

Field Operations Manual for Assessing the Hydrologic Permanence and Ecological Condition of Headwater Streams



Field Operations Manual for Assessing the Hydrologic Permanence and Ecological Condition of Headwater Streams

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PREFACE

The mission of the Ecological Exposure Research Division (EERD), National Exposure Research Laboratory (NERL), United States Environmental Protection Agency (USEPA) is to improve the scientific basis for understanding, measuring, and protecting biological integrity so that USEPA and other resource agencies can make sound, defensible environmental decisions. Our research is primarily focused on the development, evaluation, and implementation of new methods to assess ecosystem condition, to evaluate biotic responses to environmental stressors, and to predict future vulnerability of natural populations, communities and ecosystems.

This document originated from a research project, the Headwater Intermittent Streams Study (HISS), funded through the USEPA's Regional Methods (RM) Program (overseen by the Biological Advisory Committee and supported by the USEPA, Office of Science and Policy). The purpose of RM is to support development of methods needed by EPA regions, states and tribes to meet their monitoring and enforcement objectives. The widespread need for standardized methods for assessing headwater streams is apparent from the sponsorship and participation by USEPA Regional offices (1, 2, 3, 4, 5, 8, 9 and 10) and several state offices therein. The initial development of the methods was in forested headwater streams located in Indiana, Kentucky, and Ohio over 2003 and 2004. Following training workshops, state and regional teams used the methods to collect data from forested headwater streams in Illinois, New Hampshire, New York, Vermont, West Virginia, and Washington. This manual is a product of the working collaboration among EERD, regional, and state scientists. We hope that the methods described in this manual will be useful to individuals and organizations interested in monitoring and protecting headwater streams.

Florence Fulk
Acting Director
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ACRONYMS AND ABBREVIATIONS

°C	Degrees Centigrade
µm	Micrometer
µS/cm	Micro-Siemens per Centimeter
ACI	Algal Cover Index
AFDM	Ash-Free Dry Mass
BACI	Before/After and Control/Impact
BF	Bankfull
cm	Centimeter
cm ²	Square Centimeters
Cond	Conductivity
DEM	Digital Elevation Model
DI	Downstream Intermittent Site
DO	Dissolved Oxygen
DQO	Data Quality Objectives
E	Ephemeral Site
EERD	Ecosystem Exposure Research Division
EMAP	Environmental Monitoring and Assessment Program
FCSPD	Fairfax County Stormwater Planning Division
FPA	Flood Prone Area
GPS	Global Positioning System
HISS	Headwater Intermittent Streams Study
IEI	Intermountain Environmental, Inc
in	Inch
km	Kilometer
km ²	Square Kilometers
LIDAR	Light Detection and Ranging
m	Meter
m ²	Square Meters
m ³ s ⁻¹	Cubic Meters per Second
m/s	Meters per Second
mi	Miles
mi ²	Square Miles
mg/l	Milligrams per Liter
ml	Milliliter
mm	Millimeter
NaCl	Sodium Chloride
NAWQA	National Water Quality Assessment
NCDNR	North Carolina Department of Natural Resources
NCDWQ	North Carolina Division of Water Quality
NHEERL	National Health and Environmental Effects Laboratory
NRCS	National Resources Conservation Service

NRMRL	National Risk Management Research Laboratory
NYSDEC	New York State Department of Environmental Conservation
Ohio EPA	Ohio Environmental Protection Agency
OW	Office of Water
OWOW	Office of Wetlands, Oceans and Watersheds
P	Perennial Site
PC	Personal Computer
PDA	Personal Digital Assistant
PVC	Polyvinyl chloride
RHAF	Rapid Habitat Assessment Form
RM	Regional Methods
SE	Standard Error
SVL	Snout-Vent Length
Temp	Temperature
TNC	The Nature Conservancy
UI	Upstream Intermittent Site
USDA	United States Department of Agriculture
USEPA	United States Environmental Protection Agency
USFS	United States Forest Service
USGS	United States Geological Survey
UTM	Universal Transverse Mercator
VTDEC	Vermont Department of Environmental Conservation

1 INTRODUCTION

1.1 Purpose

The purposes of this manual are to: 1) document procedures that were developed and used by the United States Environmental Protection Agency's (USEPA) Ecosystem Exposure Research Division (EERD) for the assessment of the physical and biological characteristics of headwater streams; and 2) provide a catalog of procedures to other groups with an interest in headwater stream assessment. Earlier EPA field operations manuals for running waters have focused on larger systems, including wadeable streams, non-wadeable rivers, and Great Rivers (e.g., Barbour et al. 1999, Lazorchak et al. 1998, 2000, Angradi 2006). There is a growing interest in headwater streams because human activities (e.g., road building, stormwater management) frequently intersect these widespread waterbodies. There is also considerable legal debate regarding extent of jurisdictional waters under the Clean Water Act and the role or nexus of various types of headwater streams to the integrity of downstream interstate waters (Nadeau and Rains *in press*). Some states, like North Carolina and Ohio, have already begun to initiate headwater stream classification methods for regulatory purposes (Ohio Environmental Protection Agency 2002, N.C. Division of Water Quality 2005).

