



Protocol for Monitoring Aquatic Invertebrates of Small Streams in the Heartland Inventory & Monitoring Network

Natural Resource Report NPS/HTLN/NRR—2008/042

A Heartland Network Monitoring Protocol



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Heartland
Network

Natural Resource Monitoring

ON THE COVER

Herbert Hoover birthplace cottage at Herbert Hoover NHS, prescribed fire at Tallgrass Prairie NP, aquatic invertebrate monitoring at George Washington Carver NM, the Mississippi River at Effigy Mounds NM.

Protocol for Monitoring Aquatic Invertebrates of Small Streams in the Heartland Inventory & Monitoring Network

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I. Background and Objectives

Issues Being Addressed and Rationale for Monitoring Benthic Invertebrates

The condition of streams is a direct reflection of the extent of development and other human uses in the watershed. Non-point source pollution--including urban and agricultural run off, treated sewage, and changes in hydrology--threaten water quality and quantity of streams. Because of these threats, streams are among the most vulnerable natural resources in the United States (USEPA, 2006). Furthermore, streams in the Midwestern U.S. are among the most impacted because the long history of land use in this region, including extensive agriculture and development, has influenced virtually all waterbodies in this region (Hall *et al.*, 2003; Dodds *et al.*, 2004; USEPA, 2006).

The Heartland Inventory and Monitoring Program (HTLN) is a major component of the National Park Service's (NPS) strategy to improve park management through greater reliance on scientific information. The purpose of this program is to design and implement long-term ecological monitoring and provide information for park managers to evaluate the integrity of park ecosystems, including streams, and better understand ecosystem processes. Concerns over declining surface water quality have led to the development of various monitoring approaches to assess stream water quality in those systems. Benthic aquatic invertebrates are a diverse group of relatively long-lived sedentary species that often react strongly and predictably to human disturbance, making them a cost-effective and conservative tool to monitor stream water quality. Aquatic invertebrates are an important biological tool for understanding and detecting changes in stream ecosystem integrity, and they can be used to reflect cumulative impacts that cannot otherwise be detected through traditional water quality monitoring. The broad diversity of invertebrate species occurring in aquatic systems similarly demonstrates a broad range of responses to different environmental and anthropogenic stressors. Benthic invertebrates are relatively easy to collect, and they can be analyzed at many different levels of precision. They are sensitive to a wide variety of impacts that occur in the region, such as changes in chemical constituents, hydrological alterations, sedimentation and bank erosion, and land use and other changes in the watershed (Hall *et al.*, 2003). Furthermore, changes in the diversity and community structure of benthic invertebrates are relatively simple to communicate to resource managers, administrators, and park visitors because the loss of biological communities is of interest and concern to these groups. Benthic community structure can be quantified to reflect stream integrity in several ways, including the absence of pollution sensitive taxa, dominance by a particular taxon combined with low overall taxa richness, or appreciable shifts in community composition relative to reference conditions (Plafkin *et al.*, 1989; Lazorchak *et al.*, 1998; Barbour *et al.*, 1999; USEPA, 2006).

In order to assess the natural and anthropogenic processes influencing invertebrate communities, this protocol has been designed to incorporate the spatial relationship of invertebrates with their associated habitat. Local variables, such as conductivity, water

temperature, pH, dissolved oxygen, turbidity, current velocity, substrate size, and other habitat variables will be measured.

History of Invertebrate Monitoring in Midwestern NPS Parks

In the late 1980's, the National Park Service (NPS) began an intensive program to monitor water quality and invertebrate community structure in prairie streams at Agate Fossil Beds National Monument (AGFO), Homestead National Monument of America (HOME), Pipestone National Monument (PIPE), and Wilson's Creek National Battlefield (WICR) (Harris *et al.*, 1991). The initial strategy for sampling streams in these parks was presented in a "manual" by Boyle *et al.* (1990). Data collected in 1988-1989 at WICR and 1989 from PIPE, AGFO, and HOME, in addition to data collected from Herbert Hoover National Historic Site (HEHO) and George Washington Carver National Monument (GWCA), are summarized in Harris *et al.* (1991) and also presented in Appendix A. A preliminary protocol was suggested by Peterson (1996), in which data dating back to 1988 and collected under the guidance described in Boyle *et al.* (1990) were analyzed. An official invertebrate biomonitoring protocol, drawing heavily on Peterson's (1996) results, was published in 1999 (Peterson *et al.*, 1999). Although the first sampling associated with this protocol was conducted in 1988, Peterson (1996) considered 1989 as the "baseline" year, because it was the first year with "reasonably thorough sampling" (Peterson *et al.*, 1999). The Peterson protocol was implemented in 4 parks (AGFO, HOME, PIPE, and WICR), and no further monitoring was conducted at HEHO. Similarly, Peterson (1997) monitored invertebrates at GWCA in 1996 using methods similar to those described in the 1999 protocol (Appendix A), but no further monitoring was continued at this park until 2005.

Peitz and Cribbs (2005a, b, c) summarized all historic invertebrate monitoring data from HOME, PIPE, and WICR and included interpretations of the data in their reports. The various data summarized by Peitz and Cribbs indicate there has not been a precipitous decline in stream condition at these parks since monitoring was first initiated and, in general, stream condition has largely remained stable. Similarly, the monitoring data in Harris *et al.* (1991) and Peterson (1997) suggest the streams at GWCA were not degraded at the time of sampling. The data for HEHO, however, although limited and inconclusive, suggest Hoover Creek may be degraded given the low scores for taxa richness, EPT, and Shannon's index. Although not collected under NPS protocols, Foreman (2007) completed a study of water quality in Hoover Creek at HEHO that included a multihabitat assessment of the aquatic invertebrate community at four sites using IOWATER Advanced Benthic Macroinvertebrate Indexing methods. The IOWATER methods are a modification of the Environmental Protection Agency (EPA) Environmental Monitoring and Assessment Program (EMAP) protocol (Lazorchak *et al.*, 1998). The data presented by Foreman (2007) strongly suggests that Hoover Creek is degraded with respect to the invertebrate community. Summary data for AGFO (Peitz and Cribbs, 2005d) are not included here because monitoring will no longer be conducted at the park by the HTLN.

Revision of the Original Protocol

The Inventory & Monitoring (I&M) Program of the NPS had embraced high standards for monitoring protocols. HTLN staff recently completed a comprehensive review of the protocol for sampling invertebrates in small streams, the relevant documents preceding and succeeding it, and the way that the data have been collected and analyzed. These documents included a report written in 2003 by Dr. Larissa Bailey (USGS Patuxent Wildlife Research Center, Laurel, MD) that provided short-term statistical analysis and advice concerning data collected under Peterson *et al.* (1999) (unpublished report and e-mail communications). This review revealed a critical need to revise the original monitoring protocol. Morrison and Bowles (2006) presented further recommendations for modifying the original protocol to bring it in line with other existing national-level protocols, and allow for the collection of statistically robust and scientifically defensible data. A number of potential improvements have been identified and are described below that allow us to better meet program goals and objectives. The changes presented here do not preclude comparability with the early dataset (*i.e.*, 1988-2005) obtained by collaborators at Colorado State University and subsequent monitoring. Indeed, data collected during 2006-2007 (Appendix A) using the revised protocol are quite similar to the historic data. The exception is lower genus richness scores for some parks under the new protocol because members of the family Chironomidae are no longer identified to genus. A summary of the changes is shown in Table 1.

The most significant changes described in this revised protocol are the addition of monitoring at Effigy Mounds National Monument (EFMO), George Washington Carver National Monument (GWCA), Hot Springs National Park (HOSP), Herbert Hoover National Historic Site (HEHO), and Tall Grass Prairie National Preserve (TAPR). In addition, Terrell Creek (WICR) is added as an additional monitoring site. Terrell Creek became part of WICR in 2005 when the park expanded its jurisdictional boundary. AGFO is assigned to the Northern Great Plains Inventory & Monitoring Network, and as such will no longer be sampled under this protocol.

- Historically, the parks listed under the Peterson *et al.* (1999) monitoring protocol were sampled monthly for three consecutive months each summer. We are reducing the number of sampling events to one visit every three years. This will allow us to sample more streams in more parks compared to the previous protocol.
- At some parks, where two sites had been monitored on a stream (WICR and PIPE), only a single sampling site is recommended per stream. The stream segments in the respective parks are short (≤ 2 km) and monitoring two sites does not enhance the ability to detect impairment and sampling from a single stream reach is considered generally adequate to represent a stream segment (Rabeni *et al.*, 1999; Gregg and Stednick, 2000). Although Rabeni *et al.* (1999) defined a single stream reach as 20 times the stream width and encompassing approximately 2 riffle/pool sequences, stream reach as used here under this protocol effectively meets this description due to the short lengths of the streams sampled. Both historic sampling sites will be maintained at HOME because a

different collection methodology used and to provide a sample size comparable to that for other parks.

Table 1. Summary of changes between former protocol (Peterson *et al.*, 1999) and the revised protocol.

Change Made	Former Protocol	Revised Protocol
Number of parks	4	9
Sampling sites per stream ¹	1-2 per stream	1 per stream
Sampling frequency	3 times per year	Once every 3 years
Number of riffles/sites per stream	5	3
Number of samples per riffle	1	3
Total number of samples per stream per date	5-10	9
Sampling device ²	Surber sampler/Hester-Dendy	Surber sampler/Hester-Dendy
Mesh size of sampling device	263 μm	500 μm
Subsampled portion of sample	20%	25%
Metrics	Described in Data Analysis section	No major changes
Depth and current velocity	Meter stick displacement	Wading rod and flow meter
Substrate Assessment	Percentage composition estimate of silt, sand, gravel cobble within 1 m of sample	Dominant size, (Wentworth Scale) in sample frame
Stream discharge	No	Yes
Water quality	Static CORE 5 readings (hand-held meters)	Unattended hourly CORE 5 readings (datasonde)

¹ Two historic sampling sites at HOME will be maintained.

² Hester-Dendy samplers are used only at HOME.

- The mesh size of the Surber sampler is being increased to 500 μm from the original 263 μm . This change is justified because: 1) increasing the mesh size will result in little appreciable change in the number and diversity of invertebrates collected (Morin *et al.*, 2004), 2) the proposed larger mesh size is used in other national level monitoring protocols, and 3) debris and sediments often clog the finer mesh resulting in backflow from the net and reduced capture efficiency.
- The percentage of each sample that is subsampled will be increased from 20% to 25%.
- No major changes are proposed for the metrics calculated from the benthic data. However, the Hilsenhoff Biotic Index and Shannon Evenness Index will be

calculated for the genus level rather than the family level thus allowing a more precise estimate of community tolerance and evenness, respectively. Also, total density will no longer be tracked for Hester-Dendy plate samples. Although invertebrate densities may decrease when communities are exposed to certain stressors (Resh and Grodhaus, 1983, Plafkin *et al.*, 1989), they are notoriously variable under normal conditions (Chutter, 1972; Kroger, 1972) and can provide misleading results. Finally, members of the family Chironomidae are no longer identified to genus. This change only impacts genus richness and diversity scores that will now be lower than previously recorded. This change was made because processing, mounting and identifying chironomids involves considerable staff time that could not be met given other monitoring-related demands on staff time.

- Physical habitat data collection will be modified as follows:
 - Depth (cm) and current velocity (m/sec) will be measured directly in front of the collection net using a calibrated flow meter (see SOP#5 for details) attached to a wading rod rather than using the previous and highly inaccurate method of vertical displacement with a meter stick.
 - Substrate will be assessed as the dominant substrate size represented within the sampling net frame based on the Wentworth Scale (Wentworth, 1922) (see SOP#3 for details). This approach replaces the previous method of visually estimating the percentage composition of four different substrate type categories (*i.e.*, silt, sand, gravel, cobble) within 1m of the sampling site.
 - Stream discharge (m³/sec) will also be collected for each stream sampled.
 - A data sonde will be deployed for a minimum of 24 hours for each stream to collect continuous hourly CORE 5 water quality data (temperature, dissolved oxygen, pH, specific conductance, turbidity) rather than relying on static readings for each riffle using hand-held meters.
- One disadvantage of implementing this revised protocol is that by sampling only once every three years instead of annually, it will potentially take longer to detect changes or significant trends in stream condition.

Monitoring Objectives Addressed by the Protocol

Aquatic invertebrates are an important biomonitoring tool for understanding and detecting changes in ecosystem integrity over time. Therefore, the monitoring objectives of this protocol as described by DeBacker *et al.* (2005) are:

- 1) Determine the status and trends of invertebrate species diversity, abundance, and community metrics.
- 2) Relate invertebrate community to overall water quality through quantification of metrics related to species richness, abundance, diversity, and region-specific multi-metric indices as indicators of water quality and habitat condition.

II. Sampling Design

A long-term monitoring program must specify how to efficiently sample numerous parameters through space and time. An overall sampling design must contain multiple components including: 1) a spatial design -- how sample sites are located and the area of statistical inference, 2) a revisit design -- how frequently sites are sampled, and 3) a response design -- how and what data are collected.

Rationale for the Sampling Design

The streams that will be sampled under this monitoring protocol are located among six states (AR, IA, KS, MO, MN, NE) representing several EPA Level III Ecoregions (*i.e.*, Ozark Highlands, Ouachita Mountains, Flint Hills, Central Great Plains, and Western Corn Belt Plains).

State Monitoring Programs

Most of the host states for the network parks included in this protocol employ a wide variety of approaches and methodologies to assess benthic invertebrate communities and their respective relationship to water quality. In some instances, the data collected under these various programs are not directly comparable. The state of Arkansas, however, presently does not have a statewide protocol for assessing invertebrate communities in streams. The Arkansas Department of Environmental Quality presently is developing a statewide monitoring program based on EPA EMAP.

The state of Missouri uses a multi-metric index called the Stream Condition Index (SCI) that was developed by Rabeni *et al.* (1997). The SCI is based on four metrics as measures of community structure and balance. These metrics are Taxa Richness, EPT (Ephemeroptera, Plecoptera, Trichoptera) Richness, Shannon's Diversity Index, and Biotic Index (BI). These and other community metrics are described in Barbour *et al.* (1999). All metric values are normalized so that they become unitless and can be comparable and have equal influence on the SCI results. Reference data collected from throughout Missouri were used to determine a range for each metric with one of three possible scores assigned to each range. The scores are based on the lower or upper quartile of the distribution for each metric depending on whether it decreases or increases due to impairment. The scores are then used as the minimum value representative of reference conditions. The four scores are then summed to generate the SCI score. Scores range from 16-20 for not impaired, 10-14 for impaired, and 4-8 for very impaired.

The Iowa Department of Natural Resources (IDNR) employs a biological assessment of wadeable streams (Wilton, 2004) that is strongly based on the EPA EMAP approach (see discussion on this approach below). The IDNR used biological sampling data from reference sites were to develop a Benthic Macroinvertebrate Index of Biotic Integrity (BMIBI). The BMIBI is comprised of twelve metrics that reflect a broad range of aquatic community attributes, and reference site sampling data was

used to develop metric calculation formulas that transform raw individual metric values into a normalized scoring range from 0 (poor) to 10 (optimum). The normalized and combined metric scores range from 0-30 for poor, 31-55 for fair, 56-75 for good, and 76-100 for excellent.

Since 1994, the Nebraska Department of Environmental Quality (NDEQ) has employed a rotating basin approach for water quality assessment monitoring based on the EPA's Regional Environmental Monitoring and Assessment Program (REMAP). The monitoring strategy targets resources in two or three river basins annually to allow for intensive efforts to increase the identification and abatement of pollution problems. All 13 water basins in Nebraska are monitored over 5 years. Approximately 40 biological monitoring sites are selected randomly each year from the perennial streams within the water basin of interest for that year. Sample sites are selected to best represent monitoring objectives and are based on professional judgment.

The Minnesota Pollution Control Agency (MPCA) has used benthic invertebrates as indicators of human disturbance of aquatic resource integrity for the past 25 years. The MPCA approach is generally based on the methods of the USEPA and used a multihabitat sampling approach. Between 1976 and 1979 the MPCA collected invertebrate community data from 21 stream stations throughout the state to assess water quality. Between 1990 and 1992, invertebrates were collected at 45 stream stations through the Minnesota River Basin. An IBI developed by Ohio EPA for the Eastern Cornbelt Plain was used as a means for assessing aquatic resource integrity. The MPCA has biological and stream water chemistry data for several locations on Pipestone Creek (PIPE), with the most recent available data from 2004. No reports further summarizing or interpreting this data are available from MPCA.

The Kansas Department of Health and Environment (KDHE) conducts stream invertebrate monitoring as part of their Stream Biological Monitoring Program (KDHE, 2000). The KDHE approach is based on two independently collected 100-organism samples collected in the field by two scientists that are used to calculate the four metrics. The metrics used include the Kansas Biological Index (KBI), the macroinvertebrate biotic index (MBI), EPT (Ephemeroptera, Plecoptera, Trichoptera) richness, and EPT (%) abundance to assess stream integrity. Each metric is scored and assigned to one of three aquatic life-support categories: fully supporting, partially supporting, and non-supporting (Poulton *et al.*, 2007). The KDHE previously has conducted invertebrate community assessments in the Fox Creek Watershed, which includes sites on Fox Creek and its tributary, Palmer Creek. The data collected by the KDHE suggested moderate impairment in these two streams and that nitrification from animal wastes and fertilizer is the primary source of the disturbance. These data have not been published, but are available from the KDHE as part of the Neosho River Basin total maximum daily load.

Federal Monitoring Programs

Programs to monitor stream condition are used by the US Environmental Protection Agency (EPA), US Geological Survey (USGS), and the National Park Service, Heartland Inventory & Monitoring Network (HTLN).

USGS NAWQA: The general basis of the USGS National Water-Quality Assessment Program (NAWQA) program is to collect biological, physical, and chemical data at stream reaches that are subject to major natural and anthropogenic factors considered responsible for controlling water quality in a river basin. From these sampling reaches, two broad types of benthic samples are collected to characterize the invertebrate community: 1) semi-quantitative benthic samples collected from targeted habitat types (*i.e.*, richest targeted habitat or RTH), and 2) a composite qualitative sample collected from a broad variety of habitats (*i.e.*, qualitative multihabitat or QMH) from throughout the reach. The RTH theoretically supports the faunistically richest invertebrate community and is typically represented by a coarse-grained riffle or a woody snag. The semi-quantitative RTH sample consists of a series of discrete collections (Moulton *et al.*, 2002). The semi-quantitative benthic samples recommended by NAWQA are collected from the RTH (riffles in most cases) using a Slack-Surber sampler (0.25 m²) (Moulton *et al.*, 2002). The number of individual benthic samples to be collected under the NAWQA protocol depends on study objectives. The NAWQA protocol generally recommends selecting a single riffle where at least five discrete collections can be taken. Collected samples are partially processed in the field and subsequently composited into a single bulk sample. By compositing the individual samples collected from a reach, however, no estimate of variability among samples can be obtained. The NAWQA protocol allows for location of sites based on representativeness of the local area, given that the location supports project objectives. This gives the site investigator flexibility in establishing site boundaries depending on local conditions.

EPA: The EPA has two programs for assessing water quality using invertebrate communities in wadeable streams. These are the Rapid Bioassessment Protocols for Use in Streams and Rivers (Barbour *et al.*, 1999), and the Environmental Monitoring and Assessment Program-Surface Waters (EMAP) (Lazorchak *et al.*, 1998). An additional set of protocols designed for larger non-wadeable rivers (Flotemersch *et al.*, 2006) generally are not applicable to the streams in the network parks and are not further addressed here.

Rapid Bioassessment

The Rapid Bioassessment approach uses either single habitat (*e.g.*, riffles) or multi-habitat approaches. Both approaches involve collecting samples from a 100 m reach determined by the investigator to be representative of the characteristics of the stream. The single habitat approach involves sampling using a kick-net to sample approximately 1 meter in front of the net, and taking 2-3 kicks using foot agitation. The samples are then composited for analysis. Benthic metrics for analyzing data are the same or comparable to those used in this protocol (Barbour

et al., 1999). The multi-habitat approach uses 20 jabs or kicks taken from different representative habitat types within the reach using a D-frame dipnet. Samples are composited for analysis and metrics are the same or comparable to those used in this protocol (Barbour *et al.*, 1999).

EMAP

The EMAP approach focuses on evaluating ecological conditions on regional and national scales. It uses probabilistically selected sites where individual sampling sites are assessed using a transect-based design where community biological metrics are tied to habitat structure. Kick net samples collected from flowing water habitats (*e.g.*, riffles, runs) are combined into a single composite sample for the stream reach, while kick net samples collected from pool habitats are combined into a separate composite sample. The “kick net” used in the EMAP method is effectively the same net as a Slack-Surber sampler minus the frame delineating the sampling area in front of the net. Data are analyzed following Barbour *et al.* (1999) and use either multimetric or multivariate approaches. In addition, some programs use O/E (Observed/Expected) Ratio of Taxa Loss to assess invertebrate community degradation. This tool is a ratio comparing the number of taxa expected (E) to exist at a site to the number that are actually observed (O). The taxa expected at individual sites are based on models developed from data collected at reference sites. The current protocol does not use O/E ratios.

The EPA’s Wadeable Streams Assessment Program is based on the EMAP approach and is not considered separately here (United States Environmental Protection Agency, 2004a, b, c, d).

Heartland Inventory and Monitoring Network (HTLN): The HTLN developed a monitoring protocol for large rivers and their tributaries within network parks that is generally based on the NAWQA approach (Bowles *et al.*, 2007). This HTLN monitoring approach uses multiple randomly selected sampling reaches where three benthic samples are collected from each of three consecutive riffles. Samples are analyzed separately and are not composited, allowing for variability to be assessed. Also, qualitative, multihabitat samples are not collected under the HTLN protocol. Collection methods and sample processing otherwise follow NAWQA.

There are some similarities among the EPA, NAWQA, and HTLN approaches that will allow for comparison of data. In support of this statement, Peterson and Zumberge (2006) generally found no significant differences between invertebrate samples collected from riffles using the NAWQA and EMAP protocols. Also, Herbst and Silldorff (2006) reported that, while methodological uniformity is important when coordinating monitoring programs, data from multiple sources could potentially be used interchangeably and for cross-validation of assessments of stream biological integrity. Herbst and Silldorff (2006) further noted that differing bioassessment methods can yield

similar data and effectively discriminate impaired biological condition even though they have multiple differences in field and laboratory protocols.

The small streams sampling approach described here is based primarily upon that of the HTLN large rivers protocol (Bowles *et al.*, 2007). However, this protocol differs from the HTLN large rivers protocol to account for maintaining comparability with historic monitoring data, and because of limitations posed by staff size and logistical and budgetary constraints. For example, because the length of the streams inside the park boundaries is relatively short (3 km or less), this protocol will use a single sampling reach for each stream sampled, rather than multiple reaches. This approach is the same as that used by Bowles *et al.* (2007) for small tributaries that are similar in size to those in the small stream parks. Also, the sampling device proposed for use in this protocol is the Surber stream bottom sampler (0.093 m²) rather than the Slack Surber sampler (0.25 m²) recommended by the HTLN large river protocol. We propose to use the former sampler because it has been used historically for sampling invertebrates in many of the included network parks. We also will continue to use Hester-Dendy multiplate samplers at HOME because they have been used historically for this purpose, and their continued use favors comparison of data.

Spatial Design

Establishing the sample frame

This protocol focuses on aquatic invertebrate communities occurring in small, wadeable streams within the National Park Service jurisdictional boundaries at each of the included parks. Once defined, sample unit boundaries will remain fixed.

Boyle *et al.* (1990) did not specify how sites were selected, simply referring to them as “sentinel” sites. Harris *et al.* (1991) gave a written description of the locations of each site. As Peterson *et al.* (1999) later pointed out, no criteria were given for site selection. However, historical sampling sites were frequently near upstream and downstream park boundaries, and accessibility was apparently an important concern. In fact, most historical sites were located near park roads or trails, indicating convenience was an important component in site selection. Peterson *et al.* (1999) recommended continued use of these “established sites,” presumably to allow for comparisons with the earlier data. This protocol will retain the historical collection sites, but only including the downstream-most sites in parks with two historic sampling sites (with the exception of HOME). The problem with choosing sites based on such criteria is that, statistically speaking, any results obtained from such a design are applicable only to the specific locations sampled, and not the entire stream running through the park. Given the short length of the streams to be sampled, however, this shortcoming is not an overriding concern in light of maintaining comparability with historic data. Furthermore, sampling from a single stream reach (defined as 1 riffle-pool-riffle sequence) is generally considered adequate to represent a stream segment (Rabeni *et al.*, 1999; Gregg and Stednick, 2000). Gebler (2004) reported that the number of reaches required to be sampled to obtain Minimum Detectable Differences (MDD) of $\leq 20\%$, ranged in the tens

to hundreds of reaches. However, for this protocol, such a large number of sampling sites would not be feasible because of budgetary and staffing constraints, and the relatively small physical size of the streams themselves.

Sampling Sites and Reach Selection

This protocol recommends invertebrate monitoring in 18 streams located among 9 parks (Table 2). The sampling index period assigned to each stream is primarily based on an examination of the historical dataset from 1988 to 2004. Rationale for using these index periods is presented below under *Temporal Design*.

At GWCA, PIPE, and WICR, only the downstream-most historic sampling sites will be sampled. At Cub Creek (HOME), both historic sites will be maintained. Using two sampling sites at HOME is based in part on the use of a different methodology and in part because it will provide a sample size comparable to that of the other parks. For all other streams added to this revised protocol, a sampling reach was selected from the downstream-most portion of each stream within the jurisdictional boundaries of a park. The selected sampling sites were located upstream of the apparent floodplain of any larger tributaries when applicable. Riffle selection was determined *a priori*, with the three riffles sampled being those located in consecutive order upstream of the first riffle above the lower reach boundary (Fig. 1). In some cases, the first upstream riffle in a stream was located a considerable distance from the park boundary (*i.e.*, TAPR). Sampling reaches identified in this protocol are permanent, but the specific locations of the riffles sampled in a given year may move naturally due to hydrological processes. General directions to the sampling sites within a park are listed in SOP #3 (Sampling Invertebrates and Collecting Habitat Data).

Sample Placement

Placement of Surber samples within a given riffle appear to have been selected subjectively, or at best haphazardly, in previous monitoring protocols (Petersen *et al.*, 1999). This provides an obvious opportunity to introduce *bias* into the sampling. To correct for this deficiency, this protocol proposes (after Bowles *et al.* 2007) that individual Surber samples will always be taken in an upstream direction in an *a priori* alternating and equally spaced sequence (left third, middle, and right third). This arrangement is illustrated in Fig. 2. Sample sequence will be altered only if the original starting point presents a danger to the collector or if it is not accessible. Some riffles may be wider than long and, in such cases, samples can be taken from left to right in equally spaced increments.

Table 2. The streams that will be sampled in each network park, and the corresponding index period recommended for sampling.

Park	Streams sampled	UTM Coordinates (Northing, Easting)	Index Period
GWCA	Carver Creek Harkins Branch Williams Branch	4094380.11, 379254.85 4094493.46, 378963.70 4094466.25, 379268.19	May-June
EFMO	Dousman Creek	4772108.08, 645475.84	July-August
HEHO	Hoover Creek	4614462.87, 637697.89	July-August
HOME	Cub Creek	4462337.67, 684059.84	July-August
HOSP	Bull Bayou Gulpha Creek	3819096.45, 489743.19 3820036.11, 496779.10	May-June
PERI	Pratt Creek Winton Spring Branch	4033256.21, 407127.86	May-June
PIPE	Pipestone Creek	4877259.61, 714204.77	July-August
TAPR	Fox Creek Palmer Creek	4256985.51, 713944.53 4263176.10, 710907.56	August-September
WICR	Skegg's Branch Terrell Creek Wilson's Creek	4105745.65, 463391.47 4104000.832, 462818.328 4104580.870, 464167.047	May- June

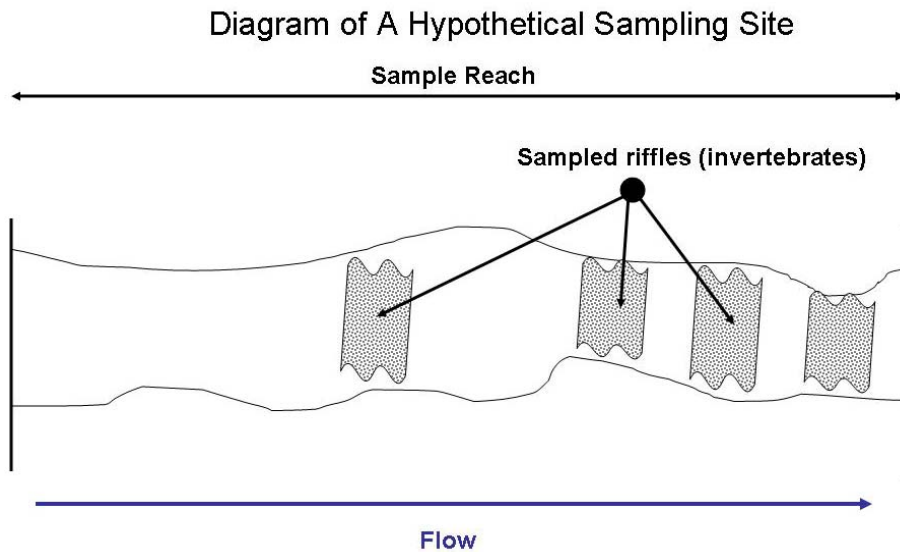


Figure 1. Riffle selection within a stream.

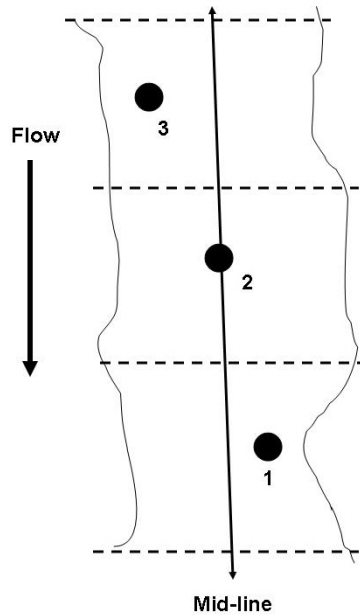


Figure 2. Placement of Surber samples within a single riffle.

For Hester-Dendy samplers, the historic sampling locations used at HOME will continue to be used. Five samplers each will be placed in the upper and lower areas (Fig. 3) with the individual samplers being placed no less than 1 m apart. Hester-Dendy samplers are prone to loss due to flooding in Cub Creek and having five sampling devices at each site is intended to offset such loss. Data from any samplers remaining at each site at the time of collection will be processed for analysis. Samplers are anchored to the bank with wire or attached to permanent structures as they are available in the stream channel.

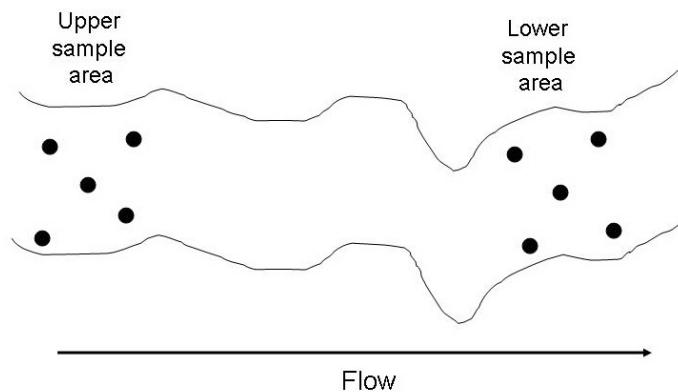


Figure 3. Placement of Hester-Dendy samplers in Cub Creek at Homestead National Monument of America.

Temporal design

The revisit design consists of a set of rotating panels for the network parks where each stream is sampled every three years (Table 3).

Table 3. Revisit design for invertebrate monitoring in small streams at Heartland I&M Network parks.

Study Park	Revisit Notation	2007	2008	2009	2010	2011	2012	2013	2014	2015	2016	2017	2018
GWCA-PIPE - WICR-	[1-2]	X			X			X			X		
EFMO-HEHO-HOME	[1-2]		X			X			X			X	
PERI-TAPR-HOSP	[1-2]			X			X			X			X

The invertebrate communities of the small streams in network parks consist of a high diversity of species in various developmental stages. Therefore, temporal consistency in sample collection is essential to reducing the natural variability in invertebrate life cycles and community structure (Rabeni *et al.*, 1997).

