Glossosomatidae	Protoptila maculata	2.0	1	SC	X
Trichoptera	Glossosomatidae	Protoptila palina	2.0	1	SC	X
Trichoptera	Glossosomatidae	Protoptila sp	2.6	1	SC	X
Trichoptera	Hydropsychidae	Ceratopsyche alhedra	0.0	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche bifida	1.0	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche bronta	2.7	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche cheilonis	1.4	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche morosa	3.2	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche piatrix	1.4	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche riola	1.4	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche slossonae	0.0	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche sp	1.4	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche sparna	3.2	4	CF	X
Trichoptera	Hydropsychidae	Ceratopsyche ventura	1.5	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche analis	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche aphanta	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche campyla	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche minuscula	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche oxa	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche pasella	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche rossi	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche sordida	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche sp	6.2	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche sp # 1	6.6	4	CF	X
Trichoptera	Hydropsychidae	Cheumatopsyche sp # 2	6.6	4	CF	X
Trichoptera	Hydropsychidae	Diplectrona metaqui	2.0	4	CF	X
Trichoptera	Hydropsychidae	Diplectrona modesta	2.2	4	CF	X
Trichoptera	Hydropsychidae	Homoplectra doringa	3.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche aerata	5.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche betteni	7.8	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche bidens	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche carolina	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche cuanis	5.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche demora	2.1	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche depravata	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche dicantha	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche frisoni	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche hageni	5.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche leonardi	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche orris	4.0	4	CF	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Hydropsychidae	Hydropsyche patera	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche phalerata	3.6	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche rossi	4.8	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche scalaris	2.1	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche simulans	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche sp	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsyche valanis	4.0	4	CF	X
Trichoptera	Hydropsychidae	Hydropsychidae (pupa)	4.0	4	CF	X
Trichoptera	Hydropsychidae	Macrostemum zebratum	3.6	4	CF	X
Trichoptera	Hydropsychidae	Paraspsyche cardis	0.0	4	CF	X
Trichoptera	Hydropsychidae	Potamyia flava	5.0	4	CF	X
Trichoptera	Psychomyiidae	Lype diversa	4.1	3	SC	X
Trichoptera	Psychomyiidae	Psychomyia flavida	2.9	3	CG	X
Trichoptera	Psychomyiidae	Psychomyia nomada	2.0	3	CG	X
Trichoptera	Psychomyiidae	Psychomyia sp	2.5	3	CG	X
Trichoptera	Odontoceridae	Psilotreta labida	0.0	0	SC	X
Trichoptera	Odontoceridae	Psilotreta rufa	0.0	0	SC	X
Trichoptera	Odontoceridae	Psilotreta sp	0.0	0	SC	X
Trichoptera	Philopotamidae	Chimarra aterrima	2.0	2	CF	X
Trichoptera	Philopotamidae	Chimarra feria	2.8	2	CF	X
Trichoptera	Philopotamidae	Chimarra obscura	2.8	2	CF	X
Trichoptera	Philopotamidae	Chimarra socia	2.8	2	CF	X
Trichoptera	Philopotamidae	Chimarra sp	2.8	2	CF	X
Trichoptera	Philopotamidae	Dolophilodes distinctus	0.8	2	CF	X
Trichoptera	Philopotamidae	Dolophilodes sp	0.8	2	CF	X
Trichoptera	Philopotamidae	Wormaldia moesta	0.7	2	CF	X
Trichoptera	Philopotamidae	Wormaldia shawnee	0.7	2	CF	X
Trichoptera	Philopotamidae	Wormaldia sp	0.7	2	CF	X
Trichoptera	Phryganeidae	Agrypnia vestita	6.0	6	SH	
Trichoptera	Phryganeidae	Banksiola dossuaria	6.0	6	SH	
Trichoptera	Phryganeidae	Phryganea sayi	6.0	6	SH	
Trichoptera	Phryganeidae	Phryganea sp	6.0	6	SH	
Trichoptera	Phryganeidae	Ptilostomis ocellifera	6.4	6	SH	
Trichoptera	Phryganeidae	Ptilostomis postica	6.4	6	SH	
Trichoptera	Phryganeidae	Ptilostomis semifasciata	6.4	6	SH	
Trichoptera	Phryganeidae	Ptilostomis sp	6.4	6	SH	
Trichoptera	Polycentropodidae	Cernotina sp	4.0	4	PR	X
Trichoptera	Polycentropodidae	Cynellus fraternus	7.3	4	CF	X
Trichoptera	Polycentropodidae	Neureclipsis crepuscularis	4.2	4	CF	X
Trichoptera	Polycentropodidae	Neureclipsis parvulus	4.2	4	CF	X
Trichoptera	Polycentropodidae	Neureclipsis sp	4.2	4	CF	X
Trichoptera	Polycentropodidae	Nyctiophylax affinis	0.9	4	PR	X
Trichoptera	Polycentropodidae	Nyctiophylax celta	0.7	4	PR	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Trichoptera	Polycentropodidae	Nyctiophylax moestus	0.3	4	PR	X
Trichoptera	Polycentropodidae	Nyctiophylax sp	0.9	4	PR	X
Trichoptera	Polycentropodidae	Phylocentropus carolinus	5.6	4	CF	
Trichoptera	Polycentropodidae	Phylocentropus hansonii	5.6	4	CF	
Trichoptera	Polycentropodidae	Phylocentropus placidus	3.5	4	CF	X
Trichoptera	Polycentropodidae	Phylocentropus sp.	4.5	4	CF	
Trichoptera	Polycentropodidae	Polycentropus barri	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus blicklei	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus centralis	3.0	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus cinereus	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus confusus	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus maculatus	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus neiswanderi	3.0	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus remotus	3.5	4	CF	X
Trichoptera	Polycentropodidae	Polycentropus sp	3.5	4	PR	X
Trichoptera	Polycentropodidae	Polycentropus sp1(short tarsus)	3.5	4	PR	X
Trichoptera	Polycentropodidae	Polycentropus sp2(long tarsus)	3.5	4	PR	X
Trichoptera	Polycentropodidae	Unidentified Polycentropodid	4.0	4		
Lepidoptera	Pyralidae	Crambus sp	5.0	3	SH	
Lepidoptera	Pyralidae	Isoparce cupressi	5.0	3	SH	
Lepidoptera	Pyralidae	Munroessa faulalis	5.0	3	SH	
Lepidoptera	Pyralidae	Munroessa gyralis	5.0	3	SH	
Lepidoptera	Pyralidae	Parapoynx obscuralis	1.0	3	SH	X
Lepidoptera	Pyralidae	Petrophila confusalis	1.8	3	SC	X
Lepidoptera	Pyralidae	Petrophila fulicalis	1.8	3	SC	X
Lepidoptera	Pyralidae	Petrophila sp	1.8	3	SH	X
Lepidoptera	Pyralidae	Pyralidae - petrophila	5.0	3	SH	
Lepidoptera	Pyralidae	Unidentified Pyralidae	8.0	3	SH	
Lepidoptera	Noctuidae	Archanara sp	5.0	3	SH	
Coleoptera	Dryopidae	Helichus basalis	4.6	5	SC	X
Coleoptera	Dryopidae	Helichus fastigiatus	4.6	5	SC	X
Coleoptera	Dryopidae	Helichus lithophilus	4.6	5	SC	X
Coleoptera	Dryopidae	Helichus sp	4.6	5	SC	X
Coleoptera	Dryopidae	Helichus striatus	4.6	5	SC	X
Coleoptera	Dryopidae	Pelonomus obscurus	5.4	5	SC	
Coleoptera	Dryopidae	Unidentified Dryopidae	5.0	5	SC	
Coleoptera	Psephenidae	Ectopria nervosa	4.2	5	SC	X
Coleoptera	Psephenidae	Ectopria sp larva	4.2	5	SC	X
Coleoptera	Psephenidae	Psephenus herricki	2.4	5	SC	
Coleoptera	Haliplidae	Halipus fasciatus	8.7	8	PH	
Coleoptera	Haliplidae	Halipus sp	8.7	8	PH	
Coleoptera	Haliplidae	Halipus triopsis	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes dietrichi	8.7	8	PH	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Coleoptera	Haliplidae	Peltodytes duodecimpunctatus	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes floridensis	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes lengi	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes muticus	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes oppositus	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes sexmaculatus	8.7	8	PH	
Coleoptera	Haliplidae	Peltodytes sp	8.7	8	PH	
Coleoptera	Ptilodactylidae	Anchytarsus bicolor	3.6	2	SH	X
Coleoptera	Scirtidae	Cyphon sp	5.0	5	SC	
Coleoptera	Scirtidae	Elodes sp	5.0	5	SC	
Coleoptera	Scirtidae	Prionocyphon sp	5.0	5	SC	
Coleoptera	Scirtidae	Scirtes sp	5.0	5	SH	
Coleoptera	Scirtidae	Unidentified Scirtid	5.0	5	SH	
Coleoptera	Helophoridae	Helophorus sp	7.6	8	SH	
Coleoptera	Hydrophilidae	Anacaena limbata	8.3	9	PR	
Coleoptera	Hydrophilidae	Berosus aculeatus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus acuminatus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus exiguus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus ordinatus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus pantherinus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus peregrinus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus sp A	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus sp B	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus sp(larvae)	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus striatus	8.4	9	PH	
Coleoptera	Hydrophilidae	Berosus youngi	8.4	9	PH	
Coleoptera	Hydrophilidae	Cercyon sp		9		
Coleoptera	Hydrophilidae	Cymbiodyta sp	8.3	9	PR	
Coleoptera	Hydrophilidae	Cymbiodyta vindicta	8.3	9	PR	
Coleoptera	Hydrophilidae	Derallus altus	9.0	9	PR	
Coleoptera	Hydrophilidae	Enochrus hamiltoni	8.8	9	PR	
Coleoptera	Hydrophilidae	Enochrus ochraceus	8.8	9	PH	
Coleoptera	Hydrophilidae	Enochrus pygmaeus nebulosus	8.8	9	PR	
Coleoptera	Hydrophilidae	Enochrus sayi	8.8	9	PR	
Coleoptera	Hydrophilidae	Enochrus sp	8.8	9	PR	
Coleoptera	Hydrophilidae	Helobata sp		9	CG	
Coleoptera	Hydrophilidae	Helochaeres maculicollis	8.3	9	PR	
Coleoptera	Hydrophilidae	Helochaeres sp	8.3	9	PR	
Coleoptera	Hydrophilidae	Helocombus bifidus	8.5	9	CG	
Coleoptera	Hydrophilidae	Helocombus sp	8.5	9	CG	
Coleoptera	Hydrophilidae	Hydrobius molaenus	8.3	9	PH	
Coleoptera	Hydrophilidae	Hydrobius sp	8.3	9	PH	
Coleoptera	Hydrophilidae	Hydrobius tumidus	8.3	9	PH	



