National Park Service
U.S. Department of the Interior

Natural Resource Program Center

# Shenandoah National Park Fish Monitoring Protocol

*Version 2.2 - DRAFT*

Natural Resource Report NPS/XXXX/NRR—20XX/XXX

**ON THE COVER**
National Park Service and Virginia Department of Game and Inland Fisheries personnel sampling the Rapidan River in Shenandoah National Park July, 2003
Photograph by National Park Service

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Natural Resource Report NPS/XXXX/NRR—20XX/XXX

Jeb Wofford and David Demarest

Shenandoah National Park
National Park Service
3655 US Hwy 211 East
Luray, VA 22835

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U.S. Department of the Interior
National Park Service
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## Protocol Revision History

All protocols, regardless of how sound they are originally, require modification as new and different information becomes available. Small changes or additions to existing methods will be reviewed in-house by Mid Atlantic Network staff while larger scale changes will require external review by appropriate experts. Document edits and updated protocol versions in the Revision History Log that accompanies the Protocol Narrative and each SOP. Log changes in the Protocol Narrative or SOP being edited only. 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 …). 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. 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. Details of protocol revision are discussed in SOP 20.

Revision History Log

| **Version #** | **Date** | **Revised By** | **Changes** | **Justification** |
|---|---|---|---|---|
| 1.0 | 1982 | Original protocol | NA | NA |
| 2.0 | 1995 | Atkinson | Sampling locations modified, more multiple pass electrofishing | See Protocol history |
| 2.1 | 2006 | Atkinson | Annual sampling changed to every other year, 5 yr. rotation changed to 8 yr., multiple passes to be used in all sampling | Required minimal increase in effort |
| | | | | |

# Contents

## Figures

Page

# Tables

# Appendices

# Executive Summary

Shenandoah National Park (SHEN), part of the Mid-Atlantic Inventory and Monitoring Network (MIDN), has identified fish populations as “high priority” for long-term monitoring under the vital signs program. Fisheries monitoring has been conducted at standardized sampling sites in Shenandoah since 1982, with an early emphasis on developing an understanding of eastern brook trout (*Salvelinus fontinalis*) population dynamics.

In 1991, fisheries monitoring was accepted into SHEN’s Inventory and Monitoring (I&M) Prototype Park sampling program. In 1995, a review by National Park Service and state fisheries managers resulted in modifications to the protocol. These modifications resulted in a reduced number of sampling sites that would be sampled annually, provided additional emphasis on collocating sampling sites with state fish monitoring locations, and resulted in changes in monitoring techniques with an emphasis on multiple pass electrofishing.

The 1995 protocol identified a suite of “primary” sites that would be monitored annually, and additional “secondary” sites that would be monitored on a five year rotation, or as financial resources allowed. Primary sites would be monitoring using multiple-pass electrofishing and secondary sites would be monitored via single pass electrofishing. Early monitoring and research results in the 1980’s noted the strong affect of underlying bedrock geology on water quality and fish diversity and, in the 1995 site selection process, sites were loosely stratified across the three major bedrock geologic types found in Shenandoah. Most streams that are sampled have one site near the park boundary, with larger watersheds having one to four additional sites upstream. Sites are generally 100m long and most are sampled without the use of blocknets. All captured fish are counted, and gamefish (namely salmonids and centrachids) are individually measured and weighed. A gross weight of non-game fish by species and a maximum and minimum length of non-game fish by species is also collected. A variety of physical habitat characteristics are collected at each site, including stream length and wetted width, substrate, gradient, etc. Water quality and quantity information is also collected during each sampling visit. As time has allowed, inventory of new streams at the park boundary has occurred using sampling methods similar to the monitoring protocol, and to date, virtually every perennial stream in Shenandoah has been sampled for fish. These sites have been added to the secondary site list.

From 1995 to 2004, protocols remained relatively unchanged. In 2004, budgetary constraints resulted in a new monitoring rotation where primary sites would now be sampled every two years, instead of the annual sampling that had occurred during the prior decade. Secondary sites are sampled every six years, instead of every five years as the 1995 protocol dictated.

# 1 Background and Objectives

## 1.1 Park Enabling Legislation

In 1926 the authorizing legislation was passed and provided for management and protection of the park. The park was subsequently established in December 1935 to protect the natural and cultural resources of the northern Blue Ridge and immediate area. On October 20, 1976, 79,000 acres in the park were designated as Wilderness and Skyline Drive was placed on the National Register of Historic Places in 1996. The park currently encompasses 199,016 total acres.

## 1.2 Park Purpose and Mission

Shenandoah National Park was established for the following purposes: 1) to protect the natural and cultural resources of the northern Blue Ridge and immediate area, 2) to have a "National Park" here, at this location, providing scenery, serving as a refuge and pleasuring ground, and including the developed visitor amenities traditionally found in other "National Parks", and 3) to construct and maintain a "sky-line drive" to provide outstanding views of the scenic and historic Shenandoah Valley and Piedmont of Virginia.

The mission of Shenandoah, is to bring about the desired future conditions of the park via the following guidelines: 1)The ecological integrity of this portion of the Blue Ridge/Central Appalachian biome is protected, maintained, and restored as appropriate, 2) Cultural landscapes, other significant cultural resources, and associated values are protected, restored as appropriate and maintained in good condition and managed within their cultural context, 3) The views of the Shenandoah Valley and Piedmont Plain, as seen from the park, are scenic and rural in character, maintained in partnership with and integrating the needs of the surrounding communities, 4) Visitors safely enjoy and are satisfied with the availability, accessibility, diversity, and quality of park facilities, services, and appropriate recreational and "re-creational" opportunities, and 5) The stories of the area and the development of the park are available; visitors and the general public learn the purpose and significance of the park.

## 1.3 Protocol History

The fisheries monitoring program at Shenandoah began in 1982 with a major emphasis on evaluating brook trout population dynamics in order to better manage the brook trout fishery. In the developing years of the program, single-pass electrofishing techniques were used to obtain relative trout numbers and size classes of brook trout and a list of other species present at one hundred and thirty-two 100 meter monitoring sites located along 46 Park streams. The monitoring effort was structured such that the west side of the park was sampled one year and the east slope was sampled the following year. There were approximately an equal number of sites on each slope of the mountain. This monitoring approach was adopted to collect relative trout population data and distribution data for other fish species in all reasonably accessible park streams known to contain brook trout. The principal assumption was that a broadly applied qualitative approach would adequately address park management needs. The principal pitfalls of the program included poor coordination with Virginia Department of Game and Inland Fisheries (VDGIF) staff on streams of mutual monitoring interest and the use of dissimilar electrofishing techniques from those used by VDGIF and other agencies. The potential of a widespread multiple-pass monitoring component was not realized during the development of the park's fisheries program.

In August, 1995, National Park Service biologists and natural resource managers from the Washington Office, Shenandoah National Park,Great Smoky Mountains National Park, and biologists from the Virginia Department of Game and Inland Fisheries (VDGIF) met to review and revise the park's fisheries monitoring program. The primary objectives of this meeting were to establish the future goals and objectives for monitoring and managing fish populations in the park and to design a monitoring program that utilized standardized techniques to sample fish populations in a suite of large, medium, and small streams, which were stratified according to the park's three predominate acid neutralizing capacity (ANC) or alkalinity levels. Secondary objectives included selecting a suite of joint sites with the VDGIF on streams of mutual interest and the use of combined crews and equipment to produce data the met the specifications of both agencies.

The revised protocol represented a sampling reduction of 58 sites and three streams from the original program. The 1995 program included 43 streams and 74 sites split between "quantitative" or multiple-pass electrofishing and "qualitative" or single-pass electrofishing components. The quantitative component included 36 sites primary sites sampled annually along 15 streams (Appendix D) loosely stratified across the Park's three dominant bedrock geologic formations and associated water chemistry ranges. Lower Lewis Run was added to the annual sites list in 2006 and is included in Appendix D.

During that 1995 review of earlier sampling protocols, stream and transect selection for inclusion in the 1995 primary component were based on the three principal park Shenandoah Watershed Assessment Study (SWAS) "research" streams, which span the three dominant geologic types including Paine Run (siliciclastic bedrock), Piney (basaltic bedrock) and Staunton (granitic bedrock) Rivers. These streams had a legacy of research and monitoring on water quality and it was assumed that these streams would continue to provide additional water quality information that would support future assessments of fish populations. Additional streams having the same bedrock and water chemistry characteristics as the three primary streams were selected in an attempt to identify gross fish population trends across the three principal bedrock substrata. Other stream and site selection considerations included linkages with and locations of other monitoring efforts including VDGIF fisheries monitoring sites, SWAS/University of Virginia (UVA) water chemistry monitoring sites, Park Research Natural Areas (North Fork Dry Run and Onemile Run), and the park's aquatic macroinvertebrate vital signs monitoring sites. Where possible, sites were collocated in an attempt to provide linkages and/or partnerships with other monitoring efforts and results. In large watersheds where annual quantitative sampling was occurring; an additional five qualitative sites (Appendix D) were added to the annual sampling schedule with a focus on large tributaries or the headwaters of the mainstem. The resulting range of streams selected included a mix of small and large streams across a variety of geologic types and spanning all of the three park administrative districts. As such, these sampling sites were deemed representative of wide ranging lotic habitats present in Shenandoah National Park.

The 1995 protocol also identified 25 streams and 34 transects that would represent the qualitative secondary component which would be sampled via single-pass electrofishing on a five year rotation. Selection criteria for qualitative sites were based on monitoring data from the pilot years of the program.

The current fish monitoring protocol reflects the standards outlined in 1995 with five exceptions. Currently, multiple pass electrofishing is completed at all sites and fish population monitoring at primary sites occurs every other year, not annually, as was originally intended. Secondary sites that were on a five year rotation are now sampled at a minimum of once at least every eight years. In addition, Lower Lewis Run was added to the list of primary sites in 2006 after serious trout population decline and ultimately, an extirpation. Lastly, 51 additional streams have been added to the secondary sampling list, each with a minimum of one transect. All sites are mapped with geology in Figures 1, 2, and 3.

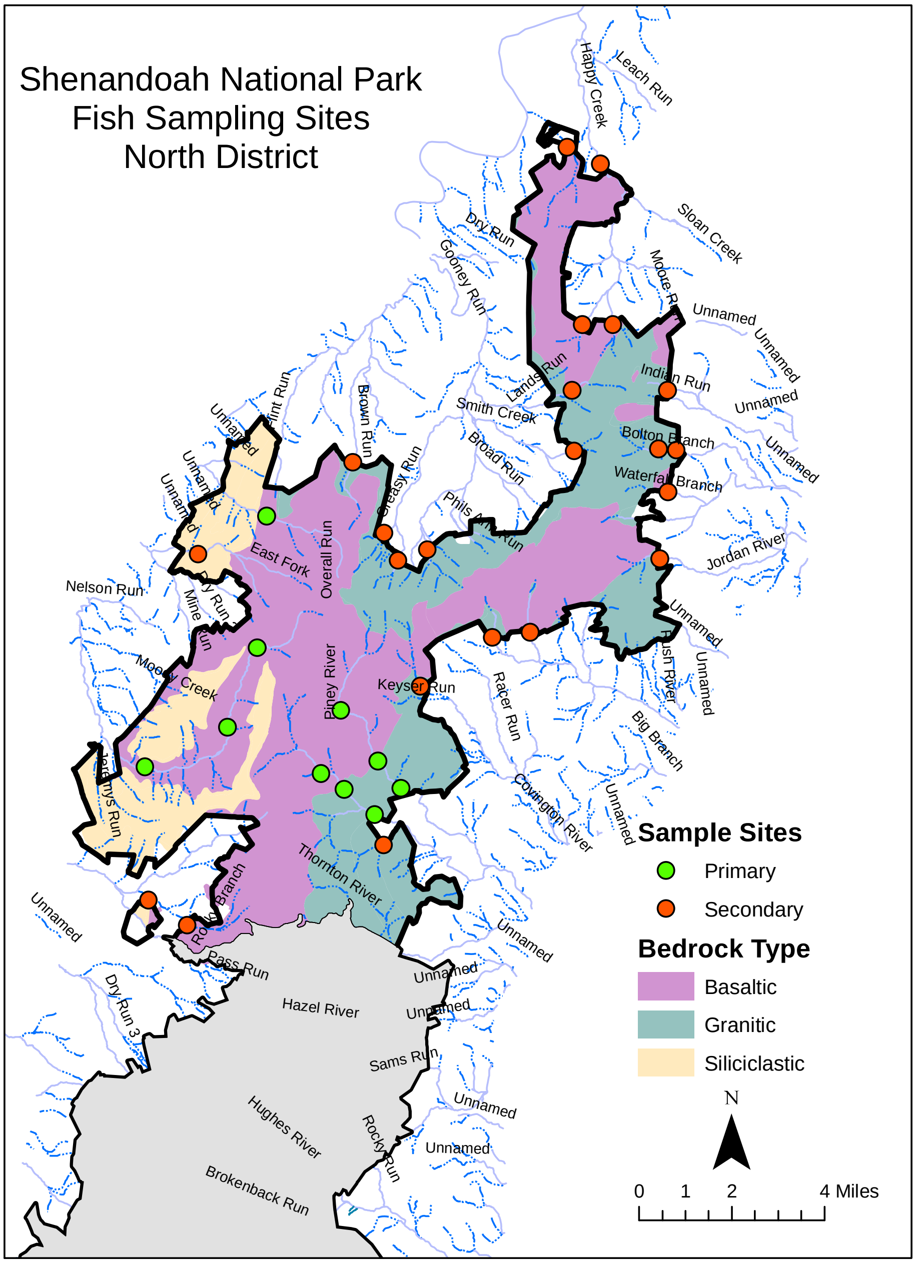


**Figure 1**. North District of Shenandoah National Park with primary and secondary fisheries monitoring sites overlaid on geologic types.

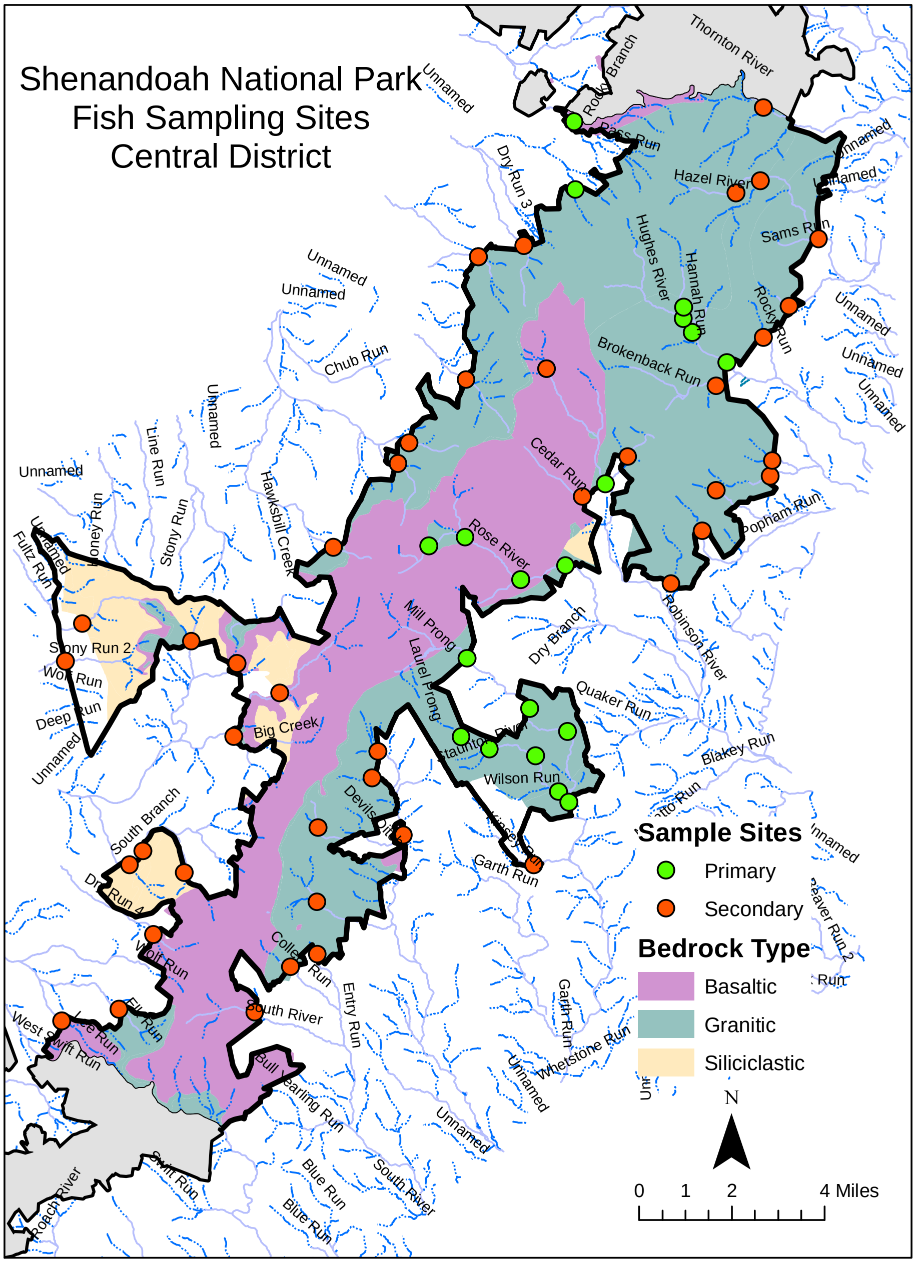


**Figure 2**. Central District of Shenandoah National Park with primary and secondary fisheries monitoring sites overlaid on geologic types.

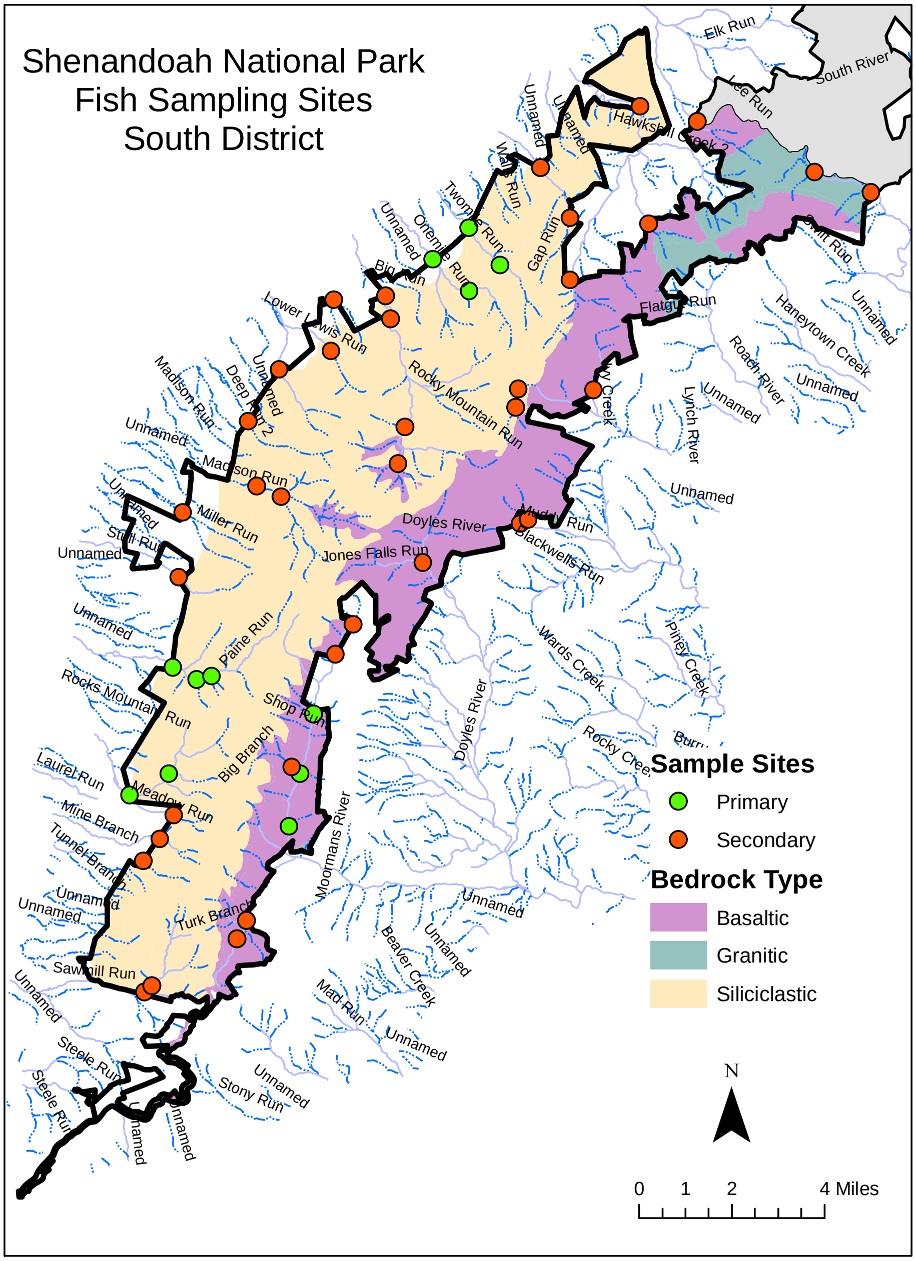


**Figure 3.** South District of Shenandoah National Park with primary and secondary fisheries monitoring sites overlaid on geologic types.

## 1.4 Environmental Setting

Shenandoah National Park was established in 1935 and covers 199,016 acres (805 square kilometers) of forested terrain on the crest of the Blue Ridge Mountains in northern Virginia. The Park has a long linear shape with a highly uneven boundary and variable width. The Park is divided east to west by the ridgetop which Skyline Drive and the Appalachian Trail follow, and into three districts north to south by U.S. Highways 33 and 211. Shenandoah is entirely contained within the Blue Ridge physiographic region and can be characterized by steep slopes, narrow ridges, and high relief (Bailey 1999). Elevations within the park range from 192 to 1231 meters (530-4050 feet) above sea level, with about 60 peaks above 914 meters (3000 feet) (Conners 1988). Low elevations in the park generally experience mild winters and hot, humid summers - average monthly temperatures range from 41.9 degrees Fahrenheit in January to 85.5 degrees Fahrenheit in July at low elevations (Vana Miller and Weeks, 2004). At higher elevations, winters are moderate and summers are relatively cool – average monthly temperatures range from 24.8 Fahrenheit in January to 65.5 Fahrenheit in July on higher ridgetops in Shenandoah (Vana Miller and Weeks, 2004). Hydrographs are generally characterized with peak stream flows in the spring and low flows occurring in late summer-fall (Lynch 1987). Shenandoah National Park contains three major bedrock units of different age and composition which can be loosely characterized as granitic, basaltic, and siliciclastic (Gathright, 1976). Each of the three main bedrock formations associated with these rock types have distinct chemical and physical properties that directly influence the surrounding landscape, including terrestrial and aquatic communities (Bulger et al 1995, Butler 2006, Southworth et al 2009, Young et al 2009).

The park's Mid-Atlantic location straddles conditions of both the Northern and Southern Appalachian Mountains allowing over 1,300 species of vascular plants to occur in the park. Ninety-five percent of Shenandoah National Park is forested with Eastern deciduous woodland. In 2005, a vegetation map was completed that divides the park into 34 vegetation communities. These include 22 upland forests and woodlands, one alluvial/riparian forest, six non-alluvial wetlands, and five outcrop communities.

Shenandoah National Park contains portions of the headwaters of three large river basins within Virginia; the James, the Shenandoah, and the Rappahannock watersheds. Streams within Shenandoah are generally 1st to 3rd order, with about 90 watersheds in the park that contain perennial stream flow. Though inherently variable, streams tend to be of relatively high gradient, with step-pool or cascade morphology (Montgomery and Buffington, 1997). Most streams are well shaded by vegetation and maintain cold or cool water temperatures during summer months. Springs and seeps provide the primary source of water for the streams that are further augmented by rain. Water quality within Shenandoah's streams is relatively good, though acid deposition and associated stream acidification are well documented (see Vana-Miller and Weeks, 2004 for review).

The relationship between bedrock lithology and stream water quality, particularly as it pertains to acid-base status and associated acid-neutralizing capacity (ANC), is well documented in the park (Lynch and Dise, 1985; Webb, 1988; Cosby et al., 1991 ). In addition, numerous studies have noted the correlation between ANC and biological parameters (Smith and Voshell, 1994; Moeykins and Voshell, 2002; Bulger et al 1999, Cosby et al 2006). These studies documented general declines in measures of ecosystem health (macroinvertebrate taxa diversity, brook trout condition factors, etc.) as ANC declined. Streams with the lowest ANC values have alkalinity ranges from 0 - 25 ueq/L (milliequivilents per liter), are associated with siliciclastic formations in the watershed, are all located along the southern half of the western slope of the park, and have the fewest number of fish species. Moderate ANC streams have alkalinity ranges from 50 - 100 ueq/L, are influenced by granitic formations, are more evenly distributed park wide, and tend to have intermediate numbers of fish species present. Streams with higher ANC have alkalinity ranges from 150 - 200 ueq/L, are influenced by basaltic bedrock, are also fairly well distributed throughout the park, and tend to have the highest number of fish species.

Forty species of fish have been documented in the park (Appendix E). Ten of these species are exceedingly rare with ephemeral populations (or individuals) that sporadically invade the downstream sections of some park waters. Common, abundant, and well distributed species include black nosed dace (Rhinichthys atratulus), brook trout, mottled sculpin (Cottus bairdi), and fantail darters (Etheostoma flabellare).

## 1.5 Rationale for Monitoring Fish Populations

Aquatic biota have long been recognized as an important resource in Shenandoah National Park, with brook trout playing a major role in the initial establishment of the park in 1920's. The relatively pristine and high elevation headwater streams in Shenandoah currently support coldwater aquatic communities that are increasingly rare in Virginia and the Southeast (EBTJV 2006). Within coldwater ecosystems, fish populations can serve as measures of aquatic ecosystem health and are frequently used to assess environmental disturbance (Simon 1999). Because salmonids are sensitive to environmental change, brook trout populations are often used as a measure of cold-water ecosystem function.

Some threats to aquatic resources in the Park are well documented -- acid deposition has long been recognized a threat to the ecological function of lotic habitats in Shenandoah (Vana-Miller and Weeks, 2004). In addition, recreational activities in the Park frequently target angling for brook trout, and public interest in fish population health is high. The relative rarity of coldwater fish resources, along with the identification of known and persistent threats, and a high level of public interest suggests that monitoring of fish populations should be important for tracking vital signs in Shenandoah National Park. An independent review by scientists as well as a review by park managers indicated that, out of 43 potential vital signs that were assessed during vital sign development for Shenandoah, brook trout and fish communities ranked as the 3rd and 5th most significant based on combined ecological, management, and policy interests (Comiskey and Callahan, 2008).