This document provides methods specifically designed for assessing the hydrologic permanence and ecological condition of headwater streams. A universal, spatially-explicit definition of a headwater stream is lacking because stream size and drainage area varies with surrounding topography and geographic location. Regardless, headwater streams are important because they are the

origins of the stream network and have unique ecological characteristics that separate them from larger, downstream waterbodies.

What are headwater streams?

Stream order is a measure of stream position within a drainage network system (Horton 1932, Strahler 1945, Shreve 1966). Headwater streams are typically considered to be first- and second-order streams (Gomi et al. 2002, Meyer and Wallace 2001), meaning streams that have no upstream tributaries (i.e., "branches") and those that have only first-order tributaries, respectively. Use of stream order to define headwater streams is problematic because stream-order designations vary depending upon the accuracy and resolution of the stream delineation (Mark 1983, Hanson 2001). Lack of agreement among maps with different mapping resolution is common when identifying headwater stream, determining stream order, and determining total stream length (Figures 1-1 and 1-2). The designation of the mainstem (central tributary) origin is typically similar between the 1:100 000 and 1:24 000 scale maps. However, the 1:24 000 maps delineate more lateral tributaries (Figures 1-1A and 1-1B) and this can result in substantial differences of headwater extent. The total stream length within the Coweeta Creek watershed (16.3 km²) in western North Carolina on a 1:500 000 scale map was only 3% of the length shown on a 1:24 000 scale map (Meyer and Wallace 2001). The smallest headwater streams are not designated as channels on topographic maps and may be difficult to discern in aerial photographs. Thus, stream-order designations based on maps are typically underestimated (Hughes

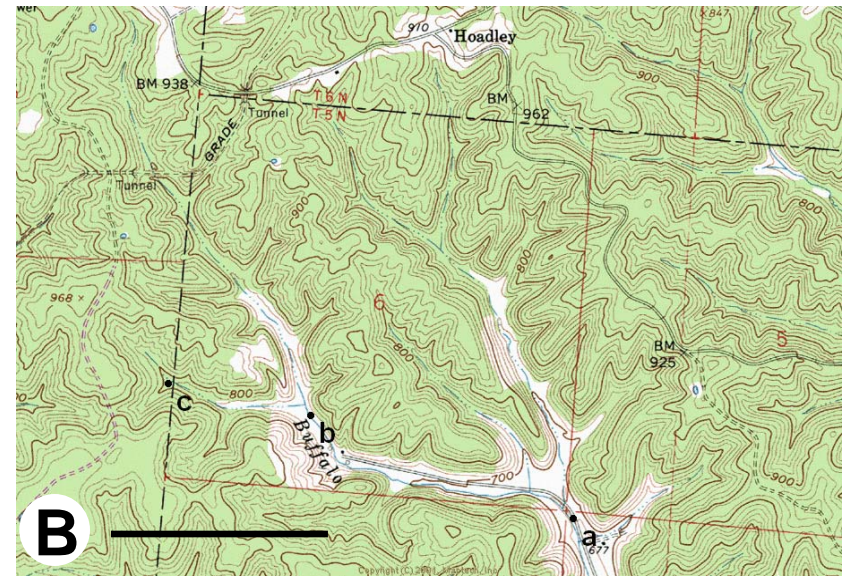
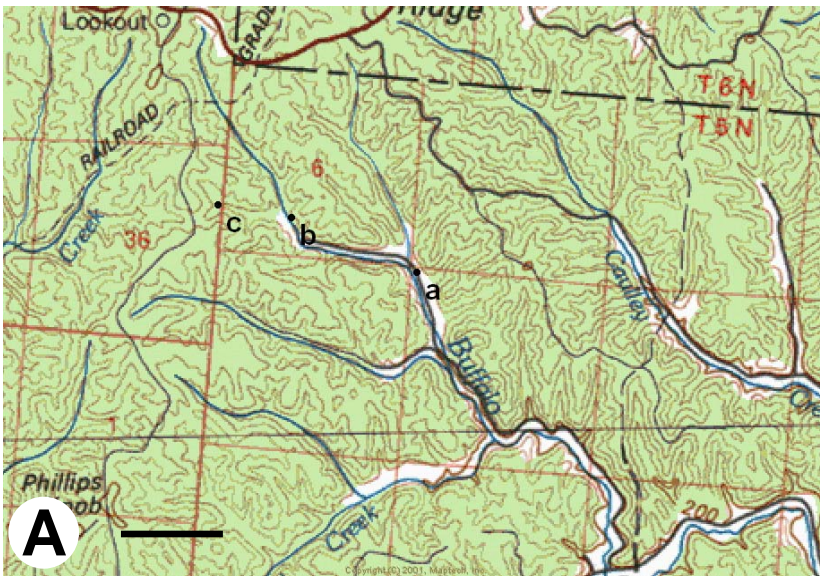