**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Coleoptera	Hydrophilidae	Hydrochara sp	8.3	9	PR	
Coleoptera	Hydrophilidae	Laccobius sp	7.3	9	PR	
Coleoptera	Hydrophilidae	Paracymus sp	8.0	9	PR	
Coleoptera	Hydrophilidae	Paracymus sp (Larvae)	8.3	9	PR	
Coleoptera	Hydrophilidae	Paracymus subcupreus	3.5	9	PR	
Coleoptera	Hydrophilidae	Sperchopsis sp	6.5	9		X
Coleoptera	Hydrophilidae	Sperchopsis tessellata	6.5	9		
Coleoptera	Hydrophilidae	Tropisternus blatchleyi blatchleyi	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus collaris	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus lateralis numbatus	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus mixtus	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus natator	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus sp	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus sp (larvae)	9.7	9	CG	
Coleoptera	Hydrophilidae	Tropisternus sp(larvae)	9.7	9	CG	
Coleoptera	Hydrophilidae	Unidentified Hydrophilid	6.3	9	PR	
Coleoptera	Noteridae	Hydrocanthus atripennis	6.9	7	PR	
Coleoptera	Noteridae	Hydrocanthus iricolor	6.9	7	PR	
Coleoptera	Noteridae	Suphisellus sp	7.0	7	CG	
Coleoptera	Staphylinidae	Micralymma sp	8.0		PR	
Coleoptera	Staphylinidae	Thinobius sp			PR	
Coleoptera	Gyrinidae	Dineutus assimilis	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus ciliatus	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus discolor	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus emarginatus	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus nigrior	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus robertsi	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus serrulatus	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus sp	5.5	6	PR	
Coleoptera	Gyrinidae	Dineutus sp(larvae)	5.5	6	PR	
Coleoptera	Gyrinidae	Gyretes iricolor	5.8	6	PR	
Coleoptera	Gyrinidae	Gyretes sp	5.8	6	PR	
Coleoptera	Gyrinidae	Gyrinus aeneolus	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus analis	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus borealis	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus discolor	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus lugens	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus marginellus	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus pachysomus	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus sp	6.2	6	PR	
Coleoptera	Gyrinidae	Gyrinus sp(larvae)	8.3	6	PR	
Coleoptera	Dytiscidae	Acilius semisulcatus	9.0	9	PR	
Coleoptera	Dytiscidae	Acilius sp	9.0	9	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Coleoptera	Dytiscidae	Agabus ambiguus	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus bifarius	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus disintergratus	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus gatates	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus glabrasellus	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus johannis	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus punctatus	9.1	9	PR	
Coleoptera	Dytiscidae	Agabus sp	8.9	9	PR	
Coleoptera	Dytiscidae	Agabus sp. A Epler	9.0	9	PR	
Coleoptera	Dytiscidae	Agabus staninus	9.0	9	PR	
Coleoptera	Dytiscidae	Bidessonotus longovalis	9.0	9	PR	
Coleoptera	Dytiscidae	Celina sp	9.0	9	PR	
Coleoptera	Dytiscidae	Copelatus glyphicus	9.1	9	PC	
Coleoptera	Dytiscidae	Copelatus sp	9.1	9	PC	
Coleoptera	Dytiscidae	Coptotomus interrogatus	9.2	9	PR	
Coleoptera	Dytiscidae	Coptotomus loticus	9.2	9	pr	
Coleoptera	Dytiscidae	Coptotomus sp.	9.3	9	PR	
Coleoptera	Dytiscidae	Coptotomus venustus	9.2	9	PR	
Coleoptera	Dytiscidae	Cybister fimbriolatus	9.1	9	PR	
Coleoptera	Dytiscidae	Cybister sp	9.1	9	PR	
Coleoptera	Dytiscidae	Deronectes sp (larvae)	4.0	9	PR	
Coleoptera	Dytiscidae	Dytiscus carolinus nimbatus	8.9	9	PR	
Coleoptera	Dytiscidae	Dytiscus hybridus	8.0	9	PR	
Coleoptera	Dytiscidae	Dytiscus sp	9.1	9	PR	
Coleoptera	Dytiscidae	Graphoderus sp	8.0	9	PR	
Coleoptera	Dytiscidae	Hydaticus sp	9.1	9	PR	
Coleoptera	Dytiscidae	Hydrochus callosus	9.0	9	PR	
Coleoptera	Dytiscidae	Hydrochus sp	6.6	9	PR	
Coleoptera	Dytiscidae	Hydropopus blanchardi	8.9	9	PR	
Coleoptera	Dytiscidae	Hydroporus oblitus gr	8.9	9	PR	
Coleoptera	Dytiscidae	Hydroporus sp	8.6	9	PR	
Coleoptera	Dytiscidae	Hydroporus sp B	8.9	9	PR	
Coleoptera	Dytiscidae	Hydroporus undalatus	8.9	9	PR	
Coleoptera	Dytiscidae	Hydrovatus platycornis	9.0	9	PR	
Coleoptera	Dytiscidae	Hydrovatus sp	10.0	9	PR	
Coleoptera	Dytiscidae	Hygrotus nubilis	9.0	9	PR	
Coleoptera	Dytiscidae	Hygrotus sp	9.0	9	PR	
Coleoptera	Dytiscidae	Ilybius biguttulus	8.0	9	PR	
Coleoptera	Dytiscidae	Ilybius oblitus	9.1	9	PR	
Coleoptera	Dytiscidae	Ilybius sp	9.0	9	PR	
Coleoptera	Dytiscidae	Laccobius agilis	8.0	9	PR	
Coleoptera	Dytiscidae	Laccophilus fasciatus	10.0	9	PR	
Coleoptera	Dytiscidae	Laccophilus gentilis	10.0	9	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Coleoptera	Dytiscidae	Laccophilus maculosus maculosus	10.0	9	PR	
Coleoptera	Dytiscidae	Laccophilus schwarzi	10.0	9	PR	
Coleoptera	Dytiscidae	Laccophilus sp	10.0	9	PR	
Coleoptera	Dytiscidae	Lioporeus pilatei	9.0	9	PR	
Coleoptera	Dytiscidae	Lioporeus sp	9.0	9	PR	
Coleoptera	Dytiscidae	Lioporeus sp1	9.0	9	PR	
Coleoptera	Dytiscidae	Lioporeus sp2	9.0	9	PR	
Coleoptera	Dytiscidae	Lioporeus triangularis	9.0	9	PR	
Coleoptera	Dytiscidae	Nebrioporus/Stictotarsus gr	9.1	9	PR	
Coleoptera	Dytiscidae	Neoporus carolinus	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus clypealis	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus dixianus	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus hybridus	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus mellitus	8.9	9	OR	
Coleoptera	Dytiscidae	Neoporus shermani	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus sp	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus straitopunctatus	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus undulatus	8.9	9	PR	
Coleoptera	Dytiscidae	Neoporus vittatipennis	8.9	9	PR	
Coleoptera	Dytiscidae	Oredytes sp	9.1	9	PR	
Coleoptera	Dytiscidae	Rhantus sp	3.6	9	PR	
Coleoptera	Dytiscidae	Thermonectus basillarus	9.0	9	PR	
Coleoptera	Dytiscidae	Unidentified Dyticid	8.0	9	PR	
Coleoptera	Dytiscidae	Uvarus granarius	10.0	9	PR	
Coleoptera	Dytiscidae	Uvarus lacustris	10.0	9	PR	
Coleoptera	Dytiscidae	Uvarus sp	10.0	9	PR	
Coleoptera	Chrysomelidae	Donacia sp	8.0	8	SH	
Coleoptera	Curculionidae	Bagous sp	10.0	10	SH	
Coleoptera	Curculionidae	Lixus sp	10.0	10	SH	
Coleoptera	Curculionidae	Unidentified Curculionid	10.0	10	SH	
Coleoptera	Elmidae	Ancyronyx variegatus	6.5	5	SC	X
Coleoptera	Elmidae	Dubiraphia bivittata	5.9	5	SC	X
Coleoptera	Elmidae	Dubiraphia quadrinotata	5.9	5	SC	X
Coleoptera	Elmidae	Dubiraphia sp (larvae)	5.0	5	SC	X
Coleoptera	Elmidae	Dubiraphia vittata	4.1	5	SC	X
Coleoptera	Elmidae	Macronychus glabratus	4.6	5	CG	X
Coleoptera	Elmidae	Microcylloepus pusillus	2.1	5	SC	X
Coleoptera	Elmidae	Optioservus immunis	2.7	5	SC	X
Coleoptera	Elmidae	Optioservus ovalis	2.4	5	SC	X
Coleoptera	Elmidae	Optioservus sp	2.4	5	SC	X
Coleoptera	Elmidae	Optioservus sp(larvae)	2.4	5	SC	X
Coleoptera	Elmidae	Optioservus trivittatus	2.7	5	SC	X
Coleoptera	Elmidae	Oulimnius latiusculus	1.8	5	SC	X