## 1.6 Monitoring Goals and Objectives

Fish population monitoring in Shenandoah National Park is nested within the larger context of the National Park Service vital signs monitoring program. Vital signs are "a subset of physical, chemical, and biological elements and processes of park ecosystems that are selected to represent the overall health or condition of park resources" (Comiskey and Callahan, 2008). Goals associated with the NPS vital signs program are:

Determine the status and trends in selected indicators of the condition of park ecosystems to allow managers to make better-informed decisions and to work more effectively with other agencies and individuals for the benefit of park resources.

1. Provide early warning of abnormal conditions of selected resources to help develop effective mitigation measures and reduce costs of management.

2. Provide data to better understand the dynamic nature and condition of park ecosystems and to provide reference points for comparisons with other, altered environments.

3. Provide data to meet certain legal and Congressional mandates related to natural resource protection and visitor enjoyment.

4. Provide a means of measuring progress towards performance goals.

Protocol objectives should provide specific measurable criteria and should articulate the focus of a monitoring plan.  Three specific monitoring objectives are identified for fish populations in Shenandoah National Park:

1. Determine long term trends in fish species abundance and biomass and community composition within selected stream sites over time.

2. Determine long term trends in condition factors and reproduction of game fish within selected stream sites over time.

3. Determine relationships between physical and biological parameters and fish community and population dynamics (species abundance, community composition, biomass, condition, reproduction, etc.) within selected stream sites over time.

4. Establish baselines and thresholds of fish community or population metrics that trigger additional research or management actions.

# 2 Sampling Design

The initial sample design was established in 1982 and it focused on collecting information on larger trout populations within Shenandoah National Park. Sampling locations were generally established at the park boundary, and site locations relied heavily upon professional opinion and the "representativeness" of the sample reach. In general, smaller streams contained one sample site and larger streams contained multiple sampling sites. Specific objectives were not defined in the initial program. The current protocol inherited much from that initial sampling design. Even though the 1995 review resulted in a program review and adjustment based largely on bedrock and associated water chemistry parameters (see below), as with the initial design, no randomization was used in determining appropriate reaches to sample, and some sample sites have been changed to be "more representative" of surrounding habitats. Due to the large number of sites involved in the current design, nearly every fish bearing watershed in the park is sampled at least once every eight years.

## 2.1 Spatial Design

The initial fish population monitoring effort began in 1982 with a design based on a full census of streams that were known to contain relatively large trout populations. Forty-six populations (i.e. streams containing brook trout) were identified, and a least one sample site per stream was selected. Though not clearly documented, it is assumed that these sites were selected because of accessibility, "representativeness", and suitability for fish sampling (e.g. stream reaches that are consistently dry in summer were not selected). Within a particular watershed, at least one site would occur at the park boundary, with additional sites occurring upstream in larger watersheds. Sites are generally approximately100m long, with actual site lengths established to capture multiple fish habitat types, and to allow for the start and end point of the reach to occur at geomorphic breaks that limit fish movement during sampling. These methods resulted in 132 sites being sampled on 46 streams. In the 1995 review, the original list of streams and sites were reviewed and adjusted as follows. Streams that span a range of ANC types (i.e. contain >50% of their watershed in one of the three principle bedrock categories), that have collocated monitoring sites for other programs, that contain exotic trout, and that are exposed to a variety of different fishing pressures were selected for inclusion as primary streams in current protocols. The sum of the selection process resulted in a suite of 18 primary streams.

Of the remaining 28 streams from the 1982 protocol, 25 were selected for the secondary component as three streams were dropped due to nearby established monitoring sites. Lastly, as time has allowed, additional boundary locations on previously unsampled streams have been inventoried for fish using methodologies outlined in this protocol. Following inventory, these sites were added to the secondary list of monitoring sites.

Once streams were selected, monitoring sites located on these streams were selected from the 1982 program of 132 sites. Site selection or retention for the revised monitoring program followed several criteria and did include the results of monitoring data from the pilot years of the program. Retained sites had single channels, were often located at the boundary, were collocated with other monitoring sites, and had adequate year around water flows. Sites that were readily eliminated included those that were closely spaced along a particular section of stream, sites with badly braided or split channels and in a few cases, sites that were devoid of fish. The revised monitoring program includes 41 primary sites along the 18 streams and 87 secondary sites along 71 streams.

The process above resulted in a suite of streams and sample sites which are loosely stratified by bedrock geology, represent a range of stream sizes, are under a variety of fishing pressures, and are widely distributed across the park.

### 2.2 Temporal Design

Currently, primary sites will be sampled once every two years while secondary sites will be sampled once every eight years. To reduce temporal variability, individual streams and sites will be sampled during a reasonably similar calendar period between May and August. A sampling differential of two to three weeks is acceptable across years but, to limit the influence of seasonal effects, streams that are typically sampled in June are not be sampled in August and vice versa.

### 2.3 Inference

Ultimately, streams and sites in Shenandoah were selected based on their “representative” status and were deemed to capture the variability of many park resource and environmental conditions. No randomization or probability sampling was involved in stream or site selection. As such, inference from this monitoring program is limited to the set of sampled sites and cannot be extrapolated to other spatial scales. That being said, because of the large number of sample sites (particularly in secondary streams), the program represents a near census of fish bearing streams, including locations where fish diversity is generally maximized (i.e. where those streams intersect the park boundary).

# 3 Field Methods and Descriptions

## 3.1 Locating Sites in the Field (SOP 4)

Specific site directions are maintained for each stream and monitoring site in the park and are organized with respect to east versus west slope locations. Site directions include written descriptions of specified road and trail routes and a U.S. Geological Survey (USGS) topographic map of the stream with clearly marked site locations. The start and endpoints of all monitoring sites are referenced by tagged live trees located on the adjacent stream bank. UTMs for each site are also available to aid in navigation if needed.

## 3.2 Water Chemistry Sampling (SOP 6)

Water chemistry sampling occurs prior to any in stream disturbance associated with fisheries monitoring and is thus the first data collected during a site visit. All water chemistry sampling associated with the fisheries monitoring program is conducted with Hydrolab® multiparameter water quality monitoring units. The “Core 4” water quality parameters are collected at each site (NPS, 2002) and spot values of water temperature, pH, conductivity, dissolved oxygen, and total dissolved solids are collected during each sampling event. To ensure data quality, the units require calibration at the beginning of each week during the field season. Procedures for calibrating the units are listed in SOP 7.

## 3.3 Discharge Sampling (SOP 11)

A Marsh-McBirney (MMB) model 2000 “Flo-Mate™” serves as the primary unit for collecting discharge data when in the field. Discharge is measured over cobble/gravel substrates with a uniform channel bed. Park staff utilize the 6/10ths flow measuring technique recommended by the USGS (Buchanan and Somers 1969) and the U.S. Forest Service (Platts et al. 1983). When feasible, measurements are taken from at least ten evenly spaced intervals at the selected cross section, perpendicular to stream flow. Discharge is collected during each site visit.

## 3.4 Habitat Sampling (SOP 9)

Habitat sampling includes a combination of actual measurements (stream width, site length, depth, and gradient) and visual estimates (stream substrates, pool/riffle ratios, riparian cover, and riparian type.). These variables are used to assist with calculations of fish density and biomass, and to provide a physical assessment of environmental attributes that may influence fish populations. Units for are in meters, percent slope, or are unitless variables (e.g. pool riffle ratios).

Habitat measurements are generally collected at 10 meter transects through the sample reach. Transects include only wetted portions of the stream channel. Wetted stream widths are recorded at each transect up to and including the end point at the upstream end of the monitoring site. Water depths are measured and stream substrates are estimated at the ¼, ½, and ¾ points across a transect. At each point (¼, ½ and ¾), the water depth is recorded to the nearest 0.01m and the dominant substrate is recorded as organics, silt (<0.06 mm), sand (0.07-2.0 mm), pebble/gravel (2.1 mm-10.0 cm), cobble (10.1-30.0 cm), boulder (>30.1 cm), or bedrock.

From the center of each transect, an observer also records the riparian cover category that exists directly above the transect. This is a visual estimate of the degree of riparian canopy cover over the wetted channel. Riparian cover is classified as 0-33%, 34-66%, or 67-100%. In addition, observers note whether the canopy consists of trees, shrubs, forbs or grasses, or that no canopy is present. Using a clinometer, observers record gradient between transects (SOP10). Between each transect an observer will also conduct a visual estimate of the relative amount of pool habitat (water with near zero velocity) and riffle habitat (water with > zero velocity) present.

### 3.5 Fish Sampling( SOP 8)

Three pass backpack electrofishing is used for all surveys. Naturally occurring geomorphic habitat breaks that limit fish movement occur at the lower and upper ends of most sample reaches and these breaks may be modified or supplemented via cobble dam construction prior to sampling. Therefore, during most electrofishing surveys (excluding select Rapidan and N.F. Moormans sites), block nets are not used. Modified habitat breaks are restored to their prior condition following sampling.

Initial settings used on electrofishers are determined by conductivity information collected during water quality sampling. The number of electrofishers required to adequately sample each reach (primarily based on stream width) has been established during prior sampling events (Appendix D) and follows recommendations set forth by the SDAFS Trout Committee (SDAFS, 1992). In general, two netters accompany each electrofisher during sampling. During an electrofishing pass, operators proceed upstream sampling all wetting habitats thoroughly and efficiently. Electrofishing procedures largely follow those outlined in NWQAP (Moulton et al 2002).

Following each electrofishing pass, captured fish are sorted to species and counted. Fish will be identified using taxonomic keys or other appropriate information. Gamefish (namely salmonids and centrachids) are each measured to the nearest mm and weighed to the nearest 0.1g. Gamefish may be anesthetized with clove oil prior to processing. For nongame fish, the smallest and largest individual per species is measured to the nearest mm. A total weight of all non-game fish by species is collected. Mortalities are also counted for each species. All captured fish are placed in holding cages located outside of the sampling area until all electrofishing is completed. At the conclusion of all the sampling, all live fish are released back into the stream in the approximate proportions and habitats from which they were captured. Any dead fish are separated out during processing and disposed of well away from the stream.

# 4 Data Management, Analysis, and Reporting

When in the field, it is critically important that the data recorder have good penmanship as the written records need to be clearly legible. Prior to leaving the site, all completed forms are reviewed to ensure there are no missing data. Upon arrival back at the office at the end of the day, completed are filed appropriately.

Specific instructions for using the park's Aquatic Database System to perform data entry, edits, and validations can be found in SOP15. Briefly, The Shenandoah National Park fish data entry program consists of three main components, the data entry application, the temporary storage databases, and the master database. Data is entered into the first temporary database using the data entry application. As a means of error checking, the data is entered a second time into a second temporary database. The two temporary databases are compared and any discrepancies can be reviewed and corrections made as needed. At the end of the field season the data in the temporary databases are reviewed by the program staff and then added to the master database (SOP16).

## 4.1 Data Analysis and Reporting (SOP17, 18)

(Adapted from Peterson et al (2008) and Dodd et al (2008)). Conclusions of ecological studies based on fisheries data and other biological, chemical, and physical data are used by resource managers to better comprehend underlying system processes and develop environmental and management policies that best serve the resource. Most analyses will be structured to address fish populations at multiple spatial scales including the sample site, stream (when more than one sample site occurs in a particular watershed), and geologic type or corresponding ANC group (<20ueq/L, 20-100ueq/L, >100ueq/L)). Primary approaches to analyzing fisheries data will rely on metric estimation (see SOP 17 for specific metrics). Metrics for analysis of fish populations will include those that assess community richness, balance, and production.

### *4.1.1 Annual Data Summaries – Control Charts (SOP 17,18)*

Data summaries have a primary audience of park program managers, superintendents, and other park staff. As such, summaries should target metrics and appropriate data displays that are easily understood and communicated. Using a subset of metrics, control charts will be employed to visualize trends and changes in fish communities. Control charts allow for the visual display of trends and thresholds for management action or further data analyses. Initially, the first ten years of data will be serve as a baseline (see Morrison, 2008) and thresholds will be defined as plus or minus two standard deviations around the ten year mean (i.e. baseline) of the statistic (see Figure 1, SOP 17 for example). During any year where thresholds are crossed, more targeted data analyses will be conducted to assess the driver behind the threshold transgression and to help determine the potential need for management action. Baselines and thresholds may be altered in the future to potentially better reflect meaningful management or biological thresholds that might not be represented in the initial 10 year mean ± 2 standard deviations of the mean.

### *4.1.2 Habitat and Water Quality Parameters*

Physical and chemical habitat measurements will be estimated using summary statistics such as means, medians, standard errors and/or confidence intervals. In the simplest presentation of data, each parameter should be estimated in each year that data are available, and confidence intervals or standard errors calculated, where appropriate. Generally, habitat and water quality parameters will be used in comprehensive analyses to assess relationships between community data and environmental characteristics.

#### *4.1.3 Comprehensive analyses and Reports*

Comprehensive trend reports and associated analyses will occur on an approximate ten year timeframe. These analyses will focus on trend detection and characterizing associations between habitat variables and fish populations characteristics. Rather than requiring a detailed list of specific data analyses, the data analysis process should be flexible enough to allow the use of newly developed statistical and analytical techniques and tailoring of analyses for a variety of audiences with a variety of questions about the aquatic resources at SHEN. Analyses may occur at a variety of temporal and spatial scales.

# 5 Personnel Requirements and Training

## 5.1 Number of Personnel Required

Most of the Shenandoah streams and sites can be optimally sampled with an electrofishing crew of nine technicians and volunteers. Since most Shenandoah streams are most efficiently sampled with two electrofishing units, this total crew size allows for a stream crew of six to eight and a fish processing crew of one to three. The stream crew consists of two shocker operators, three or four netters and one or two bucket carriers. The shore crew consists of one to three individuals responsible for sorting, measuring and weighing fish and for maintaining fresh water in the buckets. When sampling larger streams and sites, most of which are joint sites with the VDGIF, crew sizes may approach 20 or more.

## 5.2 Personnel Training in Field Procedures (SOP 2)

By late May, the full complement of biological technicians and volunteers will have arrived in the park for the summer field season. The full crew will undergo two to three days of training in the principles, use and safety of electrofishing gear, fish identification, taking fish measurements and recording data, the collection of habitat data from fish monitoring sites, CPR and first aid, and job hazard analysis discussions prior to the start of the annual monitoring schedule. All individuals are further instructed in the use of the Hydrolab to measure water chemistry parameters and flow meter equipment to measure stream discharge.

## 5.3 Personnel Safety (SOP 2)

Electrofishing in mountain stream habitats ranks among the most hazardous work activities performed by biological technicians and volunteers at Shenandoah National Park. That being said, with adequate preparation, training, the use of personal protective gear, and by being ever mindful of potential hazards during the course of the workday, all of the tasks associated with electrofishing can be accomplished in a manner that is safe to all individuals involved.

Prior to the beginning of the fisheries monitoring field schedule, most if not all of the crew will attend a one day first aid/CPR training session. Numerous job hazard analyses (JHA) specific to Shenandoah National Park will be reviewed by the crew prior to the start of the field season. These JHAs include, but are not limited to, *working in tick infested areas*, *light vehicle use*, *backcountry travel*, *electrofishing operations*, *aquatic macroinvertebrate collection, lifting and carrying, and hantavirus.* Certain JHAs may require additional safety training.

## 5.4 Sampling Conditions and Times (SOP 3)

The field component of the fisheries monitoring program typically begins in early to mid June and ends by late July to mid August. Starting and ending dates are largely determined by the frequency of rain events, the amount of total rainfall and associated water levels in the streams. Optimal conditions include low to average water flows of clear condition.

Individual streams should be sampled within the same general period of time each year to facilitate comparisons of density and biomass between years. A sampling differential of two to three weeks is acceptable from one year to the next but streams that are typically sampled in June should not be sampled in August and vice versa.

### 5.5 Required Equipment (Appendix A and B)

All of the equipment associated with fisheries monitoring is supplied and maintained by the National Park Service. A full equipment list is provided in Appendix A. Equipment is stored in three general locations (Appendix B). Electrofishing units, probes, gas/oil mix, nets, buckets and tubs are stored in the small shed at the far end of the Lower NCR building parking lot. Wading gear, packs, flow meters, measuring tapes and boards, electronic balances, gloves and clove oil are stored in the block building adjacent to the fuel pumps. GPS units, Hydrolabs® and accessories, radios, batteries, clipboards, datasheets, site directions are stored within the Lower NCR building. In addition to work equipment noted above, each individual on the crew is responsible for ensuring that their own hip boots and personal gear are packed and loaded each day.

# 6 Operational requirements

## 6.1 Roles and responsibilities

The Park Fish and Wildlife Biologist (GS-11) coordinates all aspects of the program. Implementation of the protocol is lead by a GS-07 crew leader and eight technicians or interns. The Park Data Manager (GS-11) provides leadership support and guidance on data entry, analysis, management, and archiving.

### *6.1.1 Fish and Wildlife Biologist (Program Coordinator)*

The Park Fish and Wildlife Biologist operates as the project manager to oversee all aspects of the monitoring program, including:

- Permits, scheduling, hiring, purchasing, contracting, and budget
- Maintaining and updating monitoring protocol
- Conducting or assisting in training and participating in field work as needed
- Overseeing field crew, providing logistical, administrative, and technical support
- Analyzing data and preparing reports
- Ensuring QA/QC standards are met
- Coordinating with the Park Data Manager
- Communicating results to the Park I&M Coordinator and other interested staff

### *6.1.2 Lead Biological Technician*

The lead biological technician is responsible for assisting the Fish and Wildlife Biologist with program management obligations as well as taking the lead in the day-to-day operations of the field crew. Tasks include:

- Coordinating daily field activities and ensuring equipment is in working order
- Coordinating directly with Biologist and Communication Center when conducting field work
- Conducting weekly safety briefings and offering safety reminders appropriate to current activities
- Ensuring that all field crew members are following proper data collection procedures
- Coordinating data entry into database, verification, corrections, and archiving of data from paper field sheets
- Assisting Biologist with program obligations such as hiring, , purchasing, contracting, budget, updating the protocol, analyzing data, and preparing reports

- Ensuring vehicles are properly maintained and cleaned
- Ensuring QA/QC standards are met

#### *6.1.3 Monitoring Crew Technicians and Interns*
The Crew Technicians are responsible for data collection. Tasks include:

- Collecting field data accurately as described in the SOPs
- Entering data accurately into the project database either in the field or from paper data sheets
- Ensuring all necessary equipment is available, well cared for, clean, and functional prior to each trip
- Performing work safely and efficiently

#### *6.1.4 Data Manager*
The Data Manager in the Natural and Cultural Resources Division provides oversight for all data related aspects of the project including:

- Providing database training to the crew
- Maintaining the fish population monitoring database
- Preparing maps of sample point locations
- Ensuring data management practices are met, including data backup, transfer, and archiving
- Ensuring QA/QC standards are met

### 6.2 Annual workload and field schedule
Field work and data entry, verification, and archiving is conducted primarily between mid-May and the end of September. Field planning, data analysis and report writing largely occur between October and April. Hiring of temporary employees commences in the fall and continues throughout the spring. Budget and other administrative support activities occur year round.

### 6.3 Facility and equipment needs
The field crew is based at Shenandoah National Park Headquarters and operates out of the Natural and Cultural Resource Division's Lower office. All equipment needed to conduct the field work is listed in Appendix A, and is stored in the office or nearby storage buildings. The lead technician is responsible for ensuring that all equipment is in working order and coordinating with the Biologist to replenish supplies as needed.

## 6.4 Operating costs and budget considerations

Annual operating costs are based on the costs incurred in FY2010, including personnel and supplies (Table 4). Additional costs are incurred to replace or repair equipment including electrofishers, water quality multimeters, GPS units, cameras, and vehicles.

**Table 4**. Estimated annual cost to implement Shenandoah National Park Fish Population Monitoring Protocol. The permanent staff cost estimate includes 80 hours of a GS-11 Data Manager's time, 720 hours of a GS-11 Biologist's time, and 840 hours of a GS-07 Crew Leader's time. Temporary staff costs include 960 hours of GS-05 technician time, 960 hours of GS-04 technician time, and 1920 hours SCA intern's time.

| Category | Expense (2011 dollars) |
| --- | --- |
| Permanent staff | $ 53,000.00 |
| Seasonal Staff | $54,000.00 |
| Equipment | $ 4,500.00.00 |
| Supplies | $ 3,000.00.00 |
| | |
| **Total** | **$ 114,500.00** |

# 7 Literature Cited

Example 1 2011 XXXXXXXXXXXXXX

# Appendix A: Shenandoah National Park Field Equipment List

## *FISHERIES MONITORING*

## *FIELD EQUIPMENT CHECKLIST*

**Fish Sampling**

____ Backpack Electrofishing Units
____ Anode/Cathode Probes
____ Cooler with Gas Mix
____ Tool Kit with Spark Plugs
____ Long Handled Dip Nets
____ Short Handled Dip Nets
____ Holding Cage(s)
____ Block Nets (large sites)
____ Plastic Buckets
____ Blue Plastic Tubs
____ Bucket Pack(s)
____ Back Packs
____ Clove Oil (brown Nalgene bottle)
____ Measuring Boards
____ Electronic Balances
____ 12 Extra AA and Two extra 9v batteries for balances
____ Rubber Gloves
____ Polarized Glasses
____ Fish Key (Jenkins and Burkhead)

**Flow & Physical Site Measurements**

____ 100 m tape
____ 30 m tape
____ Marsh McBirney flow meter
____ Extra batteries for MMB (2 size D)
____ Top setting metric wading rod
____ Depth stick (graduated in .01m)
____ Compass
____ Clinometer

**General Equipment**

____ GPS
____ GPS Batteries (2 “AA”)
____ 2 Hand Held Radios w/Batteries
____ First Aid Kit
____ Site Directions
____ Field Data Forms (1 set per site)
____ Site Form/H2O/Discharge
____ Gamefish Data Form
____ Nongame Fish Data Form
____ Habitat Data Form
____ Metal Clipboards
____ Pencils
____ Hip Boots
____ Drinking Water
____ Lunches

**Stream Chemical Measurements**

____ Hydrolab Multi-Sensor Unit
____ Hydrolab data Display System
____ Hydrolab data cable
____ Hydrolab weighted sensor guard
____ Hydrolab 3 extra “C” batteries

**Gear Cleaning**

____ 2 Large tubs
____ Dish detergent
____ Salt
____ Bucket with lid

## Appendix B: Shenandoah National Park Field Equipment Lists

(The list from Appendix A. is further broken out by task and location on the next three pages.)

# Clipboards/Radio/GPS

____ Hand Held Radio(s) w/Batteries
____ GPS w/Batteries
____ Metal Clipboards (2 to 3)
____ Site Directions
____ Site Form/H2O/Discharge (5 or more)
____ Nongame Fish Data Form (5 or more)
____ Habitat Data Form (5 or more)
____ Gamefish Data Form (20 per clipboard)
____ Pencils (1 per person)
____ Fish Key (Jenkins and Burkhead)
____ Fish Species Abbreviation List

Appendix B. Shenandoah National Park Field Equipment Lists (continued).

# Gas House Equipment (suburban)

_____ Packs (subtract 3 from the crew size to get the total number of packs you should have.)
_____ First Aid Kit
_____ Tool Kit (yellow pouch)
_____ Hip Boots
_____ Wading boots and stocking foots
_____ Neoprene Waders/Boots (only deep streams)
_____ Clove Oil (in **brown** Nalgene bottles)
_____ Measuring Boards (3) Eels require the big white board
_____ Electronic Balances (Check with Sup. for size needs)
_____ Rubber Gloves (bring extra)
_____ 100 m tape
_____ 30 m tape
_____ Marsh McBirney flow meter
_____ Clinometer
_____ Hydrolab Multi-Sensor Unit (Bring All Hydrolab Parts to Wet Lab On Thursday Evening)
_____ Hydrolab Data Display System
_____ Hydrolab Sensor Guard
_____ Water filter
_____ Extra AA batteries (16)
_____ Extra 9Volt batteries (2)
_____ Extra "D" batteries (2)
_____ Extra "C" batteries (3)

# Shed Equipment List (truck)

_____ Backpack Electrofishing Units
_____ Shocker Batteries (from closet)
_____ Anode/Cathode Probes
_____ Cooler with Gas Mix (if gas units are for backup)
_____ Long Handled Dip Nets
_____ Short Handled Dip Nets
_____ Holding Cage(s)
_____ Block Nets (large sites)
_____ Plastic Buckets (1 per species)
_____ Blue Plastic Tubs
_____ Bucket Pack(s)
_____ Top setting metric wading rod
_____ Depth stick (graduated in .01m)
_____ Bucket with Lid
_____ Dish Detergent
_____ Salt

# Appendix C: Shenandoah National Park Field Equipment Description

**Backpack Electrofishing Units:**

The principal electrofisher in use at Shenandoah is the Smith-Root, Model LR-24 on a backpack frame. This unit utilizes 24 volt sealed lead acid batteries which seem to hold up in our low conductivity streams.

The backup unit in use at Shenandoah is the Smith-Root, Model 15-D backpack electrofisher that was designed for use in low to medium conductivity waters. Each unit consists of a Honda model EX350 generator and a transformer box mounted on a reinforced nylon backpack frame.

**Anode/Cathode Probes:**

Probes for the backpack units consist of two 1" x 60" fiberglass poles each activated by pressing the rubber flapper forward against the pole. A magnet within the flapper will close the reed switch within the pole handle and activate the output. Each probe is also fitted with a ½" diameter aluminum frame. The anode net frame is equipped with a 3/16" mesh nylon net that is 2" deep and the cathode consists of the aluminum frame only. A "rattail" may be used for exotic fish removals.

**Cooler with Gas Mix:**

The recommended fuel mixture for the generators is 100:1 unleaded gas/small engine oil. The fuel mix should be thoroughly mixed in the metal 2 ½ gallon gas can labeled "Shocker". For daily use, the fuel mix is transported in aluminum fuel bottles of 1 liter capacity. Individual fuel bottles should be packed into the Rubbermaid® cooler labeled "Fuel" for transport in any vehicle. At the parking site, individual fuel bottles are stowed in the side pocket of an available backpack for transport to the monitoring site(s).

**Tool Kit with Spark Plugs:**

The tool kit is contained in a small yellow bag. In addition to spark plugs, the tool kit contains a scrench, a roll of electrical tape, a tube of waterproof cement, washers, bolts, nuts and other spare parts for the shockers, packs and waders.

**Long Handled Dip ("Hoop") Nets:**

Long handled dip nets consist of ½" aluminum frames with 3/16" x 18" mesh mounted on 60" fiberglass poles. Adjustments can be made to mesh depths by knotting the bottom of the net to the desired level. These are used primarily for capturing stunned fish in the stream and for transferring fish between buckets. Net replacements are SHR-1's.