Figure 1-1 Portions of a 1:100 000 (A; Ironton 30 x 60 minute quadrangle) and a 1:24 000 (B; Gallia 7.5 minute quadrangle) scale United States Geological Survey (USGS) topographic maps illustrating the upper reaches of Buffalo Creek in Wayne National Forest (Lawrence and Gallia Counties, OH). Black circles and associated letters mark corresponding points on both maps. Black horizontal bars represent 1 km. Buffalo Creek at “a” is a second-order stream on the 1:100 000 map, but is a third-order stream on the 1:24 000 map. Likewise, Buffalo Creek at “b” is considered a first-order stream on the 1:100 000 map, but is a second-order stream on the 1:24 000 map. The point marked “c” is shown as a first-order stream on the 1:24 000 map, but is not designated as a stream on the 1:100 000 map. The number of first-order streams shown upstream of “a” on the 1:100 000 map is two, whereas the 1:24 000 map has five. Field surveys of this drainage would likely find $\geq 10X$ first-order streams upstream of “a”.

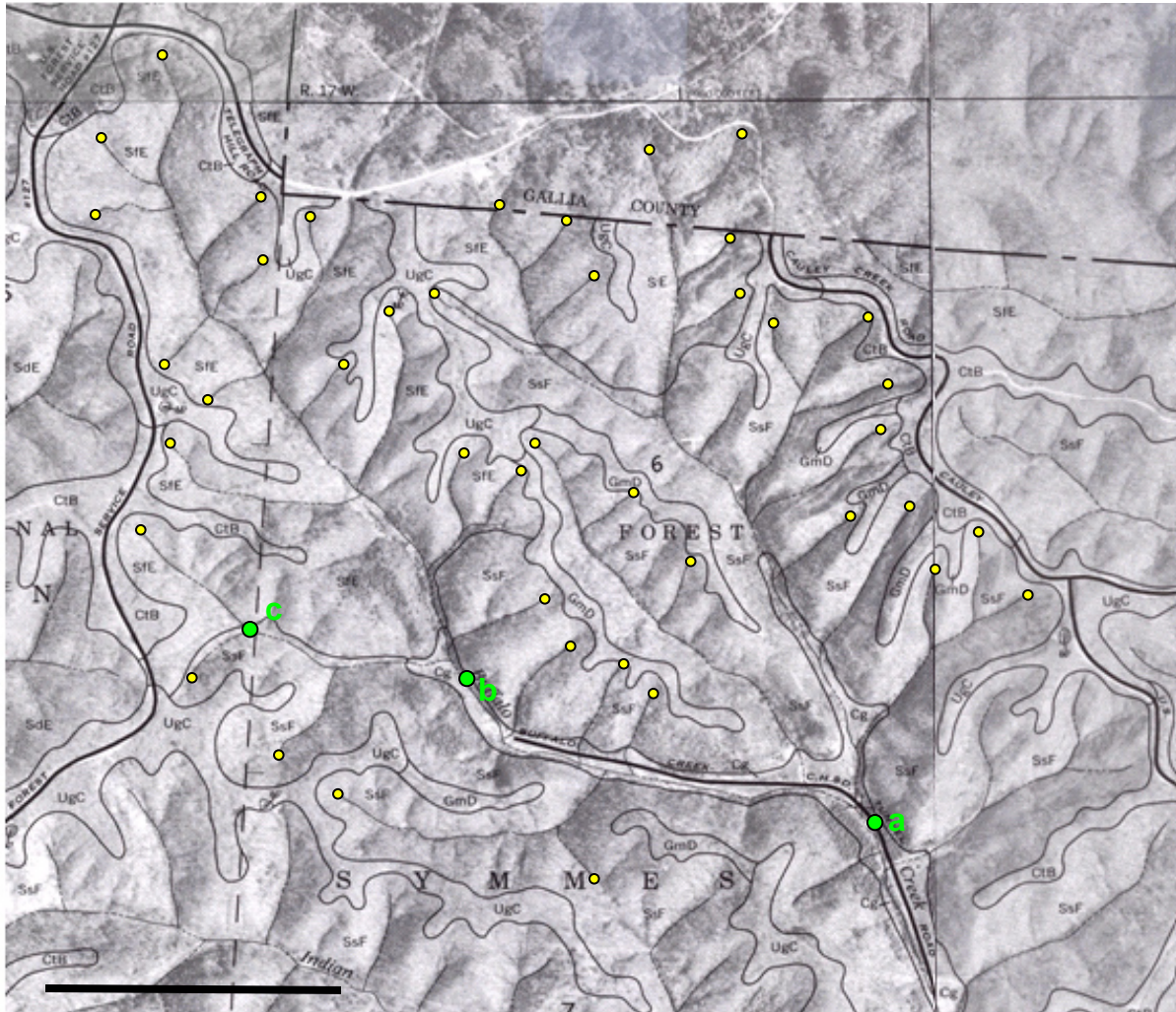


Figure 1-2 Portions of 1:15 840 United States Department of Agriculture (USDA) National Resources Conservation Service (NRCS) maps from Lawrence and Gallia Counties, OH illustrate the upper reaches of Buffalo Creek in Wayne National Forest (McCleary and Hamilton 1998). Green circles and associated letters mark corresponding points on maps in Figure 1-1. The yellow circles highlight the delineated stream origins. Black horizontal bars represent 0.5 mi (0.805 km). Buffalo Creek at “a” is a fourth-order stream, at “b” it is considered a third-order stream, and at “c” it is shown as a second-order. The number of first order streams shown upstream of “a” is 41.