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Coleoptera	Elmidae	Promoresia elegans	2.2	5	SC	X
Coleoptera	Elmidae	Promoresia sp (larvae)	2.4	5	SC	X
Coleoptera	Elmidae	Promoresia tardella	0.0	5	SC	X
Coleoptera	Elmidae	Stenelmis bicarinata	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis concinna	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis crenata	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis decorata	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis hungerfordi	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis lateralis	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis quadrimaculata	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis sandersoni	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis sexlineata	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis sp	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis sp(larvae)	5.1	5	SC	X
Coleoptera	Elmidae	Stenelmis vittipennis	5.1	5	SC	X
Coleoptera	Elmidae	Unidentified Elmids	4.2	5	SC	X
Coleoptera	Limnichidae	Lutrochus laticeps	5.0	5	SC	X
Diptera	Sacrophagidae	Blaesoxipha fletcheri	5.0	5	CG	
Diptera	Muscidae	Limnophora sp	8.4	8	PR	
Diptera	Muscidae	Unidentified Muscid	8.0	8		
Diptera	Syrphidae	Chrysogaster sp	10.0	10	CG	
Diptera	Syrphidae	Eristalis tenax	10.0	10	CG	
Diptera	Culicidae	Aedes cinereus	9.4	9	CG	
Diptera	Culicidae	Anopheles (pupae)	8.6	9	CG	
Diptera	Culicidae	Anopheles atropos	8.6	9	CF	
Diptera	Culicidae	Anopheles crucians	8.6	9	CF	
Diptera	Culicidae	Anopheles sp	8.6	9	CF	
Diptera	Culicidae	Culex pipiens	10.0	9	CF	
Diptera	Culicidae	Culex sp	10.0	9	CF	
Diptera	Chaoboridae	Chaoborus albatus	8.0	9	PR	
Diptera	Chaoboridae	Chaoborus americanus	8.5	9	PR	
Diptera	Chaoboridae	Chaoborus punctipennis	8.5	9	PR	
Diptera	Chaoboridae	Chaoborus sp	8.5	9	PR	
Diptera	Chaoboridae	Eucorethra underwoodi	8.0	9	PR	
Diptera	Chaoboridae	Mochlonyx sp	8.0	9	PR	
Diptera	Tipulidae	Antocha saxicola	4.3	5	PR	X
Diptera	Tipulidae	Antocha sp	4.3	5	PR	X
Diptera	Tipulidae	Cryptolabis sp	4.9	5	SH	
Diptera	Tipulidae	Dicranota sp	0.0	5	PR	
Diptera	Tipulidae	Dolichopeza sp	5.5	5	CG	
Diptera	Tipulidae	Gonomyia sp	5.0	5		
Diptera	Tipulidae	Helius sp	5.3	5	CG	
Diptera	Tipulidae	Hexatoma albitarsis	4.3	5	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Tipulidae	Hexatoma longicornis	4.3	5	PR	
Diptera	Tipulidae	Hexatoma sp	4.3	5	PR	
Diptera	Tipulidae	Hexatoma spinosa	4.3	5	PR	
Diptera	Tipulidae	Limnophila albipes	4.9	5	PR	
Diptera	Tipulidae	Limnophila macrocera	4.9	5	PR	
Diptera	Tipulidae	Limnophila simplex	4.9	5	PR	
Diptera	Tipulidae	Limnophila sp	4.9	5	PR	
Diptera	Tipulidae	Limonia sp	9.6	5	SC	
Diptera	Tipulidae	Megistocera longipennis	4.9	5	SH	
Diptera	Tipulidae	Molophilus sp	5.0	5	SH	
Diptera	Tipulidae	Ormosia sp	4.9	5	CG	
Diptera	Tipulidae	Pedicia sp	4.9	5	PR	
Diptera	Tipulidae	Pilaria sp	4.9	5	PR	
Diptera	Tipulidae	Prionocera sp	4.9	5	SH	
Diptera	Tipulidae	Pseudolimnophila sp	7.2	5	PR	
Diptera	Tipulidae	Tipula abdominalis	7.3	5	SH	
Diptera	Tipulidae	Tipula furca	7.3	5	SH	
Diptera	Tipulidae	Tipula sp	7.3	5	SH	
Diptera	Tipulidae	Tipula sp1	7.3	5	SH	
Diptera	Tipulidae	Tipula sp2	7.3	5	SH	
Diptera	Tipulidae	Tipula strepens	7.3	5	SH	
Diptera	Tipulidae	Unidentified Tipulid	5.0	5		
Diptera	Empididae	Chelifera sp	8.1	8	PR	
Diptera	Empididae	Clinocera sp	8.1	8	PR	X
Diptera	Empididae	Hemerodromia sp	8.1	8	PR	
Diptera	Empididae	Phyllodromia sp	8.1	8	PR	
Diptera	Empididae	Unidentified Empidid	8.1	8	PR	
Diptera	Athericidae	Atherix lantha	2.1	2	PR	
Diptera	Athericidae	Atherix sp	2.1	2	PR	
Diptera	Athericidae	Atherix variegata	2.1	2	PR	
Diptera	Blephariceridae	Bibiocephala sp	0.5	1	SC	X
Diptera	Blephariceridae	Blepharicera sp	0.0	1	SC	X
Diptera	Dixidae	Dixa notata	2.8	4	CG	
Diptera	Dixidae	Dixa sp	2.6	4	CG	
Diptera	Dixidae	Dixella sp	5.0	4	PR	
Diptera	Chironomidae	Ablabesmyia annulata	2.0	7	PR	
Diptera	Chironomidae	Ablabesmyia janta	6.0	7	PR	
Diptera	Chironomidae	Ablabesmyia mallochii gr	7.2	7	PR	
Diptera	Chironomidae	Ablabesmyia parajanta	7.4	7	PR	
Diptera	Chironomidae	Ablabesmyia peleensis	9.7	7	PR	
Diptera	Chironomidae	Ablabesmyia rhamphe	7.5	7	PR	
Diptera	Chironomidae	Ablabesmyia sp	7.2	7	PR	
Diptera	Chironomidae	Apedilum sp		7	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Apsectrotanypus sp	0.0	7		
Diptera	Chironomidae	Axarus sp	7.0	7	CG	
Diptera	Chironomidae	Brillia flavifrons	5.2	7	SH	
Diptera	Chironomidae	Brillia parva	5.2	7	SH	
Diptera	Chironomidae	Brillia sp	5.2	7	SH	
Diptera	Chironomidae	Brundiniella eumorpha	1.7	7	PR	
Diptera	Chironomidae	Brundiniella sp	3.8	7	PR	
Diptera	Chironomidae	Cardiocladius albiplumus	6.2	7	PR	X
Diptera	Chironomidae	Cardiocladius obscurus	6.2	7	PR	X
Diptera	Chironomidae	Cardiocladius sp	5.9	7	PR	X
Diptera	Chironomidae	Chironomus anthracinus gr	9.8	7	CG	
Diptera	Chironomidae	Chironomus crassicaudatus	9.8	7	CG	
Diptera	Chironomidae	Chironomus decorus gr	9.8	7	CG	
Diptera	Chironomidae	Chironomus plumosus gr	9.8	7	CG	
Diptera	Chironomidae	Chironomus riparius gr	9.8	7	CG	
Diptera	Chironomidae	Chironomus sp	9.6	7	CG	
Diptera	Chironomidae	Chironomus stigmaterus	9.8	7	CG	
Diptera	Chironomidae	Cladopelma sp	3.5	7	CG	
Diptera	Chironomidae	Cladotanytarsus sp	4.1	7	CG	
Diptera	Chironomidae	Clinotanypus pinguis	8.7	7	PR	
Diptera	Chironomidae	Clinotanypus sp	9.1	7	PR	
Diptera	Chironomidae	Coelotanypus scapularis	6.2	7	PR	
Diptera	Chironomidae	Coelotanypus sp	8.0	7	PR	
Diptera	Chironomidae	Conchapelopia aleta	8.7	7	PR	
Diptera	Chironomidae	Conchapelopia sp	8.7	7	PR	
Diptera	Chironomidae	Conchapelopia sp2	6.7	7	CG	
Diptera	Chironomidae	Constempellina sp	4.2	7	CG	
Diptera	Chironomidae	Corynoneura celeripes	6.2	7	CG	
Diptera	Chironomidae	Corynoneura sp	6.0	7	CG	
Diptera	Chironomidae	Corynoneura sp B		7	CG	
Diptera	Chironomidae	Corynoneura sp C (Epler)	6.2	7	CG	
Diptera	Chironomidae	Corynoneura sp E	6.2	7	CG	
Diptera	Chironomidae	Corynoneura taris	6.2	7	CG	
Diptera	Chironomidae	Cricotopus absurdus	5.0		CG	
Diptera	Chironomidae	Cricotopus algarum	7.0	7	SH	
Diptera	Chironomidae	Cricotopus annulator	7.0	7	SH	
Diptera	Chironomidae	Cricotopus bicinctus gr	8.5	7	SH	
Diptera	Chironomidae	Cricotopus curtus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus intersectus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus laetus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus laricomalis	7.0	7	SH	
Diptera	Chironomidae	Cricotopus lebetis		7	SH	
Diptera	Chironomidae	Cricotopus mackenziensis	7.0	7	SH	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Cricotopus sp	7.0	7	SH	
Diptera	Chironomidae	Cricotopus sp #9	7.0	7	SH	
Diptera	Chironomidae	Cricotopus sp 2 (Beck)	7.7	7	SH	
Diptera	Chironomidae	Cricotopus sylvestris gr	10.0	7	SH	
Diptera	Chironomidae	Cricotopus tremulus gr	7.0	7	SH	
Diptera	Chironomidae	Cricotopus triannulatus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus tricinctus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus trifascia gr	2.8	7	SH	
Diptera	Chironomidae	Cricotopus trifasciatus	7.0	7	SH	
Diptera	Chironomidae	Cricotopus vierriensis	4.4	7	SH	
Diptera	Chironomidae	Cricotopus/Orthocladius gr	7.1	7	CG	
Diptera	Chironomidae	Cryptochironomus blarina gr	7.4	7	PR	
Diptera	Chironomidae	Cryptochironomus fulvus gr	6.4	7	PR	
Diptera	Chironomidae	Cryptochironomus sp	6.4	7	PR	
Diptera	Chironomidae	Cryptotendipes sp	6.2	7	CG	
Diptera	Chironomidae	Demicryptochironomus sp	2.1	7	CG	
Diptera	Chironomidae	Diamesia sp	8.1	7	CG	
Diptera	Chironomidae	Dicrotendipes fumidus	7.9	7	CG	
Diptera	Chironomidae	Dicrotendipes incurvus	7.9	7	CG	
Diptera	Chironomidae	Dicrotendipes leucocelis	7.9	7	CG	
Diptera	Chironomidae	Dicrotendipes lucifer	8.0	7	CG	
Diptera	Chironomidae	Dicrotendipes modestus	8.7	7	CG	
Diptera	Chironomidae	Dicrotendipes neomodestus	8.1	7	CG	
Diptera	Chironomidae	Dicrotendipes nervosus	9.8	7	CG	
Diptera	Chironomidae	Dicrotendipes simpsoni	10.0	7	CG	
Diptera	Chironomidae	Dicrotendipes sp	8.1	7	CG	
Diptera	Chironomidae	Dicrotendipes thanatogratus	7.9	7	CG	
Diptera	Chironomidae	Diplocladius sp	7.0	7	CG	
Diptera	Chironomidae	Djalmabatista pulcher	9.3	7	PR	
Diptera	Chironomidae	Einfeldia sp	7.1	7	CG	
Diptera	Chironomidae	Endochironomus nigricans	7.8	7	SH	
Diptera	Chironomidae	Endochironomus sp	7.8	7	SH	
Diptera	Chironomidae	Endochironomus subtendens	7.8	7	SH	
Diptera	Chironomidae	Endotribelos sp		7	CG	
Diptera	Chironomidae	Epoicocladius sp	2.0	7	CG	
Diptera	Chironomidae	Eukiefferiella bavarica gr	3.7	7	CG	
Diptera	Chironomidae	Eukiefferiella brehmi gr	2.7	7	CG	
Diptera	Chironomidae	Eukiefferiella brevicealcar gr	2.2	7	CG	
Diptera	Chironomidae	Eukiefferiella claripennis gr	5.6	7	CG	
Diptera	Chironomidae	Eukiefferiella cyanea gr	2.5	7	CG	
Diptera	Chironomidae	Eukiefferiella devonica gr	2.6	7	CG	
Diptera	Chironomidae	Eukiefferiella gracei gr	3.4	7	CG	
Diptera	Chironomidae	Eukiefferiella pseudomontana	4.0	7	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Eukiefferiella rectangularis gr	3.4	7	CG	
Diptera	Chironomidae	Eukiefferiella sp	3.4	7	CG	
Diptera	Chironomidae	Euryhopsis sp		7		
Diptera	Chironomidae	Glyptotendipes barbipes	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes lobiferus	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes meridionalis	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes paripes	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes senilus	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes sp	9.4	7	SH	
Diptera	Chironomidae	Glyptotendipes sp B (Epler)	9.4	7	SH	
Diptera	Chironomidae	Goeldichironomus holoprasinus	10.0	7	CG	
Diptera	Chironomidae	Guttipelopia sp	6.3	7	PR	
Diptera	Chironomidae	Harnischia curtilamellata	9.0	7	CG	
Diptera	Chironomidae	Harnischia sp	9.1	7	CG	
Diptera	Chironomidae	Hayesomyia sp	6.2	7	PR	
Diptera	Chironomidae	Heleniella sp	0.0	7	CG	
Diptera	Chironomidae	Helopelopia sp	6.2	7	PR	
Diptera	Chironomidae	Heterotrissocladius marcidus gr	5.4	7	CG	
Diptera	Chironomidae	Heterotrissocladius sp	5.2	7	CG	
Diptera	Chironomidae	Hydrobaenus lugabris	9.5	7	SC	
Diptera	Chironomidae	Hydrobaenus pilipes	9.5	7	SC	
Diptera	Chironomidae	Hydrobaenus sp	9.5	7	SC	
Diptera	Chironomidae	Kiefferulus dux	0.0	7	CG	
Diptera	Chironomidae	Krenopelopia sp	6.2	7	PR	
Diptera	Chironomidae	Labrundinia maculata	6.0	7	PR	
Diptera	Chironomidae	Labrundinia pilosella	5.9	7	PR	
Diptera	Chironomidae	Labrundinia virescens	4.3	7	PR	
Diptera	Chironomidae	Larsia decolorata	8.3	7	PR	
Diptera	Chironomidae	Larsia indistincta	8.3	7	PR	
Diptera	Chironomidae	Larsia sp	9.3	7	PR	
Diptera	Chironomidae	Limnophyes sp		7	CG	
Diptera	Chironomidae	Lopescladius sp	1.7	7	CG	
Diptera	Chironomidae	Meropelopia americana	6.2	7	CG	
Diptera	Chironomidae	Meropelopia sp	6.2	7	CG	
Diptera	Chironomidae	Mesosmittia sp		7		
Diptera	Chironomidae	Micropsectra polita	1.4	7	CG	
Diptera	Chironomidae	Microspectra sp	1.5	7	CG	
Diptera	Chironomidae	Microtendipes caelum	6.2	7	CF	
Diptera	Chironomidae	Microtendipes pedellus gr	6.2	7	CF	
Diptera	Chironomidae	Microtendipes rydalensis gp	6.2	7	CF	
Diptera	Chironomidae	Microtendipes sp	5.3	7	CF	
Diptera	Chironomidae	Monopelopia sp	6.2	7	PR	
Diptera	Chironomidae	Nanocladius bicolor	7.2	7	CG	