**Long Handled Dip ("Stealth") Nets:**

Stealth nets utilize the smaller net frame of the anode and cathode with a 6 inch deep BDR-3 net. These are used primarily for capturing stunned fish in extremely shallow/cobble filled streams. A stealth net may not be used for bucketing fish if a hoop net is in use for netting either side of the shocker since many fish are lost transferring from a larger net to a smaller net. It is acceptable (by stream) to use an even shallower BRD-3 net (2-3 inches deep).

**Short Handled Dip ("Dinky") Nets:**

These nets consist of 3/16" x 4" mesh minnow nets with aluminum frames mounted on 24" aluminum handles. Short handled nets should only be used at the processing site for sorting and transferring fish between buckets and the electronic balances. Net replacements are BDR-4's.

**Holding Cages:**

Holding cages consist of frames constructed from ¾" PVC pipe and cemented copper fittings equipped with special ordered net boxes or cages. Each 12" x 16" x 20" x 3/16" mesh net box is equipped with a zippered top for loading, unloading and preventing the escape of captured fish. Holding cages are erected on site and submerged in shaded pool habitats such that the bottom 2/3 of the cage is submerged in the stream. These are typically located adjacent to the processing site either upstream or downstream of the monitoring site.

**Block Nets:**

These 4' x 25' non weighted nets are typically needed at the largest sites (lower Rapidan and lower Moorman's River) to adequately block the downstream end of these sections to impede fish movements in or out of the site. Associated gear includes a 100' x 3/8" length of steel cable, 3/8" cable clamps, 45 double-ended snap connectors and a 3/8" socket set with spare 9/16" sockets. These items are typically stored together in a single five-gallon bucket. The top of each net is suspended from the steel cable and the bottoms are anchored to the streambed with large rocks.

**Plastic Buckets:**

Plastic five-gallon buckets are used for temporarily storing, transporting and processing captured fish. The number needed is determined by the expected number of species at each site, expected fish volumes and the number of participating crew as based on previous experience at individual sites.

**Plastic Tubs:**

The large tubs are used for sorting large volumes of fish and for processing trout or other species captured in very large numbers, primarily to prevent overcrowding. The specific number needed is determined by anticipated volumes of trout and/or other fish based on results from previous years.

**Bucket Pack(s):**

These pack frames are typically needed to transport large numbers of buckets to and from monitoring sites.

**Back Packs:**

The specific number of packs needed on any particular day is largely dictated by the number of participating crew. For most park streams, the number of packs equals the number of crew minus three since three individuals are needed to carry the two shockers and one bucket pack. Every person on the crew will be required to transport either a shocker, bucket pack, or regular backpack loaded with sampling and/or personal gear. Generally, the larger the crew, the more packs are required to accommodate the additional personal gear. Several pack sizes and styles are available to accommodate individual needs.

**Clove Oil/Ethanol Mix:**

Clove oil mixed with ethanol in a 1:10 ratio (clove oil/ethanol) serves as a good fish anesthetic which lowers the circulatory rate and subsequent oxygen demand of fishes thus reducing handling stress. This compound is stored and transported in small (250 mL) brown nalgene bottles and is used primarily for gamefish and eels. Non-games may be transferred to a net and “cloved” for a short amount of time before weighing.

**Measuring Boards:**

All measuring boards consist of steel rulers ranging in size from 0-300mm to 0-450mm, attached to a wood trough with a wood stop at the “0” end. It is helpful to have the three existing measuring boards present at each site. Eels will take the larger white board or a 1 ½ inch pipe board.

**Electronic Balances:**

Two O-Haus balances that will measure to the nearest 0.1g will be required at all sites for processing trout and/or other gamefish. These include the 600, 1,200, and 2,000g scales. A 1.2 or 2 and 6 kg balances may also be needed at high volume sites, brown trout sites, eel sites and/or sites with other large game or nongame fish species. The 6,000g scale only reads to the nearest 0.5g. Each scale will need to be calibrated on site with the calibration weights provided. **Make sure that a full range of weights is present in the Pelican® cases for the calibration of each balance taken to the field.**

**Extra Batteries:**

Scales take AA and 9v batteries. Two 9v, and twelve or more AA should be carried every day. The Marsh-McBirney flow meter takes two D batteries and the Quanta Hydrolab unit takes three C batteries. These should be stored in a waterproof container.

**Rubber Gloves:**

These gloves are a critical safety element by guarding against electric shock. Several sizes and lengths are available but each person on or assisting the stream crew is required to wear them at all times when the electrofishing units are in operation. Gloves are “Sol-vex” gloves and are not rated for electricity. However, they do guard against most electrical accidents.

**Polarized Glasses:**

Polarized glasses are to be worn by the entire stream crew. These glasses dramatically reduce surface glare and thus enabling the crew to spot fish more readily. They also serve as eye protection from net poles and tree limbs.

**Fish Key:**

Condensed from Jenkins and Burkhead, 1994, this key represents most of the species known to inhabit park streams. Several copies are available in the clipboards, all on waterproof paper.

**100m Tape:**

The 100m/300' open reel fiberglass measuring tape is used to measure the total lengths and to mark transect locations at the monitoring sites. Each 10m interval (i.e. transect) on the tape has been marked with red electrical tape.

**30m Tape:**

This shorter (30m/100') measuring tape is used for measuring stream widths at each of the 10m cross sections and for delineating width intervals when measuring stream flow. Consider taking two when you have larger crews.

**Marsh Mcbirney Flow Meter:**

The primary unit for measuring stream velocity is the Marsh-McBirney model 2000 "Flo- Mate".

**Top Setting Metric Wading Rod:**

This device serves to position the flow meter probe at the appropriate depth in the water column for measuring stream velocity.

**Depth Stick (graduated in .01m):**

Unlike the wading rod, the depth stick is a 1.5m x 1" section of treated wood dowel that has been marked at .01m intervals for measuring channel depths at each of the 10m cross sections.

**Compass:**

While not a critical component of the fish or stream sampling effort, a hand-held compass should be included with the sampling gear for all of the interior sampling sites.

**Clinometer:**

This is a small hand-held instrument used for measuring the percent (%) gradient of stream sites. In using the clinometer, hold the instrument such that the round side-window faces to the left. The instrument is aimed by tilting it up or down until the hairline in the middle of the window is at the top edge of the point to be measured. **Make sure to read and record the percent (%) scale.**

**GPS Unit:**

GPS units currently in use are Garmin handheld units. Trimble units can be used if higher accuracy is needed. Coordinate all GPS activities with the lead biological technician.

Appendix C: Shenandoah National Park Field Equipment Description (continued)

**Hand-Held Radios w/Batteries:**

The portable radios used by park staff are Bendix-King model DPH5102Xs. These are equipped with rechargeable batteries that should be checked prior to leaving for the field. Due to the location of monitoring sites in steep watersheds, the whip antenna should be installed on one of the units for better transmission/reception. The crew should pack at least two of these portable radios with batteries in the field at all times. Careful attention needs to be paid to the repeater the radio is set on for proper reception and transmission.

**First Aid Kit:**

Items in the first aid kit include CPR mask, bandages of various sizes, an eye dressing unit, gauze, adhesive tape, iodine, ophthalmic solution, aspirin tablets, forceps, scissors, instructions and other useful items. Additional first aid material may be needed under certain circumstances or for certain crew members (e.g. “epipen”). First aid kits will contain Benadryl for allergic reactions in the backcountry. This is in direct response to two park employees and one’s wife who now have delayed anaphylactic shock (red meat allergies) due to tick bites. Items in the kit must be replaced the day they are used.

**Site Directions:**

Site directions are stored in the central file drawer labeled “Historic Electrofishing Data” within the I&M building. Electronic copies are available on the O:drive (O:\WORKING\AQUATIC\FISH DIRECTIONS). Each day, the appropriate field copy should be made on waterproof paper on which individual sites should be highlighted. The fisheries chart in the hallway should be checked for the number and type of expected species such that electronic balance and bucket needs can be planned accordingly.

**Field Data Forms:**

The full suite of data forms required for each site includes the “Site/Water Chem/Velocity” or cover form, “Gamefish Data Form”, “Nongame Fish Data Form” and “Habitat Data Form”. These should all be copied onto waterproof paper as needed each day from originals located in the central files. Multiple copies of all forms should be taken into the field each day. This is especially true for the gamefish forms since multiples are needed at almost every monitoring site in the park. Extra forms should be kept in each of the vehicles “just in case”.

**Metal Clipboards:**

At least two of the aluminum clipboards will be required at each site. Three may be required at large or high fish volume sites. Clipboards contain copies of empty and completed data forms, the fish key, site directions, a fish species code list and pencils.

**Pencils:**

All data is recorded in pencil using mechanical pencils. Always ensure that a supply of these is present in the clipboards or on person before leaving for the field.

**Hydrolab Multi-Parameter Units:**

Complete Hydrolab units consist of a number of components including the multi-sensor unit, data display system, cable, weighted sensor guard and extra “C” batteries for the Quanta should be included for emergency backup.

**Plastic Buckets:**

Plastic five-gallon buckets are used for storing and transporting soap solution for cleaning (i.e. soaking) gear between sites.

**Plastic Tubs:**

The large colored tubs are used for soaking and cleaning waders. The black rectangular tubs are used for cleaning the other gear.

**Soap:**

Dawn dish detergent or equivalent for soaking gear in the field or at the office.

**Salt:**

Salt for soaking waders at the office.

**Drinking Water:**

Each person on the crew is responsible for packing an adequate supply of drinking water for the anticipated conditions of the day. A MSR filter is provided for crews. It is particularly important to plan ahead for the longer hikes and to anticipate personal water needs accordingly

**Lunches:**

Each person on the crew has different metabolic rates and other physiological factors and must plan accordingly.

## Appendix D: Shenandoah National Park Primary and Secondary Sampling Streams with Sites, Number of Electrofishers Needed, Number of Personnel Needed, and Watershed Geology Type by Percentage.

**Primary Quantitative Sites – Two Year Rotation**

| Stream | Site #(s) | # Electrofishers | # People | % Basaltic | % Granitic | % Siliciclastic |
|---|---|---|---|---|---|---|
| Dry Run, North Fork | 2F131 | 1 | 9-10 | 0 | 100 | 0 |
| Hannah Run | 2F050 | 1 | 10-12 | 0 | 100 | 0 |
| Hogcamp Branch | 2F055 | 1 | 9-10 | 85 | 15 | 0 |
| Hughes River | 2F038, 2F039, 2F040 | 3/2/2 | 10-12 | 7 | 93 | 0 |
| Jeremy's Run | 1F007, 1FVA1, 1F118 | 2/2/2 | 10-12 | 69 | 3 | 28 |
| Lower Lewis Run | 3F105 | 1+800 meters | 9-10 | 5 | 0 | 95 |
| Meadow Run | 3F107, 3F109 | 2/2 | 10 | 0 | 0 | 100 |
| Moorman’s River, North Fork | 3F084, 3F045, 3F044 | 2-3/2/2 | 12+ | 66 | 1 | 33 |
| One Mile Run | 3F126, 3F128 | 2/2 | 10 | 1 | 0 | 99 |
| Overall Run | 1F132 | 2 | 9-10 | 68 | 17 | 15 |
| Paine Run | 3F123, 3F124, 3F125 | 3/2/2 | 10 | 0 | 0 | 100 |
| Pass Run | 2F095 | 2 | 9-10 | 44 | 56 | 1 |
| Piney River | 1F003, 1F145, 1F138 | 2/2/2 | 10-12 | 58 | 42 | 0 |
| Rapidan River | 2F093, 2F135, 2FVA4, 2FVA5 | 3-4/3/3/2 | 10-20 | 6 | 94 | 0 |
| Rose River | 2F015, 2F016, 2F017 | 2/2/2 | 10-20 | 85 | 15 | 0 |
| Staunton River | 2F072, 2F074, 2F075, 2F076 | 2/2/2/2 | 10-20 | 0 | 100 | 0 |
| Thornton River, North Fork | 1F030, 1FVA2, 1FVA3 | 2/2/2 | 10-20 | 67 | 33 | 0 |
| Two Mile Run | 3F103, 3F102 | 2/2 | 9-10 | 0 | 0 | 100 |

Appendix D: Shenandoah National Park Primary and Secondary Sampling Streams with Sites, Number of Electrofishers Needed, Number of Personnel Needed, and Watershed Geology Type by Percentage (continued).

**Secondary Sites – Six Year Rotation**

| Stream | Site #(s) | # Electrofishers | # People | % Basaltic | % Granitic | % Siliciclastic |
|---|---|---|---|---|---|---|
| Berry Hollow Run | 2F063 | 1 | 4-5 | 0 | 100 | 0 |
| Big Branch | 3F079 | 1 | 4-5 | 66 | 1 | 33 |
| Big Creek | 2F238 | 2-3 | 9-10 | 81 | 2 | 18 |
| Big Fill Hollow | 2F239 | 1-2 | 9-10 | 3 | 97 | 0 |
| Big Mine Branch | 3F202 | | | 0 | 0 | 100 |
| Big Run | 3F257, 3F021, 3F023, 3F101 | 3/3/2/1 | 12+ | 19 | 0 | 81 |
| Bolton Branch | 1F056, 1F058 | 2/1 | 9-10 | 7 | 93 | 0 |
| Broad Hollow Run | 2F067 | 1 | 4-5 | 0 | 100 | 0 |
| Brokenback Run | 2F252 | 2 | 9-10 | 6 | 94 | 0 |
| Brown Hollow Run | 1F224 | 1 | 4-5 | 42 | 58 | 0 |
| Bush Mountain Stream | 2F222 | 1 | 4-5 | 4 | 96 | 0 |
| Butterwood Branch | 1F218 | 1 | 4-5 | 0 | 100 | 0 |
| Cedar Run | 2F062 | 2 | 9-10 | 100 | 0 | 0 |
| Collet's Run | 2F241 | 1 | 4-5 | 0 | 100 | 0 |
| Conway River | 2F255 | 1-2 | 9-10 | 13 | 87 | 0 |
| Cool Spring Hollow | 3F259 | | | 0 | 0 | 100 |
| Crow Hollow | 2F240 | 1 | 4-5 | 0 | 0 | 100 |
| Deep Run | 3F265 | 1 | 4-5 | 0 | 0 | 100 |
| Devil's Ditch | 2F242 | 2 | 9-10 | 5 | 95 | 0 |
| Dorsey Hanger Hollow | 3F260 | 1 | 4 | 0 | 0 | 100 |
| Doyles River | 3F087 | 2 | 9-10 | 92 | 0 | 8 |
| Dry Run Falls | 2F243 | 1 | 4-5 | 100 | 0 | 0 |
| Dry Run, South Fork | 2F140 | 1 | 4-5 | 21 | 79 | 0 |
| Eaton Hollow | 3F261 | 1 | 4-5 | 64 | 16 | 20 |
| Elk Run | 2F244 | 1 | 4-5 | 0 | 0 | 0 |

Appendix D: Shenandoah National Park Primary and Secondary Sampling Streams with Sites, Number of Electrofishers Needed, Number of Personnel Needed, and Watershed Geology Type by Percentage (continued).

**Secondary Sites – Six Year Rotation Continued**

| Stream | Site #(s) | # Electrofishers | # People | % Basaltic | % Granitic | % Siliciclastic |
|---|---|---|---|---|---|---|
| Entry Run | 2F212 | 1 | 4-5 | 61 | 39 | 0 |
| Front Royal | 1F210 | 1 | 4-5 | 0 | 0 | 0 |
| Fultz Run | 2F122 | 1 | 4-5 | 11 | 9 | 80 |
| Gap Run | 3F204, 3F207 | 1 | 4-5 | 6 | 0 | 94 |
| Gooney Run | 1F225 | 1 | 4-5 | 33 | 67 | 0 |
| Gravel Springs | 1F219 | 1 | 4-5 | 100 | 0 | 0 |
| Greasy Run, East | 1F227 | 1 | 4-5 | 0 | 100 | 0 |
| Greasy Run, West | 1F226 | 1 | 4-5 | 0 | 100 | 0 |
| Hangman Run | 3F203 | | | 33 | 0 | 67 |
| Happy Creek | 1F228, 1F229 | 3/1 | 10-12 | 95 | 5 | 0 |
| Harris Cove | 2F245 | 1 | 4-5 | 5 | 37 | 58 |
| Hawksbill Creek | 3F262, 3F263 | 3/2 | 9-11 | 64 | 16 | 20 |
| Hawksbill Creek, East | 2F119 | 2 | 9-10 | 17 | 83 | 0 |
| Hawksbill Creek, Little | 2F110 | 1 | 4-5 | 48 | 52 | 0 |
| Hazel River | 2F071, 2F053, 2F090 | 3/2/2 | 10-12 | 0 | 100 | 0 |
| Hazel River (Runyon) | 2F139 | 1 | 4-5 | 0 | 100 | 0 |
| Heiskell Hollow (East Fork Compton Creek) | 1F231 | 1 | 4-5 | 70 | 1 | 29 |
| Hickerson Hollow | 1F230 | 1 | 4-5 | 29 | 71 | 0 |
| Indian Run | 1F232 | 1 | 4-5 | 0 | 100 | 0 |
| Ivy Creek | 3F019, 3F020, 3F220 | 2/2/1 | 9-10 | 73 | 1 | 26 |
| Jordan River | 1F233 | 1 | 4-5 | 60 | 40 | 0 |
| Kettle Canyon | 2F208 | 1 | 4-5 | 3 | 97 | 0 |
| Keyser Run | 1F234 | 1 | 4-5 | 86 | 14 | 0 |
| Kinsey Run-Not shocked yet, placeholder | 2F221 | 1 | 4-5 | 0 | 100 | 0 |
| Land's Run | 1F133 | 1 | 4-5 | 9 | 91 | 0 |

Appendix D: Shenandoah National Park Primary and Secondary Sampling Streams with Sites, Number of Electrofishers Needed, Number of Personnel Needed, and Watershed Geology Type by Percentage (continued).

**Secondary Sites – Six Year Rotation Continued**

| Stream | Site #(s) | # Electrofishers | # People | % Basaltic | % Granitic | % Siliciclastic |
|---|---|---|---|---|---|---|
| Lee Run | 2F247 | 1 | 4-5 | 60 | 40 | 0 |
| Lewis Spring Falls | 2F250 | 1 | 4-5 | 83 | 17 | 0 |
| Madison Run | 3F025 | 2 | 9-10 | 14 | 0 | 86 |
| Miller Run | 3F266 | 1 | 4-5 | 8 | 0 | 10 |
| Moormans River,S.F. | 3F082, 3F083 | 2 | 9-10 | 75 | 0 | 25 |
| Naked Creek, East Branch | 2F029 | 2 | 9-10 | 78 | 4 | 18 |
| Naked Creek, South Branch | 2F251 | 2 | 9-10 | 0 | 0 | 100 |
| Naked Creek, W.Branch (Post 1996) | 2F113 | 2 | 9-10 | 70 | 3 | 27 |
| Pocosin Hollow Run | 2F048 | 2 | 9-10 | 18 | 82 | 0 |
| Popham Run | 2F216 | 1 | 4-5 | 0 | 100 | 0 |
| Ragged Run | 2F060 | 1 | 4-5 | 0 | 100 | 0 |
| Rocky Bar Hollow Tributaries | 3F267, 3F268 | 3F267 | | 100 | 0 | 0 |
| Rocky Run | 2F217 | 1 | 4-5 | 0 | 100 | 0 |
| Rosson Hollow (Big Tributary) | 2F213 | 2 | 9-10 | 0 | 100 | 0 |
| Rosson Hollow (Medium Tributary) | 2F214 | 1 | 4-5 | 0 | 100 | 0 |
| Rush River - Waterfall Branch | 1F235 | 1 | 4-5 | 17 | 83 | 0 |
| Rush River (Big Devils Stairs) | 1F223 | 1 | 4-5 | 99 | 1 | 0 |
| Sawmill Run | 3F143, 3F144 | 2 | 9-10 | 20 | 0 | 80 |
| Shenks Hollow Run | 1F211, 1F237 | 2/1 | 9-10 | 100 | 0 | 0 |
| Smith Creek | 1F236 | 1 | 4-5 | 3 | 97 | 0 |
| South River | 2F069 | 2 | 9-10 | 100 | 0 | 0 |
| Stony Run | 2F205 | 1 | 4-5 | 16 | 2 | 82 |
| Stull Run | 3F270 | 1 | 4-5 | 0 | 0 | 100 |
| Swift Run | 3F041, 3F043 | 2 | 9-10 | 70 | 30 | 0 |
| Thornton River, South Fork | 2F036 | 2 | 9-10 | 10 | 90 | 0 |

Appendix D: Shenandoah National Park Primary and Secondary Sampling Streams with Sites, Number of Electrofishers Needed, Number of Personnel Needed, and Watershed Geology Type by Percentage (continued).

**Secondary Sites – Six Year Rotation Continued**

| Stream | Site #(s) | # Electrofishers | # People | % Basaltic | % Granitic | % Siliciclastic |
|---|---|---|---|---|---|---|
| Unnamed Robinson River Tributary | 2F215 | 1 | 4-5 | 0 | 100 | 0 |
| Upper Lewis Run | 3F264 | 1 | 4-5 | 0 | 0 | 100 |
| West Swift Run | 3F206 | 1 | 4 | 74 | 26 | 0 |
| White Oak Canyon Run | 2F009, 2F253 | 2 | 9-10 | 85 | 15 | 0 |
| Whiteoak Run | 3F271 | 1-2 | 9-10 | 14 | 0 | 86 |

# Appendix E: Shenandoah National Park Fish Species List

***SHENANDOAH NATIONAL PARK***
***FISH SPECIES LIST- 2010***

| COMMON NAME | ACRONYM | LATIN NAME | FAMILY |
|---|---|---|---|
| American Eel | AME | *Anguilla rostrata* | Anguillidae |
| Mountain Redbelly Dace | MRD | *Phoxinus oreas* | Cyprinidae |
| Rosyside Dace | RYD | *Clinostomus funduloides* | Cyprinidae |
| Longnose Dace | LGD | *Rhinichthys cataractae* | Cyprinidae |
| Blacknose Dace | BKD | *Rhinichthys atratulus* | Cyprinidae |
| Central Stoneroller | CES | *Campostoma anomalum* | Cyprinidae |
| Fallfish | FAF | *Semotilus corporalis* | Cyprinidae |
| Creek Chub | CRC | *Semotilus atromaculatus* | Cyprinidae |
| Cutlips Minnow | CUM | *Exoglossum maxillingua* | Cyprinidae |
| River Chub | RRC | *Nocomis micropogon* | Cyprinidae |
| Bluehead Chub | BHC | *Nocomis leptocephalus* | Cyprinidae |
| Common Shiner | COS | *Luxilus cornutus* | Cyprinidae |
| Satinfin Shiner[1] | SNS | *Cyprinella analostana* | Cyprinidae |
| Spotfin Shiner | SFS | Cyprinella spiloptera | Cyprinidae |
| Bluntnose Minnow | BLM | *Pimephales notatus* | Cyprinidae |
| Common Carp | CAP | Cyprinus carpio | Cyprinidae |
| Northern Hogsucker | NHS | *Hypentelium nigricans* | Catostomidae |
| Torrent Sucker | TOS | *Thoburnia rhothocea* | Catostomidae |
| White Sucker | WHS | *Catastomus commersoni* | Catostomidae |
| Margined Madtom | MAM | *Noturus insignis* | Ictaluridae |
| Brown Bullhead | BRB | Ameiurus nebulosus | Ictaluridae |
| Yellow Bullhead | YEB | Ameiurus natalis | Ictaluridae |
| Brook Trout | BKT | *Salvelinus fontinalis* | Salmonidae |
| Brown Trout | BRT | *Salmo trutta* | Salmonidae |
| Tiger Trout[2] | TGT | *Salmo X Salvelinus* | Salmonidae |
| Rainbow Trout | RBT | *Oncorhynchus mykiss* | Salmonidae |
| Mottled Sculpin | MTS | *Cottus bairdi* | Cottidae |
| Potomac Sculpin | POS | *Cottus girardi* | Cottidae |
| Rock Bass | ROB | *Ambloplites rupestris* | Centrarchidae |
| Smallmouth Bass | SMB | *Micropterus dolomieu* | Centrarchidae |
| Largemouth Bass | LMB | *Micropterus salmoides* | Centrarchidae |
| Redbreast Sunfish | RDB | *Lepomis auritus* | Centrarchidae |
| Bluegill | BLG | *Lepomis macrochirus* | Centrarchidae |
| Pumpkinseed | PKS | *Lepomis gibbosus* | Centrarchidae |
| Green Sunfish | GSF | Lepomis cyanellus | Centrarchidae |
| Johnny Darter[1] | JOD | *Etheostoma nigrum* | Percidae |
| Tesselated Darter | TED | *Etheostoma olmstedi* | Percidae |
| Fantail Darter | FAD | *Etheostoma flabellare* | Percidae |
| Greenside Darter[1] | GRD | *Etheostoma belnnioides* | Percidae |

[1] extremely rare: Three satinfin shiners found in the lower Rapidan in 1999. Three greenside darters found in Pass Run in 1998. Two johnny darters found in lower Moorman's, North Fork in 1995 prior to the 1995 flood.
[2] hybrid (progeny of female brown and male brook trout)

# Appendix F: Shenandoah National Park Fish Monitoring Data forms

## Standard Operating Procedure 1: Pre and Postseason Fisheries Tasks

Pre and postseason work largely entails visual or mechanical assessment of required equipment (Appendix A). The following list outlines the minimum requirements in order to properly prepare field equipment for use.

**Preseason Tasks Starting in February:**

1) Ensure Hydrolabs® are functioning properly.
2) Check waders for leaks or damage. Repair damage or dispose of waders.
3) Remark/repair all measuring poles, depth sticks etc. (make sure the depth is accurate as the tips may wear).
4) Calibrate both flowmeters to zero.
5) Check electrofisher batteries, order and build new if needed.
6) Make new data sheets for the current year – use waterproof paper.
7) Inventory sampling nets -- order or repair if needed.
8) Repair probes.
9) Repair/polyurethane measuring boards.
10) Check packs, mend tears, wash if needed.
11) Ensure generators are running, run dry, and fog again.
12) On April 1, put new batteries in all aquatic macro equipment (Hydrolabs/flowmeters).
13) Calibrate both flowmeters to zero.
14) On May 15th put new batteries in all of the scales for fisheries.
15) Calibrate both flowmeters to zero prior to fish work.
16) Mend block net.
17) Mend fish cages.
18) Get protocols, JHAs, and other materials ready for crew training.