and Omernik 1983), prompting some investigators to characterize such streams as zero-order streams (e.g., Brown et al. 1997). Most “blue line” designations on topographic maps are not based on field studies, but are “drawn to fit a rather personalized aesthetic”

of the cartographer (Leopold 1994) or drawn with standards that exclude a proportion of headwater channels (Drummond 1974). Moreover, large scale aerial and satellite image databases (e.g., 30-m DEM) are typically too coarse to accurately identify

most headwater channels, particularly in forested regions. Further development and more affordable application of Light Detection and Ranging (LIDAR) mapping technology provides the most promise for remotely recording the location and extent of headwater streams (e.g., Jarnagin and Jennings 2005). In addition, further work in understanding factors contributing to the evolution of stream channels will be useful for predicting the spatial distribution of headwater streams across the landscape (e.g., Montgomery and Dietrich 1988, 1992).

Headwater streams as monitoring units

Headwater streams are useful monitoring units owing to their extent (i.e., widespread and abundant), spatial scale and landscape position. Replicate streams of given treatments (e.g., types of land use/cover) and reference conditions, are more available for headwater streams because of their abundance across the landscape and relatively small watershed areas. Experimental studies are also more feasible (and ethically acceptable) in headwater streams and watersheds because they are easier to modify or perturb than downstream waterbodies (e.g., Likens et al. 1970, Wallace et al. 1999). Assessments of headwater streams can provide better resolution to diagnose cause and effect because they drain smaller areas with less land use heterogeneity than their larger counterparts. Flow of water from land to headwater channels is relatively short compared to larger rivers; therefore responses to land changes may be more rapidly detected. Because headwater streams have narrower widths and shallower depths than larger streams and rivers, a larger proportion of water flowing through headwater channels is directly contacting (and exchanging water and solutes with) the stream bed and banks at a given moment. Biogeochemical processes (e.g., denitrification) and biotic densities are

often higher in the saturated sediments of beds and banks than in the water column. This increased wetted area to water volume ratio therefore suggests that headwater channels may strongly influence downstream water quality. Lastly, because headwater streams represent the dominant interface between surrounding landscapes and downstream surface waters, further understanding of the structure and function of headwater streams will improve our ability to protect all water bodies.

Headwater streams and drying

One of the most distinctive and ecologically influential characteristics of many headwater streams is natural drying. In contrast to perennial or permanent streams that maintain continuous surface flow throughout most years, temporary streams (e.g., intermittent, ephemeral) have a recurrent dry phase(s) (Comín and Williams 1994, Uys and O'Keefe 1997, Williams 2006). Not to be confused with temporary waters are aestival water bodies (more commonly used to describe ponds than streams, but see Johansson and Nilsson 1994). Aestival habitats are characterized by being shallow and permanent, but freeze completely during the winter (Daborn and Clifford 1974). Temporary streams are the dominant form of running waters in arid and semiarid regions (Zale et al. 1989, Dodds 1997, Gasith and Resh 1999, Nanson et al. 2002), but are also common in temperate and tropical areas (e.g., Clifford 1966, Chapman and Kramer 1991, Delucchi 1988, Feminella 1996). Regardless of climatic region, headwater streams are more prone to drying than larger streams because they have smaller drainage areas for capturing recharge and generally have higher topographic elevation (McMahon and Finlayson 2003, Rivenbark and Jackson 2004, Svec et al. 2005). The rate of drying, and predictability, duration, and frequency of dry

periods vary with geographic setting and annual precipitation.

Variation in the temporal aspects of drying has been categorized by various classification schemes of temporary streams (Abell 1984, Poff and Ward 1989, Uys and O'Keefe 1997). Intermittent streams are typically identified as those that dry seasonally. During the dry season(s), frequently compounded by high evapotranspiration of watershed vegetation, the groundwater table may drop below the elevation of the streambed causing the stream to dry (Williams 2006). Ephemeral (or episodic) streams are usually dry except for several days immediately following precipitation. Surface flow in ephemeral channels is derived from surface runoff and shallow throughflow. Rather than having distinct, rigid boundaries, stream reaches classified as perennial, intermittent, and ephemeral may more accurately be described as dynamic zones within stream networks. The length or extent of these zones may be highly variable and is dictated by multiple factors (e.g., annual precipitation, evapotranspiration, land-use practices). The variable source area concept describes the dynamic zones as the expansion and contraction of flow within forested headwater systems (Hewlett and Hibbert 1967). Increases in discharge within small watersheds following a rain storm are rarely equivalent to the volume of rain fallen on the watershed. Much of the rain infiltrates into the soil and displaces subsurface water (already saturating the watershed) downslope into channels (i.e., throughflow or transitory flow). When this subsurface flow exceeds the capacity of the soil to transmit it downslope, water will be seen at the streambed surface and the wetted channel will extend upslope. Using a conservative tracer (NaCl) Genereux et al. (1993) measured the spatial and temporal variation in flow generation within a small

watershed in Tennessee. They determined that two downstream, perennial springs generated most of the flow during late summer, but as discharge increased, flow was predominantly generated from upstream, temporary reaches.