Boyle *et al.* (1990) recommended that sampling be done once in three of the four seasons: spring, summer, and fall, but gave no rationale for this approach. Peterson (1996) used data collected from a single year—1989—to assess the temporal variance in the metrics calculated. He reported two major findings: (1) for the same sample size, variance within a year was greater than variance within a single season (*i.e.*, summer), and (2) variance decreased as sample size increased. Based on these findings, Peterson recommended that, to decrease temporal variance, sampling should be done within a single season (*i.e.*, summer) rather than over the entire year, and that at least three samples be taken on different dates. In an independent assessment of Peterson (1996), Dr. Larissa Bailey (unpublished report) also recommended that sampling be conducted only once per year, within a specified time period. Dr. Bailey further specified optimal sampling periods for each prairie park, based on invertebrate community stability and family richness metrics. Bailey recommended sampling in July, August, or September, depending upon the park. Morrison and Bowles (2006) examined the complete dataset from 1988 to 2004 for all primary metrics included in this protocol. The mean values for each metric were comparable among all months sampled. Additionally, the estimates of standard error overlapped extensively, suggesting there is little difference among these metrics during

the months sampled. In order to reduce costs and increase efficiency and robustness of the community metrics that will be used in this protocol, this protocol employs a single sampling event within the index periods shown in Table 2. Also, this sampling design will control for seasonality and intra-annual effects. The ultimate choice of an index period for sampling also must consider seasonal stream flow patterns, as flows become very low or streams dry entirely in some network streams in late summer. Although the Peterson *et al.* (1999) protocol specifies that three samples are to be taken each year, frequently only one or two were in fact collected due to low stream flows (*e.g.*, Thomas *et al.*, 2002). The collection periods indicated above should ensure sufficient stream flows to allow effective sampling.

To the extent possible, temporal consistency should be maintained through successive years as well as between sample types. Samples from each stream should be collected within the shortest time frame possible (1-2 days) to minimize the effects of seasonal change. All efforts should be made to avoid collecting directly after a flood event or major disturbance. Samples should be collected only during baseflow conditions and a minimum of two weeks after flood waters recede to baseflow conditions.

Response Design

Types of Data Collected in the Field

This monitoring program will collect benthic invertebrates from stream riffles as well as associated habitat data and water quality. Habitat features are major, often limiting, determinants of invertebrate community structure and accordingly they are especially important for proper determination of biomonitoring results and assessment of ecological integrity (Barbour *et al.*, 1999). Although habitat incorporates all aspects of physical and chemical constituents and their interactions, variables such as current velocity, substrate size, embeddedness, water chemistry, sediment deposition, and presence of filamentous algae and aquatic plants play key roles in the microhabitat structure and distribution of aquatic invertebrates (Allan, 1995; Hauer and Lamberti, 1996; Rosenberg *et al.*, 2008). We propose to monitor all of the aforementioned habitat variables at our sampling sites.

Biological and environmental correlates of water quality and habitat structure compared across time are powerful tools for assessing disturbances related to natural and anthropogenic impacts on aquatic invertebrate communities, and they are useful for detecting change and elucidating patterns and trends in long-term data sets (Moulton *et al.*, 2002). For example, as habitat conditions degrade (*e.g.*, water quality decreases, embeddedness increases), degradation of the benthic invertebrate community is expected to follow. However, the relationship of cause and effect of these variables on aquatic invertebrate community structure can be difficult to assess and analyze because there often is a broad response range among the resident species based on tolerance to disturbance (Norris and Georges, 1993). Therefore, any association of community structure with these variables or their combinations must be interpreted cautiously and be based on real biological properties. These limitations withstanding, benthic community

structure, when viewed in association with environmental variables, can be an effective indicator of ecosystem change (Reice and Wohlenberg, 1993).

Sampling Devices

Choosing the appropriate sampling device is one of the most critical aspects of biomonitoring (Resh and McElroy, 1993). The physical characteristics of the stream determine the most appropriate device to use for sampling. Although a broad variety of sampling net types have been used to successfully sample stream riffles, the objectives of this protocol require continuity with the methods of Harris *et al.* (1991). Therefore, a Surber sampler (0.0929 m²) will continue to be the sampling device used to collect samples so the data will be comparable to that collected in the 1989 baseline data set, and to other historical NPS sampling. Harris *et al.* (1991) used Surber nets of differing mesh sizes depending on the stream sampled, but in this protocol we recommend using a single mesh size (500 µm) for all streams sampled.

At HOME, benthic substrate is dominated by fine sands and current velocity is slow. Because of these constraints, Surber samplers would not be effective for sampling. Instead, we are using Hester-Dendy multiplate samplers that are placed in pools or slower-moving water (*i.e.*, no riffles) to simulate stream habitats dominated by abundant woody debris. Samplers require water depth to be at least 25 cm. These samplers are composed of nine 57.76 cm² square hardboard plates separated by 3 plastic spacers and connected by a long eyebolt, ultimately providing 0.0929 m² of surface area for invertebrate colonization. Although natural woody snags are superior to Hester-Dendy samplers for assessing invertebrate communities in soft-bottomed streams (Moulton *et al.*, 2002), we propose to continue using Hester-Dendy samplers to monitor invertebrate communities at HOME. The primary justification for using these samplers rather than collecting woody snags is that these sampling devices have been used historically at the park and the goal is to produce comparable data in future monitoring efforts.

Illustrations of each sampling device are shown in SOP#3 (Sampling Invertebrates & Collecting Habitat Data).

Number of Samples

Three benthic samples will be randomly collected from each of three selected riffles, resulting in a total collection of nine separate samples per stream. This sampling approach therefore provides an estimate of intra- and inter-riffle variability. The process of collecting benthic samples is described in SOP#3. Several studies have shown that three samples per riffle are sufficient to characterize the benthic invertebrate community with respect to calculation of metrics while accounting for variability (Canton and Chadwick 1988, Bowles 1989, Mathis 2001, Usrey and Hinsey 2006).

For Cub Creek at HOME, five Hester-Dendy samplers will be deployed at each upstream and downstream sampling site for a total of 10 samples per sampling event. As previously stated above, this protocol recommends deploying five samplers because

individual samplers are prone to loss from flooding. However, data from any samplers remaining at each site at the time of collection will be processed for analysis.

Suitability of Survey Design to Meet Study Objectives

Monitoring objectives are integral to defining the sampling design. This sample design allows for assessing the integrity of invertebrate communities over time by measuring net change in certain community metrics. For assessing status and trend through time of invertebrate communities, the overall survey design was deemed suitable for several reasons:

1. *Single habitat (riffle) sampling is appropriate for long-term monitoring of benthic invertebrates.* Sampling multiple habitats provides more comprehensive information about the invertebrate fauna compared to single habitat samples (Lenat and Barbour, 1994; Moulton *et al.*, 2002). Comparability among sites is necessary for accurate bioassessments, however, and invertebrates collected from the same habitat types among sites are more similar than invertebrates collected from multiple habitats within the same site (Parsons and Norris, 1996; Rabeni *et al.*, 1997). Indeed, Rabeni *et al.* (1997) showed metric sensitivity did not increase when comparing multiple versus single habitat sampling in Missouri streams, and we contend this is true for other network streams. Therefore, single habitat sampling in riffle habitat is the focus of this protocol. For soft-bottomed streams such as Cub Creek at HOME where the substrate consists primarily of sand, Hester-Dendy samplers are judged to be an acceptable means of assessing aquatic invertebrate communities.
2. *Appropriate for all small streams in the network.* The sampling design and methods described in this protocol are applicable to all small streams located in network parks. Furthermore, the data generated from this study design will be directly comparable to those of other regional (state and federal) invertebrate monitoring programs that employ similar methodologies and rely largely on percentage-based metrics (*e.g.*, Barbour *et al.*, 1999).
3. *Easy to learn and use.* Field procedures are easy to use and repeatable over time by different sampling crews trained in these procedures. Implementation does not require extensive time or costly equipment.
4. *The sequence of sampling events and revisit design for the listed parks allows for the greatest amount of field work to be accomplished per year while minimizing cost.* Because staff available for manning field crews is limited, and there is a great distance among all network parks and travel costs associated with monitoring are high, this strategy allows cost effective monitoring for stream sites in multiple network parks.
5. *The selected approach to monitoring is advantageous over other approaches.* The study design and methods selected for this protocol allow for an integration of community attributes and further allow us to characterize temporal changes and relative site quality. Additionally, our approach will allow us to correlate invertebrate community data with land use and habitat changes potentially arising from multiple stressors.

III. Field and Laboratory Methods

Field Season Preparations, Field Schedule, and Equipment Setup

Procedures for field season preparations, including preparing a field sampling schedule and equipment setup, are described in SOP#1. Team leaders should ensure that team members have read and understand the protocol and supporting SOPs prior to sampling, and that all required equipment and supplies have been ordered and are in proper working condition. They also should check stream staff gages (<http://water.usgs.gov>) or contact park resource managers to determine if sampling sites have recently flooded. The team leaders will prepare and maintain a field notebook detailing all sampling-related activities and staff participation during monitoring trips to ensure that trip reports are complete and accurate. Finally, the team leader should ensure that all required scientific collection permits have been obtained.

Collecting Benthic Invertebrate Samples and Associated Habitat and Water Quality Data

Procedures for collecting benthic invertebrate samples and documenting habitat data are presented in SOP#3 (Sampling Invertebrates and Collecting Habitat Data), SOP#4 (Documenting CORE 5 Water Quality Variables), and SOP#5 (Measuring Stream Discharge). Work flow diagrams for collecting samples are shown in Figs. 4 and 5 below.

Three invertebrate samples will be collected from each riffle from randomly selected sample points as described in SOP#3. Samples will be collected with a Surber stream bottom sampler (500 μm mesh, 0.093 m^2). Water flow (level) should not be over the top of the net in deep riffles to prevent invertebrates dislodged from the substrate from washing over the net, and not being collected in the sample. Each discrete sample is collected while progressing in an upstream direction. Sampling procedures will be the same for each riffle sampled and whenever possible samples should be collected by the same person to limit variability in sampling techniques. For HOME, five Hester-Dendy samplers will be deployed at each of two locations, one upstream and one downstream. Samplers are deployed for 30 days to allow for sufficient colonization by invertebrates prior to harvesting.

Habitat variables will include an assessment of depth and current velocity measurements collected concurrently and immediately in front of the sampler frame. Several additional qualitative measurements of habitat condition will be taken from the area delineated by the sampler frame after it is placed securely on the stream bottom and before disturbing the substrate. These variables include visual estimates of percent embeddedness of the substrate, percent periphyton, percent filamentous algae, and percent organic material. Standard classes for all percentage estimates will be as follows: 0 = Absent (0%), 1 = Sparse (<10%), 2 = Moderate (10-40%), 3 = Heavy (40-75%), and 4 = Very Heavy (>75%). When the habitat variables have been recorded, substrate size is visually

assessed. Substrate assessments provide a unique characterization of the streambed composition at the time sampling takes place. Therefore, dominant substrate size from the area within the sampling frame of the net is visually assessed based on the standard Wentworth scale (Wentworth, 1922). The intent of the substrate assessment is to characterize the dominant substrate for individual samples and not to fully characterize all sediments present. This assessment will help us describe the prevailing microhabitat conditions that influence the structure of invertebrate communities and may help explain variability between sample points. Stream discharge will be measured at each site and preferably upstream of the sampling site after invertebrate collections have been completed. CORE 5 water quality parameters (temperature, dissolved oxygen, specific conductance, pH, turbidity) will be recorded for each stream using data loggers or sondes.

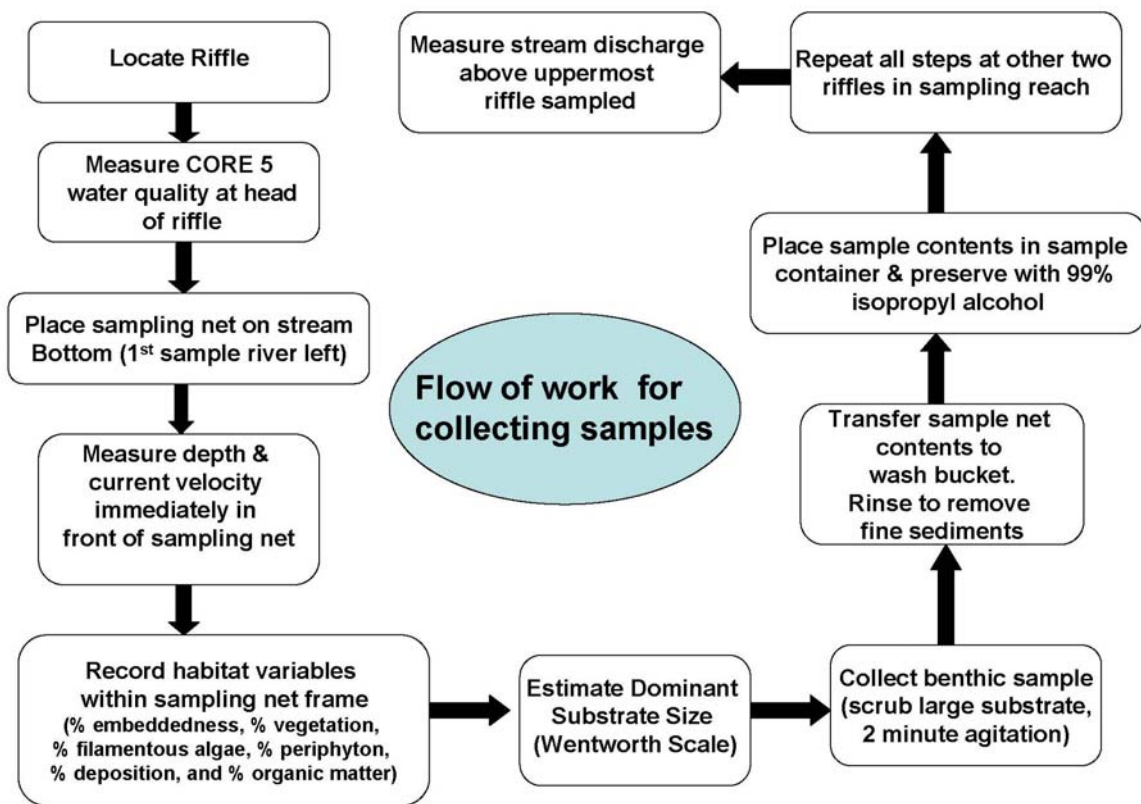


Figure 4. Flow of work diagram for collecting Surber samples.

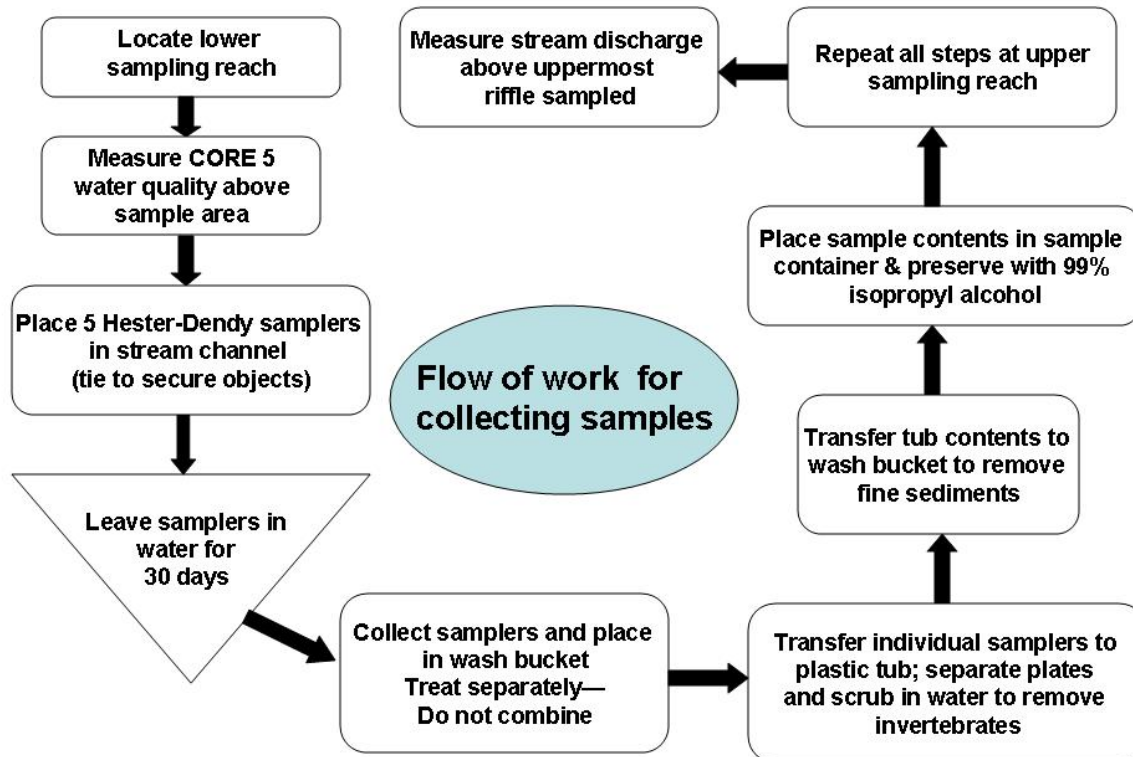


Figure 5. Flow of work diagram for collecting Hester-Dendy samples.

Benthic Sample Processing and Specimen Identification

Procedures for processing benthic samples and identifying specimens are described in SOP#6 “Laboratory Processing and Identification of Invertebrates.” Methods for preparing samples for sorting and subsampling generally follow those presented in

Moulton *et al.* (2000). A list of the aquatic invertebrate taxa known or suspected to occur in network park small streams is provided in SOP#8 (Data Analysis).

Subsampling benthic samples

The routine for subsampling benthic samples is presented in SOP#6. The method of subsampling will involve the fixed fraction approach with 25% of each sample being sorted following thorough washing, agitation, sieving, and elutriation of the entire sample (Moulton *et al.*, 2000). Additionally, a “large and/or rare” taxa component will be included where large or rare taxa that clearly are not in the sorted fraction are removed and stored in a separate vial for the purpose of reflecting accurate sample species richness

estimates and calculating specific metrics such as EPT. A fixed fraction subsampling routine was selected over a fixed count routine because some of the metrics to be calculated from samples are related to specimen density that cannot be obtained with the latter method. Subsampled fraction debris will be subjected to QA/QC analysis (SOP#6) and should be kept until QA/QC is complete for that batch of samples and the Program Leader authorizes disposal of the debris.

Sample Storage and Reference Collection

Identified samples are stored in 4 dram, glass vials with polycone caps and filled with 70% isopropyl alcohol. Specimen vials will be labeled with the taxon name, date collected, park and site names/code, and name of identifier. Organisms will be retained for at least three years and stored at the NPS HTLN facilities located at Missouri State University, Springfield, MO.

A reference collection consisting of a few representative specimens of each taxon will be prepared and stored in properly labeled vials containing 70% isopropyl alcohol. Regional or other taxonomist specialists should review the identifications for accuracy. This collection is intended to aid future identifications and training new personnel. The reference collection will be stored at the NPS HTLN facilities located at Missouri State University, Springfield, MO.

Post Season Procedures

Procedures for the end of the sample season are found in SOP#10 “Procedures and Equipment Storage after the Field Season” and are not further described here. Crew leaders will ensure all equipment is cleaned and properly stored and that all equipment is in working order prior to long-term storage.

IV. Data Management

Overview of Database Design

Step-by-step procedures for data management are described in SOP #7 “Data Management.” Microsoft Access 2003 is the primary software environment for managing stream invertebrate community data. ESRI ArcInfo 9 serves as a tool for validation of spatial data residing in Access. Data products will be posted at the NPS I&M website: <http://science.nature.nps.gov/im/monitor/protocoldb.cfm>. Metadata for stream invertebrate community monitoring will be available on the NPS I&M application server: <http://science.nature.nps.gov/im/apps/index.cfm>. QA/QC guidelines in this document are based on recommendations of Rowell *et al.* (2005) and Fancy at <http://science.nature.nps.gov/im/monitor/index.cfm> and citations therein.

The general data model for stream invertebrate community monitoring consists of two core sets of tables: invertebrate counts and habitat data. The first manages taxa attribute

data (taxa names, taxa counts, etc.) and the second associated habitat data. These core tables are linked in time and space by way of standardized event and location tables. Supporting tables include taxonomic attributes, event observers, equipment information and look-up tables linked to relevant tables to provide values for pick-lists on data-entry forms, thereby reducing possible error during data entry (see Data Verification and Validation below).

Data Entry

A number of features have been designed into the database to minimize errors that occur when field data are transcribed to the database for storage and analysis. Forms are used as portals for data entry into the database. Standardized identifiers (*e.g.*, sample location and event) are selected from a list of easily interpreted codes. Species and habitat data are entered into fields linked to appropriate tables. Look-up tables contain project-specific data and prohibit entry of data into a field if a corresponding value is not included in the look-up table. Consequently, only valid names or measures may be entered and spelling mistakes are eliminated. Species or habitat measures are selected using pick lists or by typing the beginning of the name.

Data Verification and Validation

Data verification immediately follows data entry and involves checking the accuracy of computerized records against the original source (*i.e.*, paper field records). While the goal of data entry is to achieve 100% correct entries, this is rarely accomplished. To minimize transcription errors, our policy is to verify 100% of records to their original source by staff familiar with project design and field implementation. Further, 10% of records are reviewed a second time by the Project Manager and the results of that comparison reported with the data. If errors are found in the Project Manager's review, then the entire data set is verified again. Once the computerized data are verified as accurately reflecting the original field data, the paper forms are archived at HTLN and the electronic version is used for all subsequent data activities.

Although data may be correctly transcribed from the original field forms, they may not be accurate or logical. Data validation involves checking the accuracy of computerized records against some outside standard or knowledge. For example, field records of pH should be within the range of 0 – 14 and most likely closer to 6 or 7. Likewise, a specimen count of 3,325 instead of 325 may be illogical and almost certainly incorrect, whether or not it was properly transcribed from field forms. Certain components of data validation are built into data entry forms (*e.g.*, range limits). Data validation can also be extended into the design and structure of the database. Constraints on data values might include 'no null values allowed' and 'each value must be unique'. To minimize error, values for data-entry forms have been limited to valid entries stored in the look-up tables.

Additional data validation can be accomplished during verification, if the operator is sufficiently knowledgeable about the data. The Project Manager will validate the data after verification is complete. Validation procedures seek to identify generic errors (*e.g.*,

missing, mismatched, or duplicate records) as well as errors specific to particular projects. For example, one database query detects records with a location ID from a site and a period ID from a different site. Another query counts the number of transects sampled per site to be sure all data were collected and entered.

During the entry, verification, and validation phases, the Project Manager is responsible for the data. The Project Manager must assure consistency between field forms and the database by noting how and why any changes were made to the data on the original field forms. In general, changes made to the field forms should not be made via erasure, but rather through marginal notes or attached explanations. Once validation is complete, the data set is turned over to the Data Manager for archiving and storage.

Validation of spatial data requires the cooperative effort of the project leader and GIS specialist. Spatial validation of project GPS data requires proficiency with GIS such as ArcGIS (ESRI, Inc.). GPS coordinate data are imported into the project GIS. Project shapefiles can then be compared against significant cultural and natural features such as road intersections, unusual trees, and significant hydrological features as observed from USGS DOQQ orthophotography. The project leader's familiarity with ground features will validate GPS locations.

Metadata Procedures

Metadata for project data are developed using ESRI ArcCatalog and the NPS Metadata Editor and Tools extension. These data follow the Federal Geographic Data Committee (FGDC) standards and include biological profile elements that allow integration of the metadata record with the NPS I&M NPSpecies and NatureBib databases. Metadata are then exported into text and Extensible Markup Language (xml) format. Text-based metadata are then parsed using a USGS metadata parser to check for errors in formal FGDC metadata.

Database Versions

Changes in database structure and functionality require a versioning system. This allows for the tracking of changes over time. With proper controls and communication, versioning ensures that only the most current version is used in any analysis. Versioning of archived data sets is handled by adding a two digit number separated by a period to the file name, with the first version being numbered XXXXXX1.0. Minor changes such as revisions in forms and report content should be noted by an increase of the number to the right of the period. Major changes such as migration between Access versions or database normalization across multiple tables should be indicated by an increase in the number to the left of the period. Frequent users of the data are notified of the updates, and provided with a copy of the most recent archived version.

Database Security

Secure data archiving is essential for protecting data files from corruption. No versions

of the database should be deleted under any circumstance. Monitoring databases are small and do not require significant computer drive space or resources. They represent primary data and are expensive to create and impossible to replace. Multiple backup copies of all program data are maintained at the HTLN offices, at the Wilson's Creek visitor center, and at the Missouri State University campus offices. Tape backups of the databases are made weekly. Each weekly full backup copy is maintained at the Wilson's Creek National Battlefield Visitor Center, Republic, MO. Once a month, one tape copy is stored offsite.

Currently, data are available for research and management applications on request, for database versions where all QA/QC procedures have been completed and the data have been archived. Most data requests are currently met using FTP services. Portions of the monitoring data collected under this protocol will be made available for download directly from the NPS I&M Monitoring webpage. Information related to location and persistence of species determined to be threatened or endangered will not be made available for download by the general public. In addition, metadata will be available directly from the NPS I&M NR-GIS Metadata and Data metadata server (<http://science.nature.nps.gov/nrddata>). Data requests should be directed to:

Data Manager
Wilson's Creek National Battlefield
6424 W. Farm Road 182
Republic, MO 65738-9514
(417) 732-6438

V. Analysis and Reporting

Metric Selection and Community Indices

Early biomonitoring programs tended to focus on one or two specific attributes or metrics of the community; the indicator species concept (Kremen, 1992) is an example. Individual metrics generally are chosen based on the specific and predictable response of organisms to landscape changes. Additionally, they are sensitive to a range of factors that stress biological systems and are relatively easy to measure and interpret (Karr and Chu, 1999). Barbour *et al.* (1999) lists and briefly describes many types of metrics used in assessing stream condition. Unfortunately, individual metrics in themselves often are not adequate for assessing complex systems with cumulative impacts (Karr, 1991). In comparison, multi-metric indices are designed to look at community structure through examination of multiple components of an invertebrate community and its level of change in response to disturbance. Scores of individual metrics are normalized into a single integrated score, reducing the influence of one metric on the overall score and making results less ambiguous for resource managers. Bonada *et al.* (2006), in a comparative analysis of recent bioassessment approaches, showed that multi-metric approaches rate among the best performers for 10 of 12 criteria used to test for discriminating among different kinds of human impact. Multi-metric approaches are favored by most aquatic resource agencies in the United States because they are based on

sound scientific rationale, they are simple to implement, and they are among the most sound for assessing invertebrate community structure (Lenz and Miller, 1996; Bonada *et al.*, 2006).

Peterson *et al.* (1999) concluded that invertebrate community structure could be adequately summarized by five biotic indices (*i.e.*, total density; family biotic index; Ephemeroptera, Plecoptera and Trichoptera (EPT) richness; EPT/Chironomidae ratio; and the Shannon-Weiner Index). These indices were chosen because they represented independent estimates of change with minimal redundancy in the community aspects estimated by each metric (*i.e.*, metrics were avoided if they appeared equally sensitive to the same environmental factors). This biomonitoring protocol retains most of those metrics to characterize invertebrate communities taking into account that metrics differ in their sensitivity to changes in different environmental variables. For example, some metrics may be more sensitive to changes in structural variables such as sediment grain size than to chemical and physical water quality variables (Bode and Novak, 1995; Yoder and Rankin, 1995). The exception is total density that Peterson *et al.* (1999) recommended for Hester-Dendy samples. Although total invertebrate density may decrease when communities are exposed to a stress such as water pollution or habitat alteration (Resh and Grodhaus, 1983; Plafkin *et al.*, 1989), it also can be notoriously variable under normal conditions (Chutter, 1972; Kroger, 1972) and provide misleading results. Taxa richness and genus evenness are included as additional measures of community integrity based in part on the recommendation of Rabeni *et al.* (1997). Procedures for calculating these metrics are shown in SOP # 8 (Data Analysis). Summary indices and variables will provide information to park managers on the status of the stream invertebrate communities.

EPT Richness - Ephemeroptera, Plecoptera, and Trichoptera (EPT) richness is the number of genera from these three orders represented in a sample. Members of these three insect orders are among the most ecologically sensitive taxa in streams and are considered excellent indicators of water quality (Resh and Jackson, 1993; Resh and McElray, 1993).

EPT ratio (R) - The ratio of EPT abundance (numbers of individuals) to Chironomidae (C) abundance ($R = EPT/[EPT + C]$) has also been used as a stream water quality indicator (Resh and Grodhaus, 1983). It is calculated only for replicate Surber samples. In general, EPT taxa are relatively pollution intolerant, whereas Chironomidae are generally pollution tolerant. Thus, higher values indicate better stream water quality.

Taxa richness - Taxa richness is simply the sum of the number of taxa represented in a sample replicate. Richness can be a useful criterion to describe the biological quality of a stream (Resh and Grodhaus, 1983). Low richness may indicate that a stream has been subjected to one or more stresses. This protocol uses both family level and genus richness.

Taxa diversity - Diversity is a measure of how the total number of individuals in a sample are distributed among the total species in the sample. Maximum diversity occurs in a

community when the number of individuals is distributed as evenly as possible among species (Pielou, 1966). High diversity indicates better stream quality (Resh and Jackson, 1993). This protocol uses the Shannon Index (or Shannon-Wiener diversity index) (H') to estimate taxa diversity.

Genus Evenness - This metric, also known as Shannon's Evenness Index, is a measure of how evenly the total number of individuals in a sample are distributed among the genera. Lower evenness indicates that a stream may have been subjected to disturbance and it is being populated by fewer, pollution tolerant genera. This index is calculated using the values of the Shannon Diversity index.

Biotic Indices - Biotic indices are commonly used as indicators of water quality (Resh and Jackson, 1993; Resh and McElroy, 1993). In fact, Jones *et al.* (1981), simultaneously measured invertebrate community structure and water quality variables in Missouri Ozark streams and found biotic indices to be more sensitive and less variable than diversity indices for discriminating differences in stream water quality. The Hilsenhoff biotic index (HBI) uses taxa specific (*e.g.*, family, genus) pollution tolerance values (Hilsenhoff, 1982, 1988; Lenat, 1993) to calculate index scores, which can then be related to stream water quality. This protocol uses genus level tolerance values thus allowing for a better estimate of community tolerance.

The metrics listed above are generally considered sufficiently sensitive to detect a variety of potential pollution problems in network streams. Some of the potential disturbances and the metrics that can be used to detect them include (after Doisy and Rabeni, 1999):

- Gross organic pollution- Hilsenhoff (1982) listed all previous metrics as indicators of gross organic pollution.
- Agriculturally developed catchments- Ephemeroptera and Plecoptera have shown reductions in abundance or richness (Quinn and Hickey, 1990; Lenat and Crawford, 1994).
- Increases in acidity- Taxa richness, EPT taxa, and the Shannon Diversity Index typically decrease in response to increasing acidity (Hildrew *et al.*, 1984; MacKay and Kersey, 1985). Mayflies are especially sensitive to low pH (Peterson *et al.*, 1985).
- Effects of logging and clear cutting- Stone and Wallace (1998) found that the North Carolina Biotic Index (NCBI-a modification of the Biotic Index; Lenat, 1993) was the most sensitive to this type of disturbance.
- Heavy metal pollution- Taxa richness and EPT richness (Winner *et al.*, 1980, Chadwick *et al.*, 1986) have been shown to decrease in response to this type of pollution. However, further research indicates that mayflies may decrease in richness and abundance while caddisflies increase under these conditions, resulting in a static EPT. If no difference in the EPT is found, analysis of the richness and percent composition of mayfly taxa should be considered (Doisy and Rabeni, 1999).
- Insecticides- Wallace *et al.* (1996) found that the EPT index easily detected disturbances to a stream treated with certain insecticides.

Water quality for each site can be inferred using these metrics. For metrics that decrease with increasing level of disturbance, higher metric values indicate higher water quality. For metrics that increase with increasing level of disturbance, lower metric values suggest higher water quality.

Data Analysis

In determining the appropriate statistical approaches for this monitoring protocol, it is important to take into account the primary audience of the various reports that will be produced. This audience will consist of park resource managers, park superintendents, and other park staff within the Heartland Network. Park resource managers and staff may not have an in-depth background in statistical methods, and park superintendents may have limited time to devote to such reports. Additionally, protocols such as this may provide a large amount of data on many different types of variables. Thus, it is important, to the extent possible, that core data analyses and presentation methods provide a standard format for evaluation of numerous variables, are relatively straightforward to interpret, can be quickly updated whenever additional data become available, and can be used for many different types of indicators, whether univariate or multivariate. Additionally, the type and magnitude of variability or uncertainty associated with the results should be easily discernible, and a threshold for potential management action ideally will be indicated.

There are three main statistical approaches that could be employed with data from long-term monitoring projects such as this: (1) hypotheses testing, (2) parameter estimation, and (3) application of Bayesian methods.

Bayesian methods

When analyzing ecological data, statisticians predominantly employ frequentist methods, and thus many resource managers are not familiar with the interpretation of Bayesian approaches. Bayesian methods are not widely used because they are often difficult to apply, and many researchers are not comfortable specifying subjective degrees of belief in their hypotheses (Utts, 1988; Hoenig and Heisey, 2001). Therefore a Bayesian approach is therefore not advocated as the main method of data analysis.