**Postseason Tasks Starting before the Seasonals Leave:**

1) Take batteries out of flowmeters.
2) Take batteries out of scales.
3) Inspect all equipment and tag problems.
4) Ensure there is one good set of equipment that can be quickly operational in case a problem arises during the winter.
5) Purge all gas/oil mix from generators (tank and carburetor) and oil/fog cylinders.
6) Place electrofisher batteries on chargers and rotate every 2 weeks so all batteries stay fully charged.
7) Ensure data is correct and go through with data manager before final acceptance.
8) Ensure all GPS data is downloaded and correct.
9) Check Hydrolabs every two to three weeks and replace fluids and membranes as needed.

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 2: Training and Safety

**Overview:**
By late May, the full complement of biological technicians and volunteers will have arrived in the park for the summer field season. Some of these biological technicians and volunteers will have been trained on and be proficient in the use of water chemistry and stream flow meters, having recently completed the field component of the park's annual aquatic macroinvertebrate monitoring program. The entire crew will undergo two to three days of training in CPR/First Aid, the principles, use and safety of electrofishing gear, fish identification, taking fish measurements, recording data, and the collection of habitat data from fish monitoring sites, prior to the start of the annual monitoring schedule. All individuals are further instructed in the use of the Hydrolab® to measure water chemistry parameters and flow meter equipment to measure stream discharge.

Each person on the crew will be provided the opportunity to operate an electrofishing unit under close supervision. Individuals will be instructed on the basic techniques and strategies of capturing fish with the units, operating both independently and in multiples when more than one electrofishing unit is required to adequately sample a site. During these initial electrofishing exercises, individual crew members will be evaluated for overall aptitude in the operation of the electrofishing units. Typically, the top three to five individuals in terms of demonstrated proficiency and/or potential will be selected at the discretion of the fisheries program manager or the lead biological technician to be the principal operators of the electrofishing units for the duration of the field season. This is necessary to maintain the consistency of effort and results from all of the park wide monitoring sites.

Following this initial phase, actual monitoring begins on a range of small to medium streams. Most of the streams surveyed during the first two to three weeks are small, fairly shallow, and are limited to two or three fish species. Within these small streams, it is easy to effectively sample while the biologist and lead technician provide on-the-job training in protocol sampling procedures. Several mid-sized streams with six to ten fish species and slightly more complex habitats are also included in this extended training period. During this two to three week period, the crew are closely supervised and further instructed in all aspects of the field techniques and data collection. Upon the completion of this period and prior to monitoring medium, large and joint NPS/VDGIF streams, each member of the crew will be proficient at all of the routine tasks and will be expected to anticipate work tasks and follow through with the completion of those tasks with or without specific instruction from the supervisors.

Electrofishing in mountain stream habitats ranks among the most hazardous work activities performed by biological technicians and volunteers at Shenandoah National Park. The concept of generating and applying high voltage electric current to a body of water in which the operator and a number of fellow crew members are standing seems to constitute a serious breach in fundamental operational safety. This is compounded somewhat by the long hikes over rough and steep terrain while transporting pack loads of up to sixty pounds in conditions of heat and humidity extremes just to access some of the sites. There are also venomous reptiles present, namely the northern copperhead and the timber rattlesnake, a number of biting and stinging insects and an abundance of poison ivy. With adequate preparation, training, the use of personal protective gear, and by being ever mindful of potential hazards during the course of the workday, all of the tasks associated with electrofishing can be accomplished in a manner that is safe to all individuals involved.

**General Safety Procedures**

1) Numerous job hazard analyses (JHA) specific to Shenandoah National Park will be reviewed by the crew prior to the start of the field season. These JHAs include, but are not limited to, *working in tick infested areas*, *light vehicle use*, *backcountry travel*, *electrofishing operations*, *aquatic macroinvertebrate collection, lifting and carrying, and hantavirus.* Certain JHAs may require additional safety training. Park JHAs can be found on the intranet at http://inpshen-cirsc/SHEN/SAFETY/Job%20Hazard%20Analysis/Forms/AllItems.aspx
2) As part of the initial introduction to safety, emergency contact information will be collected from each crew member and will be stored in the field vehicles. The division Secretary, Communications Center, and Supervisor, will also retain copies of emergency contact information. In addition, crew members will have an opportunity to confidentially share known health issues (e.g. allergy to bee stings) with the Biologist or Lead Technician. When permission is granted, these issues may be discussed with the entire crew.
3) Each member of the crew will be given a set of Potomac Appalachian Trail Club Maps for Shenandoah National Park. They will be expected to use these and the topo maps on the wall to know where we are going each day and routes to nearest hospitals. Routes to hospitals can be found on the intranet at http://inpshen-cirsc/SHEN/GIS/Medical%20Facilities%20Near%20SNP/Medical%20Facilities%20Near%20Shenandoah%20NP.HTM
4) The entire crew will be informed on the use, function and procedures of park radios. Each individual on the crew will know how to call for assistance in the case of an emergency and which radio channels will work best in a given location. During the field season, the crew will always have a functional portable radio on hand at all times in addition to the dash mounted vehicle radios.
5) Prior to the beginning of the fisheries monitoring field schedule, every attempt will be made to ensure that nearly every crew member has current certification of basic first aid and CPR. A general rule of thumb is at least half the members certified for the large crew and all for the small crew plus the Biologist and Lead Technician.
6) An approved first aid kit will be carried at all times when field work is conducted. This kit will be stored in a consistent location and all field personnel will be informed of that location (typically this is in the hood of the red pack). Additional first aid material may be needed under certain circumstances or for certain crew members (e.g. “epipen”). Whether “allowed” or not, all first aid kits will contain Benadryl for allergic reactions in the backcountry. This is in direct response to two park employees and one’s wife who now have delayed anaphylactic shock (red meat allergies) due to tick bites. Items in the kit must be replaced the day they are used.
7) There no substitute for good personal preparedness at the beginning of the season and at the beginning of each day. Each person on the crew is responsible for packing an adequate supply of food and water for the anticipated conditions of the day. It is particularly important to plan ahead for the longer hikes and to anticipate personal food, water and clothing needs accordingly.

**Field Techniques Training and Electrofishing Safety**

1) Principles of electrofishing and fish identification will be conducted prior to field season as well as during sampling of smaller streams at the start of the field season.
2) Office tasks, CPR/First Aid, and reviewing JHAs typically take two days. After that, the entire crew will have a half day of field training using all of the equipment associated with fisheries work at Shenandoah. At this time, there will be some fish identification training occurs on the job at individual sites in the following two weeks. Likewise, training in the use of discharge meters, water quality equipment, and habitat sampling procedures will occur on-the-job during initial monitoring efforts. Because of sample site conditions, during the initial two weeks of work, the pace is generally much slower than it will be from mid June to the end of the summer. This allows the crew to become used to working in unison and allows individuals to gain a better grasp of the physical and mental demands of the job.
3) Each person participating or assisting the electrofishing crew will be required appropriate Personal Protective Equipment as identified in the JHAs (e.g. rubber gloves, non-slip waders, polarized glasses, etc).
4) The person or persons operating the backpack electrofishing units will direct the activity of all associated netters and bucket carriers. This can be complicated by the ambient stream noise, the noise from the electrofishing units themselves, and from the combined vocal chatter of the crew.
   a.) As the crew works upstream against the stream flow and up, over or around rocks or other obstacles, it is the responsibility of the primary netters to assist the person who is wearing the backpack electrofisher in negotiating obstacles and to generally help them to maintain their balance during sampling.
   b.) When stunned fish are lodged in rock crevices or other substrates, it is imperative that the electrofisher operator(s) raise the probes out of the water before attempting to dislodge the fish by hand. The operator(s) must remain alert to the fall of a crewmember or themselves and be prepared to immediately stop electrofishing. Shenandoah's wording is "Probes Up" this can be used by anyone on the crew for a falling member **BUT** only by the electrofisher operator(s) for a netter retrieving stuck fish. This ensures that there is adequate preparation before the line of electricity is turned off.
   c.) When the probes come out of the water, your thumbs should be off of the switches and the probe ends should be pushed together. This ensures an electrical short so no current can reach a bystander.
5) All crew members are responsible for maintaining situational awareness, which largely includes identifying and communicating potential safety hazards. This also includes providing a personal safety zone about oneself and generally avoiding direct body contact with a fellow crewmember.
   a) Getting too close to a team of netters can result in being struck by a net handle.
   b) When carrying buckets full of water and/or fish, it is important to take the time necessary to ensure adequate footing to prevent a fall which could result in personal injury and/or the loss of a number of fish and associated data.
   c) Always take the time to help each other over/around obstacles.
   d) Situational awareness reduces with fatigue. Be especially aware of this towards the end of the work day.

**Revision History Log:**

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# Standard Operating Procedure 3: Sampling Conditions and Times

This SOP outlines time periods and environmental conditions when sampling should occur. Consistency of sampling times and conditions between sampling events is important to minimize seasonal associated effects and error when monitoring fish populations.

Sampling Times

1) The field component of the fisheries monitoring program typically begins in early to mid June and ends by late July to mid August. Specific annual starting and ending dates are largely determined by the frequency of rain events, the amount of total rainfall, associated water levels in streams, and budget constraints.
2) To facilitate comparisons of density and biomass between years, individual stream sites should be sampled within the same general time period each year. A temporal sampling disparity of two to three weeks between years is acceptable but larger discrepancies between sampling periods should be avoided when possible.

Sampling Conditions

1) Optimal conditions for electrofishing include low to average water flows of clear condition. Moderate to high water flows should be avoided due to increased turbidity and riffling which can decrease electrofishing efficiency and fish capture probabilities. Faster flowing water may also require higher electrofishing voltages to improve fish capture (e.g. so they remain stunned as they are quickly swept out of the field) -- this voltage increase can lead to higher fish injury in the slower sections of the site. High stream flows also increase safety risks by creating more difficult wading conditions.

2) All sampling will occur when visibility of stream fishes is maximized. As such, sampling during high turbidity (as noted above) or very early in the morning or late in the afternoon will be avoided when possible. In addition, sampling during rain events when surface disturbance or lighting conditions compromise visibilitywill not occur. Ultimately, crew leaders will determine when visibility is adequate for efficient electrofishing. During rain events, the crew leader will determine if it is worth waiting out the storm or returning the fish to the stream and reshocking the whole site another day. See the Electrofishing JHA on the Park internet for lightning storm safety at: http://inpshen-cirsc/SHEN/SAFETY/Job%20Hazard%20Analysis/Forms/AllItems.aspx

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 4: Locating Sites in the Field

When monitoring protocols involve revisiting specific sites during each sampling event, locating those sites quickly and efficiently is critical to a functional program. This SOP outlines methods of locating specific sampling sites in the field.

1) Specific site directions to each site are on file and electronic copies are available on the O:drive (O:\WORKING\AQUATIC\Site_Directions). Additionally, they are included in the Fish and Aquatic Macroinvertebrate Database. Site directions include a written description of the location of each site by number and contain information on tag trees (see step #2), including species, diameter and tag tree locations. Upper (UTT) and lower (LTT) tag trees are further referenced in the directions as left hand side (LHS) or right hand side (RHS) and direction facing up (U) or downstream (D) (e.g. UTT RHSD means that the upper tag tree is on the right hand bank as you're looking downstream). Site directions also include written descriptions of specified road and trail routes and a U.S. Geological Survey (USGS) topographic map of the stream with clearly marked site locations. In the event that access to the stream changes or tags and/or tag trees are lost, the associated site directions need to be revised accordingly. All missing or replaced tags should be noted on the datasheets.

2) The start and endpoints of all fish monitoring sites are referenced by tagged live trees located on the stream bank. Site tags are 1 ½" X 6" yellow metal strips that have "SNP Fish Transect" and the specific site number stamped on them. All tags face the stream and are generally located approximately five feet above the ground on prominent live trees. To facilitate the location of tag trees in the field, tags also face the direction of most likely approach, except in high use areas. In these areas, tags may be more discreetly hidden from view of trails. When suitable tag trees are not located at the exact start or end points, a written description will reference what portions of the stream are included in the transect (e.g. "start at tail-crest of largest pool…end at 2 ft. bedrock step").

3) Unless otherwise noted in the site directions, start and end points occur directly next to associated tag trees, usually at a geomorphic habitat break that limits fish movement (see SOP 6).

4) If needed, UTMs (NAD 83) marking all start and endpoints for monitoring sites have been collected and are on the site direction sheets as well. Additionally, all UTM's can be found in the fisheries database (see SOP 5, for use of GPS units).

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 5: Using the Global Positioning System

## Overview

This Standard Operating Procedure explains the methods that all observers should follow to learn to use Global Positioning Systems (GPS) with the Garmin GPSMAP 76CSx unit and DNR Garmin software.  GPS is used to help navigate to and locate pre-existing sites, and to document the location of new sites. All points reside on the “Aquatic” GPS or you may enter new ones if needed.  If more help is needed, see the data manager or find further directions in O:\WORKING\AQUATIC\GPS_Incoming\GPS Directions.

## Using the Garmin Unit in the Field

GPS is used to navigate to the predetermined location of new sites and to aid in the relocation of existing sites. The location of lower and upper tag trees, parking areas, and other significant features are marked and recorded as waypoints. Always be sure to carry at least one full replacement of spare batteries for the GPS receiver (2 “AA” batteries). An external antenna is recommended when using the GPS under forest canopy. The external antenna increases the reception of satellite signals and is best mounted to the top of the GPS operator’s backpack.

### *Turning on the unit*

1. Turn the unit on and wait several minutes for it to acquire satellite reception. The GPS is most accurate when it has four satellites and a 3D position. This information is displayed on the GPS information page. The location of satellites in the sky changes from day to day and thus sometimes satellite reception is more difficult to achieve. Forest canopy openings can increase the strength of satellite signals reaching the GPS unit.
2. Make sure the GPS is set to UTM NAD83. Under the main menu, select ‘Setup’, then toggle over to ‘Units’ and set the ‘Position Format’ to **UTM UPS**, the ‘Map Datum’ to **NAD83.** Hit the ‘Quit’ button, toggle over to ‘Heading’ and set the ‘North Reference’ to **True**. These settings will be saved on the GPS and should not have to be re-entered each time the unit is turned on.
3. The current estimated accuracy of the receiver is displayed on the GPS Information Page and Map Page. The accuracy fluctuates as satellite signals are gained and lost. An accuracy of + or – 10 m or less is desirable to precisely document location.

### *Navigating to a site*

1. After the receiver is turned on and has adequate reception, press the ‘FIND’ button, highlight ‘Waypoints’ and press ‘Enter’.
2. Press ‘Menu’ then ‘Find by Name’ or ‘Find by Nearest’ to find the appropriate waypoint name and press ‘Enter’.
3. On the ‘Waypoint’ screen select ‘Go to’. Type in the name of the desired point, or press ‘Page’ to scroll through all the options.
4. The Map Page or Compass Page can be used to navigate to the waypoint. The distance and bearing to the waypoint will be displayed at the top of the page. (If not, change the settings so that distance and bearing are displayed.) As the distance to the waypoint decreases, the bearing and distance numbers will begin to fluctuate as the accuracy varies. Using a hand compass in conjunction with the bearing from the GPS unit is often

the easiest way to navigate, and the task can be shared by two people. An alternative to this is to have direction of travel displayed on the screen instead of “north” and track your location to the tag tree area.

### *Marking a Waypoint*

1. The locations of the lower and upper tag trees, parking, and at times travel routes need to be recorded with GPS waypoints. This is accomplished by holding the receiver at the edge of the stream closest to the tag tree, at the vehicle or at designated intervals on a route and holding the Enter button until the ‘Mark Waypoint’ Screen appears. Note: If the Enter button or toggle button are held down too long, a MOB (man overboard) point will be collected. If this happens, just ignore it and start again.
2. The GPS automatically assigns the waypoint a number. Toggle over to the name field and once it is highlighted, press enter to change the waypoint name. Toggle up and down through the numbers and letters and left and right between characters. For the lower tag tree add an “A” to the site number. For the upper tag tree, add a “B” to the site number. Once the last letter or digit has been selected, press enter to save the name and highlight the whole name field.
3. The most accurate way to document a location is to collect a waypoint position that averages at least 100 points. To do this select ‘Menu’ and then choose ‘Average Location’. The ‘Average Location’ screen appears and measurement counts automatically begin. After the ‘Measurement Count’ reaches 100, press enter to save. Press ‘OK’ on the Mark Waypoint screen to return to the Map Page.

## 3.6 Downloading Waypoints into ArcMap GIS

Waypoints for newly established sites as well as updated waypoints for sites with inaccurate GPS locations need to be downloaded onto the office computer upon return from the field. If there is only time for a quick download to secure data off the GPS unit, don’t open ArcMap. Simply download the text file as directed below. Shapefiles can be downloaded at a later time by reloading the text file into the DNR-Garmin program.

1. Turn on the computer, open an ArcMap project (options currently located at O:\WORKING\ AQUATIC\gis\data\2010) and load the most current fisheries monitoring sites shapefile from O:\WORKING\ AQUATIC\gis\data\2010.
2. Turn on the GPS unit and insert the computer plug into the external data port located on the back of the GPS receiver.
3. Open DNR-Garmin software on the desktop (if only downloading the text file) or within an ArcMap document (if downloading text files and shapefiles).
4. At the top of the DNR-Garmin window, select ‘Waypoint’ then ‘Download’. The saved waypoints from the Garmin will now appear in the table.
5. These waypoints need to be saved twice, once as a text file and once to an existing shape file.
6. To save as a text file, under File select ‘Save To’ then select ‘File…’ The files are saved at O:WORKING/AQUATIC/GPS_incoming. The file names created are based on the letter of the GPS used and the date of download, GPSx_year-month-day_wp. Thus, a file downloaded on 06/24/2005 from GPS unit O would be named GPSO_2005-06-24_wp. Make sure the file is save as a text file, .txt.
7. To save the data in a shape file, select ‘File’, Save To’, ‘ArcMap’, ‘Shapefile Layer’ (

8. **Figure 0.1**).

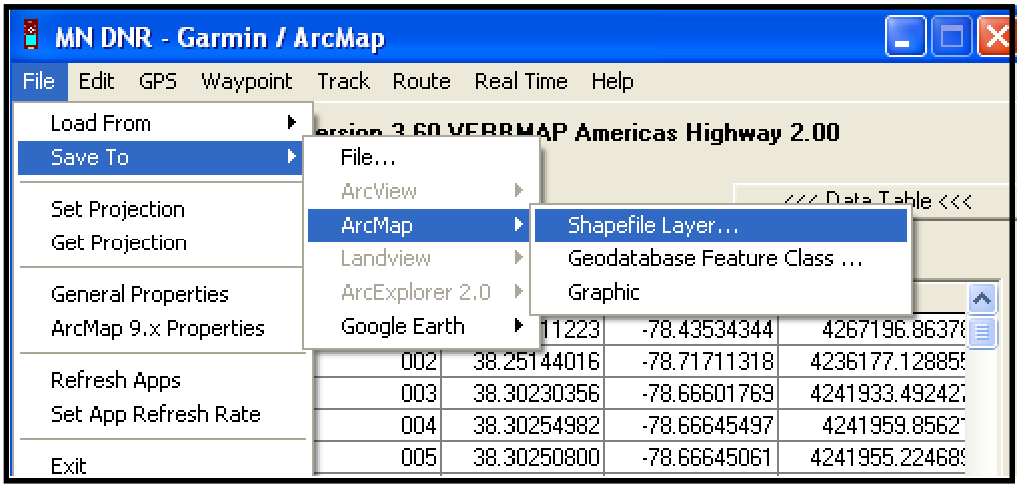


**Figure 0.1.** MN DNR – Garmin with ArcMap window showing how to save data as a Shapefile.

9. To append to an existing shapefile, browse to the shapefile and select ‘OK’. Say ‘Yes’ to append to the shapefile (Caution: ‘No’ will overwrite the existing shapefile!) To save to a new shapefile, browse to the location to be saved, name the file and select ‘OK’.
10. The data should now be saved. Load the data into ArcMap and check the attributes table to ensure that your data were properly transferred. Points should not be deleted from the GPS unit the next day to allow time for daily server backups.

## 3.7 Deleting Waypoints from the GPS Unit

The GPS receiver can hold up to 500 waypoints, but it is advisable to delete the downloaded waypoints from the GPS periodically to keep it from becoming full while in the field.

1. With the GPS turned on, press Menu twice to pull up the ‘Main Menu’ screen. Press the ‘Find’ button. Select ‘Waypoints’ then press the ‘Page’ button to open the table of all current, saved waypoints.
2. Waypoints can either be deleted individually or the entire table can be deleted at once.
3. To delete a single waypoint, toggle to the point, highlight the name and then press ‘Enter’. Select ‘Delete’ and then ‘Yes’ to delete the waypoint.
4. **Be sure that all of the waypoints have been downloaded and backed up before doing this!** To delete the entire table, Press the ‘Find’ button. Select ‘Waypoints’ then press the ‘Menu’ button. Select ‘Delete All’ to clear all of the saved waypoints from the GPS Unit.

## Suggested Reading

For more information, reference the Garmin GPSMAP 76CSx Owner’s Manual and Quick Reference Guide.

## References


Garmin GPSMAP 76CSx Owner’s Manual and Quick Reference Guide. Select ‘Manuals’ at https://buy.garmin.com/shop/shop.do?pID=351.


**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 6: Water Chemistry Sampling

**Overview**
All water chemistry sampling associated with the fisheries monitoring program is conducted with Hydrolab® multiparameter water quality monitoring units. These units provide rapid and accurate measures of water temperature, pH, conductivity, dissolved oxygen, and total dissolved solids. Hydrolab multi-parameter units have five primary components including the multi-sensor unit, data display system, cable, water storage cup, and weighted sensor guard. These units have proven to be rugged and highly reliable in the park's backcountry where many of the water chemistry measurements are taken. The units are also easy to calibrate and once calibrated, likely maintain calibration consistency for ten days following. Nevertheless, to ensure data quality, the units require calibration at the beginning of each week during the field season. Procedures for calibrating the units are listed in SOP 7. Equipment manuals can be found in the "Lower NCR" wet lab above the calibration counter.

**Hydrolab measurements should be taken prior to any of the other monitoring activities at the site that involve personnel in the stream. When placing the unit, take care not to step into the stream in the vicinity of the unit or otherwise disturbing the streambed. These considerations are necessary to capture the natural water chemistry measurements at each site prior to the disturbances that follow electrofishing events.**

<u>Hydrolab Use</u>

The following sequence should be followed when using the Hydrolab in the field:
1.) Confirm that the unit has been calibrated within the last week.
2.) Transport all unit components to a secure location adjacent to the intended sampling point.
3.) Remove the two pin covers (rubber plugs) and secure in a safe location.
4.) Remove water storage cup and replace with weighted sensor guard (for the Quanta unit, remember to remove the rubber cover off the low Ionic strength sensor -- it is filled with saturated KCl solution. Store the cover and solution in a safe location where it will not be spilled).
5.) Attach cable to data display system.
6.) Attach opposite end of cable to the multi-sensor unit.
7.) Always place the multi-sensor unit in a riffle (portion of stream with surface turbulence and greater than zero gradient) of sufficient depth to completely cover the sensors and make sure that the sensors face upstream.
8.) Turn on the data display system.
9.) Wait three to five minutes prior to recording data on the field sheets. Record the unit used, current time, water temperature, pH, conductivity, dissolved oxygen, and total dissolved solids
10.) Turn unit off and detach cable.
11.) Reconnect the two rubber plugs and coil cable on protective spool.
12.) Disconnect the weighted sensor guard and replace with full water storage cup (for the Quanta remember to replace the rubber cover on the low Ionic strength sensor, it is filled with saturated KCl solution).

13.) Stow all of the unit components in one of the packs to ensure that nothing is left at the site. The display unit should always be repacked to the top of the pack and never stowed under all of the other gear.

Hydrolab Storage

When not in use, the water storage cup filled with pH 4 buffer solution must be securely attached to the unit to protect the sensors from damage and desiccation. To prevent serious damage to the sensors, **never store sensors in distilled water**. Stream water may be used in the field if pH4 buffer solution is unavailable, but it should be replaced with pH 4 buffer solution at the earliest possible convenience back at the lab. Periodic maintenance should be scheduled throughout the year as specified in the operators manuals to further enhance the service life of the sensors. Cables associated with the Hydrolab also require careful handling and storage and should not kinked or coiled tighter than 12 inch diameter loops. The cable should always be coiled on the spool during transport and storage. To prevent fouling with debris, it is important to keep the two protective pin cover rubber plugs secured in place when the unit is not in use. Obviously, units should always be handled with care.

**Revision History Log:**

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# Standard Operating Procedure 7: Hydrolab® Calibration and Cleaning

Shenandoah National Park maintains two different Hydrolab® multi probe units for water chemistry sampling. The primary unit is a MS5 with a Surveyor 4a readout and the backup unit is a Quanta multiprobe with display. Each is calibrated and cleaned differently so separate directions for each follow. Hydrolabs® are calibrated at the beginning of each week during the monitoring season. If in doubt, see the manuals for each unit located in the Wet Lab of the Lower NCR building.

Quanta Calibration

1) Quanta Dissolved Oxygen (D.O.) Calibration

   a) Rinse probes with distilled water.
   b) Invert unit, probes up, screw on open-ended cup and fill to the bottom of the O-ring for the D.O. probe with distilled water (DO NOT OVERFILL).
   c) Dab the water off of the membrane and put the rubber top on.
   d) Turn unit ON.
   e) Wait for the D.O. to stabilize (You can do step h while waiting) and get the Temperature off the main screen.
   f) Then toggle to the CALIB menu and hit enter.
   g) Then toggle down to D.O. and hit enter.
   h) From http://www.intellicast.com/Local/Weather.aspx?location=USVA0451 get the barometric pressure and plug into formula: Inches of Hg/0.039=mm of Hg (BP) to get mm of Hg. You must then correct the BP for your altitude (A) by using the following formula: BP'=BP-2.5(A/100). **For the Lower NCR lab BP'=BP-31**
   i) Enter the corrected BP into the unit in mm of Hg using the arrow keys and press ENTER.
   j) Enter the saturated D.O number from the chart for the current water temperature (see Table 1 below). Use the temperature and go across the following chart to find your mg/l value. Enter this # and verify it.