**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Nanocladius branchicolus	5.0	7	OT	
Diptera	Chironomidae	Nanocladius distinctus	7.2	7	CG	
Diptera	Chironomidae	Nanocladius downesi	2.5	7	CG	
Diptera	Chironomidae	Nanocladius plecopterocolutherus	2.2	7	OT	
Diptera	Chironomidae	Nanocladius rectinervis	7.1	7	CG	
Diptera	Chironomidae	Nanocladius sp	7.1	7	CG	
Diptera	Chironomidae	Nanocladius spiniplenus	7.2	7	CG	
Diptera	Chironomidae	Natarsia baltimoreus	10.0	7	PR	
Diptera	Chironomidae	Natarsia sp	10.0	7	PR	
Diptera	Chironomidae	Natarsia sp # 1	10.0	7	PR	
Diptera	Chironomidae	Natarsia sp A (Epler)	10.0	7	PR	
Diptera	Chironomidae	Nilotanypus americanus	3.0	7	PR	
Diptera	Chironomidae	Nilotanypus fimbriatus	4.0	7	PR	
Diptera	Chironomidae	Nilotanypus sp	3.9	7	PR	
Diptera	Chironomidae	Nilothauma babyi	5.5	7	CG	
Diptera	Chironomidae	Nilothauma sp	5.0	7	CG	
Diptera	Chironomidae	Oliveridia sp	3.2	7	CG	
Diptera	Chironomidae	Omisus sp (Epler)	6.6	7	CG	
Diptera	Chironomidae	Orthocladius annectens	9.2	7	CG	
Diptera	Chironomidae	Orthocladius carlatus	6.4	7	CG	
Diptera	Chironomidae	Orthocladius dorenius gr	6.7	7	CG	
Diptera	Chironomidae	Orthocladius lapponicus	7.3	7	CG	
Diptera	Chironomidae	Orthocladius lignicola	3.0	7	CG	
Diptera	Chironomidae	Orthocladius mallochi	9.2	7	CG	
Diptera	Chironomidae	Orthocladius nigritus gr	0.9	7	CG	
Diptera	Chironomidae	Orthocladius obumbratus	8.8	7	CG	
Diptera	Chironomidae	Orthocladius obumbratus gr	8.5	7	CG	
Diptera	Chironomidae	Orthocladius oliveri	6.4	7	CG	
Diptera	Chironomidae	Orthocladius rivulorum	3.0	7	CG	X
Diptera	Chironomidae	Orthocladius robacki	6.6	7	CG	
Diptera	Chironomidae	Orthocladius sp	7.3	7	CG	
Diptera	Chironomidae	Orthocladius sp 1	6.4	7	CG	
Diptera	Chironomidae	Orthocladius sp 2	6.4	7	CG	
Diptera	Chironomidae	Pagastia sp	1.8	7	CG	
Diptera	Chironomidae	Pagastiella sp	2.2	7	CG	
Diptera	Chironomidae	Parachaetocladius abnobaeus	6.7	7	CG	
Diptera	Chironomidae	Parachaetocladius sp	0.0	7	CG	
Diptera	Chironomidae	Parachironomus abortivus	8.3	7	PR	
Diptera	Chironomidae	Parachironomus albimanus	9.0	7	PR	
Diptera	Chironomidae	Parachironomus carinatus	9.2	7	CG	
Diptera	Chironomidae	Parachironomus directus	9.2		PR	
Diptera	Chironomidae	Parachironomus frequens	9.2	7	PR	
Diptera	Chironomidae	Parachironomus hirtalatus	9.2	7	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Parachironomus pectinatellae	6.5	7	PR	
Diptera	Chironomidae	Parachironomus sp	9.4	7	PR	
Diptera	Chironomidae	Paracladopelma sp	5.5	7	CG	
Diptera	Chironomidae	Paracricotopus sp	4.7	7	CG	
Diptera	Chironomidae	Parakiefferiella sp	5.4	7	CG	
Diptera	Chironomidae	Parakiefferiella sp A (Epler)	5.0	7	CG	
Diptera	Chironomidae	Paralauterborniella nigrohalteralis	4.8	7	CG	
Diptera	Chironomidae	Paramerina sp	4.3	7	PR	
Diptera	Chironomidae	ParametrioXemus lundbecki	3.7	7	CG	
Diptera	Chironomidae	ParametrioXemus sp	3.7	7	CG	
Diptera	Chironomidae	Paratanytarsus sp	8.5	7	CG	X
Diptera	Chironomidae	Paratendipes albimanus	9.2	7	CG	X
Diptera	Chironomidae	Paratendipes sp	5.1	7	CG	
Diptera	Chironomidae	Pentaneura sp	4.7	7	PR	
Diptera	Chironomidae	Phaenopsectra dyari gr	6.8	7	SC	
Diptera	Chironomidae	Phaenopsectra flavipes	7.9	7	SC	
Diptera	Chironomidae	Phaenopsectra jucundus	6.8	7	SC	
Diptera	Chironomidae	Phaenopsectra obediens gp	6.8	7	SC	
Diptera	Chironomidae	Phaenopsectra punctipes gp	6.0	7	CG	
Diptera	Chironomidae	Phaenopsectra sp	6.5	7	SC	
Diptera	Chironomidae	Phaenopsectra/Tribelos sp	6.8	7	CG	
Diptera	Chironomidae	Polypedilum aviceps	3.7	7	SH	
Diptera	Chironomidae	Polypedilum bergi	7.0	7	SC	
Diptera	Chironomidae	Polypedilum fallax	6.4	7	SH	
Diptera	Chironomidae	Polypedilum flavum	5.3	7	SH	
Diptera	Chironomidae	Polypedilum halterale	7.3	7	SH	
Diptera	Chironomidae	Polypedilum illinoense	9.0	7	SH	
Diptera	Chironomidae	Polypedilum obtusum	6.8	7	SH	
Diptera	Chironomidae	Polypedilum ontario	6.8	7	SH	
Diptera	Chironomidae	Polypedilum scalaenum gr	8.4	7	SH	
Diptera	Chironomidae	Polypedilum simulans/digitifer	6.8	7	SH	
Diptera	Chironomidae	Polypedilum sp	6.8	7	SH	
Diptera	Chironomidae	Polypedilum sp C (Epler)	6.8	7	SH	
Diptera	Chironomidae	Polypedilum tritum	6.8	7	SH	
Diptera	Chironomidae	Potthastia longimanus	6.5	7	CG	
Diptera	Chironomidae	Potthastia sp	6.4	7	CG	
Diptera	Chironomidae	Procladius bellus	9.3	7	PR	
Diptera	Chironomidae	Procladius freemani	9.3	7	PR	
Diptera	Chironomidae	Procladius sp	9.1	7	PR	
Diptera	Chironomidae	Procladius sublettei	9.3	7	PR	
Diptera	Chironomidae	Prodiamesa longimanus	7.9	7	CG	
Diptera	Chironomidae	Prodiamesa sp	7.9	7	CG	
Diptera	Chironomidae	Psectrocladius psilopterus gr	3.8	7	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Psectrocladius sp	3.6	7	CG	
Diptera	Chironomidae	Psectrotanypus discolor	10.0	7	PR	
Diptera	Chironomidae	Psectrotanypus dyari	10.0	7	PR	
Diptera	Chironomidae	Psectrotanypus sp	0.0	7	PR	
Diptera	Chironomidae	Pseudochironomus articaudus	4.2	7	CG	
Diptera	Chironomidae	Pseudochironomus fluviiventris	4.2	7	CG	
Diptera	Chironomidae	Pseudochironomus prasinatus	4.2	7	CG	
Diptera	Chironomidae	Pseudochironomus sp	5.4	7	CG	
Diptera	Chironomidae	Pseudorthocladius sp	1.5	7	CG	
Diptera	Chironomidae	Pseudosmittia sp	6.0	7	CG	
Diptera	Chironomidae	Rheocricotopus nr. fucipes	6.8	7	CG	
Diptera	Chironomidae	Rheocricotopus pauciseta	6.8	7	CG	
Diptera	Chironomidae	Rheocricotopus robacki	7.3	7	CG	
Diptera	Chironomidae	Rheocricotopus sp	7.3	7	CG	
Diptera	Chironomidae	Rheocricotopus tuberculatus	7.3	7	CG	
Diptera	Chironomidae	Rheopelopia sp		7	PR	
Diptera	Chironomidae	Rheotanytarsus distinctissimus	6.4	7	CF	X
Diptera	Chironomidae	Rheotanytarsus exiguus gr	6.4	7	CF	X
Diptera	Chironomidae	Rheotanytarsus sp	6.4	7	CF	X
Diptera	Chironomidae	Saetheria sp 1 (Epler)	4.0	7	CG	
Diptera	Chironomidae	Smittia atterrma	6.0	7	CG	
Diptera	Chironomidae	Smittia sp	6.0	7	CG	
Diptera	Chironomidae	Stelechomyia perpulchra	5.0	7	SH	
Diptera	Chironomidae	Stempellina sp	0.0	7		
Diptera	Chironomidae	Stempellinella sp	4.6	7	CG	
Diptera	Chironomidae	Stenochironomus divinctus	6.4	7	CG	
Diptera	Chironomidae	Stenochironomus hilaris	6.4	7	CG	
Diptera	Chironomidae	Stenochironomus sp	6.5	7	CG	
Diptera	Chironomidae	Stictochironomus devinctus	6.7	7	CG	
Diptera	Chironomidae	Stictochironomus sp	6.5	7	CG	
Diptera	Chironomidae	Stilocladius sp	5.0	7	CG	
Diptera	Chironomidae	Sublettea sp	7.0	7	CF	
Diptera	Chironomidae	Symposiocladius sp	5.4	7	CG	
Diptera	Chironomidae	Sympothastia spinifera	5.7	7	CG	
Diptera	Chironomidae	Syndiamesa sp	5.1	7	CG	
Diptera	Chironomidae	Synorthocladius semivirens	4.7	7	CG	
Diptera	Chironomidae	Synorthocladius sp	4.0	7	SC	
Diptera	Chironomidae	Tanypus carinatus	9.2	7	PR	
Diptera	Chironomidae	Tanypus neopunctipennis	9.2	7	PR	
Diptera	Chironomidae	Tanypus punctipennis	9.2	7	PR	