Hypothesis testing

Most hypothesis testing approaches involve a null hypothesis of no difference or no change. The problem with such approach is that the hypothesis under test is thus trivial (Cherry, 1998; Johnson, 1999; Anderson *et al.*, 2000, 2001). No populations or communities will be exactly the same at different times. Thus, the primary interest of this protocol is the magnitude of change rather than change *per se*, and whether it represents something biologically important. Null hypothesis significance testing relies heavily on *P*-values, and results primarily in yes – no decisions (reject or fail to reject the null hypothesis). *P*-values are strongly influenced by sample size, however, and one

may, with a large enough sample size, obtain a statistically ‘significant’ result that is not biologically important. Alternatively, with a small sample size, one may determine that a biologically important result is not statistically significant (Yoccoz, 1991). Thus, traditional null hypothesis testing places the emphasis on the *P*-value (which is dependent on sample size) and rejection of the null hypothesis, whereas we should be more concerned whether the data support our scientific hypotheses and are practically (*i.e.*, biologically) significant (Kirk, 1996; Hoenig and Heisey, 2001).

Parameter estimation

This approach provides more information than hypothesis testing, it is more straightforward to interpret, and easier to compute (*e.g.*, Steidl *et al.*, 1997; Gerard *et al.*, 1998; Johnson, 1999; Anderson *et al.*, 2000, 2001; Colegrave and Ruxton, 2003; Nakagawa and Foster, 2004). Parameter estimation emphasizes the magnitude of effects, and the biological significance of the results, rather than making binary decisions (Shaver, 1993; Stoeckl, 1999). Moreover, trend studies should focus on description of trends and their uncertainty, rather than hypothesis testing (Olsen *et al.*, 1997). Thus, most of the data analyzed under this protocol will take the form of parameter estimation rather than null hypothesis significance testing.

Control Charts

We will also employ control charts in data organization and analysis. Control charts represent a basic summary for almost any data set, a sort of ‘quick look’ for busy managers to determine which variables are in the greatest need of more in-depth analyses or management action (Morrison 2008). Developed for industrial applications, control charts indicate when a system is going ‘out of control’, by plotting through time some measure of a stochastic process with reference to its expected value (*e.g.*, Beauregard *et al.*, 1992; Gyron, 2001; Montgomery, 2001). Control charts may be univariate or multivariate, and can represent many different types of variables. Control charts have been applied to ecological data (McBean and Rovers, 1998; Manly, 2001), including fish communities (Pettersson, 1998, Anderson and Thompson, 2004) and natural resources within the I&M program (Atkinson *et al.*, 2003). Control charts contain upper and lower control limits specifying thresholds beyond which variability in the indicator reveals a biologically important change is occurring, and warns that management may need to act. Control limits can be set to any desired level.

Multivariate control charts may also be constructed, and although some of the above-mentioned texts describe multivariate control charts (using the Hotelling T^2 statistic), this approach is only practical for a small number of variables, and assumes a multivariate normal distribution. In general, species abundances are not distributed as multivariate normal (Taylor, 1961), and traditional multivariate procedures are frequently not robust to violations of this assumption (Mardia, 1971; Olson, 1974). A new type of multivariate control chart has recently been described for use with complex ecological communities and a software application entitled *ControlChart.exe* is available for constructing these types of multivariate control charts (see Anderson and Thompson, 2004). Multivariate

temporal autocorrelation will violate the assumption of stochasticity upon which this method is based, however, it is important to test for temporal autocorrelation using Mantel correlograms prior to using this method. This new multivariate control chart appears to have promise but has not been widely applied nor thoroughly evaluated. Further evaluation of this method is warranted before being applied to the data of this protocol.

Power Analysis

A formal power analysis for this protocol was not conducted for three reasons: (1) The primary purpose of conducting a prospective power analysis is to determine whether the proposed sample size is adequate. There already exist a number of studies indicating that three samples per riffle is an appropriate number for calculation of the proposed metrics (see “Number of samples” below). Because the sample size described above is driven primarily by budget, an increase in the number of riffles sampled per reach or number of reaches could not be justified regardless of the result of any power analysis. Furthermore, in many analyses sample size will equate with number of years; in this case, analyses will simply become more powerful over time. (2) Statistical power is dependent upon the hypothesis under test and the statistical test used. Over the course of this long-term monitoring program, we will be interested in many different questions and could potentially evaluate a number of different hypotheses. Thus, there is no single ‘power’ relevant to the overall protocol. Estimating power at this point in the context of such a long-term, multifaceted monitoring program could be potentially misleading, as the test this power is based upon may rarely (or never) actually be employed. (3) Most of our data analyses will take the form of parameter estimation rather than null hypothesis significance testing. When estimating parameters, there is no associated statistical power. In general, statistical power analyses are frequently mis-used and misinterpreted in ecological contexts (Morrison, 2007), and alternative approaches to evaluating the degree of uncertainty associated with our data will be evaluated and used when applicable.

Other Statistical Tests

Although our primary approach to organizing and analyzing data will consist of multimetric indices, we do not entirely rule out the use of any statistical methods at this time. Because of the nature of this long-term monitoring program, other approaches may be appropriate at different points in time. Depending upon the needs of the resource managers and questions of interest a hypothesis testing framework may be employed. Because data from studies of aquatic insects is often not normally distributed, non-parametric approaches may be necessary. For example, if it is desirable to test for differences between riffles, non-parametric tests should be used (*e.g.*, Kruskal-Wallis Test, Friedman's Test, or Cochran's Q test). Of course, normality of the data will be evaluated prior to any tests, and transformations may be performed if useful prior to tests requiring normal distributions. These approaches and others are described in SOP#8.

Reporting

Reports summarizing monitoring data collected during the year will be prepared within one calendar year. Reports will include the individual metrics calculated for each stream. These reports will include an update on the status of the resources in addition to documenting related data management activities and data summaries. In an effort to disseminate findings in a timely manner, annual summary reports will be completed by September 30th of the year following data collection. Summary reports may be used in place of annual reports for the year data were last collected. Comprehensive trends analysis and synthesis reports will be prepared following at least 3 collecting events depending on observed impacts in the watershed and how critical summary information is for setting management goals influencing stream condition. Executive summaries will be prepared for all types of reports. Refer to SOP#9 “Data Reporting” for details on types of reports and their primary audiences, report structure and style, and review procedures.

VI. Personnel Requirements and Training

Roles and Responsibilities

The project manager is the Aquatic Program Leader for the HTLN and this person bears responsibility for implementing this monitoring protocol. Because consistency is essential to implementation of the protocol, the project manager will usually lead field data collection efforts unless technicians have several years of experience collecting the data related to this protocol as determined by the project manager. The project manager will oversee all laboratory work including all QA/QC requirements. The data management aspect of the monitoring effort is the shared responsibility of the project manager and the data manager. Typically, the project manager is responsible for data collection, data entry, data verification and validation, data summary, analysis, and reporting. The data manager is responsible for data archiving, data security, dissemination, and database design. The data manager, in collaboration with the project manager, also develops data entry forms and other database features as part of quality assurance and automates report generation. The data manager is ultimately responsible to ensure that adequate QA/QC procedures are built into the database management system and appropriate data handling procedures followed. Technicians will be responsible for field collection and laboratory processing, equipment maintenance, purchasing of supplies, and sample storage. At least one technician with taxonomic experience will be responsible for identification of specimens to the genus level.

Qualifications and Training

Training is an essential component for collection of credible data. Training for consistency and accuracy should be emphasized for both the field and laboratory aspects of the protocol. SOP#2 “Training for Field Sampling and Laboratory Processing” describes the training requirements for new technicians. The project manager should

oversee this training and ensure that each technician is adequately prepared to collect data. Taxonomic identifications may be performed by a technician with several years of experience, but initial identifications should be checked by expert taxonomists.

VII. Operational Requirements

Annual Workload and Field Schedule

Streams scheduled for sampling as listed in the revisit schedule (Table 3) will be sampled once during the appropriate index period (Table 2). Samples should be collected within the shortest time frame possible. At minimum, three people will be required to complete the field sampling portion of the protocol; however, four people make the process much more efficient. Only one site can be sampled per day under normal circumstances.

Laboratory processing time per benthic sample, including sorting, identification, and counting, requires approximately 5 hours per sample. Database entry requires an additional hour per site. These tasks will be accomplished by the Program Aquatic Ecologists and other personnel as determined by the Program Leader.

Facility and Equipment Requirements

Field and lab equipment listed in SOP#1 “Preparation for Field Sampling and Laboratory Processing” are only for one sampling crew. Beyond normal office and equipment storage space, access to a wet laboratory is required.

Startup Costs and Budget Considerations

Estimated costs for conducting invertebrate monitoring in small streams in the HTLN are shown in Table 4. Personnel expenses for fieldwork are based on a crew of three: a professional Aquatic Ecologist or Fisheries Biologist to oversee the fieldwork and data collection and to coordinate surveys, and two seasonal biological science technicians or others to assist in field data collection. Assistance with field work from other agencies is always welcome to the extent it is available. Field costs may vary somewhat from year to year depending on the skill level and size of crew. Data management personnel expenses include staff time of biological science technicians, the Aquatic Ecologist, and Data Manager.

VIII. Procedures for Protocol Revision

Revisions to both the Protocol Narrative and to specific Standard Operating Procedures (SOPs) are to be expected. Careful documentation of changes to the protocol and a library of previous protocol versions are essential for maintaining consistency in data collection and for appropriate treatment of the data during data summary and analysis. The Microsoft Access database for each monitoring component contains a field that identifies which version of the protocol was being used when the data were collected.

Table 4. Estimated costs for conducting annual monitoring at small streams in the Heartland I&M Network.

Budget Item	Estimated Costs
Personnel Costs	\$51,768
Administrative support to WICR	\$764
Overhead to Missouri State University	\$325
Field Work Travel	\$1,877
Computer Hardware & Software	\$494
Vehicle Lease	\$1,186
Field/Office Equipment	\$889
Supplies	\$593
Lab Fees	\$1,300
TOTAL	\$59,196

The rationale for dividing a sampling protocol into a Protocol Narrative with supporting SOPs is based on the following:

- The Protocol Narrative is a general overview of the protocol that gives the history and justification for doing the work and an overview of the sampling methods, but that does not provide all methodological details. The Protocol Narrative will only be revised if major changes are made to the protocol.
- SOPs, in contrast, are very specific, step-by-step instructions for performing a given task. They are expected to be revised more frequently than the protocol narrative.
- When an SOP is revised it usually is not necessary to revise the Protocol Narrative to reflect the specific changes made to the SOP.
- All versions of the Protocol Narrative and SOPs will be archived in a Protocol Library.

The steps for changing the protocol (either the Protocol Narrative or the SOPs) are outlined in SOP#11, “Revising the Protocol.” Each SOP contains a Revision History Log that should be filled out each time a SOP is revised to explain why the change was made, and to assign a new Version Number to the revised SOP. The new version of the SOP or Protocol Narrative should then be archived in the HTLN Protocol Library under the appropriate folder.

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X. Standard Operating Procedure (SOPs)

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 1: Preparation for Field Sampling and Laboratory Processing

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP provides information to prepare for the field season, including required field and laboratory equipment lists. It also provides a description for recording information on staff time spent during sampling trips, checking water levels at the parks, and obtaining collecting permits. A list of required data sheets with a brief explanation of their purpose is provided.

I. General Preparations

Prior to the field season all crew members will review the entire protocol, including SOPs. The following list includes key points to consider in preparing for the upcoming field season.

The team leader of each sampling crew must prepare a field notebook for the survey year. The notebook should contain entries for observer names, field hours and unique events that may influence how the data is reported. Information included in trip reports is based on what is recorded in field notebooks so it is imperative that they are clearly organized for ease of field note entry. Notebook entries should be recorded daily to ensure accuracy. An example of a notebook log is shown in Fig. 1.

2. Inclement weather and personnel workloads will preclude the scheduling of sampling events to specific annual dates. Sampling dates should be scheduled and logistics organized prior to the start of each field season. Monitoring efforts will require a three person crew (two people to sample and one to record data and provide general assistance). Allow at least one day per site. At HOME, one day is allotted for deploying Hester-Dendy samplers, and a second day is allotted to retrieving samplers following 30 days post-deployment.

Date	Travel time (hours)	Field time (hours)	Non-project time (hours)	Lunch (hours)
30 Sep 2006	3	8	0	0.5
1 Oct 2006	0	8	1	0.5
2 October	3	8	0	0.5
Notes: J. Smith and H. Simpson traveled to PIPE to conduct invertebrate monitoring at Pipestone Creek . Field assistance was provided by B. Jones and K. Adams. Returned to headquarters. Non-project time included discussing other projects with park staff.				

Figure 1. Example of a field note book for recording scheduling, travel and field time, and personnel information.

3. An equipment list will be compiled and equipment organized and made ready for the field season several weeks prior to the first sampling tour to make sure that all supplies are available and equipment is in working condition. This allows time to make required repairs and order replacement equipment. Inspect the sample nets and wash bucket to ensure there are no tears in the nets or screen. Ensure water quality meters can be calibrated and are properly functioning as described in SOP #4. The following lists of equipment and supplies are for field and laboratory use.

Table 1. Field equipment required for monitoring aquatic invertebrates.

Number Req.	Description
1	Surber sampler with 500µm mesh net
10	Hester-Dendy multiplate samplers (HOME only)
As needed	Nylon rope or wire for securing Hester-Dendy plate samplers
1	Nylon scrub brush
1	Hand rake or garden cultivation tool
Varies per site	Labeled sample bottles (Plastic 500-ml wide mouth bottles; 9 bottles per stretch)
Varies per # of samples	Preservative-99% isopropyl alcohol and 1-L plastic Nalgene bottles for carrying; bring extra quantities to ensure enough is available to preserve all samples. In total, approximately 1 gallon will be required per stream sampled. Label container "flammable"
2	Forceps
2	Polypropylene wash bottles
1	U.S. Standard sieve, 500-µm mesh size
2	5-gal white buckets
1	Benthic sample wash bucket with 500-µm mesh size
1	Plastic tube or pan
2	Laminated plastic sheet with Wentworth scale codes for conducting substrate assessment
1	Clip board
5	Pencils
See Table 3	Data sheets printed on waterproof paper, extra sample bottle labels and tape
1	Tape measure
1	Range finder
1	GPS unit
1	Digital camera
1	Field log book
	Directions to sample sites, sample site maps, list of GPS coordinates
1/person	Waders and boots
1/ person	Life jackets
1	Velocity meter and wading rod
1	Water quality datalogger equipped with temperature, pH, conductivity meter, dissolved oxygen, and turbidity probes
	Extra batteries for water quality meters
	Buffer solution for pH meters (pH 7 and 10)
1	Backpack for carrying equipment

Table 2. Laboratory equipment required for processing invertebrate samples.

Number Req.	Description
1	U.S. Standard sieve 500- μ m mesh sieve; marked into 4 equal-sized sections
1	Shallow white pan for sorting; 8X10 inches or other suitable size
1	Petri dish or similar sorting container
3	Multi-well Petri dishes for separating sorted insects
1	Dissecting microscope and binocular microscope
Several	Forceps (fine point)
Several	Clean plastic bottles with labels inside and out for storage of processed samples
	Preservative for sorted invertebrates (70% isopropyl alcohol)
Several	Storage vials with labels for reference collection specimens
Several	Pencils or fine point pen with waterproof ink
	Data sheets

II. Field and Laboratory Forms

Copies of field sheets and labels should be printed on waterproof paper. Data should be recorded with waterproof ink or #2 lead pencil. Example data sheets are provided as attachments to their corresponding SOPs.

Table 3. List of data sheets required for invertebrate monitoring for the sampling season.

TITLE	PURPOSE	NUMBER OF COPIES NEEDED PER PARK
Stream field sampling form	Recording physical-chemical data for riffles	1
Discharge form	Recording stream discharge	1
Sample labels	Identifying individual samples	1 per sample collected
Aquatic invertebrate identification & enumeration sheets	Recording names of identified taxa and their densities	Variable

III. Flooding and Weather

Before deploying to a sample site, staff should consult with the National Weather Service (<http://www.nws.noaa.gov/>) and Park Natural Resource Specialists to ensure that no floods have occurred in the last two weeks that could hinder sampling or affect sampling efficacy. Similar information can be obtained from the USGS (<http://water.usgs.gov/>). If flooding has occurred, sampling should not take place until two weeks after flood waters have receded.

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 2: Training for Field Sampling and Laboratory Processing

Version 1.0 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP explains the training procedures for using field equipment properly, collecting habitat data, and processing invertebrates in the laboratory. This training ensures a high level of consistency among samplers and processors. Prior to training, all personnel will review the general protocol and each SOP. Someone familiar with the protocol and experience with the sampling and processing procedures should supervise the training.

I. Field Sampling and Habitat Data Collection

Procedure:

1. Find a nearby stream to practice collecting benthic samples and habitat data. Follow the procedures outlined in SOP#3 “Sampling Invertebrates and Collecting Habitat Data” and collect several practice samples for processing in the lab. Ensure that each person is comfortable with all aspects of the sampling routine.
2. After collecting the samples, practice using the water quality instruments. Each person should be familiar with the instruments and be able to calibrate each instrument as outlined in SOP#4 “Documenting CORE 5 Water Quality Variables.” Practice discharge measurements following procedures described in SOP #5 “Measuring Stream Discharge.”
3. Once initial training is completed, annual re-training for veteran crews is not required.

II. Laboratory Processing

Procedure:

1. Follow steps outlined in SOP#6 “Laboratory Processing and Identification of Invertebrates” using practice samples collected through field sampling training.
2. Material left over after sorting is completed should be checked by someone skilled in processing benthic samples to determine recovery efficiency. The number of invertebrates recovered should be expressed as a percentage of the total number of invertebrates. The standard for recovery is 95%.

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 3: Sampling Invertebrates and Collecting Habitat Data

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP describes field procedures for selecting riffles to sample within a stretch and the process for randomly selecting sample points within a riffle. The SOP further explains procedures for collecting and processing invertebrate samples in streams, and for collecting associated habitat data. Appropriate QA/QC requirements are highlighted within the procedure descriptions.

I. Riffle Selection within a Sample Reach

1. The sample unit is a reach of contiguous stream as defined in the protocol narrative. Sampling and habitat analysis will be restricted to three riffles in this reach (two sites at HOME).
2. Locate the lower boundary of the sample reach using the UTM coordinates shown in Table 1. The following general descriptors and site maps in Appendix B also are useful for locating the sampling reaches.

EFMO

Dousman Creek. Approximately 200 m upstream from the confluence with the Yellow River.

GWCA

Carver Creek. The first sampled riffle is about 15 m downstream of the west or downstream-most visitor's trail crossing Carver Creek.

Harkins Branch. The first sampled riffle is located immediately upstream of the downstream (west) Park boundary fence.

Williams Branch. The first sampled riffle is about 130 m downstream of the west trail crossing Williams Branch.

HOME

Cub Creek. *Upstream site:* SW corner of park downstream of bridge. *Downstream site:* 50 m NE of Highway 4 bridge

HEHO

Hoover Creek. Approximately 125 m upstream of Parkside Drive in West Branch, Iowa.

HOSP

Bull Bayou. Approximately 250 m upstream of western park boundary where the stream exits the park.

Gulpha Creek. Approximately 250 meters upstream of the southern park boundary where the stream exits the park.

PERI

Pratt Creek. Approximately 100 m upstream of southern park boundary where the stream exits the park.

Winton Spring Branch. Immediately inside the south boundary of the park upstream of its confluence with Pratt Creek.

PIPE

Pipestone Creek. 25 m below Lake Hiawatha directly above Circle Trail crossing.

TAPR

Fox Creek. Approximately 600 m due west of Highway 177 immediately across from the historic house and barn.

Palmer Creek. Approximately 1,200 m upstream from confluence with Fox Creek.

WICR

Skegg's Branch. Approximately 30 m upstream of the park tour road crossing the stream.

Wilson's Creek. Located above the Old Wire Road bridge foundation upstream of the confluence with Terrell Creek. Upstream to about 25 m south of second crossing of park road loop.

Terrell Creek. The first sampled riffle is located about 70 m upstream of Hwy ZZ bridge.

Table 1. The streams that will be sampled in each network park and the UTM coordinates of the lower sampling stretch boundaries.

Park	Streams sampled	UTM Coordinates (Northing, Easting)	Index Period
GWCA	Carver Creek Harkins Branch Williams Branch	4094380.11, 379254.85 4094493.46, 378963.70 4094466.25, 379268.19	May-June
EFMO	Dousman Creek	4772108.08, 645475.84	July-August
HEHO	Hoover Creek	4614462.87, 637697.89	July-August
HOME	Cub Creek	4462337.67, 684059.84	July-August
HOSP	Bull Bayou Gulpha Creek	3819096.45, 489743.19 3820036.11, 496779.10	May-June
PERI	Pratt Creek Winton Spring Branch	4033256.21, 407127.86	May-June
PIPE	Pipestone Creek	4877259.61, 714204.77	July-August
TAPR	Fox Creek Palmer Creek	4256985.51, 713944.53 4263176.10, 710907.56	August-September
WICR	Skegg's Branch Terrell Creek Wilson's Creek	4105745.65, 463391.47 4104000.832, 462818.328 4104580.870, 464167.047	May- June

3. Riffle selection was determined *a priori*, with the three riffles sampled being those located in consecutive order upstream of the first riffle above the lower reach boundary Fig. 1.

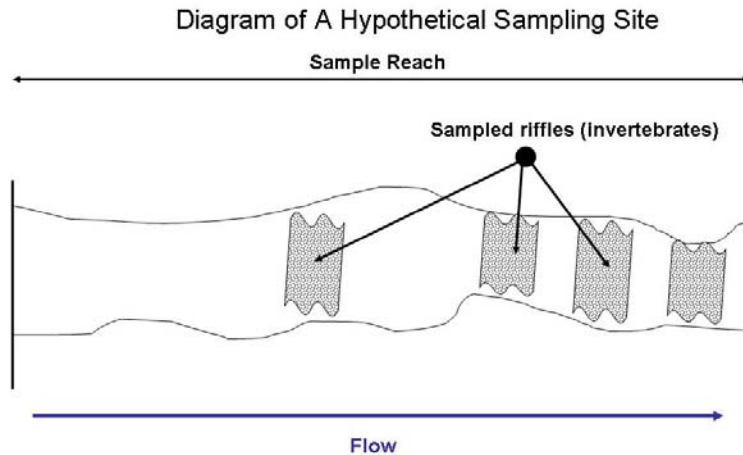


Figure 1. Diagram of riffle location within a sample reach.

- a. For Cub Creek, located at HOME, the sampling areas are the upper and lower reaches of the stream as it flows through the park (Fig. 2).

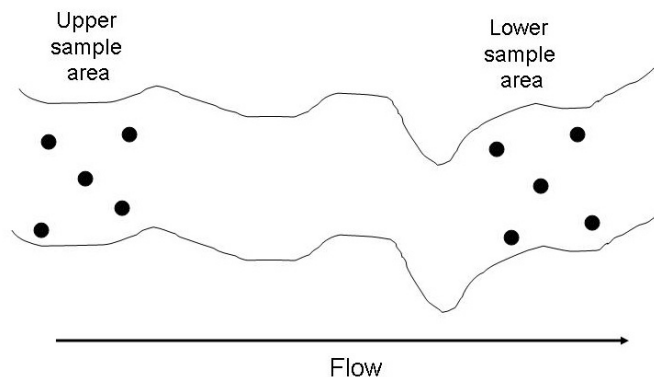


Figure 2. Diagram showing placement of Hester-Dendy samplers in Cub Creek, HOME.

- b. Specific methods for placing samplers are described below.

II. Collecting Benthic Samples and Associated Habitat Data from Riffles

Procedure:

1. These procedures apply to streams at GWCA, HEHO, PERI, PIPE, TAPR, and WICR. Instructions for HOME are shown below.
2. Prior to collecting benthic samples and taking habitat measurements, always complete data sheet information for park code and stream name (*e.g.*, PIPE, Pipestone Creek), date and time of survey, and initials of personnel who collect the samples.
3. Deploy datasonde for taking continuous CORE 5 water quality readings using the procedures described in SOP#4.

4. Collect three benthic samples per riffle. Benthic invertebrate samples will always be taken in an upstream direction. Individual benthic samples from riffles will be collected using the following *a priori* randomization procedure:

- a) The sampling area is divided into three equal portions based on the measured length of the riffle as shown in Fig. 3. First and foremost, the effective sampling area of the riffle will be based on safety of personnel, accessibility, and other pertinent factors determined by the investigators' best judgment at the time of sampling. The upper and lower riffle boundaries generally will be based on a visual assessment of gradient, velocity, and substrate characteristics. Care should be used to avoid including the deeper, downstream portion of the riffle as it transitions to deeper run habitat with more of v-shaped channel.

- b) Collect individual benthic samples starting at river left (1/4 point), then alternating to the middle, and finally the right (3/4 point). Samples will be collected at the approximate midpoint of each portion. Indicate sampling sequence on the data sheet (L, M, or R). Approximate position of the samples is shown in Fig. 3.

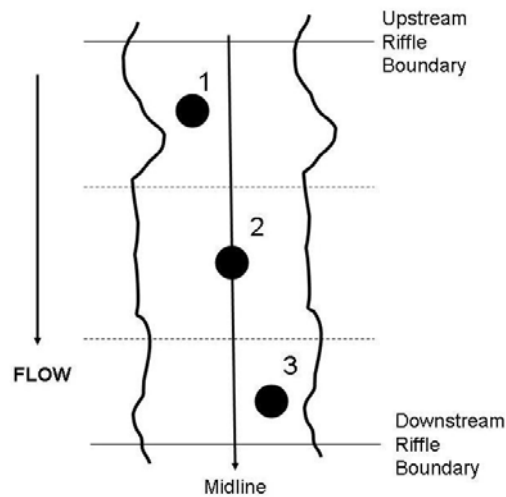


Figure 3. Diagram of sample locations within a riffle.

Note: Sample sequence should be altered only if the original starting point presents danger to the collector or if it is not accessible. Some riffles may be wider than long and, in these instances, samples can be taken from left to right in equally spaced increments.

- c) Samples will be collected with a Surber stream bottom sampler (~500 μm mesh, 0.093 m^2 sampling area) (Fig. 4). Place the sampling frame firmly against the stream substrate so that the opening is oriented directly into the stream current.

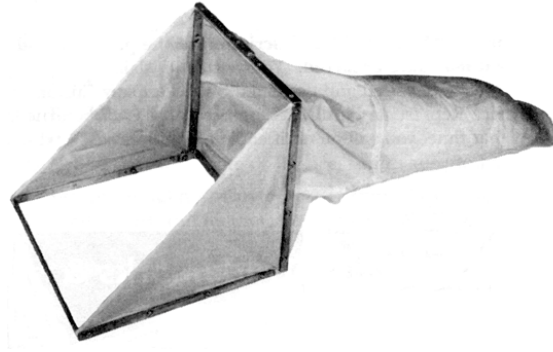


Figure 4. Surber stream bottom sampler.

5. For each benthic sample, record habitat measurements based on visual estimates from within the sample frame on the habitat data form. Percentage categories are: 0= none (0%), 1= sparse (<10%), 2= moderate (10-40%), 3= heavy (40-75%), 4= very heavy (>75).
 - a. percent embeddedness of the substrate (*e.g.*, bedrock & hardpan clay = 0% embeddedness; sand, clay, silt = 100% embeddedness)
 - b. percent periphyton
 - c. percent filamentous algae
 - d. percent sedimentation
 - e. percent organic material.

6. Record dominant substrate size within the sampling area.

Note: This technique is used to assess the dominant substrate in the sample area. It is not intended to fully characterize the substrate profile of the stream bottom. Substrate is not collected from Cub Creek at HOME since Hester-Dendy samplers are used at this park. The substrate at HOME consists primarily of sand.

- a. After the net and sample frame have been placed on the stream bottom, visually estimate the dominant substrate size as an average of three randomly (*i.e.*, blind touch) selected substrate pieces taken from within the sampling frame.
- b. The Wentworth Scale (Table 2) is used for assessing substrate size.
- c. A Wentworth Scale category field sheet (provided at the end of this SOP) may be used for rapid measurement of substrate in the field. A piece of substrate belongs to the smallest size box that it will fit through on any axis.
- d. Record the average dominant substrate size code on the field sheet.

Table 2. Substrate size classes to be used for characterizing substrate based on the Wentworth Scale.

Size Code	Particle Diameter Range (mm)	Category
1	<0.062	Silt/clay
2	0.062-0.125	Very fine sand
3	0.125-0.25	Fine sand
4	0.25-0.50	Medium sand
5	0.50-1	Course sand
6	1-2	Coarse sand
7	2-4	Fine gravel
8	4-5.7	Medium gravel
9	5.7-8	Medium gravel
10	8-11.3	Coarse gravel
11	11.3-16	Coarse gravel
12	16-22.6	Small pebble
13	22.6-32	Small pebble
14	32-45	Large pebble
15	45-64	Large pebble
16	64-90	Small cobble
17	90-128	Small cobble
18	128-180	Large cobble
19	180-256	Large cobble
20	256-362	Boulder
21	362-512	Boulder
22	512-1024	Boulder
23	>1024	Boulder
24	Bedrock	Bedrock

7. Record current velocity and depth.

Average velocity and depth are measured concurrently at each sample point, immediately in front of the sample frame (Fig. 5). Depth and velocity can be measured by a third team member while the other two members collect the benthic sample described in step 8 below. Measurements are taken using a current meter attached to a top-setting wading rod. The rod allows for quick and easy measurements of depth with incremental (cm) markings and an adjustable arm that places the current meter at the proper depth for measuring velocity (60% of the depth from the surface of the water). Velocity should be recorded in meters per second. Greater detail regarding use of current meters and the wading rod is provided in SOP#5 “Measuring Stream Discharge.”



Figure 5. Measuring depth and current velocity in front of the collecting net.

8. Benthic sample collection (Surber Sampler)

- a. While one team member holds the net firmly against the stream bottom facing the current, a second team member examines all large substrate pieces and scrubs them with a soft brush in order to dislodge any attached invertebrates so they are carried into the collecting net (Fig. 6). Forceps may be needed to remove snails and attached caddisflies. Discard the substrate outside the sampling area once scrubbing is completed. When this step is completed, a garden cultivation tool is used to agitate the entire area within the net sample frame for a timed period of 2 minutes. At the end of 2 minutes the net can be lifted from the stream bottom, but with sufficient caution so as not to spill the sample.



Figure 6. Collecting a Surber sample.

- b. Large debris or rocks should be inspected for attached organisms and then removed from the sample. Pour and rinse the contents of the net into a wash

bucket with a 500 μ m mesh sieve and rinse the sample by swirling the bucket in water using care not to submerge the bucket. Continue this process until all the fine silt and other sediments have been washed from the sample. Remove the sample contents from the wash bucket and place them in the sample container (Fig. 7). Inspect the net, bucket, and sieve for any remaining organisms and carefully place them in the sample container. Excess water in the sample container can be drained into the sieve and inspected for organisms so that the preservative is not overly diluted.



Figure 7. Transferring the sample from the wash bucket to the sample container.

- c. Once all organisms have been removed from the net, fill the jar with preservative (99% isopropyl alcohol), ensure that the container is properly labeled (example sample labels are provided at the end of this SOP), and tightly close the lid. Sample debris must be completely covered by preservative. Labels must be written in waterproof ink on 100% ragbond paper.
- d. Repeat this procedure for each discrete sample. Prior to leaving the site, recheck the samples to ensure they have been properly labeled and tightly closed, and ensure data sheets are properly completed.

9. Hester-Dendy sample collection

- a. Hester-Dendy samplers (Fig. 8) are used only at Cub Creek, HOME.



Figure 8. Hester-Dendy multiplate sampler.

- b. Hester-Dendy samplers require a deployment and retrieval trip to complete one invertebrate sample. The samplers should be spaced at least one meter apart in the deployment area. The samplers are deployed using the nylon rope, tied to the eye-bolt, and suspended from sturdy woody vegetation (*e.g.*, stable root wads) or other support, such that they are submerged at least 6 cm above the streambed, and 6 cm below the surface. Flotermersh (2006) recommends exposure periods of four to six weeks to allow for colonization of biofilm and subsequent invertebrate fauna—a 30 day exposure period is used in this protocol. Samplers are usually deployed at 1- to 3-m depths. Deployment depth is chosen so that receding or rising waters during the exposure period will not leave samplers dry, buried in mud, or too deep to retrieve. Typically, 4 or 5 Hester-Dendys are placed per sampling reach (Flotermersh 2006). Placing multiple samples per reach and compositing data also helps buffer the effects of loss from flooding.
- c. Because Hester-Dendy samplers are an entirely artificial sampling medium, habitat data as described above excluding CORE 5 data, are not collected.
- d. When the samplers are retrieved, they should be slowly lifted to the water's surface and immediately placed in a wash bucket (500 μm screen mesh) to capture any escaping invertebrates. Transfer the samplers and collected debris to a small bucket or plastic tub to facilitate scrubbing the sampler plates (Fig. 9). Inspect the bottom of the wash bucket to ensure no invertebrates remain. Remove the wing-nut that holds the plates onto the support bolt and separate the individual plates. While the sampler plates are in the plastic tub, carefully scrape or brush each plate to remove attached invertebrates. The contents that have been deposited in the tub are then sieved to remove silt and fine debris and preserved in 99% isopropyl alcohol for laboratory processing.