**Table 1, using the 1985 edition of Standard Methods for the Examination of Water and Wastewater. All Values are in mg per liter.**

| $^oC$ | mg/l | $^oC$ | mg/l | $^oC$ | mg/l | $^oC$ | mg/l | $^oC$ | mg/l |
|---|---|---|---|---|---|---|---|---|---|
| **8** | 11.843 | **14** | 10.306 | **20** | 9.092 | **26** | 8.113 | **32** | 7.305 |
| **9** | 11.559 | **15** | 10.084 | **21** | 8.915 | **27** | 7.968 | **33** | 7.183 |
| **10** | 11.288 | **16** | 9.870 | **22** | 8.743 | **28** | 7.827 | **34** | 7.065 |
| **11** | 11.027 | **17** | 9.665 | **23** | 8.578 | **29** | 7.691 | **35** | 6.950 |
| **12** | 10.777 | **18** | 9.467 | **24** | 8.418 | **30** | 7.559 | **36** | 6.837 |
| **13** | 10.537 | **19** | 9.276 | **25** | 8.263 | **31** | 7.430 | **37** | 6.727 |

2) Quanta Conductivity Calibration

a) Rinse the sensors once thoroughly with distilled water by using the calibration cup and shaking the unit to clean all of the sensors.
b) Repeat step one with a small amount of the specific conductance standard.
c) Place the unit on the ring stand with the probes pointed up.
d) Fill the calibration cup to within 1 cm of the top, making sure there are no bubbles around the sensors and cover with rubber top.
e) Turn the unit on and let it stabilize in the Standard Operating Menu. This may take 5 minutes or more.
f) Go to the CALIB mode and choose "SpC".
g) Enter the number of your conductivity standard (for the current solution temperature) using the arrow keys and press ENTER.
**h) Verify the calibration and you are done with the conductivity calibration.**

3) Quanta pH Calibration
   a) Make sure the low ionic strength reference electrode rubber cap is off.
   b) Rinse probes with distilled water; then rinse with a small amount of pH solution (the one you are calibrating for).
   c) Invert the unit on the stand.
   d) Fill cup with pH 7 buffer.
   e) Turn unit on.
   f) Wait for stable reading, (about 5 minutes).
   g) Once it is stabilized, toggle to CALIB menu and press enter.
   h) Use arrows to scroll down to pH and press ENTER: you will then be prompted for the standard (Std:).
   i) Use the arrows to change the numbers to 7.00 (the up and down arrows change the numbers and the right/left arrows change the position of the flashing digit.
   j) Press ENTER.
   k) Go back to the Standard Operating Menu.
   l) Leave the unit ON, discard pH 7.00 solution, and start the process over at step a for pH 4 and then go to Step b (or Step m after completing calibration with pH 4).
   m) Do not discard the pH 4 buffer in the cup, put some reference solution in the white rubber cap, place it on the reference probe, screw the cup on with ph4 buffer in it and go!

MS5 Calibrations

1) MS5 D.O. Calibration
   a) Rinse probes with distilled water.
   b) Make air saturated water by filling a 1 liter bottle half full of distilled water. Seal the bottle and shake vigorously for 40 seconds.
   c) Invert unit, probes up, screw on open-ended cup and fill almost to the top of the cup with air-saturated distilled water (DO NOT OVERFILL).
   d) Invert the screw cap and place on top of the cup to cover but not to create an airtight seal.
   e) Turn unit ON.
   f) Wait for the D.O. to stabilize (You can do step i while waiting).

g) Then go to the calibrate menu.
h) Then toggle down to LDO : mg/l and press Select.
i) From http://www.intellicast.com/Local/Weather.aspx?location=USVA0451 get the barometric pressure and plug into formula: Inches of Hg/0.039=mm of Hg (BP) to get mm of Hg. You must then correct the BP for your altitude (A) by using the following formula: BP'=BP-2.5(A/100). **For the Lower NCR wet lab, BP'=BP-31**
j) Enter the corrected BP into the unit in mm of Hg using the arrow keys and press Done.
k) See http://www.hydrolab.com/web/ott_hach.nsf/id/pa_videos_and_transcripts.html for additional information.

2) MS5 Conductivity Calibration
   a) Rinse the sensors twice thoroughly with distilled water by using the calibration cup and shaking the unit to clean all of the sensors pouring it out each time.
   b) Dry the sensor with a towel and cotton swab.
   c) Calibrate to "0" while the sensor is dry by doing the following.
   d) Turn the unit on and let it stabilize. This should take a minute or more.
   e) Go to the Calibrate mode and choose "SpCond : uS/cm".
   f) Enter "0" for the first conductivity standard slope.
   g) Rinse the sensors twice thoroughly with a small amount of the specific conductance standard by using the calibration cup and shaking the unit to clean all of the sensors, pouring it out each time.
   h) Place the unit on the ring stand with the probes pointed up.
   i) Fill the calibration cup to within 1cm of the top with the specific conductance standard, making sure there are no bubbles around the sensors and cover with black cap.
   j) Turn the unit on and let it stabilize. This should take a minute or more.
   k) Go to the Calibrate mode and choose "SpCond : uS/cm ".
   l) Enter the value of your conductivity standard (for the current solution temperature).
   m) Press Done.
   n) Verify the calibration and you are done with the conductivity calibration.
   o) See http://www.hydrolab.com/web/ott_hach.nsf/id/pa_videos_and_transcripts.html for additional information

3) MS5 pH Calibration
   a) Rinse probes twice with distilled water.
   b) Rinse twice more with a small amount of pH solution.
   c) Invert the unit on the stand.
   d) Fill cup with pH 7 buffer.
   e) Turn unit on.
   f) Wait for stable reading, (about 1 minute).
   g) Once it is stabilized, go to Calibrate mode and press Select.
   h) Use arrows to scroll down to pH and press Select: you will then be prompted for the standard (Std:).
   i) Use the arrows to change the numbers to 7.00 (the up and down arrows change the numbers and the right/left arrows change the position of the flashing digit.

j) Press Done.
k) Leave the unit ON, discard pH solution, and start the process over at step a for pH 4.00 and then go to Step l (or Step l following calibration with pH 4 solution).
l) Do not discard the pH4 buffer in the cup, screw the cup on with ph4 buffer in it and go!
**m) [http://www.hydrolab.com/web/ott_hach.nsf/id/pa_videos_and_transcripts.html](http://www.hydrolab.com/web/ott_hach.nsf/id/pa_videos_and_transcripts.html)**

## Hydrolab® Multiprobe Cleaning

**This information has been condensed from the Hydrolab® Manuals found in the Lower NCR wet lab.**

**The cup should be taken off of the probes and the probes and cup rinsed every Thursday and Monday morning. This will help prevent corrosion and algae from forming on the sensors. The probes should be lightly cleaned with detergent and water every two weeks and thoroughly cleaned every four weeks.**

Quanta Cleaning
To start, use a little Dawn dish detergent in the cup and shake the probes in that. Then rinse well and start the individual probe cleaning. Be sure to wash the cup as well to get rid of the algae that tends to grow in it. An old tooth brush works well for cleaning.

1) pH
    a) If the pH readings are slow, show a lot of drift, or never stabilize then the electrodes should be cleaned. (glass probe only not low ionic strength probe).

    b) Clean pH probe using rubbing alcohol on a Q-tip.

    c) If that does not help, then soak the probe in 0.1 Molar HCl for 5 minutes. Rinse the probe, then soak in pH 7 buffer for 10 minutes. (This should rarely if ever have to be done.)

    d) Refilling the reference electrode (Quanta unit only) takes 4M KCl & AgCl and 2 of the salt tablets. Only replace solution in this probe when the tablets have dissolved (about every 4-5 weeks). A good rule of thumb is every time you change the D.O. membrane change the reference solution.
        i. Start by pulling the tube off, being careful not to spill the solution on yourself.
        ii. Dump the solution in the sink and rinse the sink (not the probe).
        iii. Drop two tablets in the tube and fill with 4M KCl & AgCl.
        iv. Once you start the tube on, invert the unit and finish pressing the tube down snug allowing the air to escape through the ceramic membrane. If no air or liquid escapes, the membrane is clogged and must be replaced.

2) D.O.

a) The D.O. membrane should be changed every 3-4 weeks.
b) The gold cathode may need to be cleaned (be EXTREMLY CAREFUL) with an S.O.S. pad or fine steel wool.
c) Frequent electrolyte changes will maximize electrode life. Getting the membrane back on is tricky. No bubbles can be in the solution, and the membrane can have no wrinkles or be stretched.
    i. Start by putting the unit in the stand with the probes upright.
    ii. Pull the o-ring off which holds the membrane on.
    iii. Pull the membrane off.
    iv. Take the unit out of the stand and pour the solution out of the probe (2 Molar (M) KCl ).
    v. If the gold cathode appears tarnished, gently rub with fine steel wool.
    vi. After rubbing, fill the probe with 2M KCl (D.O. electrolyte) and dump out.
    vii. Put the unit back in the stand and fill the probe with 2M KCl making a dome with the solution while making sure there are no bubbles in the solution.
    viii. Carefully remove one clean membrane from the pack and lay it on the probe. Check to see that there are no bubbles trapped under the membrane. If there are, lift off and put more solution. You may have to do this several times.
    ix. Lay the O-ring on the membrane.
    x. Use both hands and gently push the O-ring down into its notch.
    xi. If the membrane is wrinkled, stretched, or has a bubble trapped under it, pull it off and start over.
    xii. If the membrane looks good, trim off the extra with a scalpel.

3) <u>Specific Conductance</u>
    a) The conductivity probe is on the interior of the D.O. probe. (you'll notice an oval hole on the interior)
    b) Use a Q-tip and rubbing alcohol to clean the interior of this several times. This should be sufficient.

<u>MS5 Cleaning</u>
Use only dish detergent and water with a soft brush (old toothbrush) to clean the probes and storage/calibration cups. If readings are drifting it may need to be serviced by Hach/Hydrolab. Call service technicians at 800-949-3766 opt. 2.

<u>Charging the Units</u>

The Quanta takes three "C" cell batteries. Use a quarter to twist the cap off at the base of the "handle". Change the batteries and screw the cap back on.

The MS5 Surveyor 4a **must be turned on** to charge. There are no replaceable batteries.

Storage

There is a fast growing algae in the park headquarters water system so please use pH 4 buffer solution instead of tap water in the cup for storage.

**Revision History Log:**

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## Standard Operating Procedure 8: Fish Sampling

Fish sampling in Shenandoah National Park involves multiple pass electrofishing. To prevent fish from entering or leaving the sample site during sampling, field crews will ensure that appropriate barriers to fish movement are present at the downstream and upstream end of the sample reach.

1) **Constructing a habitat break.** Upon arrival at individual monitoring sites, the upper and lower tag trees need to be located and the associated start and end points of the reach need to be identified. These points then need to be assessed for adequacy asbarriers to fish movements into or out of the site. This is important as fish movements into or out of the site should be minimized during sampling. Habitat breaks at some of the sites require additional measures such as the construction of temporary impediments to fish movement using adjacent cobble or small boulders. **Before making any modifications to habitat breaks, particularly at the upstream end, make sure that all activities associated with the use of the Hydrolab® have been completed.** Otherwise, modify each habitat break as necessary to impede fish movement. In many cases, the existing breaks are adequate and consist of relatively permanent geomorphic features (e.g. bedrock or boulder step). During assessment or construction of the upper break, it is helpful if the upstream break is temporarily visually marked for the electrofishing crew. Small rock cairns can function as a useful reference aid to electrofishing crews working upstream. This action can be accomplished expediently by one person in between the Hydrolab® sample and the first electrofishing pass.

***Block nets nets are typically needed at the largest sites (lower Rapidan and lower Moorman's River) to adequately block the downstream end of these sections to impede fish movements into or out of the site. Associated gear includes a 100' x 3/8" length of steel cable, 3/8" cable clamps, 45 double-ended snap connectors and a 3/8" socket set with spare 9/16" sockets. These items are typically stored together in a single five-gallon bucket. A cable is first stretched between two trees. Then the top of each net is suspended from the steel cable and the bottom of the net is anchored to the streambed with large rocks. The net should be visually inspected for stuck fish during or after each run.***

2) **Setting up the electrofisher.**
    a. The remainder of the crew should be busy assembling the electrofishing gear and/or setting up a fish processing site. At this point, all personnel assigned to participate in the first electrofishing pass should have the appropriate waders or hip boots on, and should have located a suitable pair of gloves and polarized glasses. The person running the backpack unit should ensure that the probes are properly attached and the battery is plugged in before putting the battery cover back on the unit. A uniform setting of 70 Hertz and a pulse width of 1 millisecond is used across all park waters – this is an established and effective setting for electrofishing. For the battery units, this is the 7% setting (edit note: this may need additional detail) and for the Generator units it is "J" on the alphabetical knob and "2" on the numerical knob. For adjustment or use of LR-24 electrofishers, see Smith-Root manual (Smith-Root, 2009).

b. When using the generator units: Prior to the first pass of the day, it is a good idea to start the generators and allow them to warm up for several minutes before entering the stream. Let them run at the high RPM setting otherwise they foul the sparkplugs extremely fast. Make sure that the fuel cap lever is set to the ON position. Locate the engine switch on the generator and set it to the choke position. After taking up the slack, pull the starter cord rapidly to start the engine. One or two pulls are usually sufficient. Then, move the engine switch from choke position to ON. The person designated to operate the unit needs to make sure that the probes are securely attached to the transformer box and that the box is plugged into the generator.

c. Voltage outputs on all units will be set to the same range as determined by stream conductivity (see table 1). All unit voltage settings will be determined by either the fisheries program manager or the lead biological technician. Volume of flow and the reaction of fish to the electrical field will dictate the final setting. All settings should remain the same through all electrofishing passes for that stream reach.

d. Table 1. The voltages listed in this table are approximate starting points for adjusting the voltage for a particular conductivity.

| **Conductivity (uS/L)** | **Approximate Voltage Setting (v)** |
|---|---|
| 10 | 1,000-1,200 |
| 20 | 900-1,000 |
| 30 | 600-800 |
| 40 | 400-500 |
| 50 | 300 |
| 60 | 300 |
| 70 | 200 |
| 80+ | 100 |

3) **<u>Electrofishing.</u>**

a. Each unit will be tested briefly at a point downstream from the lower habitat break to evaluate fish responses to the selected outputs. Following any last minute adjustments, the first pass begins immediately above the lower habitat break. On sites where two or more units are being operated together, the operators must work together as a team and remain abreast of each other in the streambed along with associated netters to the extent possible. The line abreast crew formation should methodically shock all habitats along a cross-sectional gradient while electrofishing in a manner as appropriate to minimize fish escapes downstream.. The operators set the pace of the entire stream crew and should maintain a constant forward momentum, but not move so fast that habitat is missed. The objective is three thorough and efficient passes of equal effort through the site.

b. Although some fish smaller than 40mm may be captured, in general, little effort will be spent by the electrofishing crew in the pursuit and capture of fish smaller than 40mm. This is due the difficult identification of small cyprinids, high mortality of small fish during electrofishing events, and inadequate mesh sizes on sampling gear for capture of fish less than 40mm in total length.

4) **Netting, Bucketing, and Transporting Fish.**

a. All fish encountered within the site should be netted and bucketed as quickly and efficiently as possible. Stunned fish should be netted with a rapid scooping action from below, not from above the fish. Anticipate receiving a large number of fish from the net attached to the anode as the operator will be routinely passing stunned fish to the nearest available netter. The benthic fishes including suckers, sculpin, darters, and some dace routinely become lodged in streambed substrates. These fish often have to be extracted by hand and it is imperative to make sure that the operators have cut power to the probes and raised them clear of the water before reaching for a lodged fish. Netters should never stick their hands in the water without verbal conformation with **ALL** personnel who are operating electrofishers.

b. Those persons carrying and transporting buckets are responsible for the welfare of the captured fish. Moderate volumes of live fish in the buckets should be routinely transported to the processing site. **Always work to prevent overcrowding in the buckets and overheating of the bucket water.** During the first pass, on sites of high fish volume, or at sites with water temperatures in excess of 16 °C, transports from the stream crew to the processing site may be as frequent as every ten minutes. In most cases, there will be additional personnel on site that can serve in this role. Water management and the prevention of overcrowding in the buckets is the key to fish survival. On sites where fish abundance is not as great, the water in individual buckets should be changed and replaced with fresh water from the stream about every ten minutes prior to transport to the processing site. In these cases, do the water changes at a convenient habitat break so that the operators can momentarily stop electrofishing. Water changes should not be done over the stream to prevent accidental escapes. Rather, move to a convenient location on the adjacent stream bank, pour the fish into one of the large dip nets and then dump the fish into a bucket of fresh water. If any fish manage to flip out of the net or bucket during the transfer over land, they can quickly be recovered. On sites with a large number of small fish (e.g. juvenile blacknose dace) place one net inside the other and then pour the fish from the bucket. This will trap most of the smaller fish that slip through the holes in the first net.

5) **Processing Fish.**

a. At the processing site, there will be at least one and possibly three or four large plastic tubs each containing 15 to 18 gallons of fresh stream water. When high numbers of fish are present, it is most convenient to sort fish from one or two of the large tubs. To keep the water in the tubs fresh and relatively free of debris, any transported fish should first be poured into a large dip net and then dumped into the tub(s). On sites having relatively low fish volumes, it is often more convenient to sort from the five gallon buckets, exchanging buckets with fish for empty buckets as needed for transport. Trout should be sorted into and recovered in the plastic tubs since they tend to be the larger fish, both in terms of body size and total volume in the sample. They should be processed in small lots in a 5 gallon bucket. The same principle should be applied to other species as well when large volumes of individual species are captured. The large tubs in these cases are the best line of defense against additional stress and/or mortality. Otherwise, the typically smaller volumes of other fish species can be easily accommodated, by species, in five-gallon buckets.

b. Only staff that are familiar with the species found in a study reach should be responsible for fish identification and sorting.

c. In situations where there are enough personnel for a separate processing crew, trout processing can begin once the first of the transported buckets has been sorted. To a full 5 gallon bucket pour slightly less than a quarter of a cap full of the clove oil/ethanol mix (see appendix Cfor ratio). This functions as an anesthetic to the fish, reduces stress, and greatly expedites data processing. Gently stir the water to which the clove oil has been added to allow for appropriate mixing and begin to process the fish in the order that they begin to succumb. Individual fish will typically begin to roll onto their side as the anesthetic takes effect. Some of the fish will arrive at the processing site in a stressed condition. In these cases, do not subject stressed individuals to the clove oil. Always process the most obviously stressed fish first. Getting stressed fish processed and back into fresh water will maximize their chance of survival.

d. All trout are individually measured to the nearest 1.0mm and weighed to the nearest 0.1g. For measuring total lengths, place fish on the measuring board nose first such that their nose is in contact with the wooden stop at the "0" end of the ruler. Record the total length out to the absolute tip of the caudal or tail fin. Next, make sure that the electronic balance has been tared to "0" (while thethe container being used as a pan is present on the balance), place the fish into the container on the balance and record the total weight. Due to water accumulations in the container, or wind, or other variables, the balance will likely need to be tared after each fish. Immediately following both measurements, the fish should be returned to a tub containing fresh water (designated as the "recovery bucket"). Following each pass, make the line between the rows on the gamefish data form (Appendix F) darker and extend it to the margin. This will greatly expedite distinguishing the pass during data entry while ensuring an accurate count for the number of fish to be added for data entry (edit note: should add a section of a properly filled out data sheet).

e. The nongame fish (i.e. not trout, centrachids, or ictalurids) should not be subjected to clove oil **(with the exception noted below)** since they are not processed individually. Once the entire pass has been completed, and all of the nongame species sorted, the entire catch, by species, needs to be counted. After the total count has been recorded on the nongame data form (Appendix F), determine the smallest and largest fish of each species and record or call out the length of each. Also, record or call out the total number of mortalities or dead fish by species**. Make sure that the data recorder acknowledges the receipt of each record since there may be several people processing nongame fish simultaneously.** The data recorder needs to make sure that each entry for a particular species corresponds to the current electrofishing pass being processed. Once all of the count and length data have been recorded for an individual species, a mass weight needs to be determined on the balance. Use a larger container for this since fish will be flipping about as soon as they are poured into the container for weighing. **In situations involving large numbers of individual nongame species, particularly blacknose dace, it is helpful to dip the entire group into the tub or bucket containing clove oil immediately prior to weighing.** This reduces the tendency of fish to flip themselves out of the container while the lot is being weighed. Record total mass weights for each species to the nearest 0.1g. Due to balance precision, fish weighed on the 6,000g scale can only be measured in 0.5g increments.

f. As the fish processing for each species including the trout are completed, have them immediately transported to holding cages located in the stream, outside of the transect. To reduce additional mortality during fish recovery, holding cages should be set up in areas where current velocities are extremely low and depth is maximized. All fish from all passes will be held in the holding cages until the site is completed.

6) **<u>Multiple Passes.</u>** Passes two and three and the associated transport and fish data processing will be done in a similar manner as the first pass. The primary difference is that fish volumes should dramatically decrease with each successive pass. In the event that more trout are captured during the third pass than during the second, a fourth pass will be conducted to reestablish the depletion curve. At the conclusion of all passes, all live fish are released back into the stream in the approximate proportions and habitats from which they were captured. All mortalities are separated out during processing and disposed of well away from the stream bank, preferably under a log or large rock.

**Revision History Log:**

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# Standard Operating Procedure 9: Habitat Sampling

**Overview**

Habitat sampling includes a combination of actual measurements (stream width, site length, depth, and gradient) and visual estimates (% riparian cover and type, stream substrates and pool to riffle ratio). Habitat measurements are typically taken either between or following electrofishing passes. The timing with respect to other tasks will be largely determined by the number of crew available and the complexity of individual sites. At times, crew sizes are sufficient such that a habitat crew of three can follow behind the electrofishing crew on their third electrofishing pass. Habitat measurements always start at the downstream habitat break (edit note: may need to include a glossary or definition of terms) that comprises the start point for electrofishing at each site. Readings are always taken from left to right while facing upstream. A crew of three to four including a data recorder, a measurer, and one or two people to hold each end of the 30m tape is optimal and the steps below will assume three to four individuals are involved. Required equipment includes two spooled measuring tapes (30m and 100m), a depth staff, a clinometer, a clipboard, and a habitat data form (Appendix A).

General sampling scheme

The start and end of each sampling reach will be at obvious breaks in stream habitat that are sufficient to stop or slow fish passage. Due to this, the reach may be shorter or longer than 100m. The 100m measuring tape should be extended longitudinally between the lower and upper boundaries of the site. This does not need to be done immediately, you may pull the tape in 10m intervals as you proceed from transect to transect. As the tape is run, it should be placed along the middle of the stream, following the general shape of the channel as much as possible. Habitat measurements are made at 10m intervals along each 100m stream site. If a site is longer than 100m, the 30m tape should be run for each additional segment. The overall scheme for habitat measurements is illustrated for a representative stream site in **Figure 1**. Note that transects T0 through T10 demarcate segments S1 through S10, respectively. Width, depth, riparian vegetation type , riparian vegetation % cover, and substratum are measured at each transect. Pool-riffle ratio, and % slope (gradient) are measured throughout each segment.

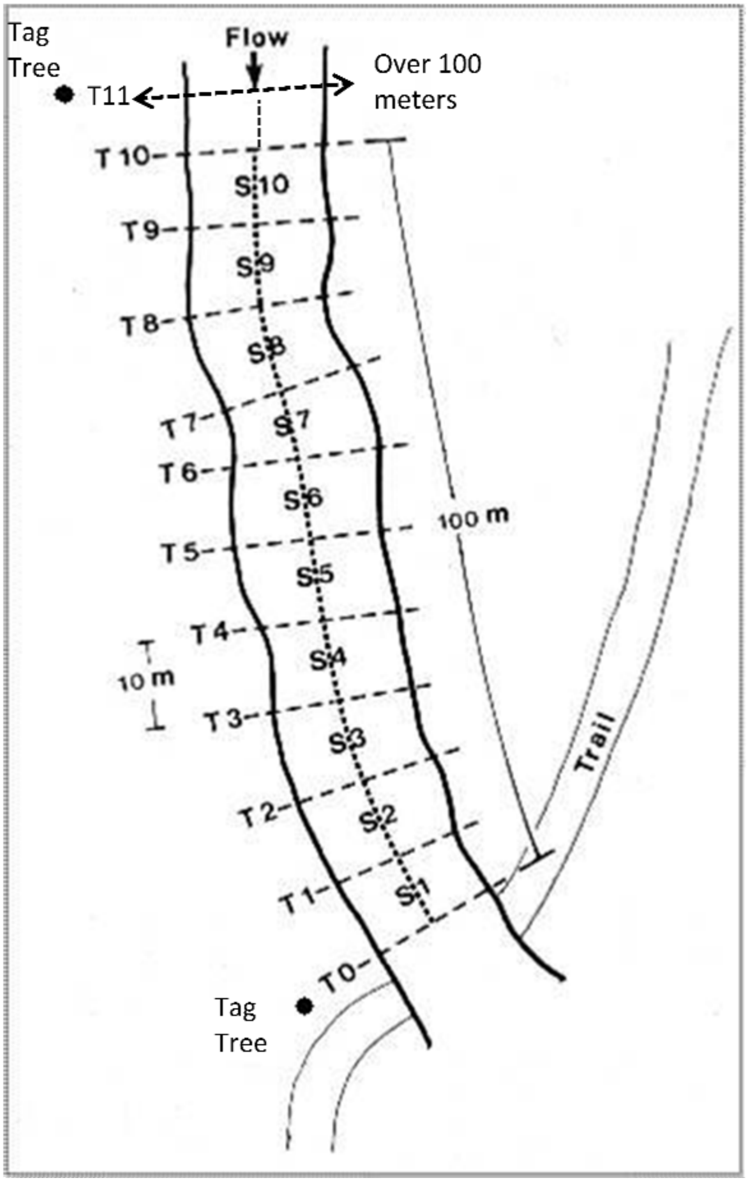


**Figure 1.** Diagram of stream reach. Width, depth, riparian vegetation, and substratum are measured at each transect. Pool-riffle ratio, and % slope (gradient) are measured throughout each segment.

Collecting Data

1) **Clinometer Use.** Prior to collecting habitat information, the technician should be familiar with the use of clinometers (see SOP 10 for details)

2) **Establishing the first transect and recording width**. To begin habitat data collection, stretch the 30m tape from **left to right (as you look upstream)** across the stream channel, perpendicular to the direction of flow at the downstream start of the sample reach. Pull any slack out of the tape and measure the waterline to waterline width (wetted width). **Wetted width will always define the left and right bounds of a transect.** Record width to the nearest 0.1m on the habitat data form in the first row that corresponds to

"zero meters" (0) in the "Distance" column.  Stream widths will be recorded at each 10m interval up to and including the habitat break at the upstream end of the monitoring site.

3) **Recording water depth and substrate.** Using the tape as a guide, water depths are measured and substrate types are estimated at ¼, ½ and ¾ distance across the channel from **left to right (as you look upstream)**.  At each point (¼, ½ and ¾), water depth is measured to the nearest 0.01 m directly below the transect (i.e. tape) using the depth staff and the dominant substrate immediately impacted by the depth staff is visually estimated according to the substrate classification chart as follows (O = organics, L = silt (<0.06mm), S = sand (0.06-2mm), PG = pebble/gravel (2.0mm-10.0cm), C = cobble (10.0-30.0cm), B =boulder (>30cm), BR = bedrock).  Surface substrates, not underlying substrates, are recorded (e.g. if the streambed and associated cobble (C), boulder (B), or bedrock (BR) substrates are covered with a layer of silt at the point of contact between the staff and the streambed, then silt or "L" is entered as the substrate value for that point.  If a layer of moss, leaf debris, sticks or woody debris, or a thick layer of algae exists at the point of contact, then organic or "O" is entered as the substrate value for that point.)  In this manner, mid-stream wood jams that are not submersed in water would be recorded as having a zero depth and a substrate of organics.   Stream depths and substrates are recorded at each 10m interval up to and including the habitat break at the upstream end of the monitoring site.