Diptera	Chironomidae	Tanypus sp	9.2	7	PR	
Diptera	Chironomidae	Tanypus stellatus	9.2	7	PR	
Diptera	Chironomidae	Tanytarsus coffmani	6.7	7	CF	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Chironomidae	Tanytarsus glabrascens gr	6.7	7	CF	
Diptera	Chironomidae	Tanytarsus guerlus gr	6.7	7	CF	
Diptera	Chironomidae	Tanytarsus sp	6.7	7	CF	
Diptera	Chironomidae	Tanytarsus sp C	6.7	7		
Diptera	Chironomidae	Tanytarsus sp D (Epler)	6.7	7	CG	
Diptera	Chironomidae	Tanytarsus sp E	6.7	7		
Diptera	Chironomidae	Tanytarsus sp G (Epler)	6.7	7	CG	
Diptera	Chironomidae	Tanytarsus sp L	6.7	7		
Diptera	Chironomidae	Tanytarsus sp M	6.7	7		
Diptera	Chironomidae	Tanytarsus sp P	6.7	7		
Diptera	Chironomidae	Tanytarsus sp S	6.7	7		
Diptera	Chironomidae	Tanytarsus sp T (Epler)	6.7	7	CF	
Diptera	Chironomidae	Thienemanniella fusca gr	6.0	7	CG	
Diptera	Chironomidae	Thienemanniella similis	5.9	7	CG	
Diptera	Chironomidae	Thienemanniella sp	5.9	7	CG	
Diptera	Chironomidae	Thienemanniella sp B	5.9	7	CG	
Diptera	Chironomidae	Thienemanniella xena	5.9	7	CG	
Diptera	Chironomidae	Thienemannimyia gr	5.9	7	PR	
Diptera	Chironomidae	Thienemannimyia nr. spD	6.7	7	PR	
Diptera	Chironomidae	Tribelos atrum	6.3	7	CG	
Diptera	Chironomidae	Tribelos fuscicorne	6.3	7	CG	
Diptera	Chironomidae	Tribelos jucundum	6.3	7	CG	
Diptera	Chironomidae	Tribelos sp	6.3	7	CG	
Diptera	Chironomidae	Tribelos/Phaenopsectra gr	6.6	7	CG	
Diptera	Chironomidae	Tvetenia bavarica gr	3.7	7	CG	
Diptera	Chironomidae	Tvetenia discoloripes gr	3.6	7	CG	
Diptera	Chironomidae	Tvetenia sp	3.6	7	CG	
Diptera	Chironomidae	Tvetenia vitracies	4.0	7	CG	
Diptera	Chironomidae	Unidentified Chironomid	5.0	7	CG	
Diptera	Chironomidae	Unidentified Larvae	5.0	7	CG	
Diptera	Chironomidae	Unidentified Orthoclad	7.0	7	CG	
Diptera	Chironomidae	Unidentified Podonominae		7	CG	
Diptera	Chironomidae	Unidentified Pupae	7.0	7		
Diptera	Chironomidae	Unidentified Tanypodinae	6.2	7	PR	
Diptera	Chironomidae	Xenochironomus festivus	7.0	7	PR	
Diptera	Chironomidae	Xenochironomus sp	7.0	7	PR	
Diptera	Chironomidae	Xenochironomus tanionotus	7.0	7	PR	
Diptera	Chironomidae	Xenochironomus xenolabis	7.0	7	PR	
Diptera	Chironomidae	Xylotopus par	6.0	7	SH	
Diptera	Chironomidae	Zalutschia sp	5.0	7	SC	
Diptera	Chironomidae	Zavrelia sp	5.3	7	CG	
Diptera	Chironomidae	Zavrelimyia sp	9.1	7	PR	
Diptera	Ephydriidae	Brachydeutera sp	9.0	9	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Ephydriidae	Ephydra sp	9.0	9	SH	
Diptera	Ephydriidae	Scatella sp	9.0	9	CG	
Diptera	Ephydriidae	Unidentified Ephydrid	9.0	9	SH	
Diptera	Dolichopodidae	Rhaphium campestre	5.0	5	PR	
Diptera	Dolichopodidae	Unidentified Dolichopodid	5.0	5	PR	
Diptera	Psychodidae	Maruina sp	10.0	10	SC	
Diptera	Psychodidae	Pericoma sp	10.0	10	CG	
Diptera	Psychodidae	Psychoda alternata	9.9	10	CG	
Diptera	Psychodidae	Psychoda sp.	10.0	10	CG	
Diptera	Ptychopteridae	Ptychoptera sp	7.0	7	CG	
Diptera	Simuliidae	Xephia sp	4.0	5	CF	X
Diptera	Simuliidae	Prosimulium magmun	2.6	5	CF	X
Diptera	Simuliidae	Prosimulium mixtum	4.0	5	CF	X
Diptera	Simuliidae	Prosimulium sp	4.0	5	CF	X
Diptera	Simuliidae	Simulium decorum	4.4	5	CF	X
Diptera	Simuliidae	Simulium slossonae	4.4	5	CF	X
Diptera	Simuliidae	Simulium sp	4.0	5	CF	X
Diptera	Simuliidae	Simulium tuberosum	4.4	5	CF	X
Diptera	Simuliidae	Simulium venustum	7.1	5	CF	X
Diptera	Simuliidae	Simulium vittatum	8.7	5	CF	X
Diptera	Simuliidae	Unidentified Simuliid	4.6	5	CF	X
Diptera	Stratiomyidae	Allognosta sp	10.0	10	CG	
Diptera	Stratiomyidae	Caloparyphus sp	10.0	10	CG	
Diptera	Stratiomyidae	Myxosargus sp	10.0	10	CG	
Diptera	Stratiomyidae	Odontomyia sp	10.0	10	CG	
Diptera	Stratiomyidae	Oxycera sp	10.0	10	SC	
Diptera	Stratiomyidae	Stratiomys sp	8.1	10	CG	
Diptera	Thaumaleidae	Thaumalea sp	5.0	5	SC	
Diptera	Tanyderidae	Protoplasm fitchii	4.3	4	PR	
Diptera	Tabanidae	Chlorotabanus sp	9.0	9	PR	
Diptera	Tabanidae	Chrysops furcatus	7.3	9	PR	
Diptera	Tabanidae	Chrysops sp	6.7	9	PR	
Diptera	Tabanidae	Haematopota(?) sp	8.0	9	PR	
Diptera	Tabanidae	Tabanus reinwardtii	9.7	9	PR	
Diptera	Tabanidae	Tabanus sp	9.2	9	PR	
Diptera	Tabanidae	Tabanus/Whitneyomyia/Atylotus sp	9.7	9	PR	
Diptera	Tabanidae	Unidentified tabanid	8.6	9	PR	
Diptera	Ceratopogonidae	Alluaudomyia sp	6.8	7	PR	
Diptera	Ceratopogonidae	Atrichopogon sp	6.5	7	PR	
Diptera	Ceratopogonidae	Bezzia sp	6.9	7	PR	
Diptera	Ceratopogonidae	Bezzia/Johnnanssenomyia/Palpolyia gr	6.9	7	PR	
Diptera	Ceratopogonidae	Bezzia/Palpomyia gr	6.9	7	PR	
Diptera	Ceratopogonidae	Ceratopogon sp	6.8	7	PR	
Diptera	Ceratopogonidae	Culicoides sp	7.7	7	PR	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Diptera	Ceratopogonidae	Dasyhelea sp	6.7	7	CG	
Diptera	Ceratopogonidae	Forcipomyia sp	6.8	7	SC	
Diptera	Ceratopogonidae	Isohelea sp	6.8	7	PR	
Diptera	Ceratopogonidae	Monohelea sp	6.8	7	PR	
Diptera	Ceratopogonidae	Palpomyia sp	6.9	7	PR	
Diptera	Ceratopogonidae	Palpomyia/Sphaeromias gr	6.9	7	PR	
Diptera	Ceratopogonidae	Probezzia sp	6.9	7	PR	
Diptera	Ceratopogonidae	Serromyia sp		7	PR	
Diptera	Ceratopogonidae	Sphaeromias sp	6.9	7	PR	
Diptera	Ceratopogonidae	Stilobezzia sp	6.9	7	PR	
Diptera	Ceratopogonidae	Unidentified Ceratopogonid	6.7	7	PR	
Diptera	Sciaridae	Unidentified Sciarid	5.0	5	SH	
Hydracarina	Unionicolidae	Unionicola sp	5.7		PC	
Hydracarina	Hydrachnidae	Unidentified Hydracarina (mite)	5.5	6	PR	
Hydracarina	Hydrodromidae	Hydrodroma sp	5.7		PR	
Amphipoda	Crangonyctidae	Crangonyx anomalus	8.0	8	SH	
Amphipoda	Crangonyctidae	Crangonyx obliquus richmondensis	8.0	8	SH	
Amphipoda	Crangonyctidae	Crangonyx sp	8.0	8	SH	
Amphipoda	Crangonyctidae	Stygobromus exilis gp	8.0	8	SH	
Amphipoda	Gammaridae	Gammarus fasciatus	9.1	8	SH	
Amphipoda	Gammaridae	Gammarus lacustris	6.9	8	SH	
Amphipoda	Gammaridae	Gammarus sp	9.1	8	CG	
Amphipoda	Talitridae	Hyaella azteca	7.8	6	CG	
Isopoda	Asellidae	Caecidotea sp	9.1	8	CG	
Isopoda	Asellidae	Lirceus fontinalis	7.9	8	CG	
Isopoda	Asellidae	Lirceus sp.	7.9	8	CG	
Decapoda	Cambaridae	Barbicambarus cornutus	4.6	6	CG	
Decapoda	Cambaridae	Cambarus bartonii cavatus	4.6	6	CG	
Decapoda	Cambaridae	Cambarus batchi	4.9	6	CG	
Decapoda	Cambaridae	Cambarus buntingi	4.9	6	CG	
Decapoda	Cambaridae	Cambarus cumberlandensis	4.1	6	CG	
Decapoda	Cambaridae	Cambarus diogenes	7.5	6	CG	
Decapoda	Cambaridae	Cambarus distans	3.9	6	CG	
Decapoda	Cambaridae	Cambarus dubius		6	CG	
Decapoda	Cambaridae	Cambarus friaufi	4.9	6	CG	
Decapoda	Cambaridae	Cambarus graysoni	4.9	6	CG	
Decapoda	Cambaridae	Cambarus ortmanni	6.2	6	CG	
Decapoda	Cambaridae	Cambarus parvovulus	3.2	6	CG	
Decapoda	Cambaridae	Cambarus robustus	4.9	6	CG	
Decapoda	Cambaridae	Cambarus rusticiformis	4.0	6	CG	
Decapoda	Cambaridae	Cambarus sciotensis	6.4	6	CG	
Decapoda	Cambaridae	Cambarus sp	4.9	6	CG	
Decapoda	Cambaridae	Cambarus sphenoides	4.9	6	CG	