Figure 9. Procedure for removing attached invertebrates from individual Hester-Dendy plates.

- 10. Record any necessary notes about the collection site or specific samples.
- 11. Take digital photographs of the sampling sites from upstream and downstream perspectives at mid-channel.



The National Park Service

Heartland I&M- Aquatic Invertebrate Monitoring Stream Field Sampling Form

Park Name: _____

Site Name/ID: _____

Date: _____

Team: _____

RIFFLE DATA

Sample #

Sampling sequence (L, M, R)

Depth (cm) (each sample)

Velocity (m/s) (@0.6 depth)

Embeddedness (%)

Vegetation (%)

Filamentous algae (%)

Periphyton (%)

Deposition (%)

Organics (%)

0 = Absent (0%), 1 = Sparse (<10%), 2 = Moderate (10-40%), 3 = Heavy (40-75%), 4 = Very Heavy (>75%)

Dominant Substrate Size

Wentworth Scale (1-24)

RIFFLE #3

3	2	1

--	--	--

RIFFLE #2

3	2	1

--	--	--

RIFFLE #1

3	2	1

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Based on a visual average estimate of three randomly selected substrate pieces for each sample

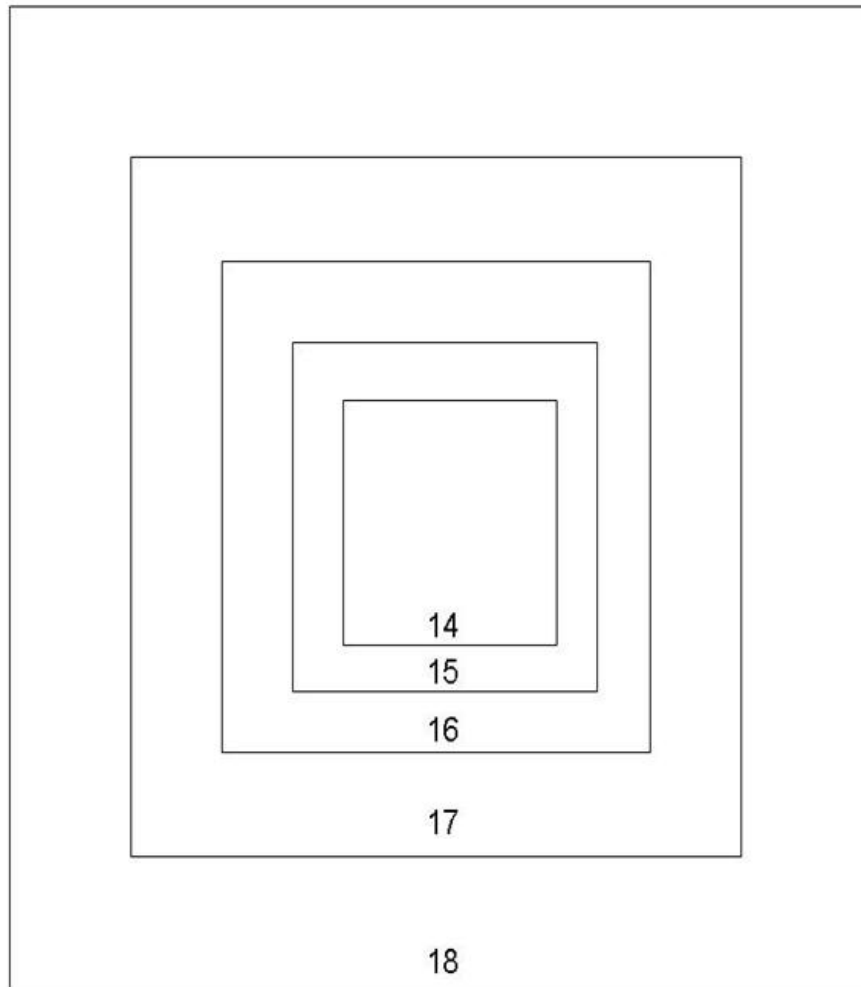
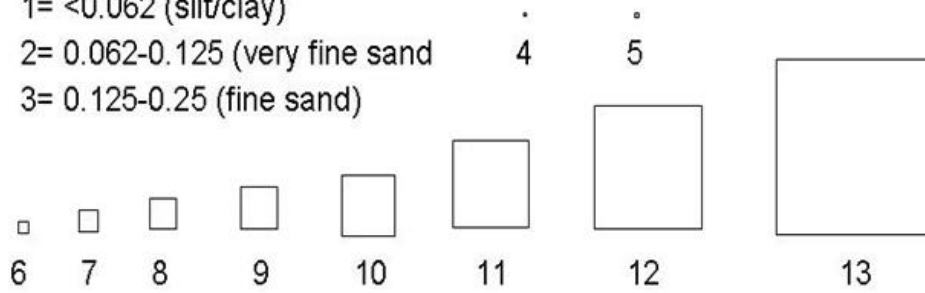
NOTES:

WENTWORTH SUBSTRATE CODES

1= <0.062 (silt/clay)

2= 0.062-0.125 (very fine sand)

3= 0.125-0.25 (fine sand)



19=180-256 mm, 20=256-362 mm, 21=362-512 mm, 22=512-1024 mm, 23= >1024 mm, 24=Bedrock

Labels for Surber samples

Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:	
Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):	
Sample Location (L, M, R):		Sample Location (L, M, R):		Sample Location (L, M, R):	
Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:	
Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):	
Sample Location (L, M, R):		Sample Location (L, M, R):		Sample Location (L, M, R):	
Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:	
Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):	
Sample Location (L, M, R):		Sample Location (L, M, R):		Sample Location (L, M, R):	
Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:	
Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):	
Sample Location (L, M, R):		Sample Location (L, M, R):		Sample Location (L, M, R):	
Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:	
Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):		Riffle Number (1, 2 or 3):	
Sample Location (L, M, R):		Sample Location (L, M, R):		Sample Location (L, M, R):	
Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:	

Labels for Hester-Dendy samples

Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:		Site Name/ID:	
Location (Up/Down)		Location (Up/Down)		Location (Up/Down)		Location (Up/Down)	
Replicate (1-5):		Replicate (1-5):		Replicate (1-5):		Replicate (1-5):	
Date:		Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:		Site Name/ID:	
Location (Up/Down)		Location (Up/Down)		Location (Up/Down)		Location (Up/Down)	
Replicate (1-5):		Replicate (1-5):		Replicate (1-5):		Replicate (1-5):	
Date:		Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:		Site Name/ID:	
Location (Up/Down)		Location (Up/Down)		Location (Up/Down)		Location (Up/Down)	
Replicate (1-5):		Replicate (1-5):		Replicate (1-5):		Replicate (1-5):	
Date:		Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:		Team Members:	
Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring		Heartland I&M Aquatic Invertebrate Monitoring	
Site Name/ID:		Site Name/ID:		Site Name/ID:		Site Name/ID:	
Location (Up/Down)		Location (Up/Down)		Location (Up/Down)		Location (Up/Down)	
Replicate (1-5):		Replicate (1-5):		Replicate (1-5):		Replicate (1-5):	
Date:		Date:		Date:		Date:	
Team Members:		Team Members:		Team Members:		Team Members:	

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 4: Documenting CORE 5 Water Quality Variables

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP addresses the equipment and methods required to measure CORE 5 water quality variables (temperature, dissolved oxygen, specific conductance, pH, and turbidity) in association with all aquatic monitoring in network parks. Detailed guidance for measuring CORE 5 parameters, including training, calibration, QA/QC, data archiving, meter specifications, field measurements, and trouble-shooting, can be found in the Documenting CORE 5 Water Quality Variables SOP located at: <http://science.nature.nps.gov/im/units/htln/fish.cfm>. This SOP is based on guidance from NPS Water Resources Division (2003) and Wagner *et al.* (2006).

I. Unattended CORE 5 measurements

Unattended CORE 5 data will be recorded using dataloggers or sondes. CORE 5 water quality parameters measured with small intervals (*i.e.*, minutes to hours) between repeated measurements are considered continuous because few, if any, significant water quality changes are likely to go unrecorded. The goal is to characterize events of short duration. Because such events are difficult to capture manually using discrete measurements, continuous monitoring is appropriate. Continuous monitoring of core parameters helps address questions concerning daily or seasonal variability, or short-term changes (*e.g.*, precipitation related events) that might not be apparent or may prevent accurate understanding of long-term data. Continuous monitoring also provides the most comprehensive temporal data set for establishing variability through time. Such information is necessary to document correlations, possible cause and effect relationships, and differentiate natural variability from anthropogenic induced change on an aquatic system. We will use loggers exclusively to collect water quality data because they provide more comprehensive data and are easily deployed in these small stream systems. Additionally, discrete samples would duplicate effort and would be considered redundant. Data logging of

CORE 5 parameters ideally will be conducted for a minimum of 24 hours for each stream sampling.

II. Analysis and Reporting

CORE 5 data will be analyzed using summary statistics (mean, median, range, standard deviation, standard error) for each site and date. This information will be presented in summary and synthesis reports to support invertebrate collection data.

III. NPStoret

Collected water quality data that has been successfully subjected to QA/QC will be exported to NPStoret (see SOP#7 Data Management). Only summary data for a site and collection period in addition to pertinent metadata will be submitted. Instructions for preparing and exporting water quality data to this archival facility can be found at the following website:

<http://nrdata.nps.gov/Programs/Water/NPStoret/>

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 5: Measuring Stream Discharge

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP is guidance for measuring discharge in small streams. The SOP describes sampling procedures, calibration, and general maintenance procedures. Field personnel should review the instruction manual for instrument-specific guidance on how to calibrate and operate meters used to measure velocity.

I. Background Information

Velocity and depth are measured using a current meter attached to a wading rod. The rod allows for quick and easy measurements of depth with incremental markings, and has an adjustable arm that places the current meter at the proper depth for measuring velocity (60% of the depth from the surface of the water) (Carter and Davidian, 1969). Some current meters have rotating cups (Pygmy and Price models) while others have a pair of electronic contacts on a small head (FLO-MATE 2000) to measure velocity. Velocity is displayed as either feet per second or meters per second.

Stream discharge (Q) is the volume of water passing a cross-section per unit of time and is generally expressed in cubic feet per second (ft^3/s) or cubic meters per second (m^3/s). Discharge is estimated by multiplying current velocity by the cross-sectional area of the desired reach (Carter and Davidian, 1969). Cross sectional area is determined by first measuring the width of the stream channel. The cross section is then divided into smaller increments (usually 15 to 20 intervals) and depth and velocity are measured at each increment. The depth and width of the interval are multiplied to get an area for each interval and then each interval area and velocity are multiplied to produce a discharge for each interval. These discharges are summed to produce a total discharge for that cross section of the stream. This process will be described in greater, step-by-step detail in the “Procedures” section.

II. Prior to the Field

1. Standard wading rods are available in both metric and English standard units (feet). This protocol will use metric units for recording data. There must be consistency in units between the settings on the velocity meter, the wading rod, and the tape measure, and the units must be clearly recorded on the data sheet. If English standard units are used in the field, the data must be converted to metric units for data entry.
2. Ensure new batteries are placed in units that require them and additional batteries are taken to the field as a back-up.
3. Calibrate velocity meters (FLO-MATE 2000 or USGS pygmy) according to instructions in the manufacturer's operations manuals. Photocopies of the operations manuals should be taken to the field.
4. Equipment maintenance and storage should follow guidance issued by the manufacturers.

III. In the Field

Discharge measurements require wading across the stream and may stir up sediments, disrupting accurate measurement of other parameters. Therefore, discharge should be the last measurement taken at a site.

Quantitative Discharge Procedure:

1. Prior to taking any measurements, the location where discharge will be measured must be determined. An ideal cross-section in the sample reach will have the following characteristics:
 - a. The stream channel directly above and below the cross-section is straight.
 - b. There is measurable stream flow, with a stream depth preferably greater than 10 cm and velocities generally greater than 0.15 meter/second.
 - c. The streambed is a uniform "U" shape, free of large boulders, woody debris, and dense aquatic vegetation.
 - d. The stream flow is laminar and relatively uniform with no eddies, backwaters, or excessive turbulence.

Note: The cross section will not likely meet all these qualifications but the best location should be selected based on these standards.
Record (or draw a diagram) on the data sheet a description of any discrepancies with the cross section.
2. Once the cross section is established, measure the width of the stream with a tape measure to the nearest 0.1 meter and secure the tape across the stream for the duration of the discharge measurement.
3. Divide the stream into equal intervals across the width of the cross section, usually 15 to 20. A minimum of 10 intervals is recommended. A velocity and depth measurement will be recorded for each interval across the stream at the center of each interval. For example,

if the stream is 10 meters wide, 10 velocity and depth measurements will be taken at one meter intervals (Fig. 1).

4. Attach the sensor to the wading rod and ensure that the sensor is securely screwed onto the rod and faces upright.

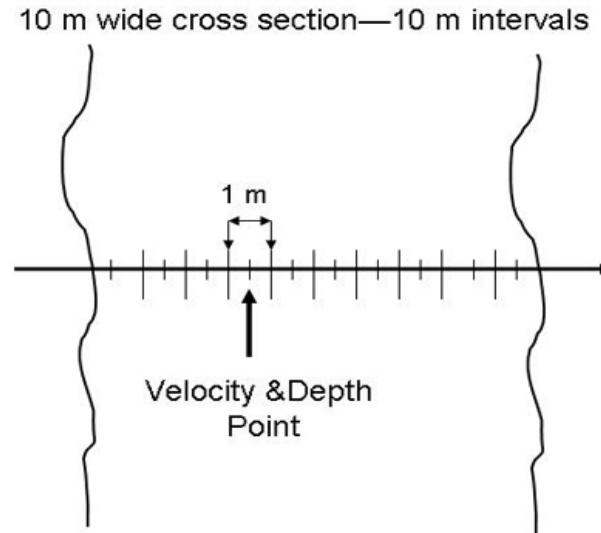


Figure 1. Cross section diagram.

5. One person should measure discharge and one person should remain on the bank, recording data. The first readings are taken at water's edge and are recorded as depth=0 and velocity=0. Proceed to the next interval and record readings. Place the wading rod as level as possible and hold perpendicular to the water level. Read depth from the wading rod to the nearest centimeter. The rod will have graduated marks along its length, with single marks indicating two centimeters, double marks indicating 10 centimeters, and triple marks indicating one-half meter increments.
6. Once depth has been read, adjust the arm of the sliding rod with the sensor attached to 60% of the water depth. The wading rod will place the sensor at 60% of the depth from the surface of the water when properly adjusted.

Note: For example, if the depth is 2.6 meters, line up the 2 on the meter scale (sliding rod) with the 6 on the tenth scale (increments on handle of fixed rod). The sensor is now located at 60% of the water depth.

7. Stand behind the sensor and make sure there is no disturbance (including the sensor cord) around the sensor that interferes with the velocity measurement. The meter may be adjusted slightly up or downstream to avoid boulders or other instream interferences.

Note: Make sure the sensor directly faces the flow of the water. This may not always be directly parallel with the stream bank; the rod and sensor may need to be turned slightly with each measurement.

8. Allow the instrument enough time to get an accurate reading--generally around a minute, or until the reading stabilizes. Watch the time bar complete two full cycles and then take the velocity reading. If something happens during the measurement, such as accidental movement of the wading rod, the reading should be repeated.
9. Call out the distance from the water's edge, the depth, and then the velocity to the person recording data. Continue moving across the stream until measurements have been taken at all intervals.

Note: If the water velocity increases greatly between intervals, additional measurements can be taken to shorten the width of the intervals within this area of high velocity. Be sure to change the interval width for these measurements in the calculation of discharge.

When finished, detach the sensor from the wading rod and place it back in the storage container for transportation. If the meter is not going to be used for several days, remove the batteries, clean the sensor, and store properly.

DISCHARGE

PARK NAME/ID: _____ STREAM NAME: _____

TEAM MEMBERS: _____ DATE: _____

	Distance Units ft ☐ m ☐	Depth Units ft ☐ cm ☐	Velocity Units ☐ ft/s XX.X ☐ m/s X.XX	
Start at River Right; Final measurement should be at river left bank				
	Distance from Bank	Depth	Velocity (@0.6 depth)	FLAG
1	0	0	0	
2				
3				
4				
5				
6				
7				
8				
9				
10				
11				
12				
13				
14				
15				
16				
17				
18				
19				
20				

Measurements are taken at 0.6 the water column depth

Flags:

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 6: Laboratory Processing and Identification of Invertebrates

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP explains procedures for processing and storing samples after field collection as well as identification of specimens. Procedures for storing reference specimens are also described.

I. Preparing the Sample for Processing

Processing procedures apply to all benthic samples. This is an important and time consuming step. Particular care should be taken to ensure that samples are being processed thoroughly and efficiently. The purpose of sorting is to remove invertebrates from other material in the sample.

Procedure:

1. Sample processing begins by pouring the sample into a USGS standard sieve (500- μ m) placed in a catch pan. The preservative should be kept and stored in a storage container for eventual rehydration of sample debris prior to QA/QC.
2. Rinse the sample contents in the sieve with tap water to flush the residual preservative. Large organic material (>2 cm) should be removed by hand and rinsed into the sieve. Each piece of debris removed from the bulk sample should be carefully inspected to ensure that all attached organisms are removed. The rinsed organic material should then be kept separate from the rest of the sample or placed in the original sample container along with the original preservative.
3. Through elutriation, the organic debris should be separated from the inorganic content (sand and gravel).

Note: Several steps are involved with the elutriation process. Begin by pouring all sample material from the sieve into a catchpan. Fill the pan with enough water

so that organic material can float to the top. Pour the water and organic material from the catchpan into a clean 500- μ m USGS-type sieve marked into four equal portions (Fig. 1). Add more water to the sample material in the catchpan and repeat these steps until all organic material has been separated from the inorganic and has been transferred into the sieve.

This may take several washes to accomplish. Carefully examine the inorganic content for the presence of remaining invertebrates (especially mollusk shells and Trichoptera cases). It is suggested that this is done under a magnified light. Add these specimens to the organic debris portion. Return the inorganic portion to the original sample container.

4. Inspect the sieve to ensure no invertebrates remain following rinsing. If present, remove any specimens and add them to the organic sample fraction.
5. The sieve should then be placed in a shallow pan of water, and the contents floated until they are evenly distributed on the pan bottom. The sieve should then be carefully lifted from the water so that contents are not redistributed. The separated sample is now ready for sorting.

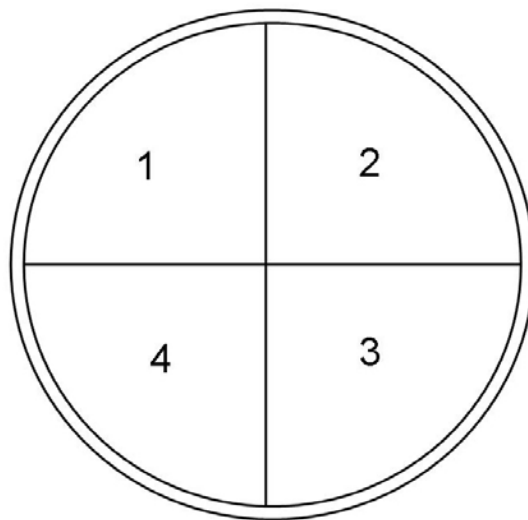


Figure 1. Diagram of a 500- μ m USGS sieve marked into quarter fractions.

Notes regarding any difficulties with sample sorting should be written on the Aquatic Invertebrate Identification & Enumeration Sheet (an example is located at the end of this SOP).

II. Subsampling

In order to ensure that the subsample adequately represents the contents of the whole sample, minimum targets of $\geq 25\%$ of the sample area and ≥ 200 organisms, if present, will be sampled.

Procedure:

1. The sorter will randomly pick one of the quarter fractions of the sample in the sieve to represent a minimum 25% subsample area using the random table below (Fig. 2). Number sequences are oriented from left to right.

3	4	1	2
4	3	2	1
1	4	3	2
3	1	4	2
2	4	1	3
1	2	3	4
1	3	2	4
1	3	4	2
2	3	1	4
2	1	4	3
4	3	1	2
2	1	3	4
4	1	3	2
2	3	4	1
3	2	1	4
4	1	2	3
4	2	3	1
4	2	1	3

Figure 2. Randomly generated integer sequences between 1 and 4.

2. Using a putty knife or similar tool, the fraction contents are carefully scraped from the sieve and added to a white sorting pan containing water. The bottom of the sieve in the area where the subsample was removed should then be carefully inspected to ensure that no invertebrates remain.
3. All samples will be sorted under a minimum 10X magnification.
4. As invertebrates are removed from the fraction, they should be counted with a hand-held enumerator. When this fraction has been completely sorted, and 200 or more organisms have been removed, no additional sorting is necessary. If less than 200 organisms were removed from the first quarter selected, the sorter should remove another randomly selected quarter fraction from the sieve and sort it. This process is repeated until a minimum of 200

organisms have been removed or the entire sample has been sorted.

5. Always completely sort the removed sample fractions regardless of how many organisms are present (*e.g.*, the first fraction removed possibly could contain 300 or more organisms, or if you have to select a second fraction and reach 200 before the sample is completely sorted you must continue sorting until all invertebrates are removed).
6. The sorter should clearly indicate on the specimen label and lab identification sheet how many fractions of the whole sample were sorted. **This information is critical for estimating benthic densities.** For example, to estimate density for the entire sample if only one quarter is sorted, the number of specimens in this fraction must be multiplied by a factor of 4; if 2 fractions are sorted the number of specimens must be multiplied by 2; if 3 fractions are sorted the number of specimens removed must be multiplied by 1.3.
7. Additionally, a “large and/or rare” taxa component is included in the subsampling routine. Large and/or rare taxa remaining in the sample that clearly were not in the sorted fraction are removed and stored in a separate vial. These specimens will be used for reflecting accurate sample diversity estimates and calculating specific metrics such as EPT.
 - a. A large and/or rare additional taxa search will be completed following the subsample routine. Any large and/or clearly rare organisms (*e.g.*, *Corydalus cornutus*, *Pteronarcys pickettii*, tabanids, tipulids, dragonfly larvae, crayfish, gordian worms, large beetles, other unusual species, *etc.*) in the sample that clearly were not in the subsampled fraction will be removed, placed in a separate vial, and labeled appropriately (Fig. 3). There may be several or no specimens depending on the sample.
 - b. Just because a creature is large does not mean it should be removed during this process. It must fit the criterion that it was not present in the subsample.
 - c. A large and/or rare collection may not be necessary for all samples.
8. Any invertebrates present in the subsampled fractions will be stored in a separate storage vial, preserved, and properly labeled (see below).
9. Organic debris from the subsampled portion will be retained in a separate container until QC checks are completed.

III. Sample preservation and labeling

1. Invertebrates removed from the bulk samples will be stored in 75-80% isopropyl alcohol.
2. Labels will be written only on rag bond paper in permanent water proof ink (both supplied). Labels written in pencil are not acceptable.
3. Label data should be printed neatly and include the following: park name (*e.g.*, PIPE), stream name (*e.g.*, Pipestone Creek), riffle number (1-3), sample location (L, M, R),

sampling date, and collector initials (Fig. 3). All of this information is on the original sample label.

4. The vial label for “large and rare” taxa should be specifically labeled as such and also include the original sample data as well.
5. Do not crowd the collected specimens excessively as it inhibits long term preservation.

<u>Bulk Sample</u>	Park Name: PIPE Site name: Pipestone Creek Transect: 1, Left Collection date: 12 July 07 Collector initials: DEB, HRD
<u>Large & Rare</u>	LARGE & RARE Park Name: PIPE Site name: Pipestone Creek Transect: 1, Left Collection date: 12 July 07 Collector initials: DEB, HRD
<u>Identification</u>	Park Name: PIPE Site name: Pipestone Creek Collection date: 12 July 07 Taxon Name: <u>Cheumatopsyche</u> Det. D. E. Bowles

Figure 3. Example specimen labels.

IV. Identification of Invertebrates

To the extent possible, all invertebrates should be identified to genus exclusive of the groups and selected conditions indicated below.

Procedure:

1. A dissecting microscope and taxonomic keys are used to identify each specimen to the genus level whenever possible. For some taxa only higher taxonomic levels can be obtained, and some of these are listed below. In most cases, an entire sample can be identified to the required level, but an occasional sample may contain early instars or damaged specimens. In such cases, the specimen should be identified to the lowest level possible, as indicated in Table 1.

Table 1. Taxonomic levels for identification when not the Genus level.

Phylum Nematoda
Phylum Nematomorpha
Phylum Annelida, Class Hirudinea, Families Glossiphonidae, Piscicolidae, Hirudinidae, Erpobdellidae
Phylum Annelida, Class Oligochaeta, Families Aeolosomatidae, Opistocystidae, Naididae, Haplotaxidae, Enchytraeidae, Lumbriculidae, Tubificidae
Phylum Arthropoda, Class Arachnoidea, Order Hydracarina
Phylum Arthropoda, Class Crustacea, Order Ostracoda
Phylum Arthropoda, Class Insecta, Order Diptera, Family Chironomidae

2. The primary keys will be Merritt *et al.* (2008) for identification of insects and Pennak (1989) for identification of non-insect invertebrates. Additional taxonomic references for specific groups include Moulton and Stewart (1996), Poulton and Stewart (1991), Stewart and Stark (1988), and Wiggins (1995). A list of taxa known or likely to occur at HTLN parks included in this protocol is included in Table 2 below. Accuracy of scientific names should be checked at the Interagency Taxonomic Information System (ITIS) website at: http://www.itis.usda.gov/advanced_search.html.
3. A running total of each taxon for each sample should be recorded on the Aquatic Invertebrate Identification & Enumeration Sheet (see below) that is signed or initialed by the identifier. Final counts of each taxon should be entered on this sheet when a sample is complete.
4. If **damaged organisms** can be identified, they are counted ONLY if:
 - (a) The fragment includes the head, and, in the case of arthropods, the thorax. For Oligochaetes, count only fragments that include a rounded end that could be a head or tail end.
 - (b) The mollusk shell (bivalve or gastropod) is occupied by a specimen.
 - (c) The specimen is the sole representative of a taxon in the sample.
5. If **early instar or juvenile** specimens can be identified, they are counted ONLY if:
 - (a) With confidence, they can be associated with one or more mature specimens that have a more developed morphology.
 - (b) The specimen is the sole representative of a taxon in the sample.

IV. Sample Storage and Reference Collection

Procedure:

1. Identified specimens are stored in vials with 75-80% isopropyl alcohol and labeled with the taxon name, date collected, park and spring name, and name of identifier. Organisms will be retained for at least three years and stored at the NPS HTLN office located at Missouri State University, Springfield, MO.
2. A reference collection consisting of a few representative specimens of each taxon should be prepared and stored in properly labeled vials containing 75-80% isopropyl alcohol. Vials should be labeled as above (Fig. 3).
3. Regional or other taxonomist specialists should review the identifications for accuracy. One set of vials should be stored at the NPS HTLN office located at Missouri State University, Springfield, MO. Additional sets of specimens should be maintained in the laboratory where identifications are performed for use as reference and training.

V. Quality Control

Procedures:

Sample processing and sorting

The following Quality Assurance and Quality Control (QA/QC) procedures (adapted from USEPA, 2004) will be used on sorted invertebrate samples.

1. Initially, a QA/QC officer (initially the Aquatic Program Leader) will use a microscope to check **all** sorted fractions from the first five samples processed by a sorter to ensure that all organisms were removed from the detritus. This will not only apply to inexperienced sorters, but also to those initially deemed as “experienced.” Qualification will only occur when sorters are consistent in achieving >90% sorting efficiency after at least five samples have been checked. Samples will be checked using 10x magnification.
2. The Aquatic Program Leader or other QA/QC Officer will calculate percent sorting efficiency (PSE) for each sample as follows:

$$PSE = A/A+B (100)$$

where A = number of organisms found by the primary sorter, and B = number of recoveries (organisms missed by the primary sort and found by the QC check).

If the sorting efficiency for each of these five consecutive samples is >90% for a particular individual, they are considered “experienced” and can serve as a QA/QC Officer. In the event that an individual fails to achieve >90% sorting efficiency, they will be required to sort an

additional five samples and his sorting efficiency will continue to be monitored. If they show marked improvement in their sorting efficiency prior to completion of the next five samples, however, and reaches the >90% sorting efficiency, the QA/QC Officer may, at their discretion, consider this individual to be “experienced.” Sorting efficiency should not be calculated for samples processed by more than one individual.

3. After individuals acquire a >90% sorting efficiency, 10% (1 out of 10) of their samples will be checked.
4. If an “experienced” individual fails to maintain a >90% sorting efficiency as determined by QC checks, QC checks will be performed on every fraction of five consecutive samples until a >90% sorting efficiency is achieved. During this time, that individual will not be able to perform QC checks.
4. Residues of sorted bulk samples should be returned to their original sample containers and rehydrated with the alcohol saved during sample preparation. All sorted samples will be kept until QC checks are complete and permission is given by the Program Leader to discard the material in accordance with NPS guidelines and policies.

Specimen Identification

1. The identifications for 10% of the samples must be checked for quality control. The QC sample will be randomly chosen.
2. Specimens from the QC samples will be analyzed by another trained and experienced individual within the same laboratory (*e.g.*, Program Leader). Percent similarity of the two samples must exceed 90%.
4. If an individual fails to maintain a >90% identification efficiency as determined by QC checks, QC checks will be performed on every identified specimen of five consecutive samples until a >90% sorting efficiency is achieved.
5. If any specimens are incorrectly identified, all specimens assigned to that taxon will be reexamined.
5. If a taxon is especially difficult to identify, specimens will be sent to a person with taxonomic expertise of invertebrates of the region. It is also important that the taxonomist maintains contact with other taxonomists through professional societies and other interactions, and stays current with the pertinent published literature.

VI. Bulk sample disposition

When the sample is finished, return the remainder of the bulk sample to its original container with its original label and old alcohol, and return to storage. Mark the top with an X or other mark to identify the sample as one that has been picked. These samples will be subjected to a QA/QC analysis by the Program Leader.

Table 2. Regional invertebrate taxa list with functional feeding groups and tolerance values.