The natural process of drying causes changes in physical and chemical conditions (e.g., loss of wetted habitat, reduced dissolved oxygen), which can exclude some species while allowing others to thrive (Boulton et al. 2000). Temporary streams may, therefore, harbor communities containing mixtures of unique endemics (i.e., locally distributed species) and opportunist cosmopolitans (i.e., widespread species). The biotic community will vary among temporary waters with duration of hydroperiod (Williams 1996) and timing of the hydrologic cycle (Boulton and Lake 1992, Fritz and Dodds 2002). The hydrologic permanence (duration and frequency of continuous surface flow) of headwater streams must be understood to avoid confounding effects of natural drying when assessing the ecological integrity or condition. Different ecological expectations are likely needed when assessing condition of perennial and temporary streams. Although the methods described in this manual were used to identify hydrologic regimes primarily in forested headwater streams, some of the methods can also serve to quantify the ecological integrity of non-forested headwater streams.

Organization of the manual

This manual is divided into three sections: 1) Assessment Design and Site Selection, 2) Physical Habitat Characterization, and 3) Biological Sampling. Sections are further divided into subsections, covering relevancy of a measure, detailed steps to collect data, lists of equipment and supplies, and alternative ways of quantifying measures (where applicable). References are provided

at the end of each subsection to aid the reader. We refer to example field sheets for recording data throughout the manual. Complete copies of these field sheets are provided at end of the manual in Appendix 1. The procedures described in this document are intended to maximize the information gained for amount of resources expended. The initial intent of most procedures described is to collect information that characterizes the hydrologic permanence of stream reaches (i.e., indicators); however, most measures are also commonly used in stream condition assessments (e.g., macroinvertebrates, substrate size).

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2 FACTORS INFLUENCING STUDY DESIGN

This section discusses some initial considerations for planning a study of headwater streams. This section is not intended to cover all possible issues when preparing a study or assessment. Rather, general options and some unique considerations for headwater streams are discussed.

Clearly stated objectives (and associated hypotheses) are important to any scientific study and should be decided before moving forward. The objectives should set the initial stage for what and how much will be measured. Therefore, the spatial and temporal scales (sampling resolution) and scope (range or extent of the study) should be determined by the data needed to meet the objectives or test the hypotheses. Logistical and economic constraints also influence the scale and scope of studies. Norris et al. (1992) point out that the objectives of most studies fall into two general categories: 1) determining values at a single location and time; and 2) comparing values from multiple locations or time periods. In the first case the goal is to provide an accurate estimate (e.g., total density), whereas the second focuses on comparing the difference of values between locations or time periods. Downes et al. (2002) identified four general objectives for assessment studies: 1) assess the ecological state of ecosystems; 2) determine if regulated criteria have been exceeded; 3) detect and quantify impacts generated by anthropogenic disturbance(s); and 4) assess the effectiveness of restoration projects. In any case, the objectives should guide the design, implementation, and analysis of the study.

Field sampling designs

After identifying the specific objectives, decisions are made regarding the study design (i.e., how, what, when and where to sample). There are two major categories for study designs: comparative and manipulative. Comparative (also called mesurative) studies have location or time period as the primary treatment(s) being investigated, where the treatment exists without the intervention of the scientists. An example of a comparative study is comparing biological and physiochemical measures among streams with different land uses or an intensity gradient of a land practice. The primary treatment of manipulative (or experimental) studies is an intervention or perturbation by the investigators. An example of a manipulative study is measuring the biological characteristics in one set of streams where large woody debris has been removed by the investigators and in another where large woody debris is left intact. Manipulative studies generally offer more control over the independent variables (and therefore greater potential to identify cause-effect relationships) than comparative studies. On the other hand, comparative studies typically offer greater realism and generality than manipulative studies. The main effects (or treatment differences) and associated variation of effects over the study duration of comparative studies are directly relevant to the systems studied. Investigators designing experimental studies should strive to apply realistic manipulations (i.e., relevant to real world situations) to experimental units. Both categories have merits and limitations that should be considered when planning a study (see Diamond 1986 for a detailed discussion).

Spatial and temporal scales of a study should match the objectives and be relevant to the

organisms and environments studied. Gotelli and Ellison (2004) identified two aspects of spatial scale that should be addressed when designing a study: the grain (size of the smallest unit of study) and the extent (total area encompassed by all units in the study). Investigators need to efficiently balance the size of the grain and extent of study with logistics and cost to effectively achieve the scope of the objectives. Temporal scale includes the time needed to collect a sample, the frequency of sampling, and the duration of the study. Hierarchical or nested designs can be used to identify variation associated with different spatial scales, and repeated measures designs assess interaction among sampling periods and treatments. Stratification of sampling by habitat type can account for variation that would otherwise be considered in the error.