**Appendix D-1. Macroinvertebrate Master Taxa List with Tolerance Value (TV), Family Tolerance Value (FamTV), Functional Feeding Group (FFG) and Clinger designations (X).**

Order	Family	Final ID	TV	FamTV	FFG	Clinger
Decapoda	Cambaridae	Cambarus striatus	4.9	6	CG	
Decapoda	Cambaridae	Cambarus tenebrosus	6.5	6	CG	
Decapoda	Cambaridae	Cambarus veteranus	4.9	6	CG	
Decapoda	Cambaridae	Fallicambarus fodiens	7.0	6	CG	
Decapoda	Cambaridae	Orconectes australis australis	5.5	6	CG	
Decapoda	Cambaridae	Orconectes australis packardi	5.5	6	CG	
Decapoda	Cambaridae	Orconectes barrenensis	5.5	6	CG	
Decapoda	Cambaridae	Orconectes bisectus	5.5	6	CG	
Decapoda	Cambaridae	Orconectes burri	4.9	6	CG	
Decapoda	Cambaridae	Orconectes compressus	5.5	6	CG	
Decapoda	Cambaridae	Orconectes cristavarius	5.5	6	SC	
Decapoda	Cambaridae	Orconectes durelli	5.5	6	CG	
Decapoda	Cambaridae	Orconectes immunis	5.5	6	CG	
Decapoda	Cambaridae	Orconectes jeffersoni	5.5	6	CG	
Decapoda	Cambaridae	Orconectes juvenilis	6.0	6	CG	
Decapoda	Cambaridae	Orconectes kentuckiensis	5.5	6	CG	
Decapoda	Cambaridae	Orconectes pellucidus	5.5	6	CG	
Decapoda	Cambaridae	Orconectes placidus	5.5	6	CG	
Decapoda	Cambaridae	Orconectes propinquus	5.5	6	CG	
Decapoda	Cambaridae	Orconectes putnami	5.5	6	CG	
Decapoda	Cambaridae	Orconectes rafinesquei	5.5	6	CG	
Decapoda	Cambaridae	Orconectes rusticus	5.9	6	CG	
Decapoda	Cambaridae	Orconectes sanborni sanborni	6.5	6	CG	
Decapoda	Cambaridae	Orconectes sp	5.5	6	CG	
Decapoda	Cambaridae	Orconectes sp(nr putmani)	5.5	6	CG	
Decapoda	Cambaridae	Orconectes tricuspis	5.5	6	CG	
Decapoda	Cambaridae	Procambarus acutus acutus	7.0	6	CG	
Decapoda	Cambaridae	Procambarus clarkii	7.0	6	CG	
Decapoda	Cambaridae	Procambarus sp	7.0	6	CG	
Decapoda	Palaemonidae	Macrobrachium ohione	5.0	5	CG	
Decapoda	Palaemonidae	Macrobrachium sp	5.0	5	CG	
Decapoda	Palaemonidae	Palaemonetes ganteri	5.0	5	CG	
Decapoda	Palaemonidae	Palaemonetes kadiakensis	5.0	6	CG	
Decapoda	Palaemonidae	Palaemonetes paludosus	5.0	6	CG	
Decapoda	Palaemonidae	Palaemonetes sp	7.1	6	CG	
Hoplonemertea	Prostomidae	Prostoma sp	8.0	8	PR	X

**Appendix D-2: KDOW Macroinvertebrate Bench Sheet**

**Station ID:**

**Stream:**

**ID By:**

**Location:**

**County:**

**Topo:**

**Collectors:**

**Method:**

**Date Collected:**

<b>Family</b>	<b>Final ID</b>	<b>Quan</b>	<b>Qual</b>	<b>Total</b>	<b>Comments</b>

**Macroinvertebrate Bench Sheet, Page 2**

**Station ID:**

**Stream:**

**ID By:**

**Date:**





**APPENDIX E-1 KY DOW FISH COLLECTION DATA SHEET**

**APPENDIX E-2 THREATENED/ENDANGERED SPECIES REPORT  
FORM**

**APPENDIX E-3 CURRENT FISH MASTER TAXA LIST**

**Appendix E -1. KY DOW Fish Collection Data Sheet**

**Site No.** \_\_\_\_\_ **Date** \_\_\_\_\_ **County** \_\_\_\_\_  
**Stream** \_\_\_\_\_ **Location** \_\_\_\_\_  
**Town nearby** \_\_\_\_\_ **Quad** \_\_\_\_\_  
**Lat/Long.** \_\_\_\_\_ **Basin** \_\_\_\_\_ **RMI** \_\_\_\_\_  
**Collector(s)** \_\_\_\_\_ **Catchment area** \_\_\_\_\_ **Order** \_\_\_\_\_  
**Collection Method** \_\_\_\_\_ **ID by** \_\_\_\_\_

Ichthyomyzon bdellium	_____	_____	Nocomis effusus	_____	_____
Ichthyomyzon castaneus (SC)	_____	_____	Nocomis micropogon	_____	_____
Ichthyomyzon fossor (ST)	_____	_____	Notemigonus crysoleucas	_____	_____
Ichthyomyzon gagei (SH)	_____	_____	Notropis sp.	_____	_____
Ichthyomyzon greeleyi (ST)	_____	_____	Notropis albizonatus (SE FE)	_____	_____
Ichthyomyzon unicuspis	_____	_____	Notropis ariommus	_____	_____
Lampetra aepyptera	_____	_____	Notropis atherinoides	_____	_____
Lampetra appendix (ST)	_____	_____	Notropis blennioides	_____	_____
Acipenser fulvescens (SE)	_____	_____	Notropis boops	_____	_____
Scaphirhynchus albus (SE FE)	_____	_____	Notropis buechanani	_____	_____
Scaphirhynchus platyrhynchus	_____	_____	Notropis hudsonius (SC)	_____	_____
Polyodon spathula	_____	_____	Notropis leuciodus	_____	_____
Atractosteus spatula (SE)	_____	_____	Notropis ludibundus	_____	_____
Lepisosteus oculatus	_____	_____	Notropis maculatus (ST)	_____	_____
Lepisosteus osseus	_____	_____	Notropis nubilus	_____	_____
Lepisosteus platostomus	_____	_____	Notropis photogenis	_____	_____
Amia calva	_____	_____	Notropis rubellus	_____	_____
Hiodon alosoides	_____	_____	Notropis shumardi	_____	_____
Hiodon tergisus	_____	_____	Notropis telescopus	_____	_____
Anguilla rostrata	_____	_____	Notropis volucellus	_____	_____
Alosa alabamae (SE)	_____	_____	Notropis wickliffi	_____	_____
Alosa chrysochloris	_____	_____	Notropis sp. "sawfin" (SE)	_____	_____
Alosa pseudoharengus	_____	_____	Opsopoeodus emiliae	_____	_____
Dorosoma cepedianum	_____	_____	Phenacobius mirabilis	_____	_____
Dorosoma petenense	_____	_____	Phenacobius uranops (SC)	_____	_____
Campostoma anomalum	_____	_____	Phoxinus cumberlandensis (ST FT)	_____	_____
Campostoma oligolepis	_____	_____	Phoxinus erythrogaster	_____	_____
Carassius auratus	_____	_____	Pimephales notatus	_____	_____
Clinostomus elongatus	_____	_____	Pimephales promelas	_____	_____
Clinostomus funduloides (SC)	_____	_____	Pimephales vigilax	_____	_____
Ctenopharyngodon idella	_____	_____	Platygobio gracilis (SC)	_____	_____
Cyprinella camura (SE)	_____	_____	Rhinichthys atratulus	_____	_____
Cyprinella galactura	_____	_____	Rhinichthys cataractae (SE)	_____	_____
Cyprinella lutrensis	_____	_____	Semotilus atromaculatus	_____	_____
Cyprinella spiloptera	_____	_____	Carpodius carpio	_____	_____
Cyprinella venusta (SC)	_____	_____	Carpodius cyprinus	_____	_____
Cyprinella whipplei	_____	_____	Carpodius velifer	_____	_____
Cyprinus carpio	_____	_____	Catostomus commersoni	_____	_____
Ericymba buccata	_____	_____	Cycleptus elongatus	_____	_____
Erimystax dissimilis	_____	_____	Erimyzon oblongus	_____	_____
Erimystax insignis (SE)	_____	_____	Erimyzon sucetta (ST)	_____	_____
Erimystax x-punctatus	_____	_____	Hypentelium nigricans	_____	_____
Hemitremia flammea	_____	_____	Ictiobus bubalus	_____	_____
Hybognathus hayi (SE)	_____	_____	Ictiobus cyprinellus	_____	_____
Hybognathus nuchalis	_____	_____	Ictiobus niger (SC)	_____	_____
Hybognathus placitus (SC)	_____	_____	Lagochila lacera (SH)	_____	_____
Hybopsis amblops	_____	_____	Minytrema melanops	_____	_____
Hybopsis amnis (SH)	_____	_____	Moxostoma sp.	_____	_____
Hypophthalmichthys molitrix	_____	_____	Moxostoma anisurum	_____	_____
Hypophthalmichthys nobilis	_____	_____	Moxostoma carinatum	_____	_____
Luxilus chrysocephalus	_____	_____	Moxostoma duquesnei	_____	_____
Lythrurus fasciolaris	_____	_____	Moxostoma erythrurum	_____	_____
Lythrurus fumeus	_____	_____	Moxostoma macrolepidotum	_____	_____
Lythrurus umbratilis	_____	_____	Moxostoma poecilurum (SE)	_____	_____
Macrhybopsis aestivalis	_____	_____	Thoburnia atripinnis (SC)	_____	_____
Macrhybopsis gelida (SH C1)	_____	_____	Ameiurus catus	_____	_____
Macrhybopsis meeki (SH C1)	_____	_____	Ameiurus melas	_____	_____
Macrhybopsis storeriana	_____	_____	Ameiurus natalis	_____	_____
Nocomis biguttatus (SC)	_____	_____	Ameiurus nebulosus	_____	_____