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
	8	C				Oligochaeta	Annelida
	10	C		Enchytraeidae	Haplotaxida	Oligochaeta	Annelida
Haplotaxis		C		Haplotaxidae	Haplotaxida	Oligochaeta	Annelida
				Naididae	Haplotaxida	Oligochaeta	Annelida
		C		Opistocystidae	Haplotaxida	Oligochaeta	Annelida
Aulodrilus	8	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Branchiura	8.4	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Ilyodrilus	9.4	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Tubifex	10	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Limnodrilus	9.8	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Potamothrix		C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Quistadrilus	10	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
	9.2	C		Tubificidae	Haplotaxida	Oligochaeta	Annelida
Eiseniella	7.3	C		Lumbricidae	Lumbricina	Oligochaeta	Annelida
Lumbriculus	8	C		Lumbriculidae	Lumbriculida	Oligochaeta	Annelida
	7.8	Pa		Erpobdellidae	Arhynchobdellida	Hirudinea	Annelida
Erpobdella		Pa		Erpobdellidae	Arhynchobdellida	Hirudinea	Annelida
Mooreobdella		Pa		Erpobdellidae	Arhynchobdellida	Hirudinea	Annelida
	6	C		Erpobdellidae	Branchiobdellida	Hirudinea	Annelida
Cambarincola	6	C		Camparinicolidae	Branchiobdellida	Hirudinea	Annelida
Placobdella	6.6	C		Glossiphoniidae	Rhynchobdellida	Hirudinea	Annelida
	7	Pr		Glossiphoniidae	Rhynchobdellida	Hirudinea	Annelida
Glossiphonia				Glossiphoniidae	Rhynchobdellida	Hirudinea	Annelida
Helobdella	8.9			Glossiphoniidae	Rhynchobdellida	Hirudinea	Annelida
Piscicola		Pa		Piscicolidae	Rhynchobdellida	Hirudinea	Annelida
					Scolecida		
				Aeolosomatidae	(subclass)	Polychaeta	Annelida
	5.7	Pa,Pr		Aeolosomatidae	Hydracarina	Arachnoida	Arthropoda
	4			Aeolosomatidae	Amphipoda	Crustacea	Arthropoda
Bactrurus		C		Crangonyctidae	Amphipoda	Crustacea	Arthropoda
Crangonyx	8	C		Crangonyctidae	Amphipoda	Crustacea	Arthropoda
Stygobromus		C		Crangonyctidae	Amphipoda	Crustacea	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Stygonectes		C		Crangonyctidae	Amphipoda	Crustacea	Arthropoda
Synurella		C		Crangonyctidae	Amphipoda	Crustacea	Arthropoda
Allocrangonyx		C		Gammaridae	Amphipoda	Crustacea	Arthropoda
Gammarus	6.9	C		Gammaridae	Amphipoda	Crustacea	Arthropoda
Hyaella	7.9	C		Hyaellidae	Amphipoda	Crustacea	Arthropoda
	8				Copepoda	Crustacea	Arthropoda
Cambarellus				Cambaridae	Decapoda	Crustacea	Arthropoda
	6	C		Cambaridae	Decapoda	Crustacea	Arthropoda
Cambarus	8.1	C		Cambaridae	Decapoda	Crustacea	Arthropoda
Fallicambarus		C		Cambaridae	Decapoda	Crustacea	Arthropoda
Faxonella		C		Cambaridae	Decapoda	Crustacea	Arthropoda
Orconectes	2.7	C		Cambaridae	Decapoda	Crustacea	Arthropoda
Procambarus	9.5	C		Cambaridae	Decapoda	Crustacea	Arthropoda
Palaemonetes	6.7	C		Palaemonidae	Decapoda	Crustacea	Arthropoda
Asellus	8			Asellidae	Isopoda	Crustacea	Arthropoda
Caecidotea	8	C		Asellidae	Isopoda	Crustacea	Arthropoda
Lirceus	7.7	C		Asellidae	Isopoda	Crustacea	Arthropoda
Cylisticus		C		Cylistidae	Isopoda	Crustacea	Arthropoda
Metroponorthus		C		Porcellionidae	Isopoda	Crustacea	Arthropoda
Porcellio		C		Porcellionidae	Isopoda	Crustacea	Arthropoda
Caucasonethes		C		Trichoniscidae	Isopoda	Crustacea	Arthropoda
Miktoniscus		C		Trichoniscidae	Isopoda	Crustacea	Arthropoda
	8			Trichoniscidae	Ostracoda	Crustacea	Arthropoda
	10			Trichoniscidae	Collembola	Insecta	Arthropoda
		C		Entomobryidae	Collembola	Insecta	Arthropoda
Folsomia		C		Isotomidae	Collembola	Insecta	Arthropoda
		C		Sminthuridae	Collembola	Insecta	Arthropoda
	5			Curculionidae	Coleoptera	Insecta	Arthropoda
Brachyvatus				Dytiscidae	Coleoptera	Insecta	Arthropoda
Coptotomus	9	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Cybister	4.6	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Desmopachria	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Dytiscus	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Graphoderus	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Hoperius		Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Hygrotus	1.9	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Ilybius	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Rhantus	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Thermonectus	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Helichus	5.4	C,Sh		Dytiscidae	Coleoptera	Insecta	Arthropoda
Postelichus	0			Dytiscidae	Coleoptera	Insecta	Arthropoda
	5			Dytiscidae	Coleoptera	Insecta	Arthropoda
Acilius		C,Sc		Dytiscidae	Coleoptera	Insecta	Arthropoda
Agabus	5	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Agabinus	5			Dytiscidae	Coleoptera	Insecta	Arthropoda
Copelatus	9.1	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Hydroporus	8.9	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Hydrovatus	3.7	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Hydaticus		Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Laccophilus	10	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Liodessus		Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Oreodytes	4.6	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
Uvarus	4.6	Pr		Dytiscidae	Coleoptera	Insecta	Arthropoda
				Elmidae	Coleoptera	Insecta	Arthropoda
Ancyronyx	6.9	C,Sc		Elmidae	Coleoptera	Insecta	Arthropoda
Cleptelmis	4			Elmidae	Coleoptera	Insecta	Arthropoda
Dubiraphia	6.4	C		Elmidae	Coleoptera	Insecta	Arthropoda
Gonielmis		C		Elmidae	Coleoptera	Insecta	Arthropoda
Heterelmis		C		Elmidae	Coleoptera	Insecta	Arthropoda
Heterlimnius				Elmidae	Coleoptera	Insecta	Arthropoda
Macronychus	4.7	C		Elmidae	Coleoptera	Insecta	Arthropoda
Microcyloepus	2.1	C		Elmidae	Coleoptera	Insecta	Arthropoda
Optioservus	2.7	C,Sh		Elmidae	Coleoptera	Insecta	Arthropoda
Stenelmis	5.4	C,Sc		Elmidae	Coleoptera	Insecta	Arthropoda
	5	F		Gyrinidae	Coleoptera	Insecta	Arthropoda
Dineutus	5.5	Pr		Gyrinidae	Coleoptera	Insecta	Arthropoda
Gyretes	3.7	Pr		Gyrinidae	Coleoptera	Insecta	Arthropoda
Gyrinus	6.3	Pr		Gyrinidae	Coleoptera	Insecta	Arthropoda
Haliplus	5			Haliplidae	Coleoptera	Insecta	Arthropoda
Peltodytes	8.5	He		Haliplidae	Coleoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Heterocerus	5	C		Heteroceridae	Coleoptera	Insecta	Arthropoda
	5			Hydrophilidae	Coleoptera	Insecta	Arthropoda
Berosus	8.6	He,C		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Chaetarthria	5.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Crenitis		C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Cymbiodyta	5.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Dibolocelus	5.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Enochrus	8.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Helobata				Hydrophilidae	Coleoptera	Insecta	Arthropoda
Hydrochara	6	C		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Helochares	5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Helocombus	5.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Helophorus	7.9	Sh		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Hydrobius	5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Hydrochus	5			Hydrophilidae	Coleoptera	Insecta	Arthropoda
Hydrophilus	4.6	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Laccobius	8	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Paracymus	7.3	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Sperchopsis	6.5	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Tropisternus	9.8	C,Pr		Hydrophilidae	Coleoptera	Insecta	Arthropoda
Hydraena		C,Sc		Hydraenidae	Coleoptera	Insecta	Arthropoda
Ochthebius		C,Sc		Hydraenidae	Coleoptera	Insecta	Arthropoda
Hydrocanthus	6.9	Pr		Noteridae	Coleoptera	Insecta	Arthropoda
Suphisellus		Pr		Noteridae	Coleoptera	Insecta	Arthropoda
	5			Lampyridae	Coleoptera	Insecta	Arthropoda
Lutrochus	2.75			Lutrochidae	Coleoptera	Insecta	Arthropoda
Ectopria	4.3	Sc		Psephenidae	Coleoptera	Insecta	Arthropoda
Psephenus	2.5	Sc		Psephenidae	Coleoptera	Insecta	Arthropoda
	5			Scirtidae	Coleoptera	Insecta	Arthropoda
Cyphon		Sh		Scirtidae	Coleoptera	Insecta	Arthropoda
Scirtes	5	Sh		Scirtidae	Coleoptera	Insecta	Arthropoda
Stenus	5			Staphylinidae	Coleoptera	Insecta	Arthropoda
	8			Staphylinidae	Coleoptera	Insecta	Arthropoda
Bledius	8			Staphylinidae	Coleoptera	Insecta	Arthropoda
Carpelimus	8			Staphylinidae	Coleoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Atherix	2.1	Pr		Athericidae	Diptera	Insecta	Arthropoda
	6	C,Pr,Sc	Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Monohelea	6		Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Probezzia		Pr	Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Atrichopogon		C	Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Bezzia	6	Pr	Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Palpomyia	6	C,Pr	Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Ceratopogon	6		Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Culicoides			Ceratopogoninae	Ceratopogonidae	Diptera	Insecta	Arthropoda
	6	C,Sc	Dasyheleinae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Dasyhelea		C,Sc	Dasyheleinae	Ceratopogonidae	Diptera	Insecta	Arthropoda
	6	C	Forcipomyiinae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Forcipomyia	6		Forcipomyiinae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Mallochohelea		Pr	Leptoconopinae	Ceratopogonidae	Diptera	Insecta	Arthropoda
Chaoborus	8.5	Pr		Chaoboridae	Diptera	Insecta	Arthropoda
Eucorethra		Pr		Chaoboridae	Diptera	Insecta	Arthropoda
	6			Chironomidae	Diptera	Insecta	Arthropoda
Acalcarella		C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Apedilum			Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Axarus	6	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Chironomus	9.8	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Cladopelma	2.5	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Cladotanytarsus	3.7	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Constempellina	4	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Cryptochironomus	7.4	Pr	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Cryptotendipes	6.1	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Demicryptochironomus	2.1	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Dicrotendipes	7.9	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Endochironomus	7.5	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Endotribelos			Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Einfeldia/Kiefferulus			Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Glyptotendipes	8.5	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Goeldichironomus	9		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Harnischia	7.5		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Hyporhygma		Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Kiefferulus	10	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Lauterborniella	5.5	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Lipiniella		C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Microchironomus	8	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Micropsectra	1.4	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Microtendipes	6.2	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Nilothauma	5.5		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Omisus			Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Pagastiella	2.6		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Parachironomus	9.2	C,Pr	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Paracladopelma	4.8	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Paralauterborniella	8		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Paratanytarsus	7.7	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Paratendipes	5.3	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Phaenopsectra	6.2	C,Sc	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Polypedilum	7.4	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Pseudochironomus	4.2	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Rheotanytarsus	6.4	F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Robackia	3.4	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Saetheria	8.1	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Stelechomyia	4.6		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Stempellina	2	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Stempellinella	5.3	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Stenochironomus	6.4	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Stictochironomus	6.7	C,Sh	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Sublettea	1.7	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Tanytarsus	6.7	C,F	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Thienemanniola			Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Tribelos	6.6	C	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Xenochironomus	7	Pr	Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Zavreliella	7		Chironominae	Chironomidae	Diptera	Insecta	Arthropoda
Diamesa	7.7	C	Diamesinae	Chironomidae	Diptera	Insecta	Arthropoda
Potthastia	4.7	C	Diamesinae	Chironomidae	Diptera	Insecta	Arthropoda
Pseudodiamesa	4.6	C	Diamesinae	Chironomidae	Diptera	Insecta	Arthropoda
Sympotthastia	5.7	C	Diamesinae	Chironomidae	Diptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Syndiamesa		C	Diamesinae	Chironomidae	Diptera	Insecta	Arthropoda
Beardius			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Brillia	5.2	C,Sh	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Brundiniella			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Cardiocladius	6.2	Pr	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Corynoneura	6.2	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Cricotopus	7.6	C,Sh	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Diplocladius	7.7	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Epoicocladius	4	C,Pr	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Eukiefferiella	4	C,Pr	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Euryhapsis			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Helopelopia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Heterotrissocladius	5.4	C,Pr	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Hydrobaenus	9.6	C,Sc	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Krenosmittia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Limnophyes			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Macropelopia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Mesocricotopus		C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Mesosmittia	7		Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Nanocladius	7.2	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Neozavrelia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Oliveridia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Orthocladius	5.8	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Paracladius			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Paracricotopus			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Parakiefferiella	5.9	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Parametriocnemus	3.7	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Paraphaenocladius	4	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Paratrachocladius			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Parorthocladius		C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Psectrocladius	3.8	C	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Pseudorthocladius		C,Sh	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Pseudosmittia	4		Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Rheocricotopus	7.3	C,Sh	Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda
Sergentia			Orthoclaadiinae	Chironomidae	Diptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Smittia	4	C	Orthocladiinae	Chironomidae	Diptera	Insecta	Arthropoda
Synorthocladius	4.7		Orthocladiinae	Chironomidae	Diptera	Insecta	Arthropoda
Thienemanniella	6		Orthocladiinae	Chironomidae	Diptera	Insecta	Arthropoda
Tvetenia	4	C	Orthocladiinae	Chironomidae	Diptera	Insecta	Arthropoda
Xylotopus			Orthocladiinae	Chironomidae	Diptera	Insecta	Arthropoda
Ablabesmyia	6.4	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Clinotanypus	9.1	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Coelotanypus	6.2	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Djalmabatista	6.4	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Krenopelopia		Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Labrundinia	5.3	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Larsia	83	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Monopelopia		Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Natarsia	8	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Nilotanypus	6	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Paramerina	2.8	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Pentaneura	4.6	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Procladius	9.3	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Psectrotanypus	10	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Tanypus	9.6	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Thienemannimyia	6	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
Zavreliomyia	9.3	Pr	Tanypodinae	Chironomidae	Diptera	Insecta	Arthropoda
	8			Culicidae	Diptera	Insecta	Arthropoda
Aedes	5.5	Pr		Culicidae	Diptera	Insecta	Arthropoda
Anopheles	9.1	F		Culicidae	Diptera	Insecta	Arthropoda
Culex	10	F		Culicidae	Diptera	Insecta	Arthropoda
Culiseta	5.5	C,F		Culicidae	Diptera	Insecta	Arthropoda
Mansonia		C,F		Culicidae	Diptera	Insecta	Arthropoda
Dixa	2.8	C,Pr		Dixidae	Diptera	Insecta	Arthropoda
Dixella		C		Dixidae	Diptera	Insecta	Arthropoda
	4			Dolichopodidae	Diptera	Insecta	Arthropoda
	6			Empididae	Diptera	Insecta	Arthropoda
Chelifera	6	Pr		Empididae	Diptera	Insecta	Arthropoda
Clinocera	6	Pr		Empididae	Diptera	Insecta	Arthropoda
Hemerodromia	6	C,Pr		Empididae	Diptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Rhamphomyia		Pr		Empididae	Diptera	Insecta	Arthropoda
Roederiodes		Pr		Empididae	Diptera	Insecta	Arthropoda
	5.5	C,Sh		Ephydriidae	Diptera	Insecta	Arthropoda
Scatella	6			Ephydriidae	Diptera	Insecta	Arthropoda
	6	Pr		Muscidae	Diptera	Insecta	Arthropoda
	10			Psychodidae	Diptera	Insecta	Arthropoda
Pericoma	4	C		Psychodidae	Diptera	Insecta	Arthropoda
Psychoda	9.9	C		Psychodidae	Diptera	Insecta	Arthropoda
Telmatoscopus		C		Psychodidae	Diptera	Insecta	Arthropoda
		He,Sh		Scathophagidae	Diptera	Insecta	Arthropoda
		Pr		Sciomyzidae	Diptera	Insecta	Arthropoda
				Simuliidae	Diptera	Insecta	Arthropoda
Cnephia	4	F		Simuliidae	Diptera	Insecta	Arthropoda
Prosimulium	2.6	F		Simuliidae	Diptera	Insecta	Arthropoda
Simulium	4.4	F		Simuliidae	Diptera	Insecta	Arthropoda
Stegopterna	5	F		Simuliidae	Diptera	Insecta	Arthropoda
	8	C		Stratiomyidae	Diptera	Insecta	Arthropoda
Allognosta				Stratiomyidae	Diptera	Insecta	Arthropoda
Caloparyphus		C		Stratiomyidae	Diptera	Insecta	Arthropoda
Euparyphus		C,Sc		Stratiomyidae	Diptera	Insecta	Arthropoda
Myxosargus		C		Stratiomyidae	Diptera	Insecta	Arthropoda
Nemotelus	7.3	C		Stratiomyidae	Diptera	Insecta	Arthropoda
Odontomyia	7.3	C		Stratiomyidae	Diptera	Insecta	Arthropoda
Oxycera		Sc		Stratiomyidae	Diptera	Insecta	Arthropoda
Stratiomys	7.3	C		Stratiomyidae	Diptera	Insecta	Arthropoda
	8.25	C		Syrphidae	Diptera	Insecta	Arthropoda
	6	C		Tabanidae	Diptera	Insecta	Arthropoda
Hybomitra		Pr		Tabanidae	Diptera	Insecta	Arthropoda
Tabanus	9.7	Pr		Tabanidae	Diptera	Insecta	Arthropoda
Chrysops	7.3	Pr		Tabanidae	Diptera	Insecta	Arthropoda
Silvius		Pr		Tabanidae	Diptera	Insecta	Arthropoda
Protoplasa	0			Tanyderidae	Diptera	Insecta	Arthropoda
	3	C		Tipulidae	Diptera	Insecta	Arthropoda
Antocha	4.6	C		Tipulidae	Diptera	Insecta	Arthropoda
Cryptolabis				Tipulidae	Diptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Dactylolabis	3.7	C,Sh		Tipulidae	Diptera	Insecta	Arthropoda
Dicranota	0	Pr		Tipulidae	Diptera	Insecta	Arthropoda
Erioptera	5.5	C		Tipulidae	Diptera	Insecta	Arthropoda
Gonomyia	5.5	C		Tipulidae	Diptera	Insecta	Arthropoda
Hexatoma	4.7	Pr		Tipulidae	Diptera	Insecta	Arthropoda
Holorusia		Sh		Tipulidae	Diptera	Insecta	Arthropoda
Lipsothrix		Sh		Tipulidae	Diptera	Insecta	Arthropoda
Limonia	10	Sh		Tipulidae	Diptera	Insecta	Arthropoda
Limnophila	4.6	Pr		Tipulidae	Diptera	Insecta	Arthropoda
Molophilus				Tipulidae	Diptera	Insecta	Arthropoda
Ormosia	4.6	C		Tipulidae	Diptera	Insecta	Arthropoda
Paradelphomyia				Tipulidae	Diptera	Insecta	Arthropoda
Pedicia	4.6	Pr		Tipulidae	Diptera	Insecta	Arthropoda
Pilaria	7			Tipulidae	Diptera	Insecta	Arthropoda
Prionocera		Sh		Tipulidae	Diptera	Insecta	Arthropoda
Pseudolimnophila	7.3	C		Tipulidae	Diptera	Insecta	Arthropoda
Rhabdomastix		C		Tipulidae	Diptera	Insecta	Arthropoda
Tipula	7.7	C,Sh		Tipulidae	Diptera	Insecta	Arthropoda
Thaumalea		Sc		Thaumaleidae	Diptera	Insecta	Arthropoda
Ameletus	7	C,Sc		Ameletidae	Ephemeroptera	Insecta	Arthropoda
	4	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Acentrella	3.6	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Acerpenna	3.7	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Apobaetis	6	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Barbaetis		C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Baetis	6	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Callibaetis	9.3	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Camelobaetidius	9.3	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Centropetium	6.3	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Cloeon	7.4	C		Baetidae	Ephemeroptera	Insecta	Arthropoda
Diphetor	5			Baetidae	Ephemeroptera	Insecta	Arthropoda
Fallceon	6	Pr		Baetidae	Ephemeroptera	Insecta	Arthropoda
Heterocloeon	2	Sc		Baetidae	Ephemeroptera	Insecta	Arthropoda
Labiobaetis	6	Pr		Baetidae	Ephemeroptera	Insecta	Arthropoda
Paracloeodes	5	Sc		Baetidae	Ephemeroptera	Insecta	Arthropoda

Plauditus	6	C	Baetidae	Ephemeroptera	Insecta	Arthropoda
Procloeon	6.3	C	Baetidae	Ephemeroptera	Insecta	Arthropoda
Pseudocentropiloides		C	Baetidae	Ephemeroptera	Insecta	Arthropoda
Pseudocloeon		C	Baetidae	Ephemeroptera	Insecta	Arthropoda
Baetisca	4	C,Sc	Baetiscidae	Ephemeroptera	Insecta	Arthropoda
Amercaenis		F	Caenidae	Ephemeroptera	Insecta	Arthropoda
Brachycercus	3.5	C	Caenidae	Ephemeroptera	Insecta	Arthropoda
Caenis	7.6	C,Sc	Caenidae	Ephemeroptera	Insecta	Arthropoda
Cercobrachys		C	Caenidae	Ephemeroptera	Insecta	Arthropoda
	4		Ephemeridae	Ephemeroptera	Insecta	Arthropoda
Ephemera	2.2	C,Pr	Ephemeridae	Ephemeroptera	Insecta	Arthropoda
Hexagenia	4.7	C	Ephemeridae	Ephemeroptera	Insecta	Arthropoda
Attenella	1	C	Ephemerellidae	Ephemeroptera	Insecta	Arthropoda
Ephemerella	1.7	C,Sc	Ephemerellidae	Ephemeroptera	Insecta	Arthropoda
Eurylophella	3	C	Ephemerellidae	Ephemeroptera	Insecta	Arthropoda
Serratella	1.9	C	Ephemerellidae	Ephemeroptera	Insecta	Arthropoda
Timpanoga	2	C	Ephemerellidae	Ephemeroptera	Insecta	Arthropoda
	4	F	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Anepeorus		Pr	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Cinygmula	4	F	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Epeorus	1.2	C	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Heptagenia	2.8	C,Sc	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Leucrocuta	0	C,Sc	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Macdunnoa	4.6	C,Sc	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Nixe	2	C,Sc	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Rhithrogena	0.4	C	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Stenonema	3.4	C,Sc	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Stenacron	7.1	C	Heptageniidae	Ephemeroptera	Insecta	Arthropoda
Isonychia	3.7	F	Isonychiidae	Ephemeroptera	Insecta	Arthropoda
	2		Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Choroterpes	2		Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Leptophlebia	6.4		Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Habrophlebiodes	6	C,Sc	Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Neochoroterpes		C,Sc	Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Paraleptophlebia	1.2	C,Sh	Leptophlebiidae	Ephemeroptera	Insecta	Arthropoda
Neoephemera		C	Neoephemeridae	Ephemeroptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Pentagenia	6.4	C		Palingeniidae	Ephemeroptera	Insecta	Arthropoda
Anthopotamus				Potamanthidae	Ephemeroptera	Insecta	Arthropoda
Ephoron	4.6	C		Polymitarcyidae	Ephemeroptera	Insecta	Arthropoda
Tortopus	5.5	C		Polymitarcyidae	Ephemeroptera	Insecta	Arthropoda
Pseudiron	5	Pr		Pseudironidae	Ephemeroptera	Insecta	Arthropoda
Siphonurus	2.6	C,Sc		Siphonuridae	Ephemeroptera	Insecta	Arthropoda
Tricorythodes	5.4	C		Tricorythidae	Ephemeroptera	Insecta	Arthropoda
Belostoma	9.8	Pr		Belostomatidae	Hemiptera	Insecta	Arthropoda
Lethocerus	4.6	Pr		Belostomatidae	Hemiptera	Insecta	Arthropoda
	8			Corixidae	Hemiptera	Insecta	Arthropoda
Corisella	6.4	Pr		Corixidae	Hemiptera	Insecta	Arthropoda
Hesperocorixa	5	Mp		Corixidae	Hemiptera	Insecta	Arthropoda
Palmacorixa	5.5			Corixidae	Hemiptera	Insecta	Arthropoda
Sigara	4.6	Mp,C		Corixidae	Hemiptera	Insecta	Arthropoda
Trichocorixa	5.5	C,Pr		Corixidae	Hemiptera	Insecta	Arthropoda
Gelastocoris	7.3	Pr		Gelastocoridae	Hemiptera	Insecta	Arthropoda
	5			Gerridae	Hemiptera	Insecta	Arthropoda
Aquarius	6.4	Pr		Gerridae	Hemiptera	Insecta	Arthropoda
Gerris	6.4	Pr		Gerridae	Hemiptera	Insecta	Arthropoda
Metrobates	6.4	Pr		Gerridae	Hemiptera	Insecta	Arthropoda
Rheumatobates	6.4	Pr		Gerridae	Hemiptera	Insecta	Arthropoda
Trepobates	6.4	Pr		Gerridae	Hemiptera	Insecta	Arthropoda
Hebrus	6.4	Pr		Hebridae	Hemiptera	Insecta	Arthropoda
Lipogomphus	5			Hebridae	Hemiptera	Insecta	Arthropoda
Merragata	7.3	Pr		Hebridae	Hemiptera	Insecta	Arthropoda
Hydrometra	7.3	Pr		Hydrometridae	Hemiptera	Insecta	Arthropoda
Mesovelgia	6.4	Pr		Mesoveliidae	Hemiptera	Insecta	Arthropoda
Pelocoris		Pr		Naucoridae	Hemiptera	Insecta	Arthropoda
Nepa	4.6	Pr		Nepidae	Hemiptera	Insecta	Arthropoda
Ranatra	7.5	Pr		Nepidae	Hemiptera	Insecta	Arthropoda
Buenoa	5.5	Pr		Notonectidae	Hemiptera	Insecta	Arthropoda
Notonecta	5.5	Pr		Notonectidae	Hemiptera	Insecta	Arthropoda
Neoplea	5.5	Pr		Pleidae	Hemiptera	Insecta	Arthropoda
Steinovelgia		Pr		Pleidae	Hemiptera	Insecta	Arthropoda
	10			Saldidae	Hemiptera	Insecta	Arthropoda

Micracanthia	5.5	Pr	Saldidae	Hemiptera	Insecta	Arthropoda
Pentacora	6.4	Pr	Saldidae	Hemiptera	Insecta	Arthropoda
Saldula	6.4	Pr	Saldidae	Hemiptera	Insecta	Arthropoda
	7		Veliidae	Hemiptera	Insecta	Arthropoda
Microvelia	6.4	Pr	Veliidae	Hemiptera	Insecta	Arthropoda
Paravelia	5	F	Veliidae	Hemiptera	Insecta	Arthropoda
Platyvelia		Pr	Veliidae	Hemiptera	Insecta	Arthropoda
Rhagovelia	5		Veliidae	Hemiptera	Insecta	Arthropoda
Pyroderces	5		Cosmopterigidae	Lepidoptera	Insecta	Arthropoda
		He	Cossidae	Lepidoptera	Insecta	Arthropoda
Nepticula		He	Nepticulidae	Lepidoptera	Insecta	Arthropoda
			Nectuidae	Lepidoptera	Insecta	Arthropoda
Archanara	5		Noctuidae	Lepidoptera	Insecta	Arthropoda
Bellura		He	Noctuidae	Lepidoptera	Insecta	Arthropoda
Simyra		He	Noctuidae	Lepidoptera	Insecta	Arthropoda
Nymphula	7	He	Pyalidae	Lepidoptera	Insecta	Arthropoda
Parapoynx	5	He	Pyalidae	Lepidoptera	Insecta	Arthropoda
Petrophila	5	Sc	Pyalidae	Lepidoptera	Insecta	Arthropoda
Schoenobius		He	Pyalidae	Lepidoptera	Insecta	Arthropoda
Archips		Sh	Tortricidae	Lepidoptera	Insecta	Arthropoda
Bactra			Tortricidae	Lepidoptera	Insecta	Arthropoda
Chauliodes	4	Pr	Corydalidae	Megaloptera	Insecta	Arthropoda
Corydalus	5.6	Pr	Corydalidae	Megaloptera	Insecta	Arthropoda
Nigronia	5.8	Pr	Corydalidae	Megaloptera	Insecta	Arthropoda
Sialis	7.5	Pr	Sialidae	Megaloptera	Insecta	Arthropoda
Climacia	6.5	Pr	Sisyridae	Neuroptera	Insecta	Arthropoda
Aeshna	6.4	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
Anax	6.4	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
Basiaeschna	7.7	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
Boyeria	6.3	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
Epiaeschna	3.7	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
Nasiaeschna	8	Pr	Aeshnidae	Odonata	Insecta	Arthropoda
	5		Calopterygidae	Odonata	Insecta	Arthropoda
Hetaerina	6.2	Pr	Calopterygidae	Odonata	Insecta	Arthropoda
Argia	8.7	Pr	Coenagrionidae	Odonata	Insecta	Arthropoda
Amphiagrion	2.8	Pr	Coenagrionidae	Odonata	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Calopteryx	8.3	Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
Chromagrion		Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
Coenagrion	9			Coenagrionidae	Odonata	Insecta	Arthropoda
Enallagma	9	Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
Ischnura	9.4	Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
Nehalennia		Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
Telebasis	9	Pr		Coenagrionidae	Odonata	Insecta	Arthropoda
				Coenagrionidae	Odonata	Insecta	Arthropoda
Cordulegaster	6.1	Pr		Cordulegastridae	Odonata	Insecta	Arthropoda
	7	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Arigomphus	6.4	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Dromogomphus	6.3	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Erpetogomphus	5.5	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Gomphus	6.2	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Hagenius	4	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Lanthus	2.7	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Ophiogomphus	6.2	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Progomphus	8.7	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Stylogomphus	4.8	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Stylurus	4	Pr		Gomphidae	Odonata	Insecta	Arthropoda
Archilestes	6.4	Pr		Lestidae	Odonata	Insecta	Arthropoda
Lestes	6.4	Pr		Lestidae	Odonata	Insecta	Arthropoda
	9			Libellulidae	Odonata	Insecta	Arthropoda
Epithea	7	Pr	Corduliinae	Libellulidae	Odonata	Insecta	Arthropoda
Helocordulia		Pr	Corduliinae	Libellulidae	Odonata	Insecta	Arthropoda
Neurocordulia	4	Pr	Corduliinae	Libellulidae	Odonata	Insecta	Arthropoda
Somatochlora	8.9	Pr	Corduliinae	Libellulidae	Odonata	Insecta	Arthropoda
Brachymesia		Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Celithemis	3.7	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Dythemis	3.7	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Erythemis	7.7	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Erythrodiplax		Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Leucorrhinia	6.4	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Libellula	9.8	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Orthemis	4.6	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Pachydiplax	9.6	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Pantala	6.4	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Perithemis	10	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Plathemis	10	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Sympetrum	7.3	Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Tramea		Pr	Libellulinae	Libellulidae	Odonata	Insecta	Arthropoda
Didymops	5.5	Pr	Macromiinae	Libellulidae	Odonata	Insecta	Arthropoda
Macromia	6.7	Pr	Macromiinae	Libellulidae	Odonata	Insecta	Arthropoda
Tachopteryx	3.7	Pr		Petaluridae	Odonata	Insecta	Arthropoda
	5	C		Tetrigidae	Orthoptera	Insecta	Arthropoda
	2			Capniidae	Plecoptera	Insecta	Arthropoda
Allocapnia	2.8	Sh		Capniidae	Plecoptera	Insecta	Arthropoda
Nemocapnia		Sh		Capniidae	Plecoptera	Insecta	Arthropoda
Paracapnia	0.2	Sh		Capniidae	Plecoptera	Insecta	Arthropoda
Alloperla	1.4	Pr		Chloroperlidae	Plecoptera	Insecta	Arthropoda
Haploperla	1.4	C,Pr		Chloroperlidae	Plecoptera	Insecta	Arthropoda
				Leuctridae	Plecoptera	Insecta	Arthropoda
Leuctra	0.7	Sh		Leuctridae	Plecoptera	Insecta	Arthropoda
Zealeuctra	0	Sh		Leuctridae	Plecoptera	Insecta	Arthropoda
	0			Nemouridae	Plecoptera	Insecta	Arthropoda
Amphinemura	3.4	Sh		Nemouridae	Plecoptera	Insecta	Arthropoda
Prostoia	6.1	Sc,Sh		Nemouridae	Plecoptera	Insecta	Arthropoda
Shipsa	0.3	Sc,Sh		Nemouridae	Plecoptera	Insecta	Arthropoda
Attaneuria	2.75	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
				Perlidae	Plecoptera	Insecta	Arthropoda
Acroneuria	1.4	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
Agnetina	1.4	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
Neoperla	1.6	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
Paragnetina	1.8	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
Perlesta	0	C,Pr		Perlidae	Plecoptera	Insecta	Arthropoda
Perlinella	0	Pr		Perlidae	Plecoptera	Insecta	Arthropoda
	2			Perlodidae	Plecoptera	Insecta	Arthropoda
Clioperla	4.8	Pr		Perlodidae	Plecoptera	Insecta	Arthropoda
Cultus	2			Perlodidae	Plecoptera	Insecta	Arthropoda
Helopicus	5.75	Pr		Perlodidae	Plecoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Hydroperla	2.75	Pr		Perlodidae	Plecoptera	Insecta	Arthropoda
Isoperla	3.2	C,Pr		Perlodidae	Plecoptera	Insecta	Arthropoda
Pteronarcys	1.7	Sh		Pteronarcyidae	Plecoptera	Insecta	Arthropoda
	2			Taeniopterygidae	Plecoptera	Insecta	Arthropoda
Strophopteryx	2.5	Sh		Taeniopterygidae	Plecoptera	Insecta	Arthropoda
Taeniopteryx	2.8	C,Sh		Taeniopterygidae	Plecoptera	Insecta	Arthropoda
Brachycentrus	1	F		Brachycentridae	Trichoptera	Insecta	Arthropoda
Micrasema	0.6	Sh		Brachycentridae	Trichoptera	Insecta	Arthropoda
Phylocentropus	5.6	F		Dipseudopsidae	Trichoptera	Insecta	Arthropoda
	0			Glossosomatidae	Trichoptera	Insecta	Arthropoda
Agapetus	0	C,Sc		Glossosomatidae	Trichoptera	Insecta	Arthropoda
Glossosoma	1.5	C,Sc		Glossosomatidae	Trichoptera	Insecta	Arthropoda
Protoptila	2.8	Sc		Glossosomatidae	Trichoptera	Insecta	Arthropoda
Helicopsyche	0	Sc		Helicopsychidae	Trichoptera	Insecta	Arthropoda
	4			Hydropsychidae	Trichoptera	Insecta	Arthropoda
Ceratopsyche	1.4	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
Cheumatopsyche	6.6	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
Diplectrona	2.2	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
Hydropsyche	4	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
Macrostemum	3.6	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
Potamyia	5	F		Hydropsychidae	Trichoptera	Insecta	Arthropoda
	4			Hydroptilidae	Trichoptera	Insecta	Arthropoda
Agraylea	8	He,Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Dibusa		Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Ithytrichia	4	Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Hydroptila	6.2	He,Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Leucotrichia	4.3	C,Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Mayatrichia	6			Hydroptilidae	Trichoptera	Insecta	Arthropoda
Neotrichia	2	Sc		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Ochrotrichia	6.8	C,He		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Oxyethira	3	C,He		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Stactobiella	2.75	Sh		Hydroptilidae	Trichoptera	Insecta	Arthropoda
Lepidostoma	1	Sh		Lepidostomatidae	Trichoptera	Insecta	Arthropoda
	4			Leptoceridae	Trichoptera	Insecta	Arthropoda
Ceraclea	2.3	C,Sh		Leptoceridae	Trichoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Leptocerus	4.6	Sh		Leptoceridae	Trichoptera	Insecta	Arthropoda
Mystacides	3.5	C,Sh		Leptoceridae	Trichoptera	Insecta	Arthropoda
Nectopsyche	4.1	C,Sh		Leptoceridae	Trichoptera	Insecta	Arthropoda
Oecetis	5.7	He,Pr		Leptoceridae	Trichoptera	Insecta	Arthropoda
Setodes	0.9	C,Pr		Leptoceridae	Trichoptera	Insecta	Arthropoda
Trienodes	3.7	Sh		Leptoceridae	Trichoptera	Insecta	Arthropoda
	4			Limnephilidae	Trichoptera	Insecta	Arthropoda
Frenesia	0	Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Hesperophylax	3	He,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Hydatophylax	2.3	C,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Ironoquia	7.3	C,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Leptophylax		He,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Limnephilus	2.75	He,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Platycentropus	4	Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Pseudostenophylax	0	C,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Pycnopsyche	2.3	C,Sh		Limnephilidae	Trichoptera	Insecta	Arthropoda
Molanna	3.9	C,Sh		Molannidae	Trichoptera	Insecta	Arthropoda
Marilia		Sh		Odontoceridae	Trichoptera	Insecta	Arthropoda
Psilotreta	0	C,Sc		Odontoceridae	Trichoptera	Insecta	Arthropoda
	3			Philopotamidae	Trichoptera	Insecta	Arthropoda
Chimarra	2.8	F		Philopotamidae	Trichoptera	Insecta	Arthropoda
Wormaldia	0.4	F		Philopotamidae	Trichoptera	Insecta	Arthropoda
Agrypnia	4.6	Sh		Phryganeidae	Trichoptera	Insecta	Arthropoda
Banksiola		Sh		Phryganeidae	Trichoptera	Insecta	Arthropoda
Phryganea	4.6	Sh,Pr		Phryganeidae	Trichoptera	Insecta	Arthropoda
Ptilostomis	6.7	Sh,Pr		Phryganeidae	Trichoptera	Insecta	Arthropoda
				Polycentropodidae	Trichoptera	Insecta	Arthropoda
Cernotina	4.6	Pr		Polycentropodidae	Trichoptera	Insecta	Arthropoda
Cyrnellus	7.4	F		Polycentropodidae	Trichoptera	Insecta	Arthropoda
Neureclipsis	4.4	F,Sh		Polycentropodidae	Trichoptera	Insecta	Arthropoda
Nyctiophylax	6			Polycentropodidae	Trichoptera	Insecta	Arthropoda
Polycentropus	3.5	C,Pr		Polycentropodidae	Trichoptera	Insecta	Arthropoda
	0			Psychomyiidae	Trichoptera	Insecta	Arthropoda
Lype	4.3	Sc		Psychomyiidae	Trichoptera	Insecta	Arthropoda
Paduniella	0	C,Sc		Psychomyiidae	Trichoptera	Insecta	Arthropoda