A critical aspect of a field study is the sample size needed to effectively test a hypothesis or to provide an acceptable level of confidence around estimates of resource condition. Often the emphasis for condition surveys is to estimate the proportion of a resource among classes of condition (e.g., Diaz-Ramos et al. 1996). Condition classes reflect categories of ecological integrity and are measured with indicators representing various physical and biological parameters. Thresholds separating condition classes are typically set by regulatory standards. The formula for estimating the standard error for a proportion is:

$$\hat{\sigma}_p = \sqrt{\frac{p(1-p)}{n}}$$

where p is the proportion of a population representative of a class and n is the total population size (i.e., sample size). By assuming a proportion that results in the largest estimate of the standard error of the proportion ($p = 0.5$), one can visualize that

standard error decreases asymptotically with increasing sample size (Figure 2-1).

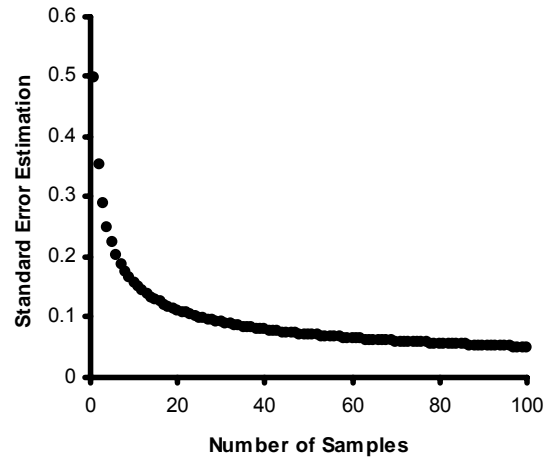


Figure 2-1 Relationship between sample size and standard error estimations assuming proportions are equal among populations.

Therefore, confidence around estimates also increases with higher sample size, but investigators need to balance sampling cost and acceptable level of confidence when designing surveys.

In hypothesis testing, power analysis can be useful for determining the appropriate number of replicates to provide sufficient statistical power for an expected effect size (the detectable difference between treatments) and natural variation (Peterman 1990, Fairweather 1991, Foster 2001). Statistical power measures the probability of correctly rejecting the null hypothesis when in fact it is false (converse of the probability of Type II error). Power is generally described as:

$$Power \propto \frac{ES \cdot \alpha \cdot \sqrt{n}}{s}$$

where ES is effect size, α is the a priori significance level (Type I error probability), n is the sample size, and s is the standard deviation among replicate units. This relationship indicates that for a given effect

size and level of variability, power increases with higher study unit replication; however, with that in mind, increasing sample size can enable detection of very small effects that may not be ecologically significant. Larger effect sizes are more likely to be detected than smaller ones with the same sample size and level of variability. The actual formulae for calculating power or deriving appropriate sample size or minimum detectable effect size will vary with statistical test and test statistic (see Cohen 1988, Zar 1998). Effect size may be derived from previous studies, regulatory thresholds, or convention (e.g., order of magnitude). Expected variation can be taken from the literature or pilot studies. An appropriate a priori level of statistical power will vary depending upon the objectives of the study. For example, failing to detect an environmental impact where one exists (i.e., Type II error) may have greater consequences than detecting an impact that does not exist (Type I error), therefore greater power may be desired to protect against a Type II error (see Peterman 1990, Di Stefano 2003). Frequently, cost (time and money) is a critical factor governing sample size. Mapping the study beforehand (estimating time and costs) will help determine the feasibility of the study design. Designing an effective study is balancing effect size, sample size, and cost to meet the study objective.

Randomization should be used whenever feasible to ensure unbiased data collection. Random or probabilistic site selection produces a representative sample of the population(s) targeted under the study objectives, so that results can be more confidently extrapolated to the overall population from which the selected sites were randomly chosen. In contrast, targeted sampling focuses the effort toward a specific problem. The difficulty with randomized site selection is the *a priori* knowledge of the

entire population of possible sites or sampling points within the bounds of the study objective. If the scope of the objectives is narrow and the population is known (e.g., water bodies within Central Park), probabilistic sampling is more feasible compared to broader scales where the population is uncertain (e.g., spring seeps of Kentucky). The scope of a study will be narrowed under most circumstances because of the inability to account for the entire population of potential sites. Time of data collection is rarely randomly selected because of the stochastic nature of streams; however, seasonal sampling is usually desired. Index periods are typically determined by the logistics of sampling and the life history of targeted biota.

There are practical difficulties associated with large scale experiments, including the need for a large number of independent replicates to overcome natural variability among replicate study units. A study design that is increasingly used in stream research is the Before/After and Control/Impact (BACI; Stewart-Oaten et al. 1986, Carpenter et al. 1989, Downes et al. 2002). In this design, one or more control sites and one or more impact sites are simultaneously sampled multiple times, both before and after the manipulation to the impact site(s). The difference in parameters measured between the control and impact at each time period represents a replicate unit for the Before and After treatments. Underwood (1991, 1992) strongly advocated the incorporation multiple randomly selected control sites in the design to overcome the possibility that the control and impact sites may have naturally different trends in the measured parameters. Further issues and concerns about BACI designs are reviewed by Smith et al. 1993, Osenberg et al. 1994, and Downes et al. 2002. An in-depth discussion of specific statistical designs is

outside the scope of this manual. A few relevant texts for ecological studies include: Clarke and Warwick (2001), Gotelli and Ellison (2004), Scheiner and Gurevitch (1993), Quinn and Keough (2002), and Underwood (1997).