Appendix E-1 KY DOW Fish Collection Data Sheet

Ictalurus furcatus			Etheostoma baileyi		
Ictalurus punctatus			Etheostoma barbouri		
Noturus elegans			Etheostoma barrenense		
Noturus eleutherus			Etheostoma bellum		
Noturus exilis (SE)			Etheostoma bison		
Noturus flavus			Etheostoma blennioides		
Noturus gyrinus			Etheostoma caeruleum		
Noturus hildebrandi (SE)			Etheostoma camurum		
Noturus miurus			Etheostoma chienense (SE FE)		
Noturus nocturnus			Etheostoma chlorosomum		
Noturus phaeus (SE)			Etheostoma cinereum (SC)		
Noturus stigmosus (SC)			Etheostoma crossopterum		
Pylodictus olivaris			Etheostoma flabellare		
Esox americanus			Etheostoma flavum		
Esox lucius			Etheostoma fusiforme (SE)		
Esox masquinongy			Etheostoma gracile		
Esox niger (SC)			Etheostoma histrio		
Umbra limi (ST)			Etheostoma kennicotti		
Osmerus mordax			Etheostoma kantuckeense		
Oncorhynchus kisutch			Etheostoma lynceum (SE)		
Oncorhynchus mykiss			Etheostoma maculatum (ST)		
Salmo trutta			Etheostoma microlepidum (SE)		
Salvelinus fontinalis			Etheostoma microperca		
Salvelinus namaycush			Etheostoma n. nigrum		
Percopsis omiscomaycus (SC)			Etheostoma n. susanae (SE)		
Aphredoderus sayanus			Etheostoma obeyense		
Amblyopsis spelaea (SC)			Etheostoma oophylax		
Forbesichthys agassizi			Etheostoma parvipinne (SE)		
Typhlichthys subterraneus (SC)			Etheostoma percnum (SE FE)		
Lota lota (SC)			Etheostoma proeliare (ST)		
Fundulus catenatus			Etheostoma pyrrhogaster (SE)		
Fundulus chrysotus (SE)			Etheostoma rafinesquei		
Fundulus dispar (SE)			Etheostoma rufilineatum		
Fundulus notatus			Etheostoma s. sagitta		
Fundulus olivaceus			Etheostoma s. spilatum		
Gambusia affinis			Etheostoma sanguifluum		
Labidesthes sicculus			Etheostoma simoterum		
Menidia beryllina (ST)			Etheostoma smithi		
Culaea inconstans			Etheostoma spectabile		
Cottus bairdi			Etheostoma cf. spectabile "Cumberland"		
Cottus carolinae			Etheostoma cf. spectabile "Headwater"		
Morone chrysops			Etheostoma cf. spectabile "Sheltoewe"		
Morone mississippiensis			Etheostoma squamiceps		
Morone saxatilis			Etheostoma stigmaeum		
Ambloplites rupestris			Etheostoma cf. stigmaeum "Bluegrass"		
Centrarchus macropterus			Etheostoma cf. stigmaeum "Longhunt"		
Lepomis sp.			Etheostoma swaini (SE)		
Lepomis auritus			Etheostoma tippecanoe		
Lepomis cyanellus			Etheostoma tecumsehi (ST)		
Lepomis gibbosus			Etheostoma variatum		
Lepomis gulosus			Etheostoma virgatum		
Lepomis humilis			Etheostoma zonale		
Lepomis macrochirus			Etheostoma zonistium		
Lepomis marginatus (SE)			Perca flavescens		
Lepomis megalotis			Percina burtoni		
Lepomis microlophus			Percina caprodes		
Lepomis miniatus (ST)			Percina copelandi		
Lepomis symmetricus			Percina evides		
Micropterus coosae			Percina macrocephala (ST)		
Micropterus dolomieu			Percina maculata		
Micropterus punctulatus			Percina oxyrhyncha		
Micropterus salmoides			Percina phoxocephala		
Pomoxis annularis			Percina sciera		
Pomoxis nigromaculatus			Percina shumardi		
Elassoma zonatum			Percina squamata (SE)		
Ammocrypta clara (SE)			Percina stictogaster		
Ammocrypta pellucida (SC)			Percina vigil		
Ammocrypta vivax (SH)			Stizostedion canadense		
Crystallaria asprella (SH)			Stizostedion vitreum		
Etheostoma asprigene			Aplodinotus grunniens		

**APPENDIX E-2**

**THREATENED/ENDANGERED SPECIES REPORT FORM**  
**Kentucky Division of Water**  
**Water Quality Branch**

Species collected: \_\_\_\_\_ Date collected: \_\_/\_\_/\_\_  
# collected: \_\_\_\_\_ Released? Yes No: If no, explain \_\_\_\_\_  
Stream: \_\_\_\_\_ Site #: \_\_\_\_\_ Basin: \_\_\_\_\_  
County: \_\_\_\_\_ Location: \_\_\_\_\_  
Quad: \_\_\_\_\_ Collected/Identified by: \_\_\_\_\_  
Collection method: \_\_\_\_\_  
Comments: \_\_\_\_\_

**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Petromyzontidae	Ichthyomyzon bdellium		X						
Petromyzontidae	Ichthyomyzon castaneus		X						
Petromyzontidae	Ichthyomyzon fossor		X						
Petromyzontidae	Ichthyomyzon gagei		X						
Petromyzontidae	Ichthyomyzon greeleyi		X						
Petromyzontidae	Ichthyomyzon unicuspis		X						
Petromyzontidae	Lampetra aepyptera		X						
Petromyzontidae	Lampetra appendix		X						
Petromyzontidae	Lamprey ammocoete		X						
Petromyzontidae	Lamprey spp.		X						
Acipenseridae	Acipenser fulvescens		X					X	
Acipenseridae	Scaphirhynchus albus		X					X	
Acipenseridae	Scaphirhynchus platyrhynchus		X					X	
Polyodontidae	Polyodon spathula		X			X		X	
Lepisosteidae	Atractosteus spatula	TC	X						X
Lepisosteidae	Lepisosteus oculatus	TC	X						X
Lepisosteidae	Lepisosteus osseus	TC	X						X
Lepisosteidae	Lepisosteus platostomus	TC	X						X
Lepisosteidae	Lepisosteus spp.	TC	X						X
Amiidae	Amia calva	TC	X						X
Hiodontidae	Hiodon alosoides		X		X				
Hiodontidae	Hiodon tergisus		X		X				
Anguillidae	Anguilla rostrata		X						
Clupeidae	Alosa alabamae		X					X	
Clupeidae	Alosa chrysochloris		X						
Clupeidae	Alosa pseudoharengus								
Clupeidae	Dorosoma cepedianum		X	X					
Clupeidae	Dorosoma petenense		X	X					
Cyprinidae	Campostoma anomalum	MIN	X						
Cyprinidae	Campostoma oligolepis	MIN	X						
Cyprinidae	Carassius auratus	MIN	X	X			X		
Cyprinidae	Clinostomus elongatus	MIN	X		X	X		X	X
Cyprinidae	Clinostomus funduloides	MIN	X		X	X		X	X
Cyprinidae	Ctenopharyngodon idella	MIN							
Cyprinidae	Cyprinella camura	MIN	X		X	X			X
Cyprinidae	Cyprinella galactura	MIN	X		X	X			X
Cyprinidae	Cyprinella lutrensis	MIN	X	X					
Cyprinidae	Cyprinella spiloptera	MIN	X		X				X
Cyprinidae	Cyprinella venusta	MIN	X		X				X
Cyprinidae	Cyprinella whipplei	MIN	X		X				X
Cyprinidae	Cyprinus carpio	MIN		X			X		
Cyprinidae	Ericymba buccata	MIN	X	X					
Cyprinidae	Erimystax dissimilis	MIN	X		X	X		X	X
Cyprinidae	Erimystax insignis	MIN	X		X			X	X

**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Cyprinidae	Erimystax x-punctatus	MIN	X		X	X		X	X
Cyprinidae	Hemitremia flammea	MIN	X		X				X
Cyprinidae	Hybognathus hayi	MIN	X						
Cyprinidae	Hybognathus nuchalis	MIN	X						
Cyprinidae	Hybognathus placitus	MIN							
Cyprinidae	Hybopsis amblops	MIN	X		X	X		X	X
Cyprinidae	Hybopsis amnis	MIN	X		X				X
Cyprinidae	Hypophthalmichthys molitrix	MIN							
Cyprinidae	Luxilus chrysocephalus	MIN	X				X	X	
Cyprinidae	Lythrurus fasciolaris	MIN	X		X				X
Cyprinidae	Lythrurus fumeus	MIN	X				X		
Cyprinidae	Lythrurus umbratilis	MIN	X		X			X	X
Cyprinidae	Macrhybopsis aestivalis	MIN	X		X	X			X
Cyprinidae	Macrhybopsis gelida	MIN	X		X	X			X
Cyprinidae	Macrhybopsis meeki	MIN	X		X	X			X
Cyprinidae	Macrhybopsis storeriana	MIN	X		X				X
Cyprinidae	Nocomis biguttatus	MIN	X	X		X		X	X
Cyprinidae	Nocomis effusus	MIN	X		X	X		X	X
Cyprinidae	Nocomis micropogon	MIN	X		X	X		X	X
Cyprinidae	Notemigonus crysoleucas	MIN	X	X			X		
Cyprinidae	Notropis albizonatus	MIN	X		X	X		X	X
Cyprinidae	Notropis ariommus	MIN	X		X	X		X	X
Cyprinidae	Notropis atherinoides	MIN	X	X				X	
Cyprinidae	Notropis blennioides	MIN	X		X			X	X
Cyprinidae	Notropis boops	MIN	X		X			X	X
Cyprinidae	Notropis buchmanii	MIN	X		X				X
Cyprinidae	Notropis hudsonius	MIN	X		X				
Cyprinidae	Notropis leuciodus	MIN	X		X	X			X
Cyprinidae	Notropis ludibundus	MIN	X	X					
Cyprinidae	Notropis maculatus	MIN	X						X
Cyprinidae	Notropis nubilus	MIN	X		X				
Cyprinidae	Notropis photogenis	MIN	X		X	X		X	X
Cyprinidae	Notropis rubellus	MIN	X		X	X		X	X
Cyprinidae	Notropis shumardi	MIN	X						
Cyprinidae	Notropis sp.	MIN	X						
Cyprinidae	Notropis spp. (sawfin shiner)	MIN	X		X	X		X	X
Cyprinidae	Notropis telescopus	MIN	X		X	X			X
Cyprinidae	Notropis volucellus	MIN	X	X					
Cyprinidae	Opsopoeodus emiliae	MIN	X	X		X			
Cyprinidae	Phenacobius mirabilis	MIN	X		X			X	X
Cyprinidae	Phenacobius uranops	MIN	X		X	X		X	X
Cyprinidae	Phoxinus cumberlandensis	MIN	X	X		X		X	
Cyprinidae	Phoxinus erythrogaster	MIN	X	X		X		X	
Cyprinidae	Pimephales notatus	MIN	X	X			X		
Cyprinidae	Pimephales promelas	MIN	X	X			X		