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Psychomyia	3.3	C,Sc		Psychomyiidae	Trichoptera	Insecta	Arthropoda
Rhyacophila	0.8	Pr		Rhyacophilidae	Trichoptera	Insecta	Arthropoda
Neophylax	1.6	Sc		Uenoidae	Trichoptera	Insecta	Arthropoda
Hydra	5	C/P		Hydridae	Hydroida	Hydrozoa	Cnidaria
	0	F		Unionidae	Unionida	Bivalvia	Mollusca
Dreissena	8	F		Dreissenidae	Veneroidea	Bivalvia	Mollusca
Corbicula	6.3	F		Corbiculidae	Veneroidea	Bivalvia	Mollusca
Pisidium	6.8	F		Sphaeriidae	Veneroidea	Bivalvia	Mollusca
Sphaerium		F		Sphaeriidae	Veneroidea	Bivalvia	Mollusca
Sphaerium/Musculium	7.7	F		Sphaeriidae	Veneroidea	Bivalvia	Mollusca
Campeloma	6.7	Sc		Viviparidae	Architaenioglossa	Gastropoda	Mollusca
Cipangopaludina		Sc		Viviparidae	Architaenioglossa	Gastropoda	Mollusca
Lioplax		Sc		Viviparidae	Architaenioglossa	Gastropoda	Mollusca
Viviparus	6	Sc		Viviparidae	Architaenioglossa	Gastropoda	Mollusca
Laevapex	7.3	Sc		Ancylidae	Bassommatophora	Gastropoda	Mollusca
Ferrissia	6.9	Sc		Ancylidae	Bassommatophora	Gastropoda	Mollusca
Rhodaemea		Sc		Ancylidae	Bassommatophora	Gastropoda	Mollusca
Pseudosuccinea	7.2	Sc		Lymnaeidae	Bassommatophora	Gastropoda	Mollusca
	6	Sc		Lymnaeidae	Bassommatophora	Gastropoda	Mollusca
Fossaria	6	Sc		Lymnaeidae	Bassommatophora	Gastropoda	Mollusca
Lymnaea	6	Sc		Lymnaeidae	Bassommatophora	Gastropoda	Mollusca
	8	Sc		Physidae	Bassommatophora	Gastropoda	Mollusca
Physa	8	Sc		Physidae	Bassommatophora	Gastropoda	Mollusca
Physella	9.1	Sc		Physidae	Bassommatophora	Gastropoda	Mollusca
Helisoma	6.5	Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Menetus	8.4	Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
	7	Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Gyraulus	8	Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Carychium		Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Planorbella		Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Planorbula		Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
Promenetus		Sc		Planorbidae	Bassommatophora	Gastropoda	Mollusca
	5	Sc		Valvatidae	Heterostropha	Gastropoda	Mollusca
Valvata	8	Sc		Valvatidae	Heterostropha	Gastropoda	Mollusca
Pomatiopsis		Sc		Pomatiopsidae	Mesogastropoda	Gastropoda	Mollusca

Genus/Species	Tolerance Value	FFG	Subfamily	Family	Order	Class	Phylum
Bithynia	5	Sc		Bithyniidae	Neotaenioglossa	Gastropoda	Mollusca
Antrobia		Sc		Hydrobiidae	Neotaenioglossa	Gastropoda	Mollusca
	7	Sc		Hydrobiidae	Neotaenioglossa	Gastropoda	Mollusca
Fontigens		Sc		Hydrobiidae	Neotaenioglossa	Gastropoda	Mollusca
Elimia	2.5	Sc		Pleuroceridae	Neotaenioglossa	Gastropoda	Mollusca
Goniobasis		Sc		Pleuroceridae	Neotaenioglossa	Gastropoda	Mollusca
Leptoxis	1.6	Sc		Pleuroceridae	Neotaenioglossa	Gastropoda	Mollusca
Lithasia		Sc		Pleuroceridae	Neotaenioglossa	Gastropoda	Mollusca
Pleurocera	6	Sc		Pleuroceridae	Neotaenioglossa	Gastropoda	Mollusca
	5	Pa					Nematoda
	5	Pa					Nematomorpha
		Pa		Gordiidae			Nematomorpha
	4	C		Stenostomidae	Catenulida	Turbellaria	Platyhelminthes
Macrocotyla		C,Pr		Dendrocoelidae	Tricladida	Turbellaria	Platyhelminthes
Dugesia	7.5	C,Pr		Planariidae	Tricladida	Turbellaria	Platyhelminthes

Aquatic Invertebrate Identification & Enumeration Sheet

Page ____ of ____

Site Name: _____
 Riffle No., Sample No. _____
 Sample date: _____
 Subsampled portion: 25 50 75 100

State: _____
 Date of Det: _____
 Determiner(s): _____

ORDER (or higher)	FAMILY	GENUS	SPECIES	TALLEY/COUNT	Tot Cnt	RC
Diptera						
Coleoptera						
Ephemeroptera						
Plecoptera						
Trichoptera						
Amphipoda						
Other						

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 7: Data Management

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP explains procedures for data management of Effigy Mounds NM, George Washington Carver NM, Herbert Hoover NHS, Homestead NM of America, Hot Springs NP, Pea Ridge NMP, Pipestone NM, Tallgrass Prairie NP, and Wilson's Creek NB. Historical data collected for Agate Fossil Beds NM (AGFO) under Peterson *et al.* (1999) are included in this database, but no new data will be added following the FY 08 sampling season data. It includes a general description of the data model - a continuation of the model implemented from Peterson *et al.* (1999) and subsequently standardized by the NPS I&M program as the Natural Resource Database Template (NRDT) (NPS 2006). It describes procedures for data entry, data verification and validation, and data integrity for the primary invertebrate data model adapted from Bowles *et al.* (2007).

Data management can be divided into (a) the initial design phase that involves defining the data model, its entities and their relationships and (b) the procedures necessary to implement the database. Microsoft (MS) Access 2003 is the primary software used for maintaining invertebrate community data. Water quality data will be stored in the National Park Service's NPStore database. Environmental Systems Research Institute (ESRI) ArcInfo 9.2 is used for managing spatial data associated with field sampling locations. Data products derived from this project will be available at the NPS I&M Data Store and EPA Store National Data Warehouse. QA/QC guidelines in this document are based on recommendations of Rowell *et al.* (2005) and citations therein.

I. Data Model

The initial data model implemented from Peterson *et al.* (1999) identified core tables (sites and events) to link invertebrate count data through a habitat table. This model was later revised pursuant to the NRDT developed by the NPS I&M program to be used as the database model for

storing vital signs monitoring data in MS Access. The template has a core database structure that standardizes location and observation data to facilitate the integration of datasets across the I&M program. Developed in MS Access, the database allows users to enter, edit, display, summarize, and generate reports as well as integrate with other Natural Resource data systems such as NPStoret.

NPS Water Resource Division (WRD) has designed the NPStoret database to facilitate archiving NPS data in the EPA Storet database. NPStoret is a series of Access-based templates patterned after the NRDT and includes data entry templates and an import module. It supports the core data management objectives of data entry and verification/validation in a referentially constrained environment as in related locations, events, and primary data elements (NPS/WRD 2007).

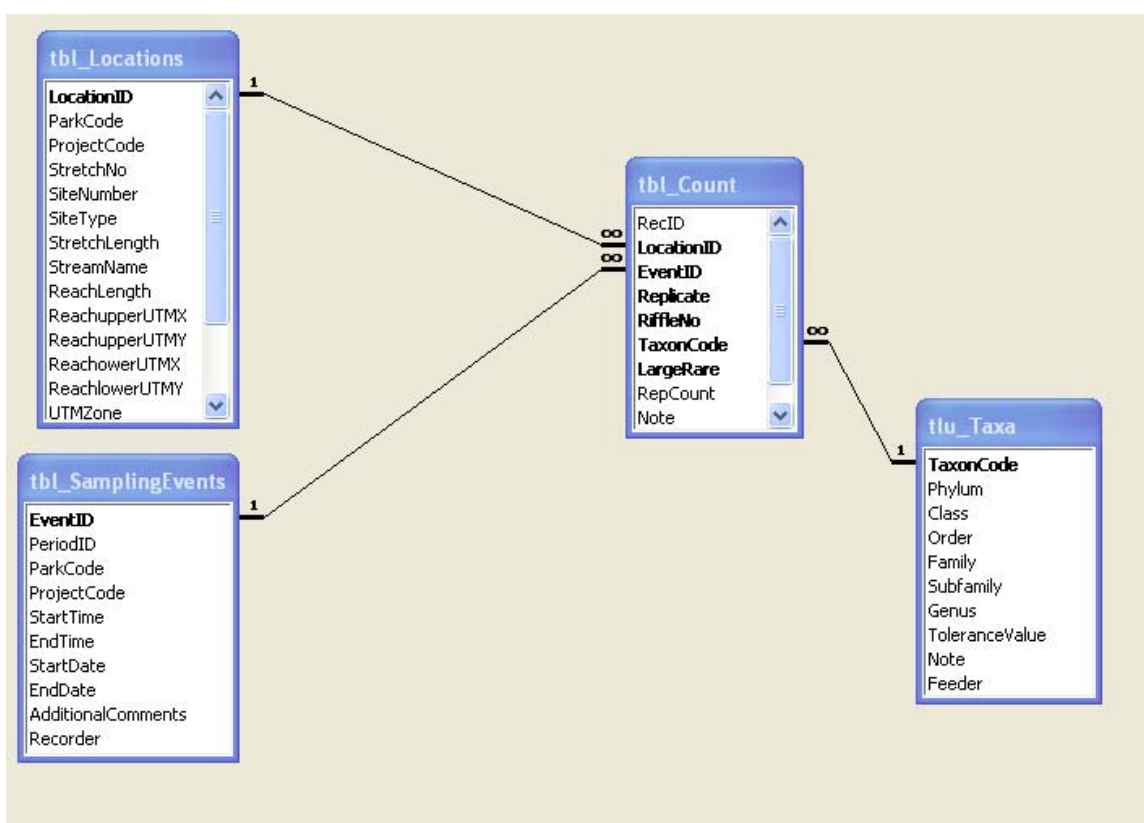


Figure 1. Data model for stream invertebrate community monitoring. Note tbl_Locations tracks GPS spatial and non-spatial field data while tbl_SamplingEvents tracks events. The data entry forms (below) follow these two levels of information organization.

A generalized NRDT entity relationship diagram of the invertebrate community database is given in Fig. 1. The locations (tbl_Locations) and sampling events (tbl_SamplingEvents) tables are the two core tables and contain general information pertaining to the field sample occasion (the where and when of the sample). This includes information such as date and time, site name,

UTM coordinates, and park/project codes. Detailed information pertaining to the community sampled is maintained in tbl_Count (invertebrate count data). Other tables include habitat data (e.g., substrate, discharge) and associated lookup tables (e.g., Wentworth substrate codes, taxonomic data).

II. Data Preparation

Quality assurance and quality control procedures related to data recording are important components of any project. Sampling data (*i.e.*, sample methods, effort, weather/water quality conditions, and species abundance data) are recorded and checked for completeness either before leaving a site or within 24 hours of data recording. This will aid in verification and validation of the data after entry into the database. To prevent the complete loss of field form data due to unforeseen circumstances (*i.e.*, fire or flood in the workplace), all field sheets are photocopied and a hard copy located in a separate location as the original. Field sheets are scanned into a computer and electronic copies of the data sheets stored on the HTLN server located at Missouri State University, Springfield, MO. This will ensure that at least one copy of the field sheets is available for data entry and verification.

III. Data Entry

Data entry commences from a main switchboard (Fig. 2). A preliminary set of forms define the sampling occasion and requires the input of location, period, and event IDs *prior* to entry of additional data. The other forms address the details of invertebrate occurrence, habitat, observers, etc. and have data entry instructions. Once all fields for the preliminary set of forms have been completed, data can be entered for the remaining forms. Additional forms document sampling personnel for each occasion and their specific hours related to the project (sampling hours, travel hours, *etc.*).

Several features are “built-in” to form properties that enable the user to maximize data entry efforts while minimizing error. These include data input masks for ease of viewing multi-part data (*i.e.*, park/project codes and date in PeriodID, LocationID, or EventID), “fill-in-as-you-type” to automatically complete a field, limiting input values to known ranges (or restricting null values) or providing “drop-down boxes”, highlighting data entry boxes being edited, and tab indexes to control the order of data entry. Forms also contain fields that require data input or are constrained to the properties and integrity of related tables.

Figure 2. Stream invertebrate community database switchboard. Note the primary data entry form button (top).

Entering Sampling Occasion Data

Procedure:

1. Open the Locations form (Fig. 3, top) to enter the LocationID.
2. Select the Park Code from the drop down box.
3. Enter the specific Site Number.
4. Click the “Generate LocationID” button to generate a LocationID and enter additional location information (Fig. 3, bottom). An input mask formats LocationID to separate Park Code, Project, and LocationID with underscores.

LocationID Data Entry Form

1. Choose Park Code and enter Site # (required).

Park Code: Site Number: eg. 01

2. Verify and generate a LocationID

LocationID Data Entry Form

Location ID

Park Code: Project:

SiteNumber:

Add additional data for this specific sampling location.

Stream Name: County:

Panel No:

Reach Length:

Lower Boundary of Reach UTMX:

Lower Boundary of Reach UTM Y:

Site Description

Figure 3. Location data entry forms for stream invertebrate data.

5. Open the Sampling Period form (Fig. 4, top), select park code, and use the calendars to select the start and end dates of the sampling period time frame to develop a PeriodID.

- Clicking the “Generate Sampling PeriodID” button passes the user data to a secondary form where the user enters the Season and accepts (or revises) the Protocol Version and then clicks the “Verify” button (Fig. 4, bottom).

PeriodID Data Entry Form

1. Choose Park Code.

Park Code:

2. Select Start Date and End Date

Start Date

Oct 2007

Sun	Mon	Tue	Wed	Thu	Fri	Sat
30	1	2	3	4	5	6
7	8	9	10	11	12	13
14	15	16	17	18	19	20
21	22	23	24	25	26	27
28	29	30	31	1	2	3
4	5	6	7	8	9	10

End Date

Oct 2007

Sun	Mon	Tue	Wed	Thu	Fri	Sat
30	1	2	3	4	5	6
7	8	9	10	11	12	13
14	15	16	17	18	19	20
21	22	23	24	25	26	27
28	29	30	31	1	2	3
4	5	6	7	8	9	10

3. Click to generate a PeriodID

PeriodID Data Entry Form

Park Code: Project Code:

Period ID:

Start Date: End Date:

Enter Season and verify the Protocol Version used for this Sampling Period

Season:

Protocol Version:

Figure 4. Sampling period forms for stream invertebrate community data.

7. Open the Sampling Events forms (Fig. 5, top), select the appropriate Park Code and PeriodID, then use the calendars to select the start and end dates of the sampling event to develop an EventID.
8. Clicking the “Generate Sampling EventID” button passes the user data to a secondary form where the user enters additional event data (Fig. 5, bottom).

EventID Data Entry Form

1. Choose Park Code and PeriodID

Park Code: Period ID:

2. Select Start Date and End Date

Start Date

Oct 2007

Sun	Mon	Tue	Wed	Thu	Fri	Sat
30	1	2	3	4	5	6
7	8	9	10	11	12	13
14	15	16	17	18	19	20
21	22	23	24	25	26	27
28	29	30	31	1	2	3
4	5	6	7	8	9	10

End Date

Oct 2007

Sun	Mon	Tue	Wed	Thu	Fri	Sat
30	1	2	3	4	5	6
7	8	9	10	11	12	13
14	15	16	17	18	19	20
21	22	23	24	25	26	27
28	29	30	31	1	2	3
4	5	6	7	8	9	10

3. Click to generate an EventID

EventID Data Entry Form

Park Code: Period ID:

Start Date: End Date: Project Code:

Event ID:

Add additional data for this specific sampling event.

Start Time: End Time: Recorder:

Additional Comments:

Figure 5. Sampling event forms for stream invertebrate data.

After inputting sampling occasion data (locations, periods and events), the user can begin to enter additional stream invertebrate community data. The following demonstrates data entry for the additional data and can be used in any order.

Entering Invertebrate Counts

Procedure:

1. Click the “Enter Invertebrate Counts and Additional Data” button on the main switchboard to enter invertebrate data.
2. Select the appropriate Park, LocationID, Riffle #, Replicate, EventID, and % Subsampled (Fig. 6) and click the “Open Count Form” button to enter occurrence data (Fig. 7).

Macroinvertebrate Occasion Data Entry Form

Prairie Aquatic Invertebrate Enumeration Form

These must be selected first.

Park: WICR

LocationID: WICR_AQUBUG_07

Riffle #: 1

Replicate: L

EventID: WICR_AQUBUG_2007-OCT-25

% Subsampled: 25

Open Count Form

Return to previous form

Figure 6. Preliminary data entry form for invertebrate counts. Data are selected from drop down boxes and upon clicking the “Open Count Form” are passed to the invertebrate count data form (Fig. 7).

3. Select the Taxa from the drop down box. Upon selection, the TaxonCode remains visible and the remaining values are hidden (inset of TaxonCode, Phylum, *etc.* added to demonstrate selection).
4. After selecting Taxa, enter the count.
5. Clicking the “Continue” button will prompt for the next Taxa.

Macroinvertebrate Data Entry Form

LocationID: WICR_AQUBUG_07 Rifle #: 1 Replicate: L EventID: WICR_AQUBUG_2007-OCT-25 You cannot modify these data

Taxa: 1. Select Taxa

TaxonCode	Phylum	Class	Order	Family	Genus
LYMN	Mollusca	Gastropoda	Bassomatophora	Lymnaeidae	
LYMNF0	Mollusca	Gastropoda	Bassomatophora	Lymnaeidae	Fossaria
PHYS	Mollusca	Gastropoda	Bassomatophora	Physidae	
PHYS0PH	Mollusca	Gastropoda	Bassomatophora	Physidae	Physa
PLAD	Mollusca	Gastropoda	Bassomatophora	Planorbidae	
VALV	Mollusca	Gastropoda	Heterostropha	Valvatidae	
ANCYFE	Mollusca	Gastropoda	Limnophila	Ancylidae	Ferissia

You cannot modify these data

Exit

Figure 7. Taxa count form for invertebrate data.

Entering Habitat and Discharge Data

Habitat data are entered by clicking the “Enter Habitat Data” button on the main switchboard and selecting the appropriate button for habitat type (see Fig. 8). The user is then presented with selection boxes (LocationID, EventID, *etc.*) for the appropriate habitat type and this data is passed to subforms where specific replicate data is entered. When finished entering habitat data, the user will be returned to the initial habitat form and may exit the form by clicking the “Return to previous menu” button.

Entering Water Quality Data

Water quality data collected by unattended CORE 5 data loggers (sondes) are uploaded from the logger using the manufacturer’s accompanying software program and saved in MS Excel. Data are then edited to correct any missing data due to logger maintenance (down time) and validated to determine if the data meet the expected range requirements or critical limits. CORE 5 water quality summary data are then entered into NPStoret by using the direct data entry templates for each characteristic by station and visit (*i.e.*, location and event). Coordinate data for logger locations are collected in accordance with the current HTLN spatial data collection techniques and entered into NPStoret. An NPStoret database is then sent to the WRD staff on an annual basis for initial QA/QC and subsequent upload into the WRD master copy of the EPA STORET.

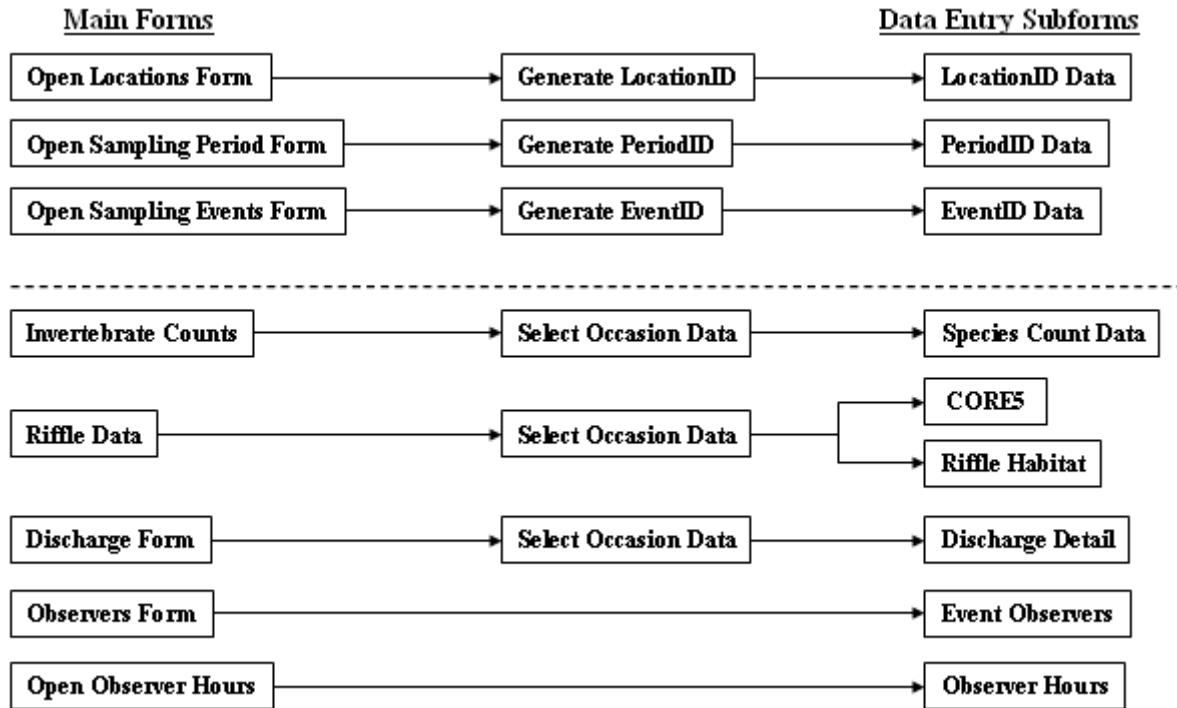


Figure 8. Outline of stream community data forms. Note: Forms are selected from the main switchboard and data entered into data entry subforms.

IV. Data Verification

Data verification immediately follows data entry and involves checking the accuracy of computerized records against the original source, usually paper field records. Data tables are queried to produce specific sets of data (*i.e.*, one season of invertebrate data or habitat data, *etc.*) and exported to Excel worksheets (via the “QAQC Data” and “Export Data” buttons, Fig. 2). These worksheets can then be compared to the original source (*i.e.*, field data sheets) by staff familiar with project design and field implementation in order to verify 100% of the records to identify missing, mismatched, or redundant records. A review of 10% of records is then conducted by the Project Manager and the results of that comparison reported with the data. If errors are found in the Project Manager’s review, then the entire data set is verified again.

Procedure:

1. Print pertinent data.
2. Compare 100% of the data with original field forms.
3. Reconcile errors in database.
4. Recheck 10% of records.

V. Data Validation

Although data may be correctly transcribed from the original field forms, they may not be accurate or logical. For example, field crews collect data on a given occasion (location and event) and the records residing in the database should reflect this. At any given occasion a query of these data should reflect these conditions and confirm the coincident (*i.e.*, relational) nature of the database. Missing or mismatched records can then be corrected.

As annual data are amassed in the database, validation is conducted via query and comparison among years to identify gross differences. For example, species A may be recorded at a location in a given year, but not in previous years, thus representing a possible new locality. Once verification and validation are complete, the data set is turned over to the Data Manager for archiving and storage. The data can then be used for all subsequent data activities.

Procedure:

1. Query pertinent data to insure resident data match existing protocols for each parameter.
2. Archive validated database.

Spatial Data Validation

Spatial validation of sample coordinates can be accomplished using the ArcMap component of ArcGIS. Coordinate data are maintained in the Access database and can be added to an ArcMap project and compared with existing features (*i.e.*, park boundaries, USGS Digital Orthophoto Quarter Quadrangles, National Hydrography Dataset hydrography, *etc.*) to confirm that coordinate data are valid.

Procedure:

1. Develop testing project within ArcMap constrained to appropriate UTM zone and projection (15N, NAD83).
2. Add park unit boundaries and any necessary spatial data (roads, water, contour, *etc.*).
3. Add relevant site coordinate data to testing project and validate against known features.
4. Correct mistakes, if necessary, and re-validate.

VI. Metadata Procedures

Biological and spatial metadata are developed in accordance with guidelines established in Rowell *et al.* (2005). Metadata for project data are developed using the NPS Database Metadata Extractor (a MS Access add-in), ESRI ArcCatalog 9, and the NPS Metadata Editor and Tools extension. Metadata follow the Federal Geographic Data Committee (FGDC) standards and include biological profile elements that allow integration of the metadata record with the NPS I&M NPSpecies and NatureBib online databases. Metadata are then parsed using a USGS metadata parser to check for errors. Once metadata are complete the database and metadata record can be uploaded to the NPS I&M online databases (see section X. Data Availability).

Procedure:

1. Document all resident MS Access database tables and field descriptions based on project protocols.
2. Use the Metadata Extractor add-in to harvest database metadata and export data to extensible markup language (.xml) file format.
3. Document the database in NatureBib acquiring a Bibkey ID and upload desktop NPSpecies data to online database.
4. In ArcCatalog, select the Metadata Editor NPS_Basic_Edit stylesheet to enter the required metadata elements. When complete, import the Integrated Taxonomic Information System (ITIS) generated taxonomic data into the current metadata file.
5. Parse the data using the USGS metadata parser and correct errors where necessary.

VII. File Organization

The various databases, reports, and GIS coverages used and generated by the Heartland Network create a large number of files and folders to manage. Poor file organization can lead to confusion and data corruption. As a standard data management technique, files pertaining to the project are managed in their own folder: Analysis, for data analysis; Data, for copies of archived data as well as data sheets; Documents, for supporting materials related to the project; and Spatial info, for various spatial data. The database is managed in the Databases folder and contains prior versions of the database in a subfolder.

VIII. Version Control

Prior to any major changes of a data set, a copy is stored with the appropriate version number. This allows for the tracking of changes over time. With proper controls and communication, versioning ensures that only the most current version is used in any analysis. Versioning of archived data sets is handled by adding a floating-point number to the file name, with the first version being numbered 1.00. Each major version is assigned a sequentially higher whole number. Each minor version is assigned a sequentially higher .1 number. Major version changes include migrations across Access versions and complete rebuilds of front-ends and analysis tools. Minor version changes include bug fixes in front-end and analysis tools. Frequent users of the data are notified of the updates, and provided with a copy of the most recent archived version.

IX. Backups

Secure data archiving is essential for protecting data files from corruption. Once a data set has passed the QA/QC procedures specified in the protocol, a new metadata record is created using the NPS Metadata Tools and Editor (in ArcCatalog). Backup copies of the data are maintained at both on- and off-site locations. An additional digital copy is forwarded to the NPS I&M Data Store. Tape backups of all data are made at regular intervals in accordance with current HTLN backup standard operating procedures. Tape backups will be made at least once per week, with tapes permanently archived semi-annually.

Procedure:

1. Create metadata record pursuant to data archiving.
2. Backup data.
3. Store backup copies on- and off-site and forward a copy to the I&M Data Store.
4. Administer regularly scheduled backups of data.

X. Data Availability

Currently, data are available for research and management applications for those database versions where all QA/QC has been completed and the data have been archived. Data can be transferred using ftp or by e-mail (where files are smaller than a few megabytes). Monitoring data will become generally available for download directly from the NPS I&M Data Store. Metadata for the stream invertebrate database are developed using ESRI ArcCatalog 9.2 and the NPS Metadata Tools and Editor extension and will be available at the NPS I&M GIS server (<http://science.nature.nps.gov/nrdata/>). Water quality data will be stored at the EPA STORET National Data Warehouse (EPA 2007) and be publicly accessible via the Internet. Additionally, data requests can be directed to:

Heartland I&M Network
Attn. Data Manager
Wilson's Creek National Battlefield
6424 W. Farm Road 182
Republic, MO 65738-9514
417-836-5313

Aquatic Invertebrate Monitoring Protocol for Small Streams in the Heartland I&M Network. Heartland I&M Network and Prairie Cluster Prototype Monitoring Program

SOP 8: Data Analysis

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP describes the metrics to be calculated for invertebrate data collected from small streams of the Heartland I&M Network, the use of control charts, and other potential analytic approaches.

I. Habitat and Water Quality Data

Summary statistics of habitat and water quality variables will provide information to park managers on the relative condition of the stream in relation to the invertebrate community metrics.

1. Once estimates for all parameters have been obtained for each sample, averages, ranges, and a measure of variability among individual samples (standard deviation) and riffles (standard error of the mean) are obtained for each sampling reach.
2. Categories for percentage class intervals are first converted to median values to estimate percent coverage for each variable (*i.e.*, 0 = 0%, 1 = 5%, 2 = 25%, 3 = 57.5%, 4 = 87.5%). Mean percent for each habitat variable is then calculated for each riffle and reach.