4) **Recording riparian vegetation type and % cover.** Riparian vegetation data can be collected efficiently by the data recorder.  From the center of the transect, the data recorder should determine the type and estimate the percent of vegetative cover directly over the transect.   The estimate includes all vegetation that extends over the wetted width of the stream above the transect.  Vegetation is differentiated between trees (T), shrubs (S), grasses/forbs(GF), or none (N).  Percent cover is estimated at >66% (coded as 3 on data sheet), 33-66% (coded as 2), <33% (coded as 1) or 0% (coded as 0).  When vegetation type is recorded as "none" (N), it should always be accompanied by a 0% (0) determination, or vice-versa.  Riparian cover will be recorded at each 10m transect up to and including the habitat break at the upstream end of the monitoring site.

5) **Recording gradient.** After wetted width, substrate, depth, and riparian vegetation have been recorded for the transect, the 100m tape should be secured at the site start point in the center of the channel.  With the endpoint securely anchored, the 100m tape should now be extended upstream at mid-channel.  Each 10m interval on the tape is marked with a strip of red electrical tape so that it can be readily distinguished at a distance.  At 10m, any slack should be pulled out of the tape from the upstream end.  When multiple channels are encountered, run the tape up the center of the "island" (for discussion of islands, see step #9).  Using a clinometer, gradient should be measured (in %) within the segment (e.g. for the first segment, between the 0 and 10m transect, see SOP 10 for details).    **It is important to note that the gradient value measured from the 0 meter transect of the monitoring site must be recorded in the 10m distance row on the data form.** The space for gradient on the "0" distance row is crossed out for this purpose.  Gradient measurements will be recorded in each segment up to the habitat break at the upstream end of the monitoring site.  The 10m offset on the data form will provide a

gradient value that corresponds to the last recorded distance measured at the upstream end of the monitoring site.

6) **<u>Recording pool habitat.</u>** Following gradient estimation, the data recorder walks upstream through the first 10m section and conducts a visual estimate of the relative amount of pool versus riffle habitat. Pool habitat is defined as wetted habitat with an apparent water velocity of 0. Riffle habitat would constitute all other portions of the channel. Pool and riffle habitat are estimated as a percent of the total wetted channel, but are recorded as integers between 0-10. As such, the relative amounts of pools versus riffles for each 10m section should always be two numbers that add up to ten. **Note that pool and riffle habitat data also have a 10m offset on the data form.** Therefore, the first recorded values for these habitats must be recorded in the "10" distance row on the data form. The spaces for "pool" and "riffle" on the "0" distance row are crossed out for this purpose. As for gradient, the 10m offset on the data form will provide a set of habitat values that corresponds to each 10m section including the last measured section at the upstream end of the monitoring site.

7) **<u>Establishing the next transect.</u>** At this point, the entire habitat crew should be assembled at the 10 m distance point along the monitoring site. Begin another transect by stretching the 30m tape across the stream channel, making sure it intersects the 100m tape at the appropriate 10m interval. Again, all transects are perpendicular to stream flow. The same sequence for measuring or estimating stream channel width, depths, substrates, riparian cover type, percent riparian cover, gradient and habitats must be followed at each subsequent 10m distance interval as described above.

8) **<u>Ending the sample reach.</u>** Monitoring sites rarely end right at a 10m distance interval. It is important to record the total length of the site to the nearest 0.1m in the distance column following the last full 10m section measured. For example, if the last full 10m section was at the "100" distance point and there are still several meters remaining between that point and the actual upstream habitat break (endpoint where electrofishing sampling ended), measure to the endpoint and record the total length (e.g.104.7) in the "110" distance cell. Be sure to cross out the "110" and record the actual distance just above it in the remaining space within that cell. Gradient and habitat measurements are continued up to the endpoint and channel width, depths, substrates and riparian cover are measured at the endpoint transect as described above.

9) **<u>Encountering Islands.</u>** An additional important consideration at some of the monitoring sites is the procedure for defining and measuring habitat parameters around islands. Islands are defined as dry portions (in general, greater than 1m in width) of a transect that support rooted vegetation. When proceeding upstream past an island, the 100m tape should always be extended up the center of the island. At a transect that intersects an island, the width of wetted channel on each side of the island is summed for a total wetted channel width. When measuring wetted width, a chart at the bottom of the habitat data form contains a special set of columns and rows to assist in recording information from multiple wetted channels (see Habitat Form, Appendix F). Make sure to note the specific distance point(s) where islands are present in the "Dist." column of the island chart. The

clip or endpoint of the tape should be positioned on the left-hand stream bank and the tape should be extended to the right as noted above. The left-hand channel will be “channel A” and the right hand channel will be “channel B”. The data recorder should traverse the tightly stretched tape and record the waterline to waterline width of each channel as well as the width of the island. Individual widths for Channel A and Channel B and the island, as well as the total wetted width (i.e. sum of Channel A and Channel B widths) are recorded in the island chart on the habitat form. The total width is then also recorded in the main table for the corresponding transect.

Depth and substrate measurements will be taken in the same order across the stream from left to right but will be split between the dominant and subdominant channels. The dominant channel will be determined by a visual judgment of discharge. Two depth and two substrate measurements will be recorded in the dominant channel regardless of whether it is channel A or B and the remaining one set of depth and substrate measurements will be recorded in the least dominant channel. For recording single values in side channels, take measurements from the center of that channel. The dominant channel should be visually split into thirds with measurements recorded at 1/3 and 2/3 width intervals. **The resulting data are recorded in the main table under the ¼, ½, and ¾ columns as at single channel sites.** All measurements are still taken from left to right facing upstream. At the time of data entry into the main fisheries database, individual channel widths and the island width can be referenced in the “notes” field for each associated distance point.

**Revision History Log:**

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## Standard Operating Procedure 10: Using a Clinometer Percent Slope of the Stream Channel

**This method takes two people. Normally, the data recorder also takes the percent slope readings.**

**1.)** Before starting habitat measurements, the clinometer user should find his or her eye level on the others in the group. All gradients are recorded in %, which is found on the right hand scale of the clinometer. To determine eye-level height between crew members, both individuals should stand on a level surface. The clinometer user then looks through the clinometer and notes the height (e.g. facial feature) on the opposing person that corresponds with a 0% mark on the clinometer's scale. This height should be the clinometers user's "target" or "aim point" when estimating gradients between transect. If the clinometer users aim point is over the head of a short person, **DO NOT SIGHT ON THAT PERSON**. Switch roles or sight on another in the group.

**2.)** Habitat transects are 10 meters apart, so the clinometer user stands in the center of the lower transect (must be in water, not on a rock), while the person they are sighting on stands in the center of the next transect upstream (must be in water, not on a rock).

**3.)** Look through the clinometer with one eye (keeping both open) and focus on the other persons aim point. (You should see a line in the clinometer, and this should be superimposed on your aim point.)

**4.)** While this line is imposed on the aim point, read the number on the right side (%) of the clinometer.

**5.)** If the person you are aiming at is standing in deep water, it is possible that you may get a negative number. Record this as such. All other records should be positive.

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# Standard Operating Procedure 11: Discharge

**Overview**
Stream discharge can dramatically influence the nature and composition of fish habitats at each monitoring site and can also affect sampling error. One set (cross section) of discharge measurements are collected at every site between electrofishing runs, or following the completion of all fisheries . Discharge is calculated by measuring the width, depth, and water velocity within subsections across a transect of the stream channel (see Figure 1). This SOP provides a summary of equipment and techniques to quantify stream flow at monitoring sites.

<u>Flow meters</u>
The park has two Marsh-McBirney (MMB), model 2000 “Flo-Mate™” flow meters for collecting discharge information. Each unit consists of three components including the display box, cable with electromagnetic sensor (probe) and a USGS top setting (metric) wading rod. The display box and cable/probe are permanently connected. The probe should be attached to the wading rod for operational use only. The units are shock resistant, waterproof, and generally durable. The Flo-Mate can be re-calibrated following instructions in the operator’s manual (located in the Wet Lab of Lower NCR).

<u>Selecting a transect location to measure discharge.</u>
One of the most critical factors in measuring water flow is selecting the appropriate location for the discharge cross section. The specific cross section location may be variable from year to year and is based on field conditions during the time of sampling. The ideal cross section is a single channel where flow is restricted, and water depth and substrate are uniform. Bedrock is an ideal substrate for the cross section because flow occurring below the substrate will be zero. Avoid cross sections with split channels, obvious subterranean flows, exposed boulders or other large substrates, areas with large subsurface obstructions, large areas of slack flow along banks, eddies, or sections where the center of the stream is flowing at a much faster rate than the edges. At some sites, optimal locations do not exist, especially during low flow conditions. In these cases, it may be necessary to temporarily modify the streambed or channel by removing rocks to ensure a more uniform and measurable flow. If the channel bed is modified, restore the channel to its prior condition following the discharge measurement.

<u>Establishing intervals across the discharge transect</u>
Measurements must be taken from at least ten evenly spaced intervals at the selected cross section, perpendicular to stream flow. In selecting the interval spacing, start by dividing the entire cross section into tenths. Select an interval that is easy to compound across the channel to the nearest 0.1m. Consider the example of a cross section with a total width of 3.7m. Dividing by ten would result in intervals of 0.37. Rounding down to 0.30 would ensure at least 10 measurements across the transect. Every effort will be made to select a portion of channel where at least 10 measurements of stream depth and velocity can be collected. During instances of very low flow, or in very small stream channels, this may be impossible. In those cases, collect as many measurements at 0.1 intervals as allowed by the stream channel (e.g. a 0.4 m transect would allow for 4 measurements of depth and velocity). In even rarer instances, the best location for flow measurements may be in a split channel. In this case, get at least eight measurements

from each channel and note it on the datasheet.  This will have to be dealt with separately in the database.

Using the flow staff.

The wading rod or flow staff is calibrated to utilize the 6/10ths flow measuring technique recommended by the USGS (Buchanan and Somers 1969) and the U.S. Forest Service (Platts et al. 1983).  This method results in measurements of water velocity $6/10^{ths}$ down from the water surface, which is the average water velocity in the column under ideal conditions.  An examination of the top setting rod, while reading the instructions below, will provide an understanding of the mechanics of the top setting rod.  By aligning the sliding decimeter scale (on the round rod) with the fixed centimeter scale (marked on the fixed portion at the top of the rod), the probe can be rapidly set to 6/10ths of the recorded depth at all of the sampling intervals. The fixed portion (octagonal shaped) of the rod is graduated in 0.02m increments.  Each 0.02m increment is designated by a single line, the first of which is located above the base plate or stand.  From the base plate, each 0.10m increment is designated by a double line and each 0.50m increment is designated by a triple line.  To set the probe at 6/10ths depth, read the water depth as determined from the single, double or triple lines on the octagonal shaped rod and slide the round rod up until the graduations on the round rod match the estimated depth.  For example, at a depth of 0.15m (2 ½ single lines above the first set of double lines), align the "1" on the sliding round rod between the "4" and "6" on the fixed scale and the probe will be set at 6/10ths or approximately 0.06m above the streambed.  The two scales are aligned by depressing the button at the top of the rod and sliding the small (round) rod up or down to the appropriate setting.

Collecting data.

Once the location and interval for measuring flow has been established, do the following:

1.) Stretch the 30m tape from left to right across the stream channel, perpendicular to the direction of flow.   Pull any slack out of the tape and secure it at both ends.

2.) Mark the discharge device used on the datasheet.

3.) Measure the waterline to waterline width.  Record width to the nearest 0.1m in the space provided on the "Discharge Measurements" data sheet (Appendix A)).  From the measured channel width, determine an appropriate sample interval following the criteria outlined above and fill in the distances on the data sheet.

4.) Once the sampling intervals have been determined, attach the probe to the wading rod with the thumbscrew.

5.) You will see on the data form, that at point 0, depth and flow have already been recorded as 0.  This is because point 0 corresponds with the starting stream bank.  Start actual measurements by placing the wading rod at the first interval beyond the "0" distance, and make sure the flow meter bulb is in the water facing upstream (the bulb should be perpendicular to the measuring tape).

6.) Turn the flow meter on and read/record the depth on the flow staff.  Ensure that the velocity readings are in M/S and the Period is set to 6.  **Note: It is not necessary to switch the unit**

**on and off for each measurement, just keep the probe submerged throughout the entire sequence.**

7.) Set the wading rod for the depth at that location according to the instructions above. **Always stand at least 1 ft. on the downstream side of the probe to avoid interfering with the velocity measurements.**

8.) Once the probe is in position facing upstream, allow the reading in the display box to stabilize (at least six seconds) before calling out the value to the data recorder. You should see the period counter at the bottom of the screen cycle through once. The resulting number represents flow in meters per second at that point. Repeat steps 4 through 8 until flow measurements at all of the subsequent intervals have been completed.

9.) The final interval should be recorded as depth 0 and velocity 0 as this will correspond with the "ending" stream bank width.

For a more thorough discussion of this procedure, see Buchanan and Summers, 1969 ( http://pubs.usgs.gov/twri/twri3a8/)

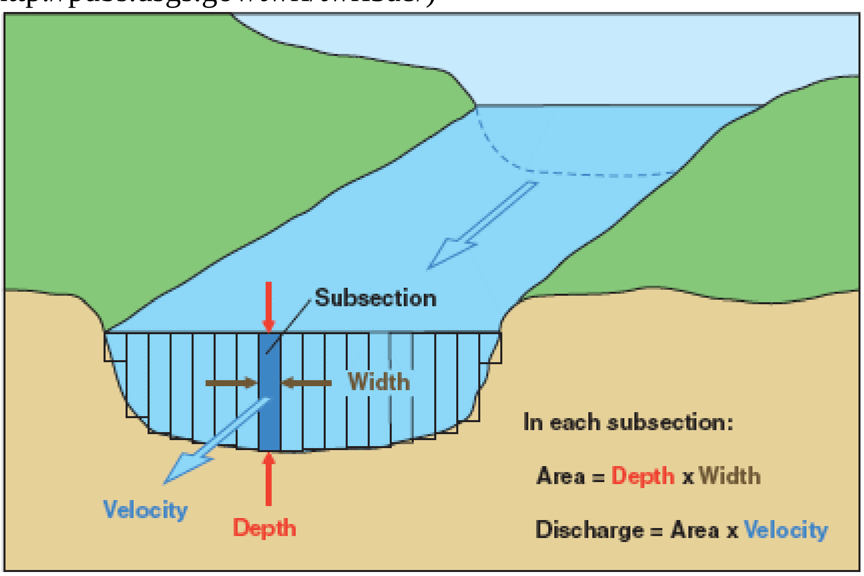


Figure 1. Schematic of discharge measurements. Discharge is calculated by measuring the width, depth, and water velocity at a number of subsections across a transect. Discharge for each subsection is then summed for a total measurement of discharge.

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# Standard Operating Procedure 12: Site Photography

**Overview**
Digital photos are inexpensive and relatively easy to capture but can take a significant amount of time to process and manage. A few steps in the field and lab can make it easier down the road. Typically, a single photograph is taken at the lower end of the site to help document the conditions present at the time of sampling. Wide angle digital photos taken of the sites should feature the bottom of the transect looking upstream capturing the water level, stream side vegetation and some reference for the location of the start of the transect (tape measure or clipboard, see Figure 1). The two best times to take this photograph are either as the Hydrolab® is stabilizing, or just before habitat measurements are initiated. The photograph is taken to capture T0 in the lower portion of the pictures frame. This SOP documents proper field and office techniques. It is up to the photographer to know and understand the owner's manual for the camera being used.

**Before going in the field:** Make sure the battery has enough charge to make it through the week and pack the camera. Also ensure that the date and time is set properly on the camera

**In the field:** Before establishing the first transect, take a picture. This can be done with habitat measurements or while the Hydrolab is stabilizing.

1) Prior to photographing a site, take a close up photograph of the datasheet showing the SiteID and date information (when renaming it will help clarify if there is some confusion).
2) Photographs should be taken by standing approximately four meters below the first transect (T0) with the camera at eye level.
3) The picture is taken looking upstream, ensuring that the full width of the bottom transect (T0) of the reach is captured at the bottom of the pictures frame. Ensure that a clipboard or tape is included at the photo where T0 occurs for scale and to mark the site start point in the photo.
4) On larger streams you will have to stand further downstream (> than four meters) to view the whole T0 transect.
5) It is unnecessary to keep a photo log unless more than one camera is used during the week.

**At the Office:** Downloading, file naming, and camera file deleting.

1) At the office, download the incoming pictures to O:\WORKING\AQUATIC\Aquatic_Images\Camera_Downloads.
2) Rename the pictures to reflect the following: Site number _month-day-year_picture number. Always start with "001" for the first picture at a site and count up if there is more than one picture per site. In the following example, two pictures were taken at 2F300 on 10/20/2010. The pictures would be renamed from the arbitrary number assigned by the camera to 2F300_10-20-2010_001 and 2F300_10-20-2010_002. **Note:** If you're confused about which site the picture belongs to, check the photo file properties or ask the Biologist or Lead Technician. Once the site pictures are renamed they should be moved to O:\WORKING\AQUATIC\Aquatic_Images\Site_Images\(year sampled).

3) If, after renaming, there are additional pictures of the crew working in the field, those should be moved to the file O:\WORKING\AQUATIC\Aquatic_Images\Working_Pictures\(year sampled). O:\WORKING\AQUATIC\Aquatic_Images\Site_Images\(year sampled) is strictly for pictures of the site…not for additional photos of crew, organisms, etc.
4) Confirm that the picture was successfully renamed, and open the file to be sure it is functional.
5) Once the pictures are downloaded to the computer, renamed, and confirmed, you may delete those pictures (BUT ONLY THOSE THAT WERE DOWNLOADED) off of the camera.

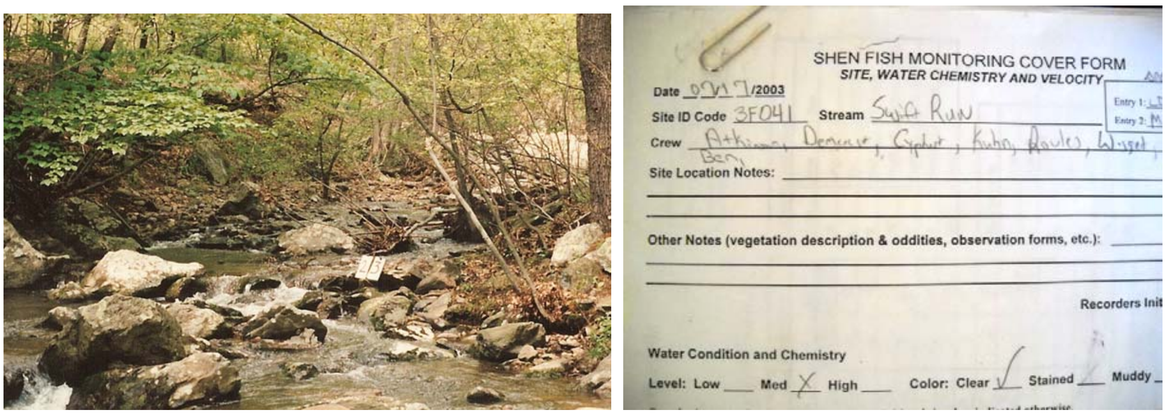


Figure 1. Example of field photograph and site form photograph.

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## Standard Operating Procedure 13: Voucher Specimens

This Standard Operating Procedure explains how to collect and preserve fisheries voucher specimens for Shenandoah National Park. In the event that a new species for Shenandoah has been collected or specimens need to be retained for lab identification, use the following procedures. (Note that these are not to be regarded as museum specimens.)

**Procedures:**

1.) In the field, keep the fish alive as long as possible or get in a plastic bag and on ice as soon as possible. Usually someone on the crew will have an ice pack in their lunch and this will suffice. Otherwise, a water bottle or bucket can be used to hold multiple individuals. We do not carry ice, bags, or bottles of formalin in the field so the crews must improvise.

2.) At the office, fix the specimen(s) in fresh formalin in a plastic bottle for 5-7 days. Specimens larger than 200mm should be slit (2-3cm) on the lower right side to allow formalin to enter the abdominal cavity, or formalin may be injected via a syringe.

3.) The specimen bottle must be labeled as formalin and a temporary label should have the collector, date, stream, and site number.

4.) Following 5-7 days in formalin, the specimen should be soaked in tap water until formalin smell is barely perceptible. With larger fishes, the tap water may need to be changed twice a day to speed dilution of formalin.

5.) Transfer the specimen(s) to 70% ethanol and a glass jar after 2-4 days in water.

6.) Once the fish is in alcohol, a more permanent label should be made with the following information: Collector, Date, Stream, Site number or UTMs, County, State, NPS unit, Topo quad name, Common name, Genus name, Species name.

7.) Add the voucher to the list in the Biologists office and in O:\WORKING\AQUATIC\Collections\SHEN Fish Collection Master List and store in the "fish" cabinets in the "lower NCR" herbarium. These are not intended to be museum specimens but are retained for proper identification and teaching as aids for future employees.

**Revision History Log:**

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# Standard Operating Procedure 14: Equipment Cleaning Procedures and Practices for Biosecurity

**Background:**
This document gives instructions for disinfecting gear that has come in contact with stream water and also provides some common sense measures to maintain clean gear in the field. This SOP was developed in direct response to increasing concern regarding the spread of *Didymosphenia geminate* (didymo) but pertains to other aquatic exotic species as well. Care must be taken to prohibit poor equipment hygiene, which can transfer exotics quickly as individuals/crews move between water bodies in the park. Additional care should be taken when gear is used outside park boundaries and then returned for use in the park. There are five known didymo populations within 100 miles of Shenandoah National Park. Three of those are in free flowing streams (West Virginia) and two are in tailwater systems (Virginia, Maryland). This SOP covers Fish Population Monitoring as well as Aquatic Macroinvertebrate Monitoring and therefore, certain gear references may be irrelevant to any one particular protocol.

**Standard guidelines are to:**

a) Use clean, disinfected gear in each stream (each site if practical)
b) Sample upstream sites prior to downstream sites when practical
c) Carry duplicate gear if needed
d) If practical don't use felt soled waders
e) Remove dirt and debris from equipment between sites
f) Dry buckets and tubs between sites
g) Clean/disinfect equipment at least daily if not more often (see below for appropriate cleaning solutions/steps)
h) Dry all equipment as often as possible
i) Packs will be hung, propped open and dried as long as possible between watersheds.
j) Buckets and tubs will be pulled apart each night and air dried. At the least, they will be disinfected in salt or soap solutions between major watersheds (James, Rappahannock, and Shenandoah River watersheds).
k) Sampling in streams can occur in up to four different watersheds in a single a day. All gear will be duplicated to the extent possible and one set will be soaked/cleaned while the crew is sampling a site with a "clean" set.
l) Ensure that when other crews borrow waders that they are responsible for cleaning

**<u>Steps to Clean Gear</u>**

**1. Choosing a staging area for cleaning gear**

Choose a fairly level area out of the way of traffic and other work groups that is close to a water source. This will ensure a safe place to work. In most cases, these areas are predetermined by the Biologist or Lead Technician.

**2. Washing containers**

a) Two to three 20 gallon “muck buckets” are used for the salt solutions and one or two 30x26” trays are used for the detergent solutions.
b) Measure out the amount of product you want for the volume of water needed and pour into the wash container. (A little more product is better than too little (see chart or directions below))
c) Measure the amount of water needed and pour into the tub or use a tub that has been marked for this purpose so water can run straight in from the hose.

**3. 5% Salt Wash (for use with non-metallic equipment)**

**Mix –** Two cups of salt per 2.5 gallons of water or one pound salt per 5 gallons of water in one to two of the large colored tubs. Stir until salt is dissolved. A second or third tub should be onsite for rinsing. Waders, buckets, and other non metallic gear will be soaked at least daily in this solution.

a) Gloves should be washed in the salt solution first. They should be filled with solution individually, and then allowed to soak in the solution for **at least one minute** before draining and rinsing.
b) The tapes, measuring trays, and other small items may be soaked with the gloves for **at least one minute.**
c) Waders and wading boots should be scrubbed to get the dirt off and soaked in salt solution (**at least one** for non felt items and **at least 30 minutes** for felt items). It is almost impossible to soak buckets and tubs, so they are rotated in the solution and allowed to sit for a couple of minutes before rinsing. The waders will have to be weighed down to stay submerged.
d) Rinse items well with tap water and then put away to dry. Do not rinse equipment over soaking tubs.
e) Rinse your hands well following exposure to the salt solution.

**4. 5% Detergent Wash (for use with metal equipment)**

**Mix -** 1 cup of Dawn or Palmolive dish detergent per gallon of water. Macroinvertebrate sampling equipment, nets, and probes will be disinfected with soap solution at least daily or air dried. *Note: Organic/green type detergents have been found ineffective at killing didymo so please use the specified products.*

a) Metal gear should be soaked in dish detergent since salt is corrosive. Electrofishing probes, nets, measuring boards, depth sticks, Surber Sampler and non-felt waders should be soaked in detergent solutions for **at least one 1 minute.**
b) It is almost impossible to soak buckets and tubs are rotated in the solution and allowed to sit for a couple of minutes before rinsing. (also see item “d” below)
c) The Portable Invertebrate Box Sampler (PIBS) and felt waders should be soaked for **at least 30 minutes**.
d) All soaked gear will be rinsed well with tap water and then put away to dry. Do not rinse equipment over soaking tubs. Buckets should be spread out upside down, not stacked.

**5. Air Drying –** To be effective, items must be air dried for 48 hours after they are dry to the touch. Buckets and tubs will be pulled apart each night and air dried. At the least, they will be disinfected in salt or soap solutions between major watersheds (James, Rappahannock, and Shenandoah River watersheds). Packs will be hung, propped open and dried as long as possible between watersheds.

6. **Freezing –** Can be used for any porous item, but space is limited and in most cases impractical.

7. **Use in the Field**
For cleaning equipment in the field 5 gallon buckets with lids can be used to store solutions. It takes about 4 gallons to adequately soak the Portable Invertebrate Box Sampler in the large black tray. Equipment should only be rinsed in stream water that the equipment is intended to be used in.