Special considerations for headwater streams

Headwater streams are narrower, shallower, have higher drainage density, and are more likely to dry than larger streams and rivers. Their position in the network also makes many headwater streams more responsive to precipitation, so lag time is shorter between precipitation and peak discharge. Notable exceptions to this are spring-fed streams, where deep and more stable groundwater discharge can dominate the hydrologic regime. Depending upon the geographic location, headwater streams may have higher gradients and therefore the repeating habitat units are typically more closely spaced than wadable streams. Reach lengths for ecological assessment are typically scaled to the channel width (e.g., Barbour et al. 1999, Lazorchak et al. 1998, Moulton et al. 2002). Following this convention, reach lengths of headwaters are shorter than those needed for larger perennial streams and rivers. Multiple reaches or longer reaches may be required for studies using multiple indicators or assessment approaches (i.e. amphibian surveys, tracer additions, etc.). If multiple reaches are used, they should be as close as possible given the sampling or logistical limitations. They should have similar channel dimensions and levels of permanence, avoiding influences by intervening tributary confluences. Higher drainage density affords the opportunity to have nearby replicate streams for studies, but also may result in frequent discontinuities (e.g., abrupt changes in substrate size) at tributary confluences (Rice et al. 2001, Brenda et al. 2004). Unique sampling methods are often required for headwaters because the low

flows prevent use of many conventional sampling devices. For example, core samples are preferred for headwater invertebrate sampling rather than Surber or other net samplers that require sufficient flow to carry dislodged debris into the net. Estimates of flow permanence are critical and may be the master variable influencing headwater communities. Measures of channel dimension and substrate size may provide critical insight into the typical flow regime or degree of permanence at a site and should be included in any headwater assessment.

Time of year for sampling is critical in temporary headwaters because precipitation and evapotranspiration has a relatively strong influence on stream discharge. Historic hydrological data are rare for headwater streams because most gauges are positioned on wadable streams and large rivers. Discharge data from downstream gauges can provide an integrated measure of precipitation and evapotranspiration for a basin. The utility of gauging data from downstream locations will depend upon their distance from headwater sites, their position relative to reservoirs (where levels may reflect not solely precipitation, but recreational and socioeconomic use), and changes in watershed land cover. In addition, many gauges on intermediate size streams and rivers have been retired, and therefore problematic for developing stage relationships with headwater sites. However, long-term precipitation records may serve as surrogate for flow. The seasonal and interannual variation in precipitation and hydrologic observations provide the likelihood of flowing conditions. While year-round sampling (both dry and wet seasons) over several years may be optimal for categorizing or assessing a headwater site, researchers are rarely afforded such opportunities. For shorter-term studies, sampling should take place during the driest

and wettest periods of the year to assess extreme conditions. If sampling is restricted to one visit, headwater index periods will typically be during the spring when flow is higher, and most aquatic organisms can be collected.

The gradual change in environmental conditions (e.g., lower dissolved oxygen, higher temperatures) as temporary habitats dry can be as critical to understanding mechanisms influencing biotic response as the duration and frequency of drying. Disturbances (disrupting force) or perturbations (sequence of disrupting force and system response) have been classified as either pulse or press events (Bender et al. 1984, Glasby and Underwood 1996). A pulse disturbance is characterized by a short and sharply delineated event (relative to the time scale of the response measure, Figure 2-1a), whereas a press disturbance has a continuous and constant level that is relative long-lasting (Figure 2-1b). In contrast to pulse and press disturbances, environmental conditions for many organisms worsen over time as streams dry (Slack and Feltz 1968, Towns 1985, Ostrand and Wilde 2004). Lake (2000, 2003) characterized this difference by conceptualizing that drying or drought was a “ramp” disturbance (Figure 2-1c). As the sequence of physicochemical changes progresses, greater stress is placed upon inhabitants, causing more taxa to succumb or emigrate over time. Rather than a steady sequence of physicochemical changes of a “ramp”, Boulton (2003) argues that the sequence of changes may be better characterized as a series of “steps” (Figure 2-1d), wherein critical thresholds cause substantial shifts in wetted habitat (e.g., drying of riffles, subsurface habitat). Differences between the ramp and stepped models may be to some extent dependent upon the hierarchical scale through which the drying

process is approached (Stanley et al. 1997). Some changes may be more apparent at small spatial or temporal scales, but undetectable at larger scales.