II. Multi-metrics

Several metrics or biological criteria will be used to analyze data collected under this protocol. Once estimates for all metrics have been obtained for each sample, averages and a measure of

variability (standard error of the mean) are calculated both within and among riffles (*i.e.*, site average).

Richness and Diversity Measures

1. Family Richness. This metric is calculated as the sum of families represented in replicate samples. Taxa richness generally increases with improving water quality, habitat diversity, or habitat suitability (Rabeni *et al.*, 1997).
2. Genus Richness. This is calculated as the number of invertebrate genera present in a sample. Lower richness may indicate impairment.
3. Genus Evenness. This metric is a measure of how evenly the total number of individuals in a sample are distributed across the genera. It is calculated as:

$$J' = H' / \ln(\text{genus richness})$$

Where H' is Shannon-Weiner Diversity.

Lower evenness indicates that a stream may have been subjected to disturbance and it is being populated by fewer, pollution tolerant genera.

4. EPT Richness. This metric is calculated as the total number of genera in the insect orders Ephemeroptera, Plecoptera, and Trichoptera (EPT). Lower richness may indicate that a stream is impaired. In general, the majority of taxa in these three orders are pollution intolerant.
5. EPT Ratio. The ratio of Ephemeroptera, Plecoptera, and Trichoptera (EPT) abundance (density) to Chironomidae abundance is calculated for replicate samples. Members of the EPT orders are generally considered pollution sensitive organisms and a decrease in their total number may indicate impairment. The Chironomidae often are more tolerant of disturbance and increases in their density may signal impairment. Calculate this metric as:

$$R = \text{EPT} / [\text{EPT} + \text{C}]$$

Where EPT is the abundance of EPT taxa and C is the abundance of Chironomidae.

6. Shannon Index (or Shannon-Weiner Index). This measure of taxa diversity assesses how the total number of individuals in a sample are distributed among the total species in the sample. The higher the score, the greater the degree of uncertainty that the next species will be the same as the previous one. High diversity generally implies better stream condition and it normally decreases with declining water quality because of reductions in both richness and evenness. It is calculated for each replicate sample as:

$$H' = -\text{SUM}\{p_i * \ln(p_i)\}$$

or

$$H = - \sum_{i=1}^S p_i \log p_i$$

where p_i is the density of species i (Shannon, 1948). This index is calculated for both the family and genus levels. While any base may be used, the natural log (ln) is the most common.

Pollution Tolerance Measures

1. Hilsenhoff Biotic Index (BI). This index was first developed by Hilsenhoff (1982) and subsequently modified by Hilsenhoff (1988). Each taxon is assigned a pollution tolerance value related to its assumed or known tolerance of water quality degradation. Tolerance values for taxa will follow Hilsenhoff (1982) and Lenant (1993) and are presented in SOP#6, Table 1. The HBI increases with increasing impairment.

Calculate as:

$$HBI = \sum n_i a_i / N$$

Where N is the total number of individuals in the sample, n is the total number of individuals for the i th genus, and a is the tolerance value for the i th genus.

III. Control Charts

The construction and interpretation of control charts is covered in many texts focusing on quality control in industry (*e.g.*, Beauregard *et al.*, 1992; Gyrya, 2001; Montgomery, 2001). The application of control charts for ecological purposes, however, is relatively straightforward. The use of control charts in environmental monitoring is discussed in texts by McBean and Rovers (1998) and Manly (2001), although not in as great detail as the texts referenced above focusing on industrial applications. Many different types of control charts could be constructed, depending upon the type of information desired. For example, control charts can be used to evaluate variables or attributes (*i.e.*, count or frequency data), and focus on measures of central tendency or dispersion.

Most traditional control charts assume that observations come from a normal distribution, or that data can be transformed to normality. In industry, control limits are often set at a distance of 3 standard deviations on either side of the centerline (Wetherill and Brown, 1991; Beauregard *et al.*, 1992; Montgomery, 2001). Thus, assuming a normal distribution centered at the centerline, the control limits would encompass 99.73% of the distribution.

Control limits may be constructed so as to contain any desired proportion of the distribution (*i.e.*, representing $[1-\alpha]$ confidence intervals for any α). In this case, choosing control limits is equivalent to specifying a critical region for testing the hypothesis that a specific observation is statistically different from the proposed centerline value. It is crucial that the centerline value is

representative of the true population parameter. Control limits could also be based on probabilistic thresholds other than confidence intervals (*e.g.*, McBean and Rovers, 1998).

If the observations cannot be assumed to come from a normal distribution, there are several options available beyond simple transformations of data. One option is to create subgroups of consecutive samples, and then use the subgroup averages, which will be approximately normally distributed in accordance with the central limit theorem (see Beauregard *et al.*, 1992; Montgomery, 2001). It is possible to construct control charts based on other distributions (*e.g.*, a Poisson distribution as in Atkinson *et al.*, 2003), and construct analogous confidence limits, as long as the distributions are known. Distribution-free confidence limits may also be calculated, although these will usually be relatively wide and less sensitive to changes (Conover, 1999).

It is not absolutely necessary to use values from a statistical sampling process to determine centerlines and thresholds for action. One can also subjectively choose a centerline value as the desired state and set threshold limits to match an acceptable amount of variability for the variable of interest. It is crucial to realize that this approach has no statistical basis, and thus probabilities cannot be readily associated with the observations. This application also has a precedent in industry. Such charts, which plot observations without relevance to an underlying distribution, have been termed ‘conformance charts’. Threshold values, which may be subjective, are termed ‘action limits’ (Beauregard *et al.*, 1992). If taking this approach, one should be very familiar with the system in question, and preferably select values that are defensible based on the data.

Although control charts have potentially wide applicability, each application may be different. A generic process for control chart construction is provided below, although decisions will always have to be made and an analyst familiar with control charts should ideally be consulted.

Steps in constructing a univariate control chart (see Fig. 1):

1. Determine the parameter of interest. This may be any of the metrics presented above.
2. After several years of data are available, plot the values of the parameter of interest (on the y-axis) against time (on the x-axis).
3. Determine a “center-line” value for this parameter, which could represent a mean of the observations, a target value, or some other value. Determining an appropriate center-line contains inherent pitfalls, and an analyst who is familiar with control charts should be consulted.
4. Establish control limits around the center-line. It is possible that only an upper control limit, or only a lower control limit, or both will be necessary, depending upon the parameter of interest and management concerns. Control limits may be based on a probability distribution and thus allow one to make statistical inferences, or they may be based on target levels set by management. Once again, determining appropriate control limits can be tricky, especially if statistical inferences are desired, and an analyst who is familiar with control charts should be consulted.

5. Continue to plot values of the parameter of interest over time as new data become available. If an observation exceeds the control limit(s), this is indicative of the potential need for management action, or a more focused study.

Control charts should be constructed after several years of data are available, and updated annually. This use of a control chart is not based on any statistical distribution, and thus no statistical inferences are made, it is simply a management tool. Additional control charts can be constructed from other variables of interest as described above.

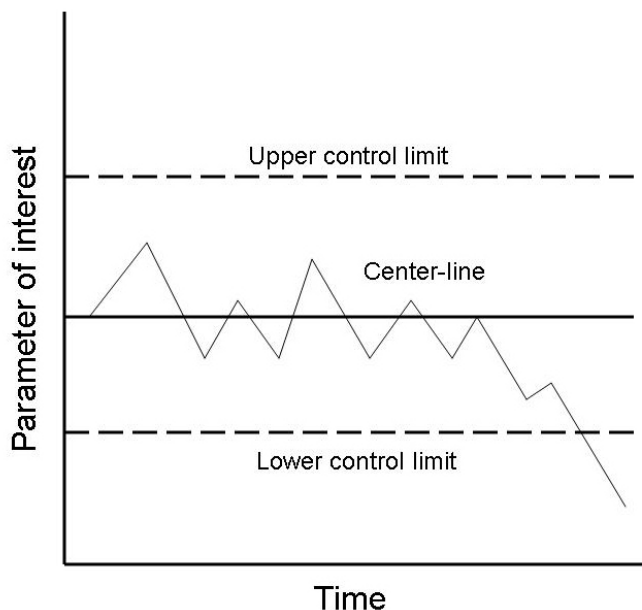


Figure 1. Generic univariate control chart.

IV. Other methods

If a hypothesis testing approach is deemed appropriate, many tests may be employed, depending upon the question being asked and the structure of the data. For example, a Kruskal-Wallis ANOVA may be used to test for significant differences between riffles using metric scores from each of the pseudoreplicates. If there is reason to compare more than two variables among samples, Friedman's non-parametric two-way analysis of variance should be used. Alternatively, Cochran's Q is particularly useful for measuring changes in frequencies (proportions) across time.

Quantitative Similarity analyses can be performed on each possible pairing of samples to produce a similarity matrix. Then spatial and temporal analysis with previously collected data can be done using a Mean Similarity Analysis (Van Sickle, 1997) or Wilcoxon's matched pairs test.

A Detrended Correspondence Analysis (DCA) may be used to look for spatial relationships. This statistic can be used to produce a two-dimensional graph where samples of similar taxonomic composition are near each other and dissimilar entities are far apart. Equal distances

in the ordination correspond to equal differences in species composition. Samples from the previous years can be compared over time and samples can be compared from different locations in the same year. In addition, environmental gradients can be inferred from the species composition data by performing a Spearman rank correlation between the rankings of the sample scores on the first two axes and rankings of the environmental variables (water quality and habitat data).

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SOP 9: Data Reporting

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP gives instructions for reporting on collected invertebrate data. It also describes the procedure for formatting a report, the review process, and distribution of completed reports. Efficient reporting of monitoring results is critical in assisting park Resource Managers in management decisions.

I. Report Format

Template

The report template for regional natural resource technical reports should be followed (<http://www.nature.nps.gov/publications/NRPM/index.cfm>). Natural resource reports are the designated medium for disseminating high priority, current natural resource management information with managerial application. The natural resource technical reports series is used to disseminate the results of scientific studies in the physical, biological, and social sciences for both the advancement of science and the achievement of the National Park Service's mission.

Style

Standards for scientific writing as recommended in the CBE Style Manual (1994) should be followed. Reports should be direct and concise. Refer to CBE Style Manual Committee (1994), Mack (1986), Goldwasser (1999), Strunk and White (1999), and Day and Gastel (2006).

II. Types of Reports and Review Procedure

Table 1. Summary of types of reports produced and review process. Adapted from DeBacker *et al.*, 2005.

Type of Report	Purpose of Report	Primary Audience	Review Process	Frequency
Status Reports for Specific Protocols	Summarize monitoring data collected during the year and provide an update on the status of selected natural resources. Document related data management activities and data summaries.	Park resource managers and external scientists	Internal peer review by HTLN staff	Every 3 years
Executive Summary of Annual Reports for Specific Protocols	Same as Annual Status Reports but summarized to highlight key points for non-technical audiences.	Superintendents, interpreters, and the general public	Internal peer review by HTLN staff	Simultaneous with Annual Status Reports
Comprehensive Trends and Analysis and Synthesis Reports	Describe and interpret trends in individual vital signs. Describe and interpret relationships among observed trends and park management, known stressors, climate, <i>etc.</i> Highlight resources of concern that may require management action.	Park resource managers and external scientists	Internal peer review by HTLN staff	Following at least 3 sampling events per park
Executive Summary of Comprehensive Trends and Analysis and Synthesis Reports	Same as Comprehensive Trends and Analysis and Synthesis Reports, but summarized to highlight findings and recommendations for non-technical audiences.	Superintendents, interpreters, and the general public	Internal peer review by HTLN staff	Simultaneous with Comprehensive Trends Analysis and Synthesis Reports

III. Distribution Procedure

Annual reports will be provided to the respective parks where aquatic invertebrate monitoring was done. Additionally, a copy will be kept on file with the HTLN office of the National Park Service, Republic, Missouri, and made available to all interested parties upon request. All data collected by the HTLN is public property and subject to requests under the Freedom of Information Act (FOIA). Reports are converted to Adobe .pdf files and posted on the HTLN website: (<http://science.nature.nps.gov/im/units/htln/aquabugs.cfm>).

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SOP 10: Procedures and Equipment Storage After the Field Season

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This Standard Operating Procedure explains procedures that all field observers using this Invertebrate Monitoring Protocol should be familiar with and follow after the field season is completed.

Procedures:

Equipment

1. Clean and repair all equipment prior to return to the proper storage areas.
2. Check sampling nets and determine if new nets must be ordered prior to the next field season.
3. Remove batteries from all equipment.

Paperwork and Reports

1. All reference manuals should be re-shelved on their appropriate bookshelf. Other reference materials and extra data sheets should be filed in their appropriate filing cabinet. Clean the insides and outsides of all vehicles used in the field.
2. At the end of each field season, after all sampling has been completed, the project manager will file a trip report with the data manager outlining hours worked, field-crew members and their responsibilities on the project, and any unique situations encountered. This information is incorporated in the database and used during data analysis, and it may be useful in identifying causes for discrepancies and inconsistencies in the data.

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SOP 11: Revising the Protocol

Version 1.00 (6/2/2008)

Revision History Log:

Previous Version #	Revision Date	Author	Changes Made	Reason for Change	New Version #

This SOP explains how to make changes to the Monitoring Protocol Narrative and accompanying SOPs, and how to track these changes. Anyone asked to edit the Protocol Narrative or SOPs must follow this outlined procedure in order to eliminate confusion in how data is collected and analyzed.

Procedures:

1. This Monitoring Protocol and accompanying SOPs uses sound methodologies for collecting and analyzing aquatic invertebrate data. However, all protocols require editing as new and different information becomes available. Required edits should be made in a timely manner and appropriate reviews undertaken.
2. All edits require review for clarity and technical soundness. Small changes or additions to existing methods will be reviewed in-house by the HTLN staff. If a complete change in methods is sought, however, an outside review is required. Regional and national staff of the National Park Service with familiarity in aquatic invertebrate research and data analysis will be utilized as reviewers. Also, experts in aquatic invertebrate research and statistical methodologies outside of the Park Service will be used in the review process.
3. Document edits and protocol versioning in the Revision History Log that accompanies the Protocol Narrative and each SOP. Log changes in the Protocol Narrative or SOP being edited. Version numbers increase incrementally by hundredths (*e.g.*, version 1.01, version 1.02, ...*etc.*) for minor changes. Major revisions should be designated with the next whole number (*e.g.*, version 2.0, 3.0, 4.0 ...*etc.*). Record the previous version number, date of revision, author of the revision, identify paragraphs and pages where changes are made, and the reason for making the changes along with the new version number.

4. Inform the Data Manager about changes to the Protocol Narrative or SOP so the new version number can be incorporated in the Metadata of the project database. The database may have to be edited by the Data Manager to accompany changes in the Protocol Narrative and SOPs.
5. Post new versions of the protocol on the Heartland Network internet website and forward copies to all individuals with a previous version of the affected Protocol Narrative or SOP.

Appendix A. Historic benthic invertebrate monitoring data (1988-2007) for selected parks in the Heartland Inventory & Monitoring Network

Table 1. Mean invertebrate metrics collected from Hoover Creek, HEHO, 1989-1990. Data are from Harris *et al.* (1991). N= number of samples collected.

Metric	Hoover Creek
N	5
Density (no./m ²)	2145
Genus Richness	18
EPT Richness	4
Shannon's Index	1.26

Table 2. Mean invertebrate metrics collected from Carver Creek, GWCA, 1989-2007. Data for 1989 and 1996 are from Harris *et al.* (1991) and Peterson (1997), respectively. Hilsenhoff Biotic Index was based on family-level scores prior to 2005. N= number of samples collected.

Carver Creek					
Metric	1989	1996	2005	2006	2007
N	15	5	15	9	9
Family Richness	n/a	14.60 (1.78)	14.2 (1.09)	15.33 (0.5)	16 (1.12)
Genus Richness	33	n/a	15.87 (1.17)	17 (0.68)	17.56 (1.12)
EPT Richness	11	n/a	7.4 (0.40)	6.89 (0.56)	6.89 (0.56)
EPT Ratio	n/a	0.48 (0.07)	0.38 (0.04)	0.48 (0.05)	0.68 (0.06)
Shannon Index	2.14	1.84 (0.12)	1.74 (0.8)	2.11 (0.07)	0.79 (0.02)
Shannon Evenness Index	n/a	n/a	0.64 (0.03)	0.74 (0.02)	2.26 (0.08)
Hilsenhoff Biotic Index	n/a	4.75 (0.09)	5.23 (0.99)	4.23 (0.12)	4.62 (0.13)

Table 3. Mean invertebrate metrics collected from Williams Branch, GWCA, 1989-2007. Data for 1989 are from Harris *et al.* (1991). N= number of samples collected.

Williams Branch				
Metric	1989	2005	2006	2007
N	15	15	9	9
Family Richness	n/a	11.8 (0.62)	15.33 (0.85)	14 (0.62)
Genus Richness	37	13.87 (0.69)	17 (1.03)	15.44 (0.85)
EPT Richness	15	5.33 (0.39)	5.89 (0.42)	6 (0.37)
EPT Ratio	n/a	0.48 (0.07)	0.52 (0.05)	0.68 (0.05)
Shannon Index	2.29	1.80 (0.07)	2.04 (0.08)	2.03 (0.08)
Shannon Evenness Index	n/a	0.74 (0.04)	0.72 (0.02)	0.79 (0.01)
Hilsenhoff Biotic Index	n/a	5.54 (0.11)	4.30 (0.5)	4.44 (0.17)

Table 4. Mean and standard error (in parentheses) of invertebrate metrics collected from Harkins Branch, GWCA, 1996-2007. Data for 1996 are from Peterson (1997). Hilsenhoff Biotic Index was based on family-level scores prior to 2005. N= number of samples collected.

Harkins Branch				
Metric	1996	2005	2006	2007
N	5	10	9	9
Family Richness	17.80 (2.03)	10.6 (0.87)	14 (1.28)	15.11 (1.12)
Genus Richness	n/a	12.1 (0.87)	15.11 (1.44)	16.11 (1.21)
EPT Richness	n/a	6.4 (0.43)	7.33 (0.94)	7.56 (0.58)
EPT Ratio	0.51 (0.05)	0.51 (0.06)	0.65 (0.08)	0.79 (0.02)
Shannon Diversity Index	2.03 (0.08)	1.88 (0.12)	1.99 (0.09)	2.27 (0.06)
Shannon Evenness Index	n/a	0.74 (0.04)	0.74 (0.02)	0.83 (0.01)
Hilsenhoff Biotic Index	4.87 (0.12)	5.04 (0.14)	4.82 (0.32)	4.30 (0.50)

Table 5. Mean and standard error (in parentheses) of invertebrate metrics collected from Cub Creek, HOME 1989-2007. N= number of samples collected. Data prior to 2005 are from Peitz and Cribbs (2005a). Hilsenhoff Biotic Index was based on family-level scores prior to 2005. Genus richness for 2005-2007 does not include Chironomidae genera.

Year	1989	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007
N	10	10	20	20	20	25	20	20	30	30	27	19	12
Density	3298.2 (1005.4)	1422 (33.37)	3232 (967.89)	4947.3 (979.78)	3805.7 (794.98)	2414.2 (380.05)	2651.2 (921.96)	2872 (220.8)	2774 (412.89)	3414.7 (937.31)	n/a	n/a	n/a
Genus Richness	11.9 (1.30)	12 (1.6)	20.4 (1.93)	15.6 (1.39)	19.88 (3.1)	14 (0.57)	13.18 (2.56)	13.26 (0.84)	15.29 (1.24)	15.45 (0.76)	7.89 (3.63)	6.53 (0.39)	8.83 (0.46)
EPT Richness	0.4 (0.2)	5.3 (0.3)	5.1 (0.95)	2.85 (0.29)	7.15 (0.83)	4.84 (0.69)	5.13 (0.53)	4.62 (0.68)	5.02 (0.71)	3.1 (0.26)	3.63 (0.27)	1.47 (0.16)	3.75 (0.37)
Shannon Index (Genus)	1.2 (0.15)	1 (0.05)	2.07 (0.14)	1.84 (0.15)	1.84 (0.19)	1.45 (0.06)	1.81 (0.29)	1.5 (0.12)	1.73 (0.14)	1.99 (0.11)	1.23 (0.05)	1.46 (0.06)	1.41 (0.06)
Shannon Evenness Index	0.49 (0.08)	0.4 (0)	0.69 (0.04)	0.68 (0.04)	0.62 (0.05)	0.55 (0.03)	0.72 (0.07)	0.6 (0.03)	0.63 (0.04)	0.73 (0.03)	0.61 (0.02)	0.82 (0.03)	0.65 (0.03)
Hilsenhoff Biotic Index	7.75 (0.04)	4.27 (0.02)	5.81 (0.5)	5.76 (0.35)	4.6 (0.09)	4.57 (0.14)	4.91 (0.35)	4.92 (0.18)	5.02 (0.15)	5.62 (0.27)	6.51 (0.05)	6.43 (0.18)	6.55 (0.10)

Table 6. Mean and standard error (in parentheses) of invertebrate metrics collected from Pipestone Creek, PIPE 1989-2007. N= number of samples collected. Data prior to 2005 are from Peitz and Cribbs (2005b). Hilsenhoff Biotic Index was based on family-level scores prior to 2005. Genus richness for 2005-2007 does not include Chironomidae genera.

Year	1989	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007
N	10	10	30	30	30	30	30	30	30	20	19	17	9
Family Richness	8.8 (0)	6.6 (2.2)	5.27 (1.57)	3.33 (0.27)	9.43 (0.96)	9.33 (1.02)	6.87 (0.74)	8.4 (0.88)	6.83 (1)	6.6 (0.73)	7.37 (0.50)	8.41 (0.45)	10 (0.73)
Taxa (Genus) Richness	12.5 (0.3)	9.2 (3)	8.33 (2.45)	4.22 (0.53)	15.03 (1)	14 (1.49)	10.73 (1.09)	13.87 (0.94)	9.6 (1.2)	11.4 (1.19)	9 (0.59)	9.18 (0.36)	11.44 (0.78)
EPT Richness	3.1 (0.1)	3.2 (0)	2.67 (1.23)	1.23 (0.2)	6.2 (0.38)	4.53 (0.47)	4.2 (0.21)	5.7 (0.51)	4.23 (0.42)	5.45 (1.07)	3.84 (0.31)	3.65 (0.19)	5 0.17
EPT Ratio	0.63 (0.05)	0.62 (0.13)	0.36 (0.06)	0.53 (0.11)	0.32 (0.05)	0.23 (0.06)	0.4 (0.03)	0.4 (0.05)	0.77 (0.09)	0.65 (0.05)	0.74 (0.04)	0.21 (0.03)	0.58 (0.06)
Shannon Index (Family)	1.66 (0.03)	1.44 (0.27)	0.94 (0.17)	0.9 (0.1)	1.16 (0.14)	1.09 (0.16)	1.19 (0.13)	1.3 (0.1)	1.24 (0.15)	1.28 (0.12)	n/a	n/a	n/a
Shannon Index (Genus)	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	1.49 (0.07)	1.24 (0.06)	1.73 (0.08)
Genus Evenness	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	0.68 (0.03)	0.59 (0.05)	0.71 (0.03)
Hilsenhoff Biotic Index	5.32 (0.14)	4.85 (0.14)	5.12 (0.21)	4.67 (0.33)	5.34 (0.12)	5.59 (0.11)	5.38 (0.1)	5.17 (0.07)	4.43 (0.32)	4.65 (0.13)	4.13 (0.18)	5.52 (0.08)	5.64 (0.16)

Table 7. Mean and standard error (in parentheses) of invertebrate metrics collected from Wilson's Creek, WICR, 1988-2007. N= number of samples collected. Data prior to 2005 are from Peitz and Cribbs (2005c). Hilsenhoff Biotic Index was based on family-level scores prior to 2005. Genus richness for 2005-2007 does not include Chironomidae genera.

Year	1988	1989	1990	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007
N	10	20	10	20	30	20	20	10	30	30	30	30	30	18	9
Family Richness	10 (1)	12.5 (0.52)	9.6 (0.2)	10.8 (1.1)	12.17 (1.45)	11.8 (1)	8.93 (0.71)	5.8 (0.2)	8.57 (0.47)	8.53 (0.26)	7.73 (0.28)	7.23 (0.71)	9.97 (0.45)	9.5 (0.42)	10.78 (0.52)
Taxa (Genus) Richness	12 (1.4)	16.4 (0.83)	13.6 (0.2)	20.95 (1.44)	18.77 (1.79)	18.5 (1.48)	14.5 (1.12)	10.3 (0.3)	14.63 (0.61)	13.4 (0.75)	12.6 (0.78)	9.87 (1.04)	17.63 0.83)	13.56 (0.46)	14.89 (0.54)
EPT Richness	3.9 (0.3)	3.1 (0.68)	1.4 (0)	5.15 (0.44)	4.57 (0.84)	5.65 (0.43)	3.1 (0.71)	2.5 (0.1)	4.6 (0.2)	3.83 (0.28)	3.83 (0.51)	1.87 (0.38)	3.57 (0.22)	2.47 (0.21)	3.78 (0.52)
EPT Ratio	0.61 (0.09)	0.26 (0.03)	>0.01 (>0.01)	0.27 (0.06)	0.25 (0.08)	0.56 (0.02)	0.37 (0.1)	0.11 (0.03)	0.39 (0.02)	0.28 (0.04)	0.37 (0.13)	0.33 (0.08)	0.29 (0.04)	0.57 (0.04)	0.18 (0.03)
Shannon Index (Family)	1.47 (0.16)	1.09 (0.09)	0.72 (0.01)	1.15 (0.08)	1.23 (0.08)	1.62 (0.08)	1.27 (0.08)	0.83 (0.06)	1.45 (0.07)	1.38 (0.04)	1.35 (0.05)	1.43 (0.11)	1.42 (0.04)	n/a	n/a
Shannon Index (Genus)	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	1.94 (0.04)	2.03 (0.06)	1.93 (0.14)
Shannon Evenness Index	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	0.71 (0.01)	0.77 (0.01)	0.72 (0.05)
Hilsenhoff Biotic Index	5.33 (0.13)	5.61 (0.09)	6.61 (0.11)	5.85 (0.18)	5.94 (0.19)	5.5 (0.05)	6.17 (0.31)	6.61 (0.08)	5.77 (0.11)	5.87 (0.18)	5.83 (0.35)	5.24 (0.25)	5.98 (0.10)	5.37 (0.15)	6.72 (0.69)

Table 8. Mean and standard error (in parentheses) of invertebrate metrics collected from Skegg's Branch, WICR, 1988-2007. N= number of samples collected. Data prior to 2005 are from Peitz and Cribbs (2005c). Hilsenhoff Biotic Index was based on family-level scores prior to 2005.

Year	1988	1989	1990	1997	1999	2001	2002	2003	2004	2005	2006	2007
N	5	10	5	10	10	15	10	5	20	5	9	9
Family Richness	12 n/a	17.3 (0.3)	17 n/a	17.3 (1.1)	12.5 (0.7)	10.87 (0.74)	8 (0.2)	8.2 n/a	8.68 (1)	9.78 (0.74)	11 (0.85)	11.89 (0.59)
Taxa (Genus) Richness	14 n/a	23.5 (0.5)	23.8 n/a	25.8 (0.6)	20 (1.8)	18.73 (0.53)	15.7 (1.3)	14 n/a	11.88 (2.08)	18.56 (1.56)	20 (1.5)	21.67 (1.42)
EPT Richness	3.8 n/a	6.2 (0.4)	5.6 n/a	5.1 (0.5)	2.8 (0.4)	4.87 (0.18)	2.8 (0.2)	4.4 n/a	2.45 (0.9)	3 (0.37)	3.89 (0.48)	3.78 (0.52)
EPT Ratio	0.72 n/a	0.55 (0.16)	0.58 n/a	0.27 (0.03)	0.29 (0.09)	0.45 (0.02)	0.33 (0.05)	0.45 n/a	0.44 (0.1)	0.36 (0.06)	0.28 (0.04)	0.18 (0.03)
Shannon Index (Family)	1.92 n/a	1.92 (0.02)	1.86 n/a	1.96 (0.02)	1.7 (0.09)	1.76 (0.12)	1.35 (0.07)	1.24 n/a	1.7 (0.12)	0.72 (0.04)	n/a	n/a
Shannon Index (Genus)	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	2.31 (0.09)	2.34 (0.10)	2.0 (0.12)
Shannon Evenness Index	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	n/a	0.80 (0.02)	0.77 (0.02)	0.66 (0.03)
Hilsenhoff Biotic Index	5.81 n/a	5.69 (0.11)	5.70 n/a	6.12 (0.19)	5.5 (0.12)	5.21 (0.09)	5.41 (0.09)	6.24 n/a	4.95 (0.07)	4.39 (0.29)	5.49 (0.10)	5.72 (0.22)

Table 9. Mean and standard error (in parentheses) of invertebrate metrics collected from Terrell Creek, WICR, 2006-2007. N= number of samples collected.

Terrell Creek		
Year	2006	2007
N	9	9
Family Richness	15.89 (1.12)	14.78 (0.94)
Taxa (Genus) Richness	25.44 (1.99)	22.44 (1.33)
EPT Richness	7.78 (0.68)	4.56 (0.58)
EPT Ratio	0.43 (0.04)	0.15 (0.04)
Shannon Index (Genus)	2.69 (0.10)	2.19 (0.10)
Shannon Evenness Index	0.83 (0.02)	0.71 (0.03)
Hilsenhoff Biotic Index	5.00 (0.18)	5.30 (0.20)

Appendix B. Maps showing the lower sampling reach boundaries of streams locations in network parks.

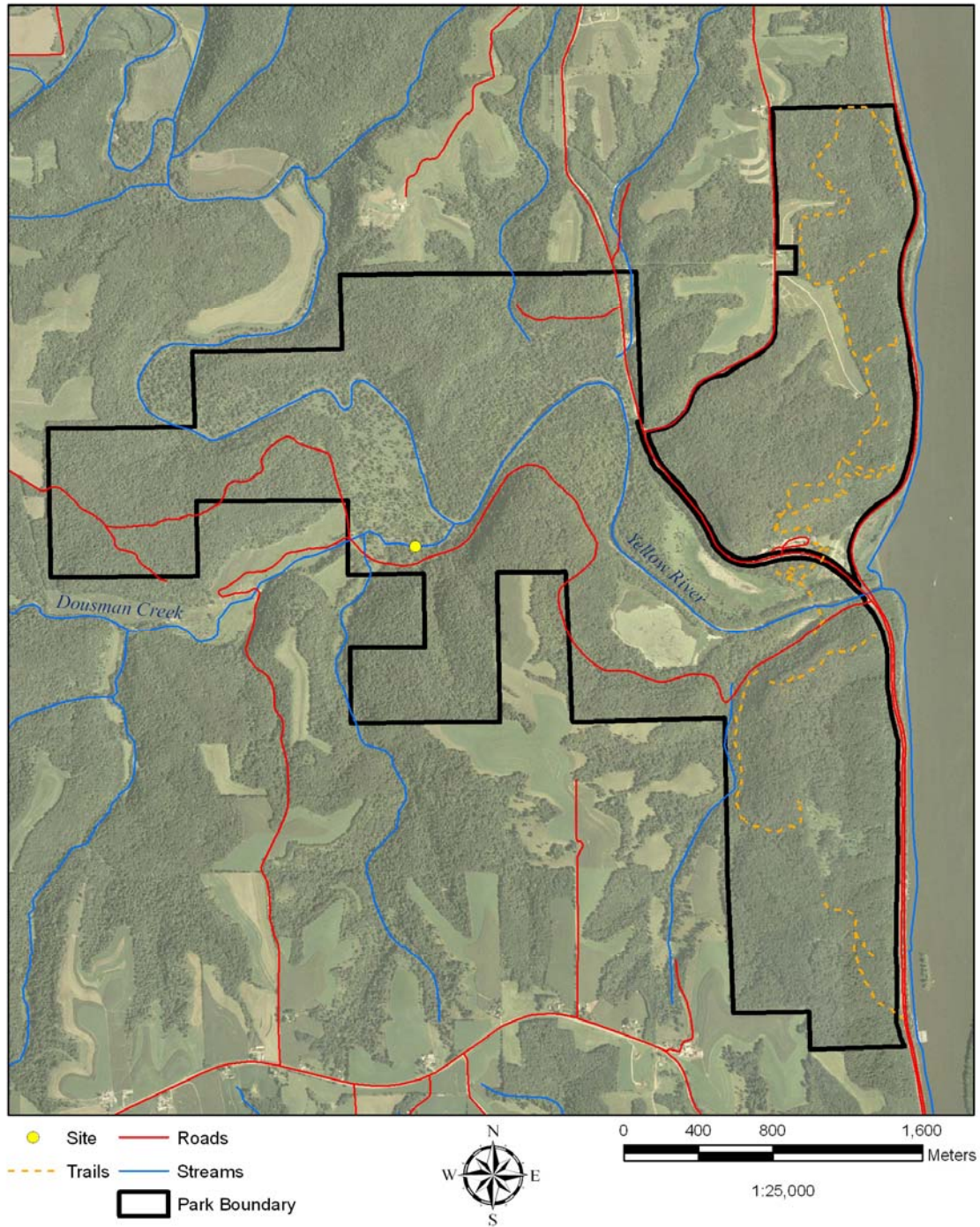


Figure 1. Map showing the approximate lower sampling reach boundary for Dousman Creek, EFMO.