8. **Solution Degradation (i.e. how long can we use it?)**
Dish detergent will degrade with sunlight so the mixed solution should be kept no longer than a week. With care, the salt solution may be able to be used for as many as three weeks depending on how dirty the water gets. Additional water and detergent/salt will need to be added as the week progresses. Salt may harm grass over time even though the waders and other gear will be rinsed daily in the same location so chose an area with a harder surface for salt solutions while detergent may be used almost anywhere.

9. **Disinfection Options–** February 2010 (adapted from Biosecurity New Zealand/ National Institute of Water and Atmospheric Research Ltd.)

| **Treatment** | **% Solution** | **Formula** | **Duration** | **Gear to treat** |
|---|---|---|---|---|
| Dish detergent (Dawn or Palmolive) | 5% | 1 cup per gallon of water | Greater than a Minute<br>30+ Minutes for felt | All gear |
| Salt | 5% | 2 cups per 2.5 gallon water OR one pound per 5 gallons water | Greater than a Minute<br>30+ Minutes for felt | Non-metalic items |
| Drying | | | Greater than 48 Hours | All gear |
| Freezing | | | Until frozen solid | Porous gear |

**Biosecurity Issues**

- 5% Dish detergent is effective against Viral Hemorrhagic Septicemia (VHS), didymo, other aquatic plants, quagga/zebra mussels, New Zealand Mud Snails, and many other exotic aquatic species.
- 5% Salt is effective against all but VHS.
- Air drying of clothes is listed as 48 hours after material is dry to the touch.
- Freezing is listed as an alternative but space is limited and freezing time to “solid” is time consuming and most of our gear needs to be dry to use.

- 2% chlorine bleach is effective against all pests, but is extremely harsh on equipment for daily use and is toxic to aquatic systems. For these reasons, bleach is not considered to be a viable alternative.

**References:**
**http://www.biosecurity.govt.nz/pests/didymo/research#ds**

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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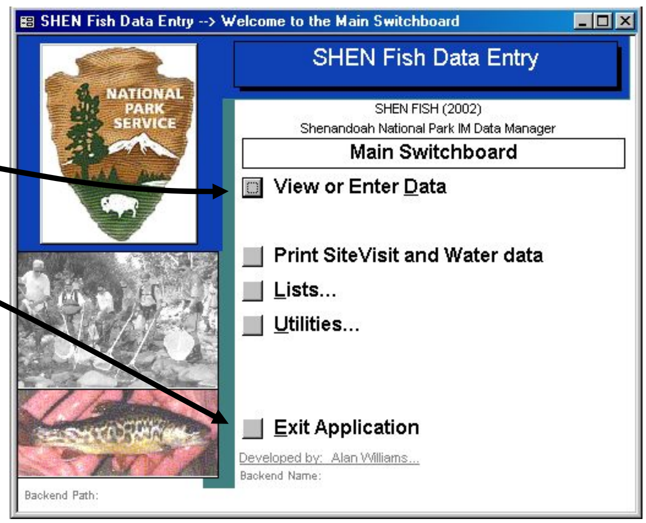
# Standard Operating Procedure 15: Data entry, editing, and verification

**Overview**
The Shenandoah National Park fish data entry program consists of three main components, the data entry application, the temporary storage databases, and the master database. Data is entered into the first temporary database using the data entry application. Then to help provide accurate data the data is entered a second time into a second temporary database. The two temporary databases are compared and any discrepancies can be reviewed and fixed. At the end of the field season the data in the temporary databases are reviewed by the program staff and then added to the master database.

Data entry –
1) **Overview:**
To start data entry press
<View or Enter Data>

To quit press
<Exit Application>

The other buttons are used by the data or program managers

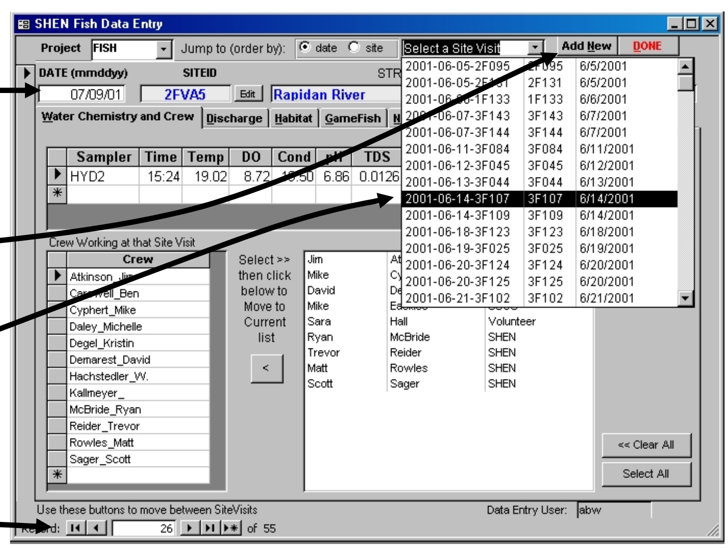



2) **The main data entry screen** has basic date and location information at the top, followed by a series of tabs with different types of data on each tab.

To enter data for a new date and site press the <Add New> button.

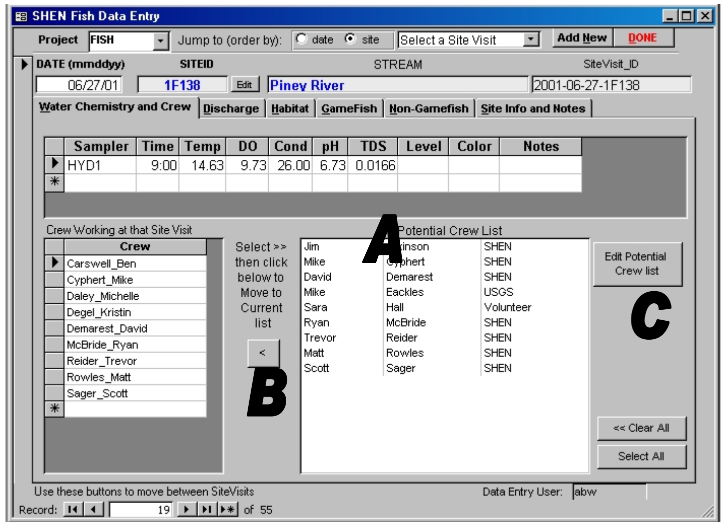
To search for data that has already been entered use the pull down list.

You can also use the navigation buttons at the bottom of the form to move between records.




Data is automatically saved as you go, row by row, so there is no need to save mannualy.

3) **Water Chemistry:** fill in the information for each of the fields from the data sheet.

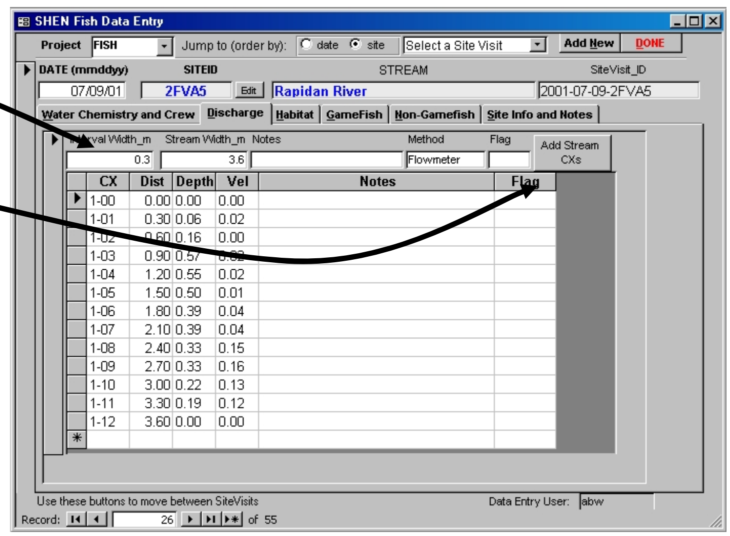



4) **Crew:** Select the members of the crew that were present from the right side (A) then press the "move" button(B). If you need to add someone to the list use the "Edit" button (C).

5) **Discharge:**
Enter the stream width and the interval that was selected.

Then press the <Add Stream…> button and the CX and Dist data should be filled in by the computer.

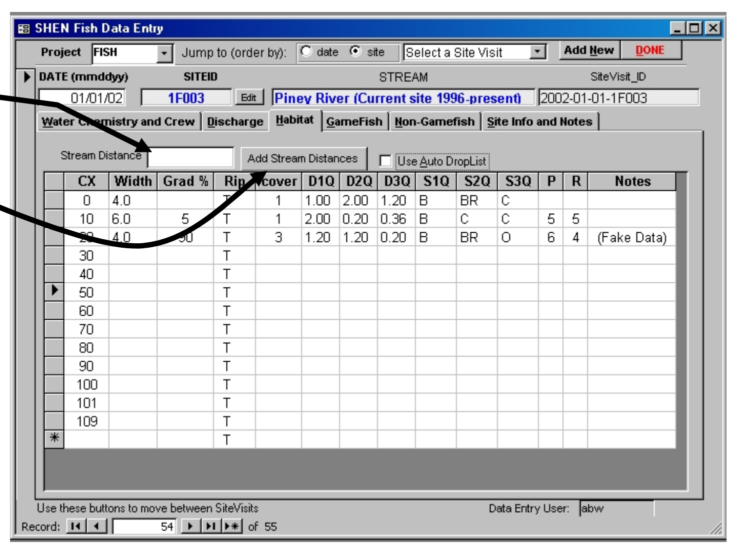
After that data is confirmed enter in the depth and velocity for each distance.




6) **Habitat:**
Enter the total length of the transect.

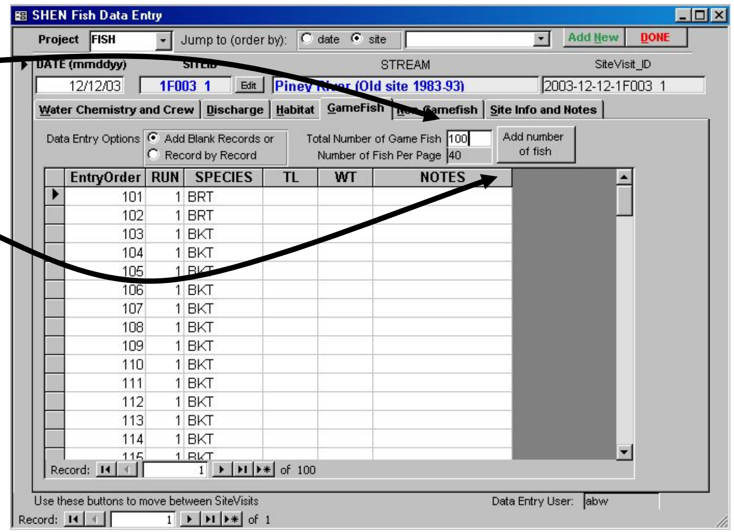
Then press the <Add Stream…> button and the CX data should be filled in by the computer.

After that data is confirmed enter the rest of data for each of the CX row of data.



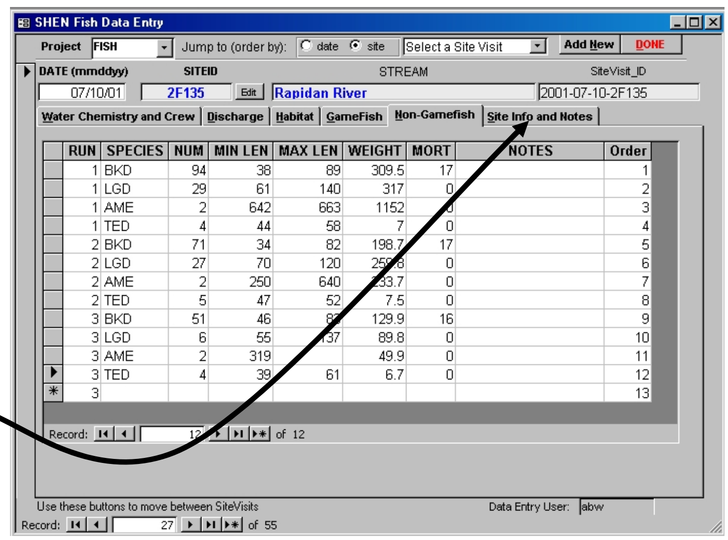
7) **Game Fish:**
Enter the total length of the transect.

Then press the <Add number…> button and the EntryOrder data should be filled in by the computer. This should be a number where the last two digits match the line number of the data sheet and the first 1 or 2 digit(s) is the page number.
The last Run and Species entered will be carried down so be sure to confirm they are correct.




6) **Non-Game Fish:**
Enter the data for each run from the data sheet. The last Run entered will be carried down so be sure to confirm it is correct for each line.

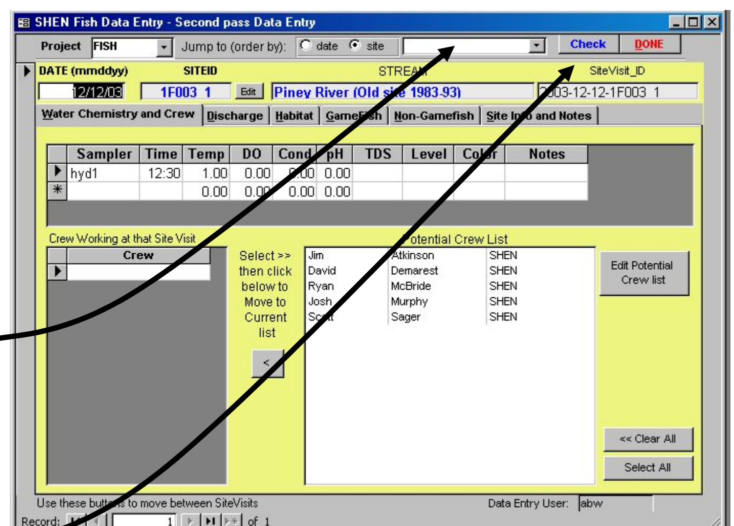
If there are generic notes about the that day at that site including, data collection, equipment problems, water level, fishing signage etc, enter it in the space provided on the last tab.




| RUN | SPECIES | NUM | MIN LEN | MAX LEN | WEIGHT | MORT | NOTES | Order |
|---|---|---|---|---|---|---|---|---|
| 1 | BKD | 94 | 38 | 89 | 309.5 | 17 | | 1 |
| 1 | LGD | 29 | 61 | 140 | 317 | 0 | | 2 |
| 1 | AME | 2 | 642 | 663 | 1152 | 0 | | 3 |
| 1 | TED | 4 | 44 | 58 | 7 | 0 | | 4 |
| 2 | BKD | 71 | 34 | 82 | 198.7 | 17 | | 5 |
| 2 | LGD | 27 | 70 | 120 | 259.8 | 0 | | 6 |
| 2 | AME | 2 | 250 | 640 | 233.7 | 0 | | 7 |
| 2 | TED | 5 | 47 | 52 | 7.5 | 0 | | 8 |
| 3 | BKD | 51 | 46 | 83 | 129.9 | 16 | | 9 |
| 3 | LGD | 6 | 55 | 137 | 89.8 | 0 | | 10 |
| 3 | AME | 2 | 319 | | 49.9 | 0 | | 11 |
| 3 | TED | 4 | 39 | 61 | 6.7 | 0 | | 12 |
| 3 | | | | | | | | 13 |

7) **Second pass data entry:**
Follow the same basic procedures for entering data during both data entry passes.

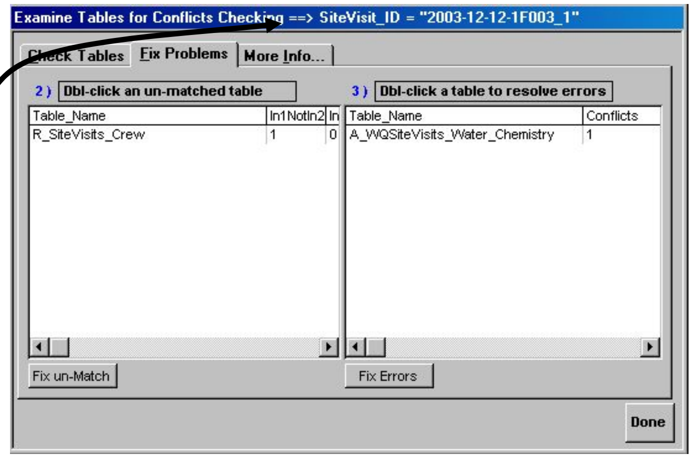
The main differences on the 2$^{nd}$ pass are:
**a)** The form will be yellow indicating a 2$^{nd}$ pass.
**b)** You will not be entering the Date or site data again, rather you will be choosing from the list of already entered dates and sites.
**c)** Once you have completed entering all of the data for a site press the <Check> button to compare it to the



first pass.

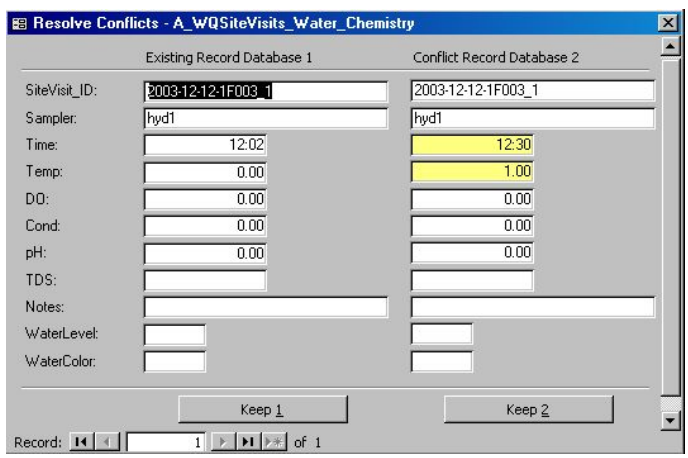
8) **Checking** the $1^{st}$ and $2^{nd}$ data entry pass**:**
After pressing the <Check> button (see above) wait a moment and this form will apear.

At the top should show the date and site it is checking.
If there are any listings in the 2) or 3) areas then each of thoses tables will need to be fixed. To fix a table select the table name and press the button below or just double click the table name.




9) **Un-Matched Records:**
Each table will have a custom form. Check the records in question against the data sheets. Either keep or delete each one.

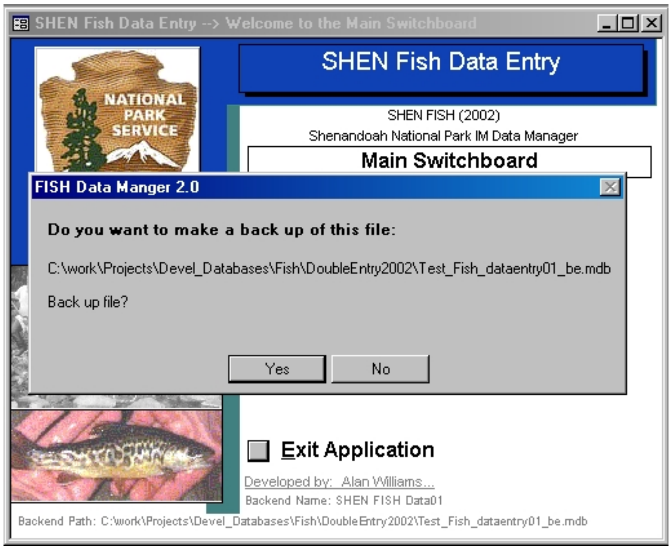

10) **Resolve Errors:**
Each table will have a custom form. Yellow fields will indicate the problem data. Check the records in question against the data sheets. Keep either the record from the $1^{st}$ or $2^{nd}$ pass.




When you have fixed all of the problems the form should close returning you to the "**Checking**" form. Fix all of the tables then press <Done>.

11) **Exiting and Backing up:**

When you are done entering data for that day you should return to the Switchboard by closing the data entry form using the <Done> button. Then press the <Exit Application> button. You will be asked if you want to make a back up of a file, press <Yes>. This saves a copy of the data from the network to your local computer.




For help contact the data manager:
Phone: 540-999-3500 x3431

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 16: Data management and archiving

This SOP gives step-by-step instructions for managing the data collected in the Fisheries Monitoring program. This SOP describes the procedure for field checking data, data entry, data verification and correction, and database organization and documentation. Accurate collection and stewardship of data are critical to the success of the fisheries monitoring program. Ensuring data quality must be a priority during crew training, and throughout the data collection, entry, verification and validation process.

### 16.1 Responsibilities for quality control

The fisheries biologist and field crew leader are responsible for ensuring that crew members:

1) can correctly identify fish species,
2) understand all field protocols,
3) understand data fields and values on paper field forms and in the database.

Correct taxonomic identification and documentation of fish species is an important aspect of monitoring. Frequent quality control assessments should be made by the fisheries biologist or lead technician, which will consist of field-checking crew member identifications and the quality of their entered data.

Crew members are responsible for learning species identification, field protocols, and all data fields and values on paper field forms and in the database. In addition, each crew member is responsible to do the following whenever they are unsure or unclear about the protocol:

1) Do not guess.
2) **<u>Ask</u>** the crew leader or other crew members while in the field.
3) Ask fisheries biologist.
4) Take copious and clear notes about the uncertainty.
5) Follow through to securing guidance and correcting the uncertainty.
6) Err on the side of collecting too much data.

### 16.2 Data collection

Field crews record data on paper field forms (see Appendix H). Carry datasheets within an enclosed clipboard. **Print legibly** all information on datasheets using pencil. There may be some crew members who will not be able to record data due to poor handwriting skills. Waterproof/all-weather copies of blank datasheets should be carried into the field at all times. All corrections to the datasheet should be made by crossing through the error. Information should never be erased and incorrect or older information should never be overwritten. Entering data in the field into hand-held data loggers and computers eliminates the need to transfer data from paper field forms into the project database, and thus may reduce data entry errors. However, field crews at SHEN have experimented with various data entry tools over the years and have always migrated back to recording data on paper.

Regardless of the data collection method, **all data values shall be called out and repeated** when one crew member is measuring and one crew member is recording. The measurer shall clearly call out the value to be recorded. The recorder shall clearly repeat the value aloud as it is recorded, to ensure that the value was heard and recorded accurately. If the measurer does not hear a response from the recorder, the measurer shall repeat calling out the measurement until a response is obtained. There should be no extraneous chat from the rest of the crew while data is being recorded. This distracts from the data taking process and will result in erroneous data.

### 16.3 Field checking data

Field checking the data is the process of reviewing all paper data sheets before leaving the site to ensure all fields are complete and no obvious errors exist. A single person is needed to complete this step. Usually this task is completed by the field crew leader, but any field member who is knowledgeable of species in the park and the monitoring protocol, and has an eye for detail can also complete this task. Datasheets should be field checked by someone other than the data recorder.

On each datasheet, the person conducting the field check should ask the following:

a. Is the header information complete – date, site number, stream name, crew?
b. Is the site name used on the datasheet match the one used on the tree tag?
c. Are multiple pages of the same datasheet numbered with site name and date filled in also?
d. Are all species codes and numbers legible?
e. Are all species codes valid codes on the park list?
f. Do all species codes make sense for the given habitat?
g. Are there any notes in the margins of the datasheets that need to be addressed, moved, or crossed out to eliminate future confusion?

Once it has been determined that the datasheet is complete, the initials of the crew member who field checked the datasheet should be added to the datasheet in the margin at the bottom right corner of each page.

### 16.4 Post-processing of field datasheets in office

After the site has been sampled, the datasheets go into the appropriate file cabinet drawer in the office. If questions or unknowns exist for the site, the datasheets should be flagged and filed appropriately. If possible, questions should be dealt with before leaving the office that day.

### 16.5 Electronic Files

The electronic files for this program can be found on the O: Drive in the working directory under “O:\WORKING\AQUATIC\”. There should be several directories with files for the program. Data management files are located in the “O:\WORKING\AQUATIC\Database\” directory (Figure 2). Electronic data files including the databases, scanned datasheets and photo monitoring images are stored in a directory structure on a file server with appropriate access levels granted to users. Prior to a data entry session an automated backup of the back-end data file is made and stored on the server to allow “roll-back” if database corruption is encountered. A scheduled back-up utility makes copies of changed files to a server in a different building at the SHEN HQ campus nightly. Annually these back-up directories are evaluated and archived or replaced by a “fresh” copy of the directory.

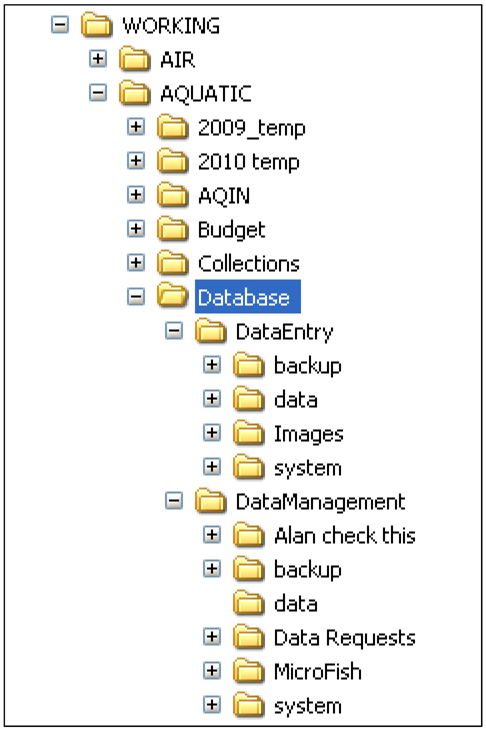


**Figure 2. O Drive directories.**

### 10.6 Database System

The Shenandoah National Park fish data system consists of three main components, the data entry application, the temporary storage databases, and the master database. Field data is entered into the first temporary database using the data entry application. Then to help assure accurate data the same data is entered a second time into a second temporary database. The two temporary databases are compared and any discrepancies can be reviewed and fixed. At the end of the field season, the data in the temporary databases are reviewed, spot checked, and then added to the master database by the program staff (see below). An ESRI shapefile is maintained to house the point locations of the sample sites. The shapefile can be joined to data exports in ArcGIS to display attributes of the sites spatially.

The current SHEN fisheries monitoring database was developed in Microsoft Access 95 but has been converted and updated to the current version of MS Access. This database predates the NPS I&M Natural Resources Database Template (NRDT) but many of the design principles are present.