**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Cyprinidae	Pimephales spp.	MIN	X	X			X		
Cyprinidae	Pimephales vigilax	MIN	X	X					
Cyprinidae	Platygobio gracilis	MIN	X				X		
Cyprinidae	Rhinichthys atratulus	MIN	X				X	X	
Cyprinidae	Rhinichthys cataractae	MIN	X		X			X	X
Cyprinidae	Semotilus atromaculatus	MIN	X	X			X		
Catostomidae	Carpiodes carpio	SUC	X	X					
Catostomidae	Carpiodes cyprinus	SUC	X	X					
Catostomidae	Carpiodes velifer	SUC	X	X					
Catostomidae	Catostomid fry	SUC	X				X	X	
Catostomidae	Catostomus commersoni	SUC	X				X	X	
Catostomidae	Catostomus sp.	SUC	X				X	X	
Catostomidae	Cycleptus elongatus	SUC	X		X	X		X	X
Catostomidae	Erimyzon oblongus	SUC	X		X				X
Catostomidae	Erimyzon sucetta	SUC	X	X					X
Catostomidae	Hypentelium nigricans	SUC	X		X			X	X
Catostomidae	Ictiobus bubalus	SUC	X	X					
Catostomidae	Ictiobus cyprinellus	SUC	X	X					
Catostomidae	Ictiobus niger	SUC	X	X					
Catostomidae	Lagochila lacera	SUC	X					X	X
Catostomidae	Minytrema melanops	SUC	X		X			X	X
Catostomidae	Moxostoma anisurum	SUC	X		X	X		X	X
Catostomidae	Moxostoma carinatum	SUC	X		X	X		X	X
Catostomidae	Moxostoma duquesnei	SUC	X		X	X		X	X
Catostomidae	Moxostoma erythrum	SUC	X		X			X	X
Catostomidae	Moxostoma macrolepidotum breviceps	SUC	X		X			X	X
Catostomidae	Moxostoma poecilurum	SUC	X		X			X	X
Catostomidae	Moxostoma sp.	SUC	X		X			X	X
Catostomidae	Moxostoma valenciennesi	SUC	X		X			X	X
Catostomidae	Thoburnia atripinne	SUC	X		X	X		X	X
Ictaluridae	Ameiurus catus			X					
Ictaluridae	Ameiurus melas		X	X			X		
Ictaluridae	Ameiurus natalis		X	X			X		
Ictaluridae	Ameiurus nebulosus		X	X			X		
Ictaluridae	Ameiurus spp.		X	X					
Ictaluridae	Ictalurus furcatus		X	X					
Ictaluridae	Ictalurus punctatus		X	X					
Ictaluridae	Noturus elegans	MAD	X		X	X			
Ictaluridae	Noturus eleutherus	MAD	X		X	X			
Ictaluridae	Noturus exilis	MAD	X		X	X			
Ictaluridae	Noturus flavus	MAD	X		X	X			
Ictaluridae	Noturus gyrinus	MAD	X		X	X			
Ictaluridae	Noturus hildebrandi	MAD	X		X	X			
Ictaluridae	Noturus miurus	MAD	X		X	X			
Ictaluridae	Noturus nocturnus	MAD	X		X	X			



**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Ictaluridae	Noturus phaeus	MAD	X		X	X			
Ictaluridae	Noturus stigmosus	MAD	X		X	X			
Ictaluridae	Pylodictus olivaris	TC	X						X
Esocidae	Esox americanus vermiculatus	TC	X						X
Esocidae	Esox lucius								
Esocidae	Esox masquinongy	TC	X						X
Esocidae	Esox niger	TC	X						X
Umbridae	Umbra limi		X				X		
Osmeridae	Osmerus mordax								
Salmonidae	Oncorhynchus kisutch								
Salmonidae	Oncorhynchus mykiss					X			
Salmonidae	Salmo trutta								
Salmonidae	Salvelinus fontinalis								
Salmonidae	Salvelinus namaycush								
Percopsidae	Percopsis omiscomaycus		X		X	X			
Aphredoderidae	Aphredoderus sayanus		X		X				
Amblyopsidae	Amblyopsis spelaea		X						
Amblyopsidae	Forbesichthys agassizi		X						
Amblyopsidae	Typhlichthys subterraneus		X						
Gadidae	Lota lota		X					X	
Fundulidae	Fundulus catenatus		X		X			X	
Fundulidae	Fundulus chrysotus		X		X				
Fundulidae	Fundulus dispar		X		X				
Fundulidae	Fundulus notatus		X		X				
Fundulidae	Fundulus olivaceus		X		X				
Poeciliidae	Gambusia affinis		X				X		
Atherinidae	Labidesthes sicculus		X		X				
Atherinidae	Menidia beryllina		X		X				
Gasterosteidae	Culaea inconstans								
Cottidae	Cottus bairdi	COT	X		X	X			
Cottidae	Cottus carolinae	COT	X		X	X			
Moronidae	Morone chrysops		X						
Moronidae	Morone chrysops x M. saxatilis								
Moronidae	Morone mississippiensis		X						
Moronidae	Morone saxatilis								
Centrarchidae	Ambloplites rupestris	SUN	X			X			X
Centrarchidae	Centrarchus macropterus	SUN	X						X
Centrarchidae	Elassoma zonatum	SUN	X		X				X
Centrarchidae	Lepomis auritus	SUN					X		
Centrarchidae	Lepomis cyanellus	SUN	X				X		
Centrarchidae	Lepomis gibbosus	SUN	X						X
Centrarchidae	Lepomis gulosus	SUN	X						X
Centrarchidae	Lepomis humilis	SUN	X						X
Centrarchidae	Lepomis macrochirus	SUN	X				X		
Centrarchidae	Lepomis macrochirus X L. cyanellus						X		

**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Centrarchidae	Lepomis macrochirus X L. megalotis	SUN							
Centrarchidae	Lepomis marginatus	SUN	X						X
Centrarchidae	Lepomis megalotis	SUN	X						X
Centrarchidae	Lepomis microlophus	SUN	X						X
Centrarchidae	Lepomis miniatus	SUN	X			X			X
Centrarchidae	Lepomis sp.	SUN	X						
Centrarchidae	Lepomis symmetricus	SUN	X						X
Centrarchidae	Micropterus coosae								
Centrarchidae	Micropterus dolomieu	TC	X						X
Centrarchidae	Micropterus punctulatus	TC	X						X
Centrarchidae	Micropterus salmoides		X						
Centrarchidae	Micropterus spp.		X						
Centrarchidae	Pomoxis annularis	TC	X						X
Centrarchidae	Pomoxis nigromaculatus	TC	X						X
Percidae	Ammocrypta clara	DAR	X		X	X		X	
Percidae	Ammocrypta pellucida	DAR	X		X	X		X	
Percidae	Ammocrypta vivax	DAR	X		X	X		X	
Percidae	Crystallaria asprella	DAR	X		X	X		X	
Percidae	Etheostoma asprigene	DAR	X		X				
Percidae	Etheostoma baileyi	DAR	X		X	X		X	
Percidae	Etheostoma barbouri	DAR	X		X	X			
Percidae	Etheostoma barrenense	DAR	X		X	X			
Percidae	Etheostoma bellum	DAR	X		X	X			
Percidae	Etheostoma bison	DAR	X		X			X	
Percidae	Etheostoma blennioides	DAR	X		X			X	
Percidae	Etheostoma caeruleum	DAR	X		X			X	
Percidae	Etheostoma camurum	DAR	X		X	X		X	
Percidae	Etheostoma chienense	DAR	X		X	X			
Percidae	Etheostoma chlorosomum	DAR	X		X				
Percidae	Etheostoma cinereum	DAR	X		X	X			
Percidae	Etheostoma crossopterum	DAR	X		X	X			
Percidae	Etheostoma flabellare	DAR	X		X				
Percidae	Etheostoma flavum	DAR	X		X	X		X	
Percidae	Etheostoma fusiforme	DAR	X		X	X			
Percidae	Etheostoma gracile	DAR	X		X				
Percidae	Etheostoma histrio	DAR	X		X	X		X	
Percidae	Etheostoma kantuckeense	DAR	X		X			X	
Percidae	Etheostoma kennicotti	DAR	X		X			X	
Percidae	Etheostoma lynceum	DAR	X		X			X	
Percidae	Etheostoma maculatum	DAR	X		X	X		X	
Percidae	Etheostoma microlepidum	DAR	X		X	X			
Percidae	Etheostoma microperca	DAR	X		X	X			
Percidae	Etheostoma nigrum	DAR	X		X				
Percidae	Etheostoma obeyense	DAR	X		X	X			
Percidae	Etheostoma oophylax	DAR	X		X	X			

**Appendix E-3 Master Species List**

Family	Species	Fish_Type	Native	OMN	INS	INT	TOL	SL	WC
Percidae	Etheostoma parvipinne	DAR	X		X	X			
Percidae	Etheostoma percnum	DAR	X		X	X			
Percidae	Etheostoma proeliare	DAR	X		X	X			
Percidae	Etheostoma pyrrhogaster	DAR	X		X	X			
Percidae	Etheostoma rafinesquei	DAR	X		X	X			
Percidae	Etheostoma rufilineatum	DAR	X		X	X			
Percidae	Etheostoma sagitta	DAR	X		X	X			
Percidae	Etheostoma sanguifluum	DAR	X		X	X			
Percidae	Etheostoma simoterum	DAR	X		X	X		X	
Percidae	Etheostoma smithi	DAR	X		X	X		X	
Percidae	Etheostoma sp.	DAR	X		X	X		X	
Percidae	Etheostoma spectabile	DAR	X		X			X	
Percidae	Etheostoma squamiceps	DAR	X		X	X			
Percidae	Etheostoma stigmaeum	DAR	X		X	X		X	
Percidae	Etheostoma swaini	DAR	X		X	X		X	
Percidae	Etheostoma tecumsehi	DAR	X		X			X	
Percidae	Etheostoma tippecanoe	DAR	X		X	X		X	
Percidae	Etheostoma variatum	DAR	X		X	X		X	
Percidae	Etheostoma virgatum	DAR	X		X	X			
Percidae	Etheostoma zonale	DAR	X		X			X	
Percidae	Etheostoma zonistium	DAR	X		X	X		X	
Percidae	Perca flavescens		X						
Percidae	Percina burtoni	DAR	X		X	X		X	
Percidae	Percina caprodes	DAR	X		X			X	
Percidae	Percina copelandi	DAR	X		X	X		X	
Percidae	Percina evides	DAR	X		X	X		X	
Percidae	Percina macrocephala	DAR	X		X	X		X	
Percidae	Percina maculata	DAR	X		X			X	
Percidae	Percina oxyrhynchus	DAR	X		X	X		X	
Percidae	Percina phoxocephala	DAR	X		X	X		X	
Percidae	Percina sciera	DAR	X		X	X		X	
Percidae	Percina shumardi	DAR	X		X			X	
Percidae	Percina squamata	DAR	X		X	X		X	
Percidae	Percina stictogaster	DAR	X		X	X		X	
Percidae	Percina vigil	DAR	X		X	X		X	
Percidae	Stizostedion canadense	TC	X						X
Percidae	Stizostedion vitreum	TC	X						X
Sciaenidae	Aplodinotus grunniens		X						

COT= sculpin, DAR= darter, MAD= madtom, MIN= minnow, SL= simple lithophilic spawner, SUC= sucker, SUN= sunfish, TC= top carnivore, OMV=omnivore, INS=insectivore, INT=intolerant, TOL=tolerant, WC=Water Column

## **APPENDIX F-1 FISH COLLECTED FOR FISH TISSUE ANALYSIS**

**Appendix F-1  
FISH COLLECTED FOR FISH TISSUE ANALYSIS**

Site No. \_\_\_\_\_ Date \_\_\_\_\_ County \_\_\_\_\_  
 Stream \_\_\_\_\_ Location \_\_\_\_\_  
 Town nearby \_\_\_\_\_ Quad \_\_\_\_\_  
 Lat./Long. \_\_\_\_\_ Basin \_\_\_\_\_ RMI \_\_\_\_\_  
 Collection Method \_\_\_\_\_ Catchment area \_\_\_\_\_ Order \_\_\_\_\_  
 Collector(s) \_\_\_\_\_

Name of fish	Length	Weight	Fillet type: (RF LF WB)