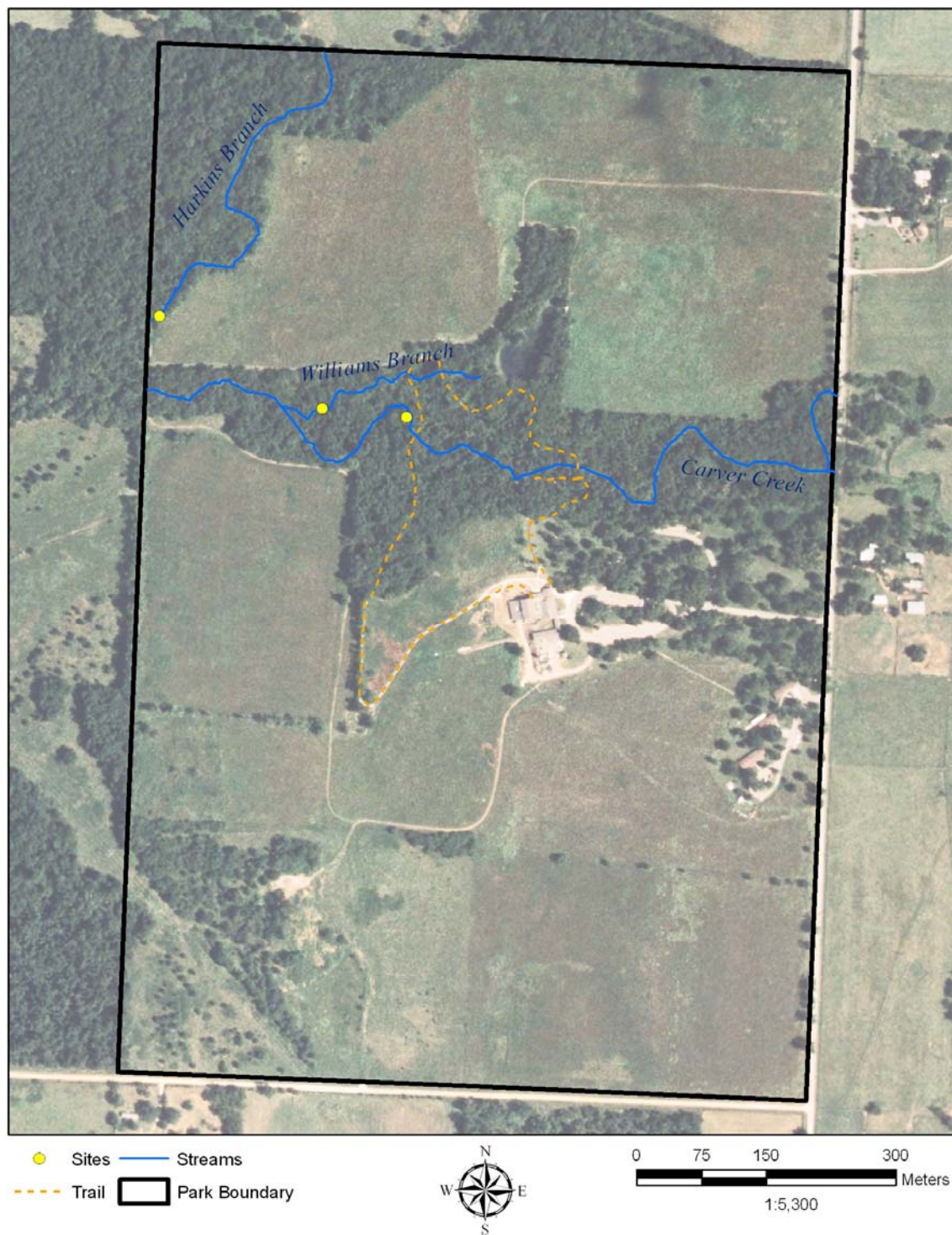


Figure 2. Map showing the approximate lower sampling reach boundaries for Carver Creek, Williams Branch and Harkins Branch, GWCA.

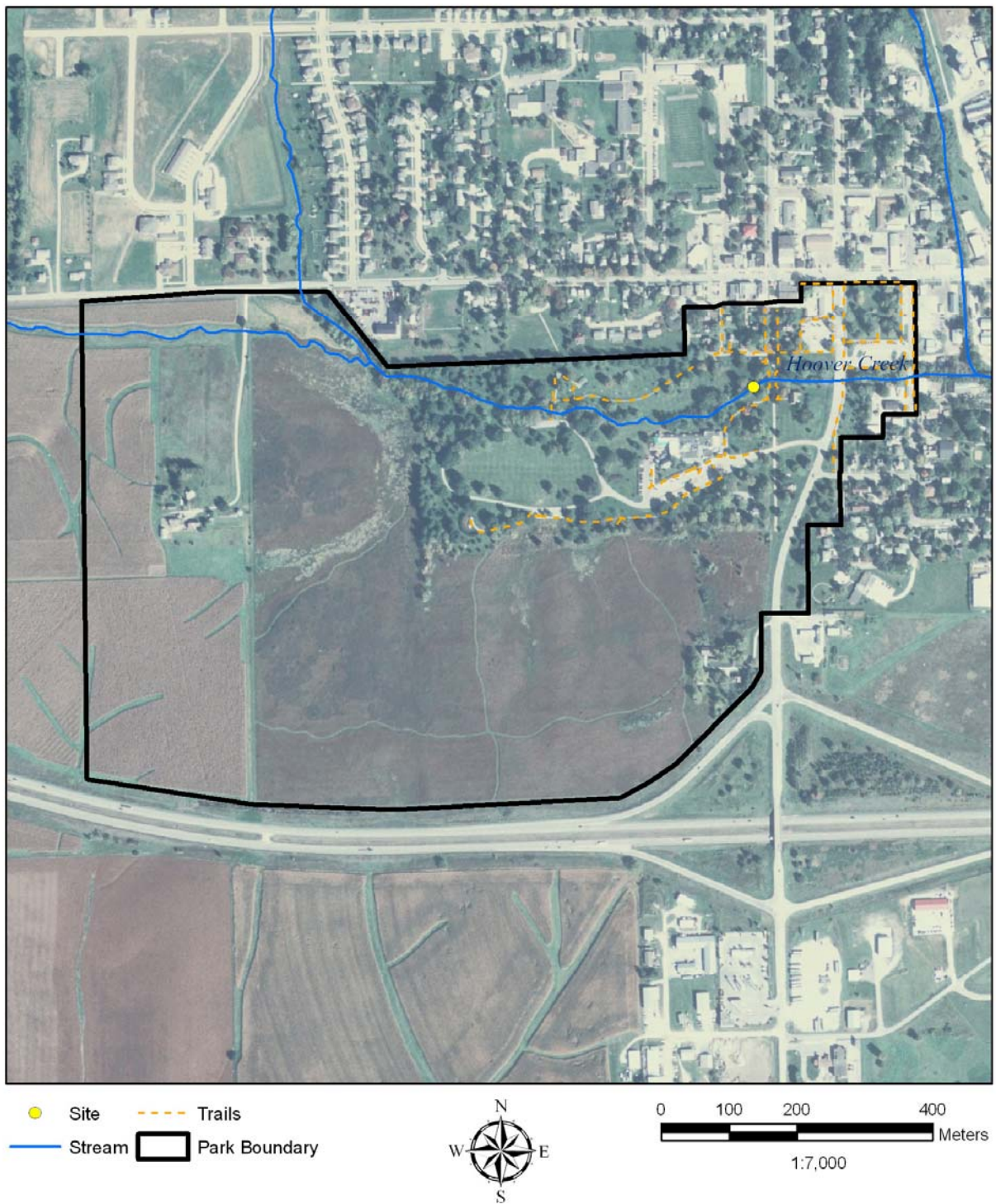


Figure 3. Map showing the approximate lower sampling reach boundary for Hoover Creek, HEHO.

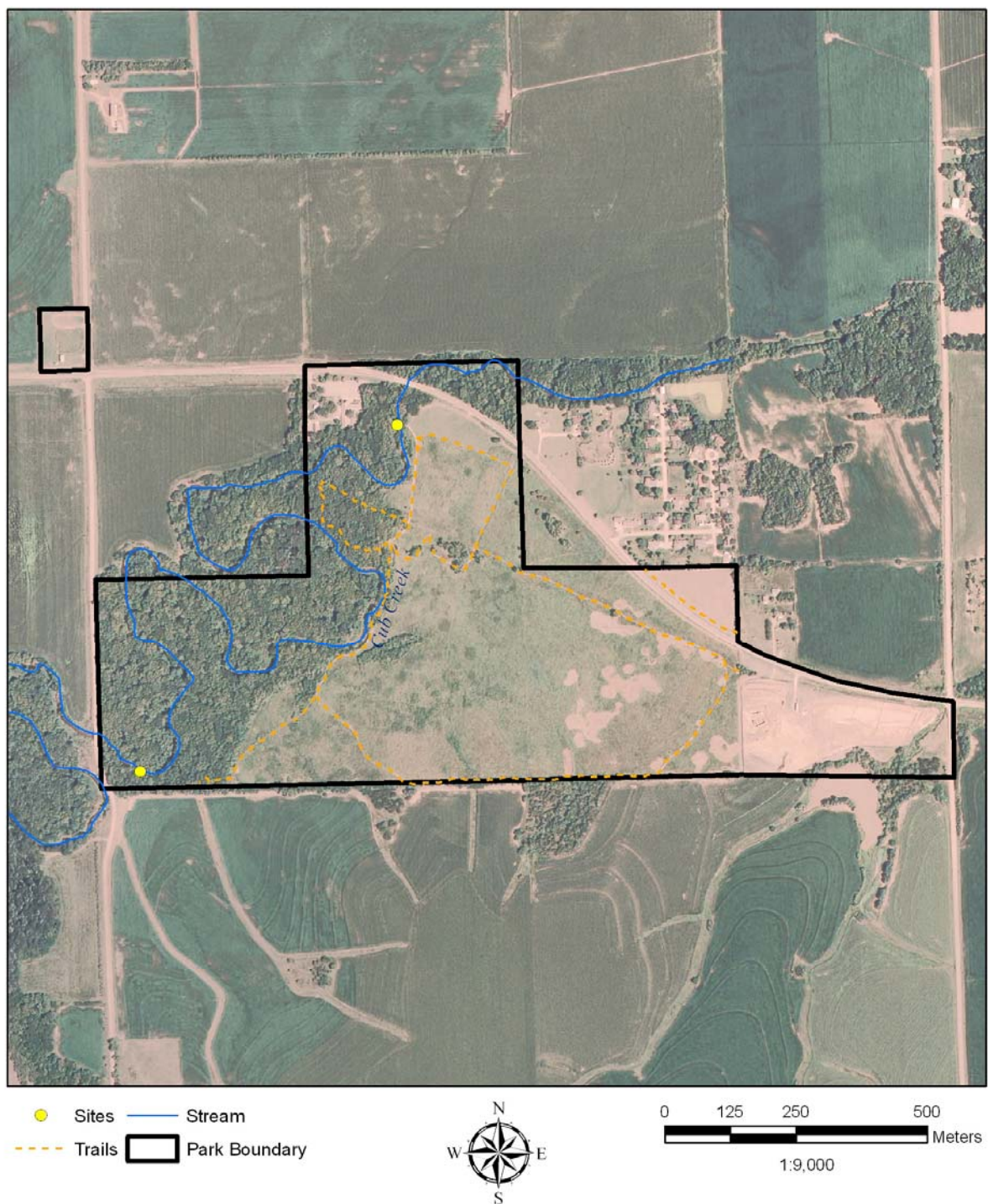


Figure 4. Map showing the approximate upper and lower sampling locations for Cub Creek, HOME.

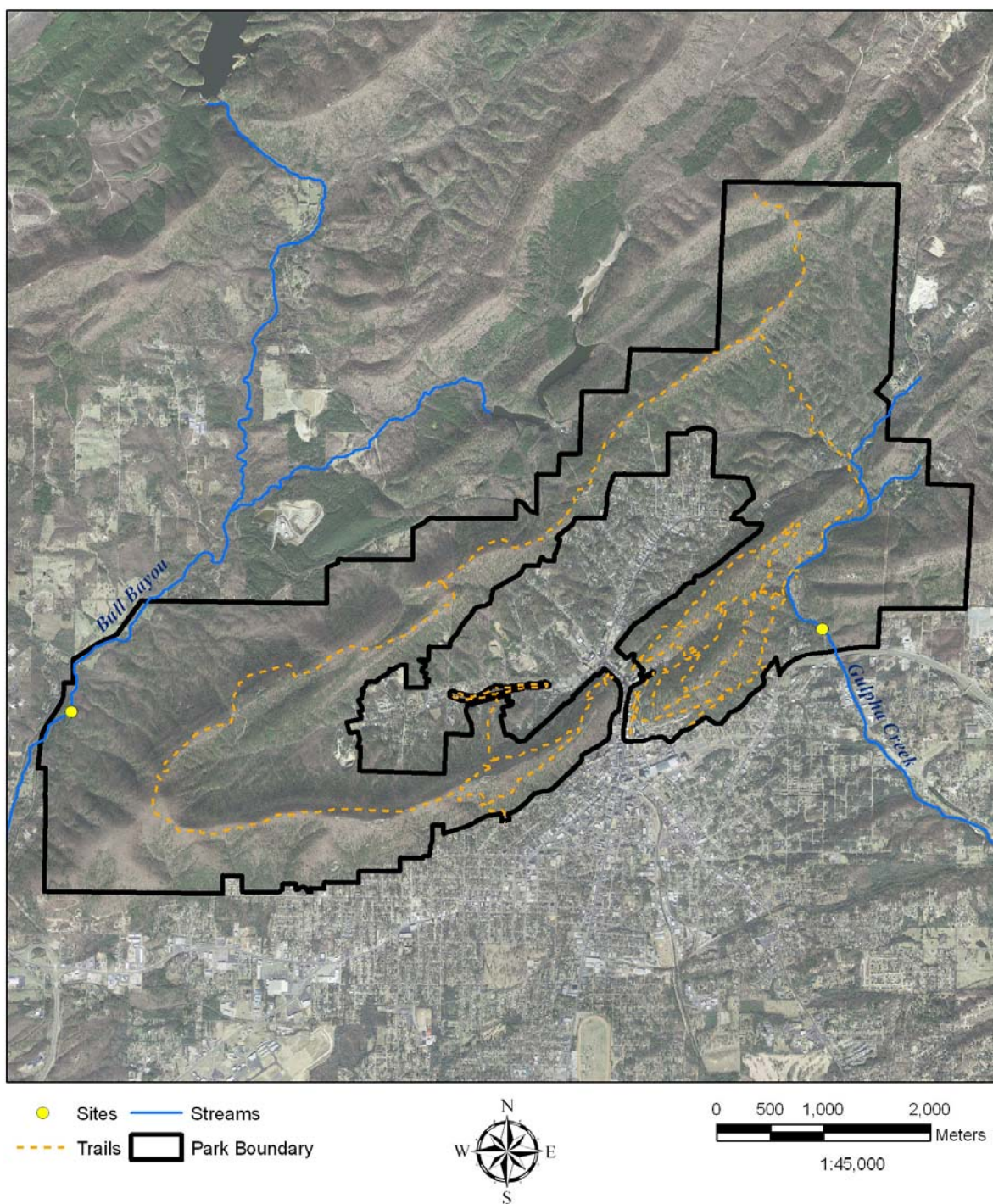


Figure 5. Map showing the approximate lower sampling reach boundaries for Bull Bayou and Gulpha Creek, HOSP.

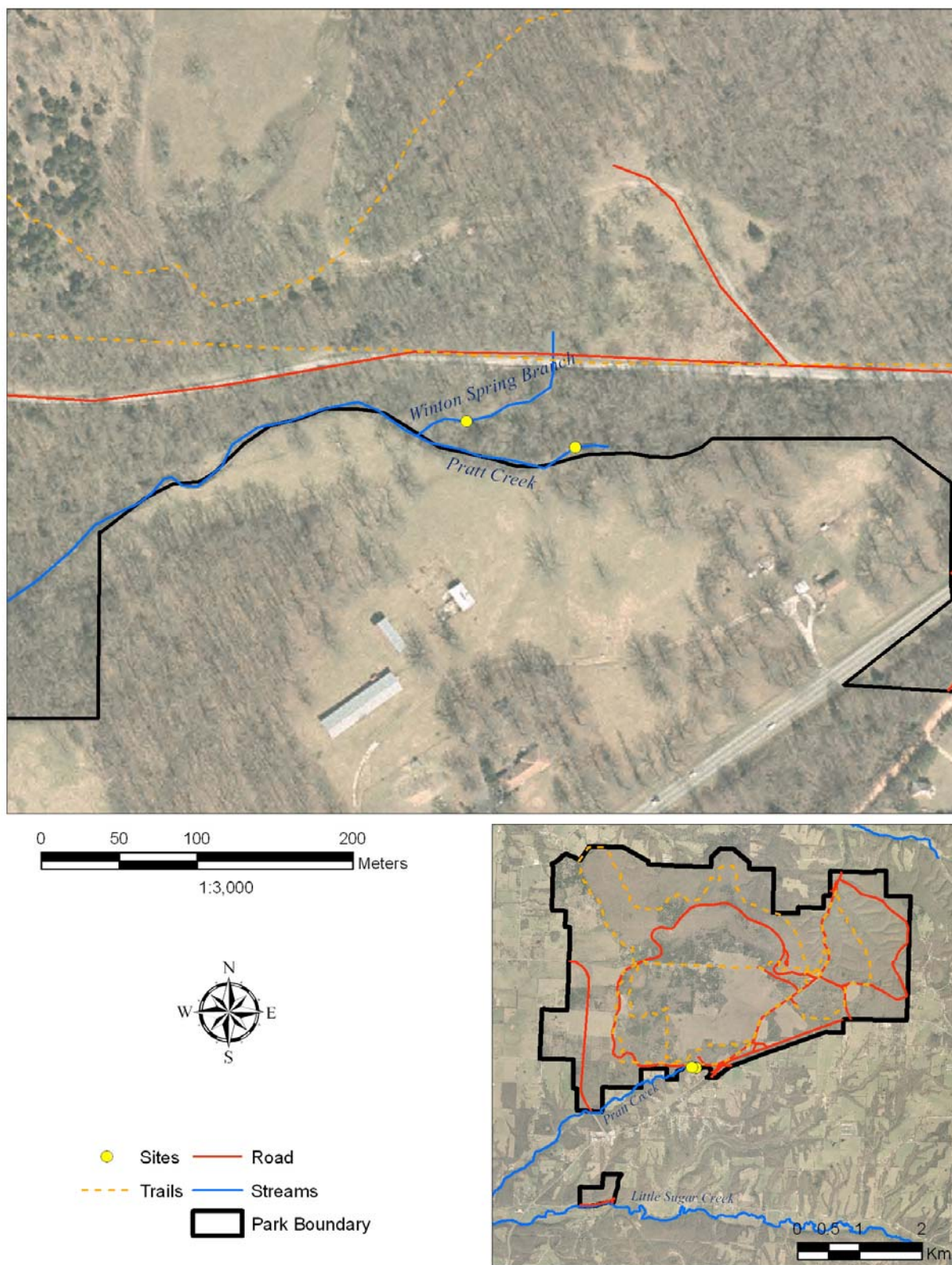


Figure 6. Map showing the approximate lower sampling reach boundaries for Pratt Creek and Winton Spring Branch, PERI.

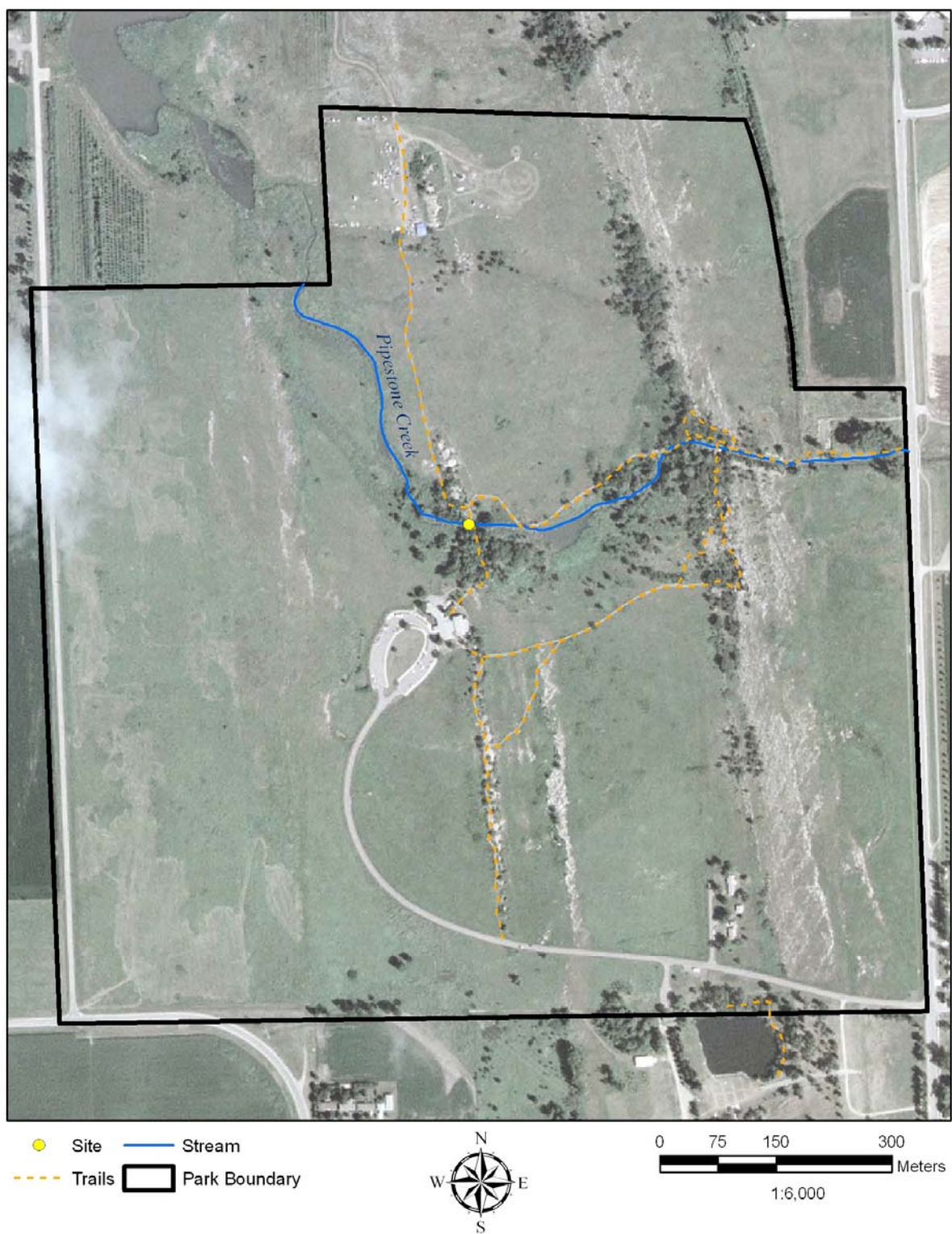


Figure 7. Map showing the approximate lower sampling reach boundary for Pipestone Creek, PIPE.

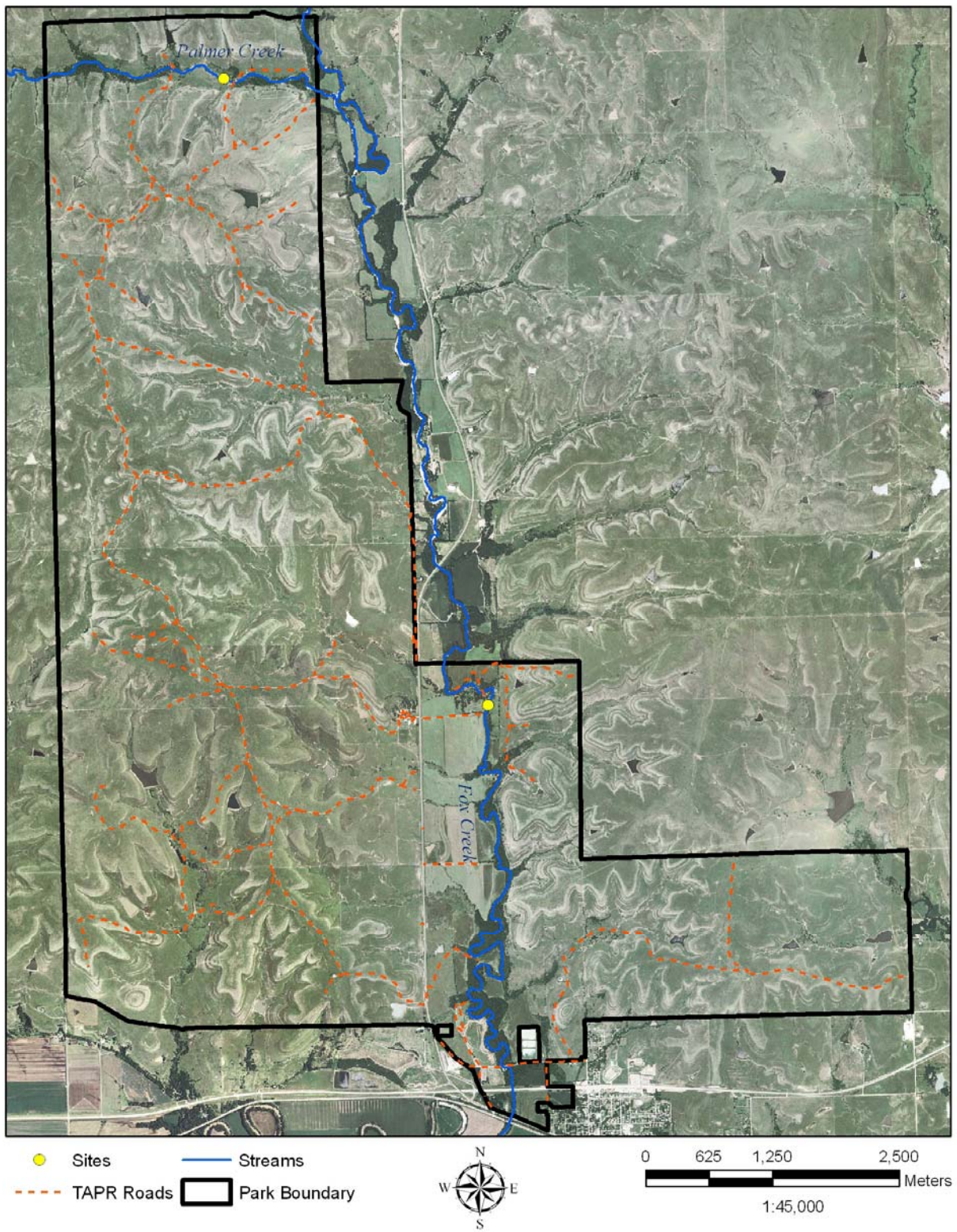


Figure 8. Map showing the approximate lower sampling reach boundary for Fox Creek and Palmer Creek, TAPR.

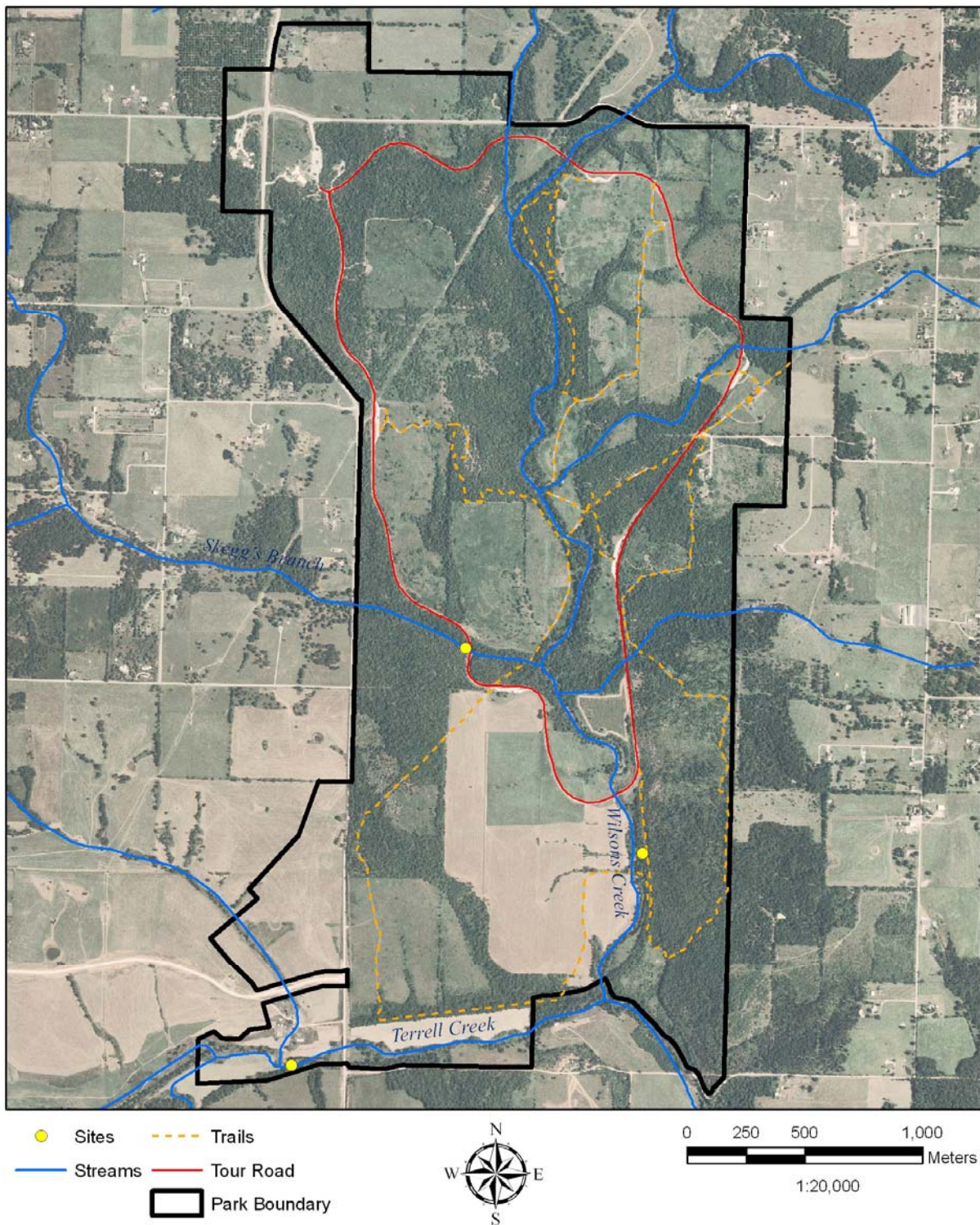


Figure 9. Map showing the approximate lower sampling reach boundaries for Wilson's Creek, Skegg's Branch, and Terrell Creek, WICR.

The NPS has organized its parks with significant natural resources into 32 networks linked by geography and shared natural resource characteristics. HTLN is composed of 15 National Park Service (NPS) units in eight Midwestern states. These parks contain a wide variety of natural and cultural resources including sites focused on commemorating civil war battlefields, Native American heritage, westward expansion, and our U.S. Presidents. The Network is charged with creating inventories of its species and natural features as well as monitoring trends and issues in order to make sound management decisions. Critical inventories help park managers understand the natural resources in their care while monitoring programs help them understand meaningful change in natural systems and to respond accordingly. The Heartland Network helps to link natural and cultural resources by protecting the habitat of our history.

The I&M program bridges the gap between science and management with a third of its efforts aimed at making information accessible. Each network of parks, such as Heartland, has its own multi-disciplinary team of scientists, support personnel, and seasonal field technicians whose system of online databases and reports make information and research results available to all. Greater efficiency is achieved through shared staff and funding as these core groups of professionals augment work done by individual park staff. Through this type of integration and partnership, network parks are able to accomplish more than a single park could on its own.

The mission of the Heartland Network is to collaboratively develop and conduct scientifically credible inventories and long-term monitoring of park “vital signs” and to distribute this information for use by park staff, partners, and the public, thus enhancing understanding that leads to sound decision making in the preservation of natural resources and cultural history held in trust by the National Park Service.

www.nature.nps.gov/im/units/htln/



The Department of the Interior protects and manages the nation’s natural resources and cultural heritage; provides scientific and other information about those resources; and honors its special responsibilities to American Indians, Alaska Natives, and affiliated Island Communities.

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