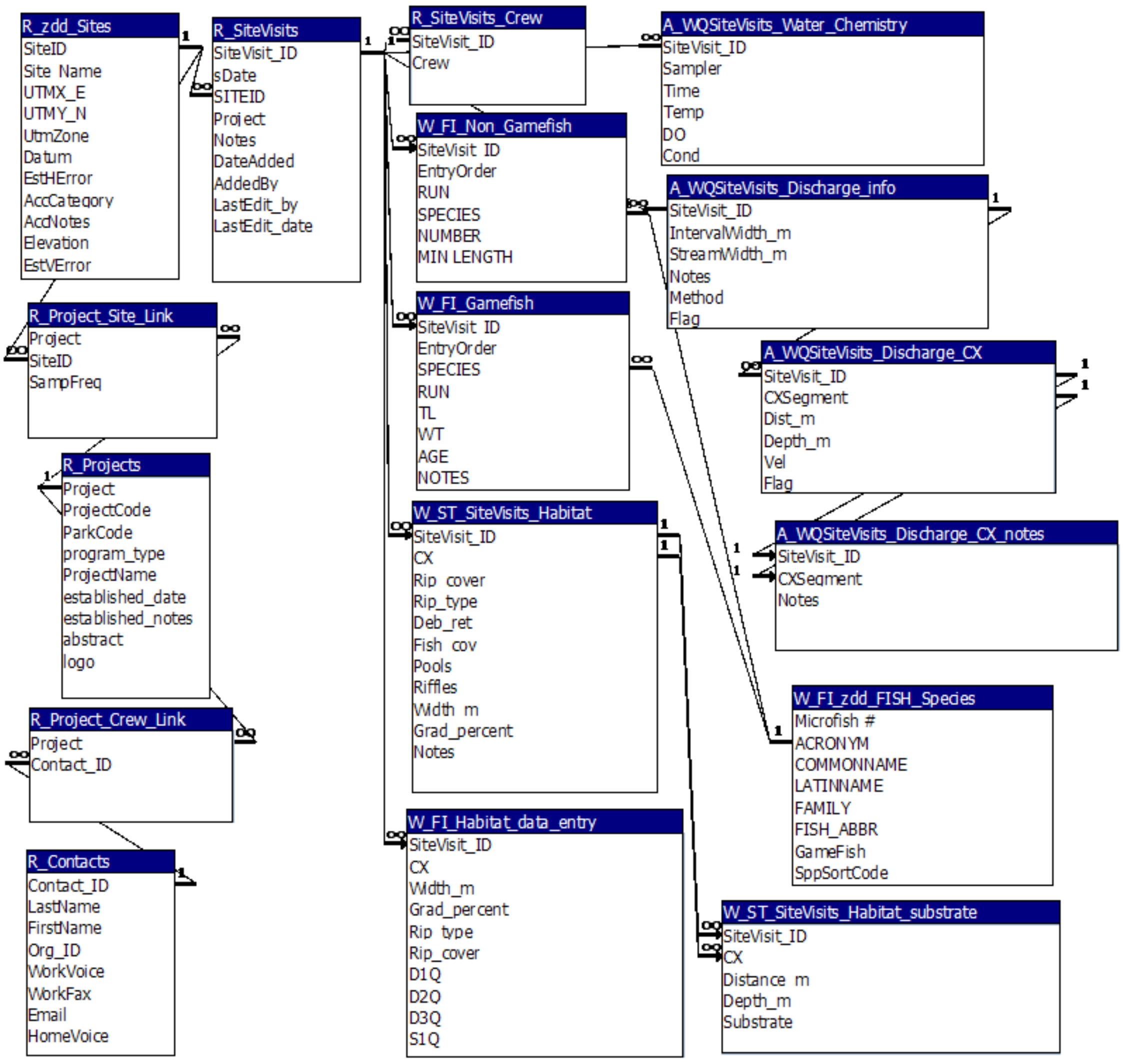



**Figure 3. Back-End table design and relationships for the fisheries database.**

## 16.7 Data Entry and Verification

Data entry is typically accomplished by teams of two people working together. One person calls out the data and the other types it in. The database can be found on the O: Drive in the working directory for the program in the Database\DataEntry\ directory. There should be several copies of the database available here so several teams of two can perform data entry at the same time.

Data is entered into the first temporary database using the data entry application. Then to help verify that the data entered matches accurately what has been written on the datasheet, data the data is entered a second time into a second temporary database by a different data entry team. The two temporary databases are compared by the application and any discrepancies are reviewed and fixed (see SOP 15 for a detailed guide to data entry).

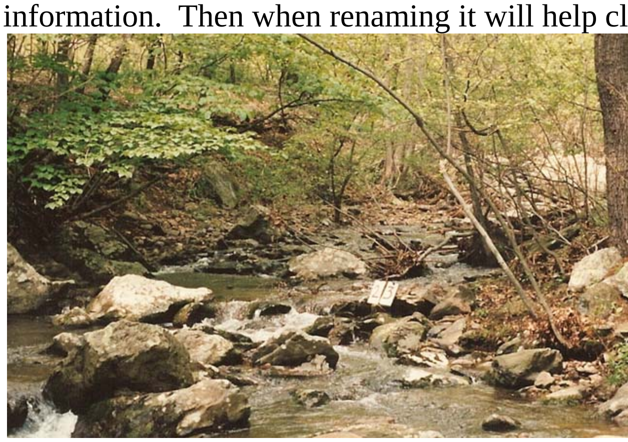
## 16.8 Scanning Field datasheets

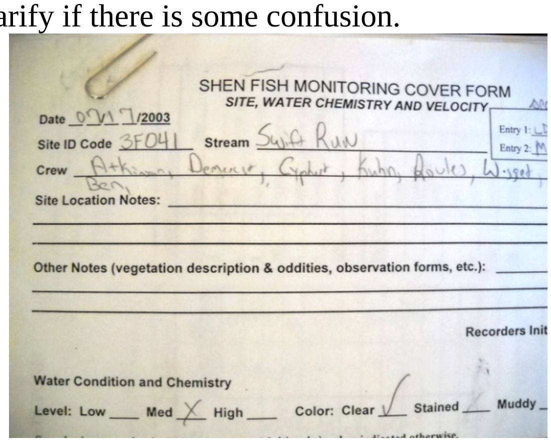
All of the field datasheets will be scanned to provide a permanent back up of the raw data. This should be done after the site data has been entered twice (and reviewed) in order to ensure all site and data entry notes are captured for future reference. Use the Panasonic DP-2330 copier/printer/scanner in the hallway of the NCR Lower building for scanning data sheets. Press the “scan/file” button on the far left. Using the touch-pad, press O_Drive New > Scanner settings: Resolution= 600, Contrast=Middle, Type=Text/Photo. Load the stack of pages to be scanned face up with the top of the documents to the left side of the scanner (or going into the document feeder head first) making sure corners are not folded and sheets are flat. Press the green button and wait for the prompt that the scan is complete then proceed to the next set of documents to scan.

After documents have been scanned, log into a computer and go to “O:\WORKING\OTHER\ScannerJobs\” . Search for new scanned documents with the scanner generated names like, “080023142ec9-100908112916-0000.pdf”. Open these, confirm the site and date information for the file, close the file then rename. Rename each of the files using the format, “SiteID_YYYY-MM-DD_Date Sampled.pdf” (example, 1F114_2008-07-02.pdf). Double check that all sites that were scanned were saved and re-named.Then move the pdf files into the O:\WORKING\AQUATIC \Database\Scanned_Datasheets\ <SiteID> folder. After all this is completed, file the site folders after the “Scanning Complete – To be Filed” section of the fish data file drawer ( edit note: is this section complete?).

**16.9 Digital Photos**

Digital photos are inexpensive and relatively easy to capture but can take a significant amount of time to process and manage. A few steps in the field can make it easier down the road. First make sure the date time is set properly on the camera prior to going into the field. Then either include a “slate” with date and location information in the photo or just prior to taking a site photo take a photo of a “slate” or a close up of the datasheet showing the SiteID and date information. Then when renaming it will help clarify if there is some confusion.





SHEN FISH MONITORING COVER FORM
SITE, WATER CHEMISTRY AND VELOCITY

Date __/__/2003
Site ID Code 3F041 Stream
Crew
Site Location Notes:
Other Notes (vegetation description & oddities, observation forms, etc.):
Recorders Init
Water Condition and Chemistry
Level: Low __ Med X High __ Color: Clear √ Stained __ Muddy __

Wide angle digital photos taken of the sites should feature the bottom of the transect looking upstream capturing the water level, stream side vegetation and some reference for the location of the start of the transect (tape measure).

Daily or at least weekly, digital cameras with site photos should be downloaded and organized by date and camera into temporary directories to facilitate renaming and filing in permanent locations (example, O:\WORKING\AQUATIC \Aquatic_Images\Camera_Downloads\<date-camera>). Later, when time allows, photos should be renamed to directories such as O:\WORKING\AQUATIC \Aquatic_Images\Site_Images\<SiteNumber>\ <SiteID> using the filename format, “SiteID_YYYY-MM-DD_Date Sampled.jpg” (example,O:\WORKING\AQUATIC \Aquatic_Images\ 2F072\2F072_2010-06-30_A.jpg

**16.10 End of Season - Database Tasks and Data Quality Control (QC)**
Following data entry, 100% verification, scanning of field datasheets, and renaming of site photos for the current season a QC data review will be conducted to identify additional potential mistakes. This QC step should include, running a set of predefined QC summaries to catch common errors, omissions, and problems. The summaries will include:

- List of sites entered in the database checked against the crew leader’s notes and the schedule for the year.
- List of renamed digital photos checked against photo logs or visited site list
- List of scanned datasheets checked against folders of entered data and database
- Identify out-of-range values
- Search for missing values
- Evaluate outliers

After identified issues are addressed or corrected then the data records can be Labeled “Certified” the records can be locked buy the program lead. Future edits to the data will need to “un-lock” the record and describe why in the notes then relock the record after edits are made. Then data in the temporary / seasonal databases are added to the master database using pre-programmed macros Figure 4. The master database is used for all data summaries and analysis.

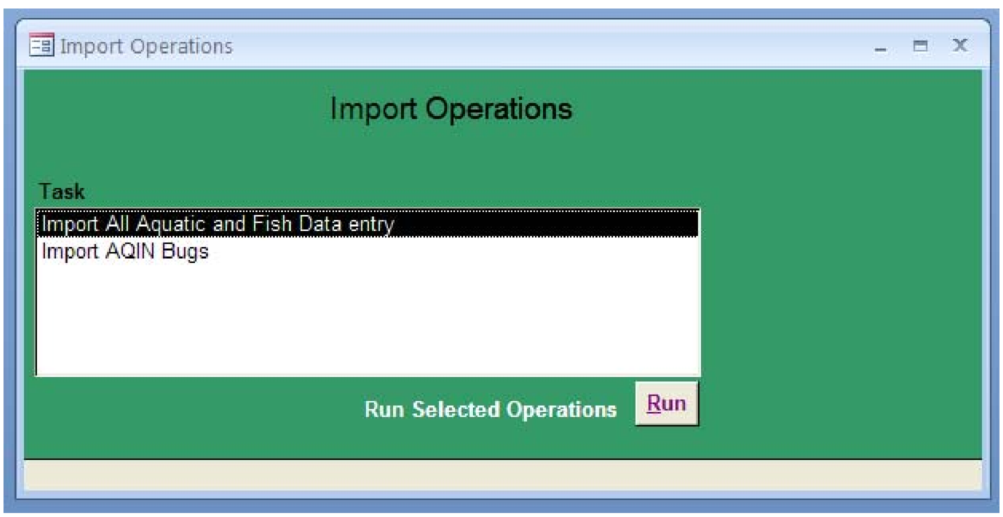



**Figure 4. Import data operations form.**

**16.11 Metadata Procedures**
Any dataset compiled by SHEN must be accompanied by metadata. This includes both spatial and non-spatial datasets. For metadata associated with geospatial data, we abide by Executive Order 12906, which mandates that every federal agency document all new geospatial data it collects or produces using the Federal Geographic Data Committee (FGDC) Content Standard for Digital Geospatial Metadata (CSDGM; www.fgdc.gov/metadata/contstan.html). All GIS data layers will be documented with applicable FGDC and NPS metadata standards. SHEN will also

generate FGDC-style metadata for non-spatial datasets that meet this standard, without the geospatial-specific elements.

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 17: Data analysis

Adapted from Peterson et al (2008) and Dodd et al (2008).

Conclusions of ecological studies based on fish and other biological, chemical, and physical data are used by resource managers to better comprehend underlying system processes and develop environmental and management policies that best serve the resource.

Annual data summaries will be produced during every year that monitoring is conducted. These summaries will provide visual displays of select metrics, largely in the form of control charts (see below). Annual summaries will not include formal analyses of trend or in-depth data reviews unless control chart thresholds are crossed. In-depth data analyses will occur every ten years or when thresholds indicate further data investigations are prudent. Ten year analyses will include formal assessment of trend and more rigorous statistical review.

This primary audience for most data analyses (and reports – see SOP 18) will consist of park resource managers, superintendents, and other staff who may not have an in depth background in statistical methods or may have limited time for evaluation of these analyses. Thus, to the extent possible, it is important that core data analysis and presentation methods are relatively straightforward to interpret, provide a standard format for evaluation of numerous variables, can be quickly updated whenever additional data become available, and work for many different types of indicators, whether univariate or multivariate. Control charts can fill this need. Additionally, the type and magnitude of variability or uncertainty associated with the results should be somewhat intuitive, and it may be necessary to indicate a threshold for potential management action. More detailed analysis of the data (comprehensive trend reviews) generally will benefit from the use of multiple data analysis methods.

Primary approaches to analyzing fish data will rely on metric estimation. Potential metrics and estimated variables for analysis of stream fish communities include:

**1. Species richness (number of species).** Species richness will be calculated for the sample site.

**2. Simpson's Diversity Index (D).** Simpson's Diversity Index will be calculated for the sample reach. Simpson's Diversity Index is preferable to the Shannon diversity index and will be used for data analysis because this index is independent of sample size. Simpson's Diversity Index is calculated with the formula shown below.

$$D = \Sigma ((n^2 - n) / (N^2 - N))$$

*n = total number of individuals of a fish species, and N = total number of fish of all species.*

**3. Catch per area (#individuals/100m2).** Catch per area (Ci) will be calculated for each captured species at the reach scale. For game fish, catch per area will also be calculated for each identifiable age class (generally age-0 and age 1+, as identified by length-frequency analysis).

Based on 3-pass depletion, catch will be estimated using maximum likelihood methods as described in Van Deventer (1985).

*Ci / A*

*Ci = catch of individuals of species i (either as a whole or by age)*

*A = area of sample reach*

**4. Biomass.** Biomass per area (BPAi; kg/ha) will be calculated for each captured species at the reach scale. For game fish, biomass per area will also be calculated for differing age classes (generally age-0 and age 1+, as identified by length-frequency analysis). To calculate biomass for game fish, simply sum the biomass of all weighed individuals by species and divide by the sample reach area. Calculate biomass per area for non-game species (Bi) via the following:

*BPAi = WiNi / A*

*Wi = average weight of fish species i(either as a whole or by age)*

*Ni = number of individuals of species I (either as a whole or by age)*

*A = area of the sample reach*

**5. Size Structure.** Size structures of fish populations within the community can be indicative of a disturbance or resource problem. A community with primarily larger fish indicates that there is little recruitment to keep the community self-sustaining. A community with primarily smaller fish could indicate inadequate resources (*e.g.*, food resources) or environmental stresses (e.g., acidification) that limit growth. Average length and weight (and ranges) for each gamefish species at the reach will be calculated. Averages will also be calculated for differing age classes (generally age-0 and age 1+, as identified by length-frequency analysis). For non-game fish, only average weight will be calculated (length of individual non-game fish is not collected).

**6. Mean Relative Weight.** Mean relative weight for each gamefish species in the reach. Mean relative weight (Wege and Andersen, 1978) will be calculated for each gamefish species using established length-specific weight predicted from a published weight-length regression for the species (Blackwell et al. 2000; Hyatt and Hubert, 2001).

**7. Percent Composition**. Percent composition by biomass will be calculated for each species in the reach. To calculate this:

*First, calculate individual biomass for each species (Bi):*

*Bi = WiNi*
*Wi = average weight of fish species i*

*$Ni$ = number of individuals of species i*

*Second, calculate biomass per area for each species (BPAi) and total biomass per area (BPAt):*

$BPAi = Bi / A$

$BPAt = \Sigma BPAi$

*$A$ = area of the sample reach*

*Lastly, calculate percent composition for each species (Ci)*

$Ci = BPAi / BPAt * 100$

*EDIT NOTE: Consider providing example of each metric*

**Habitat and Water Quality Parameters**

Physical and chemical habitat measurements will be estimated using summary statistics such as means, medians, standard errors and/or confidence intervals. In the simplest presentation of data, each parameter should be estimated in each year that data are available, and confidence intervals or standard errors calculated, where appropriate. For parameters where percentage cover class categories are used (such as canopy cover) the median value of the cover class will be used in calculating means and standard errors. For those parameters where presence/absence data are collected, a percentage will be calculated for the reach.

**Annual Data Summaries – Control Charts**

Using several of these estimated variables listed above, control charts can be employed to visualize trends and changes in fish communities (Morrison, 2008). The construction and interpretation of control charts is covered in many texts focusing on quality control in industry (Beauregard and others, 1992; Gyrna, 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 the texts by McBean and Rovers (1998) and Manly (2001), although not as detailed 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 (for example, count data, richness, or biomass), to focus on measures of central tendency or dispersion, and in univariate or multivariate analyses.

Control charts allow for the visual display of trends and thresholds for management action or further data analyses. Initially, the first ten years of data will be serve as a baseline (see Morrison, 2008) and thresholds will be defined as plus or minus two standard deviations around the ten year mean (i.e. baseline) of the statistic (see figure 1 for example). Depending on the expected fish response to perturbation/stress, thresholds may occur above, below, or both above and below baseline values. During years where thresholds are crossed, more targeted data analyses may be conducted to attempt to assess the driver behind the threshold transgression and to help determine the potential need for management action. Baselines and thresholds may be altered in the future to potentially better reflect meaningful management or biological thresholds that might not be captured in the mean plus or minus two standard deviations.

Although some of the above-mentioned texts discuss the use of multivariate control charts (using the Hotelling T2 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 recently has been described for use with complex ecological communities (Anderson and Thompson, 2004). 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, and it is important to test for temporal autocorrelation using Mantel correlograms prior to using this method.

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.

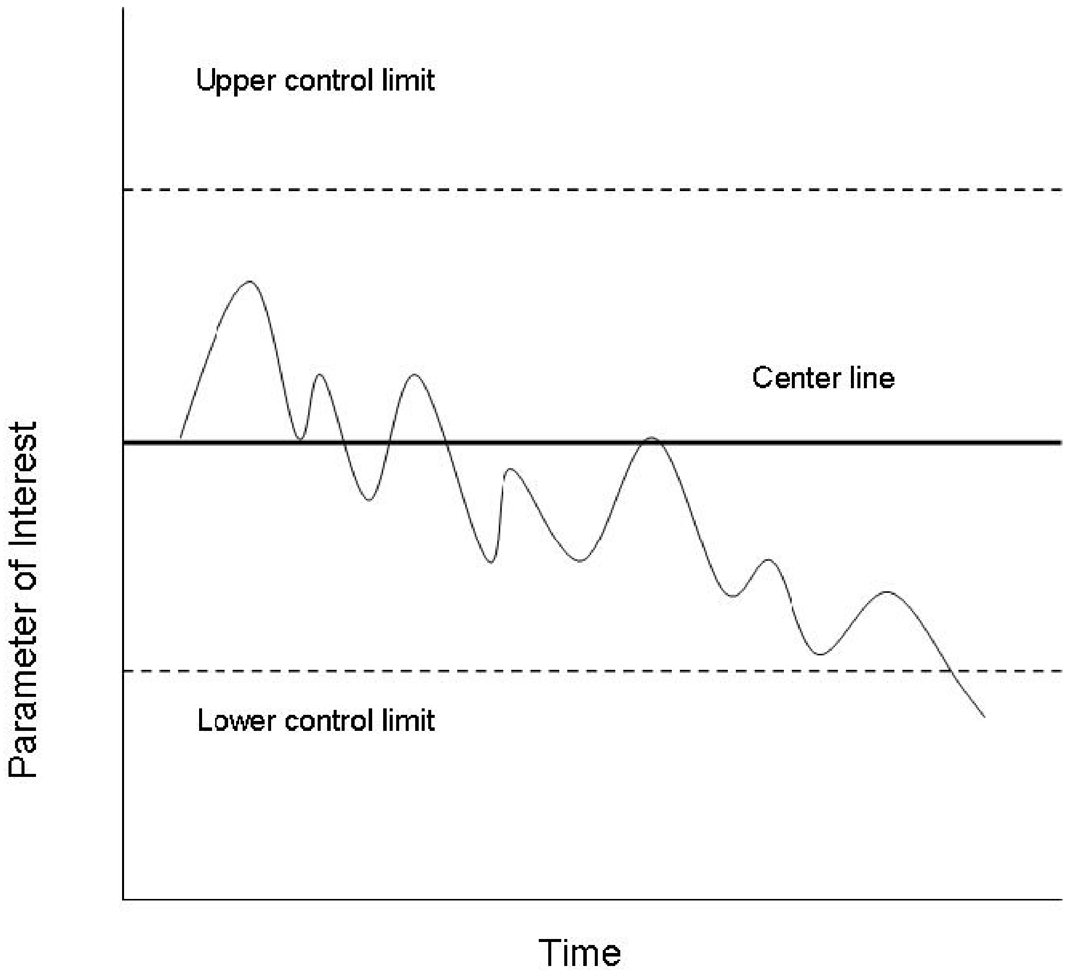


Figure 1. A univariate control chart.

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

1. Determine the parameter of interest. This may be any of the metrics presented above.

2. After several data points are available, plot the values of the parameter of interest (on the y axis) against time (on the x-axis).

3. Determined 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 data points are available, and updated regularly. Additional control charts can be constructed from other variables of interest as described above.

**Comprehensive analyses**

Comprehensive analyses will provide a more formal analysis of trend and will occur every 10 years. Rather than requiring a detailed list of specific data analyses, the data analysis process should be flexible enough to allow the use of newly developed statistical and analytical techniques and tailoring of analyses for a variety of audiences with a variety of questions about the aquatic resources at Shenandoah.

In general, comprehensive analyses will include multivariate and univariate assessments of community and environmental data. Univariate trend tests such as a seasonal Kendall test will be used to assess trends in univariate measures. Depending on the metric of interest, additional trend tests will be used with appropriate transformations. Multivariate analysis is another frequently used analysis technique and involves methods used to explain variability in community data and to identify the environmental variables that best explain, and have an assumed responsibility for, the variability measured (Gauch, 1982; Jongman and others, 1995; Everitt and Dunn, 2001; Timm, 2002). Multivariate techniques elicit a hypothesis from the biological data rather than disproving a null hypothesis. Two commonly used multivariate techniques include: ordination (such as principal components analysis, canonical correspondence analysis, and detrended correspondence analysis) and classification (such as two-way indicator species analysis). Detailed discussion of these methods can be found in several texts (Gauch, 1982; Jongman and others, 1995; Everitt and Dunn, 2001; Timm, 2002).

References:

Edit note: need to add citations

**Van Deventer, J.S. and W.S. Platts. 1985.** A computer software system for entering, managing, and analyzing fish capture data from streams. Research Note INT-352, Intermountain For. & Range Research Station. U.S.D.A. Forest Service. Ogden, UT.

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 18: Reporting

Adapted from Dodd et al (2008).
This SOP gives instructions for reporting on macroinvertebrate community data and associated stream habitat and water quality collected at Shenandoah National Park (SHEN). The SOP 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.

**Report Format**
The report template for national and regional natural resource technical reports will be followed. The templates for natural resource reports can be found at: (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 mission. Standards for scientific writing as recommended in the CBE Style Manual (1994) should be followed. Reports should be direct and concise. For style, refer to NPS guidance (http://www.nature.nps.gov/ParkScience/archive/PDF_Downloads/Editorial%20Style%20Guide%20Revised%208-4-2008/Editorial_Style_Guide_Revised_8-4-2008.pdf).

**Types of Reports and Review Procedure**
A variety of reports may be produced at different times during a monitoring program, such as following a monitoring year, or following acquisition of new information. In general, two types of reports will be completed for the aquatic macroinvertebrate monitoring program – annual reports using the Natural Resource Data Series (NRDS) format, and comprehensive reports every five to ten years using the Natural Resource Technical Reports (NRTR) (see Table 1 for details)

Table 1. Summary of types of reports produced and review process.

| Type of Report | Purpose of Report | Primary Audience | Review Process | Frequency |
|---|---|---|---|---|

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|---|---|---|---|---|
| Annual Status Reports for Specific Protocols | Summarize monitoring data collected during the year and provide an update on the status of selected natural resources (see data analysis for metrics).<br>Document related data management activities and data summaries. | Park resource managers and external scientists | Internal peer review by SHEN staff | Every year monitoring is conducted. |
| 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 SHEN 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, *etc*.<br>Highlight resources of concern that may require management action. | Park resource managers and external scientists | Internal peer review by SHEN staff | Every 5-10 years |
| 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 SHEN staff | Simultaneous with Comprehensive Trends Analysis and Synthesis Reports |

**Distribution Procedure**

Annual reports will be provided to the interested park staff and the Superintendent's Office. Reports will be made available to all interested parties upon request, posted on the MIDN website (http://science.nature.nps.gov/im/units/midn/index.cfm), and also posted to the national Natural Resource Technical Reports webite (http://www.nature.nps.gov/publications/nrpm/nrtr.cfm). Park staff will ensure that copies are included in Shenandoah's central files and in the Natural and Cultural Resources Library. Data collected by Shenandoah National Park is public property and subject to requests under the Freedom of Information Act (FOIA).

**References**

Dodd, H.R., D.G Peitz, G.A. Rowell, D.E. Bowles, and L.M. Morrison. 2008. Protocol for Monitoring Fish Communities in Small Streams in the Heartland Inventory and Monitoring Network. Natural Resource Report NPS/HTLN/NRR - 2008/052. National Park Service, Fort Collins, Colorado.

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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# Standard Operating Procedure 19: Revising the Protocol

This Standard Operating Procedure explains how to make changes to the Fisheries Monitoring Protocol Narrative for Shenandoah National Park, Virginia and accompanying SOPs, and tracking these changes. Observers asked to edit the Protocol Narrative, or any one of the SOPs need to follow this outlined procedure in order to eliminate confusion in how data is collected and analyzed. All observers should be familiar with this SOP in order to identify and use the most current methodologies.

**Procedures:**

1. The Fisheries Monitoring Protocol Narrative for Shenandoah National Park, Virginia and the accompanying SOPs have attempted to document the current methodologies for collecting and analyzing fisheries data. However, all protocols will ultimately require editing as new and different information or methods become 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 Mid Atlantic Network staff. However, if a complete change in methods is sought, an outside review is required. Experts in fisheries research and statistical methodologies will be utilized 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 only. 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 …). 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 on the inter-net and forward copies to all individuals with a previous version of the effected Protocol Narrative or SOP.

**Revision History Log:**

| Prev. Version # | Revision Date | Author | Changes Made | Reason for Change | New Version # |
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**National Park Service**
**U.S. Department of the Interior**

**Natural Resource Program Center**
1201 Oakridge Drive, Suite 150
Fort Collins, CO 80525

www.nature.nps.gov