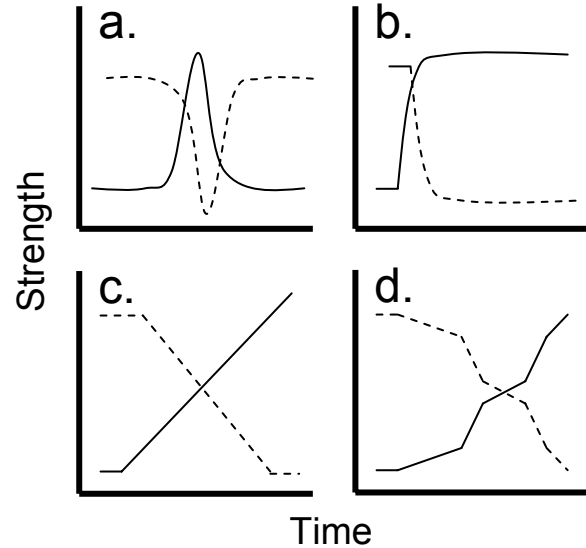


Figure 2-2 Types of disturbance (solid) and responses (dashed) in streams: pulse (a), press (b), ramp (c) and stepped (d). Based on figures from Lake (2003) and Boulton (2003).

Wetted area and volume are reduced initially in the drying sequence that leads to increased isolation of the wetted area from stream banks (contraction toward the deeper flowpaths in the channel) and between habitat units (contraction to pools). As discharge declines, flow may at first become braided between larger emergent substrates, then become limited to strong upwelling zones along the channel, and then finally cease altogether, leaving surface water to remain only in deep pools. These remaining pools shrink by evaporation and the hyporheic habitat (subsurface zone between the surface water and groundwater) dries if water deficit continues. The rate of channel drying varies with channel gradient, degree of exposure (to wind and sun), evapotranspiration by

watershed vegetation, soil moisture status, and permeability or infiltration capacity from the surrounding watershed. Vegetation cover, type, and succession stage can also influence headwater stream hydrology (Bosch and Hewlett 1982). For example, annual stream flow is typically lower in streams draining conifers because of higher annual interception (and subsequently evaporation) of precipitation and higher transpiration loss at the beginning and end of the growing season than hardwoods (e.g., Swank et al. 1988). Streams draining limestone or “karst” geology retain surface water for shorter periods than streams draining geologic materials with lower hydraulic conductivity and effective porosity (e.g., sandstone and clay). Many of these factors will also influence the timing of flow commencement following precipitation (Blyth and Rodda 1973, Day 1978, de Vries 1995) or leaf abscission (Doyle 1991).

As previously mentioned, headwaters have a distinct bioassessment advantage because the small watershed areas make stressor identification more straight-forward. However, the timing of sample collection relative to the resumption of flow or start of drying is critical. The diversity, abundance, and biomass of benthic organisms increase and community composition shifts with time following the resumption of flow (Peterson 1987, Boulton and Lake 1992, Fritz and Dodds 2002, 2004). The rate of assemblage recovery varies with magnitude, duration, and extent of drying, particularly in relation to the permanence history (i.e., flow predictability) of streams. Resilience will likely vary among assemblage types and biological parameters (e.g., abundance, biomass) because of differences in the recovery mechanism (i.e., resistance vs. colonization), vagility, and growth rates. For purposes of bioassessments, samples should be collected near the peak of recovery from drying to maximize the number

of indicator taxa present and biotic index or metric discrimination among condition categories.

Minimizing impacts associated with sampling

The potential for impacting streams during sampling is higher for headwater streams compared to larger streams and rivers, and therefore requires special consideration. Small wetted areas mean that sample collection and geomorphic measurements can potentially disturb a large portion of the local channel with potential adverse effects downstream. Individual substrates (e.g., cobble, small woody debris) that are inconsequential in larger streams and rivers may provide important geomorphic functions in headwater streams. Channel alteration caused by sampling may be more persistent in small streams than in larger channels because the power associated with flood events that resets channels is typically lower. Sampling in an upstream direction is typical in larger streams, and it is especially important when working in headwaters to minimize trampling the stream reach during assessments. Because headwater streams are small and positioned at the tips of stream networks, oversampling of unique populations and species is a concern. Headwater streams, particularly those that are spring-fed, often contain endemic taxa (Hubbs 1995, Ferrington 1995, Myers et al. 2001). Rather than further the endangerment of these unique communities, sampling protocols should provide information for their conservation.

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- 2.1 Study design for comparing across stream reaches with varying hydrologic permanence.**
- This section describes a specific study design used for comparing among headwater stream reaches varying in hydrologic permanence. The objectives were: 1) to characterize biological and physical features of reference headwater streams across a gradient of hydrologic permanence (frequency and duration of drying) and 2) to identify indicators of hydrologic permanence. The study focused on headwater streams in intact forests to limit potentially confounding effects of land use on hydrology. Streams were sampled in Indiana, Illinois, Kentucky, Ohio, New Hampshire, New York, Vermont, Washington, and West Virginia. The drainage area of study sites was restricted to basins $\leq 2.92 \text{ km}^2$ (1 mi²) that corresponded to the upper boundary of streams measured. For assessment purposes, Ohio EPA is using this

drainage area size to distinguish “Primary Headwater Habitat Streams” from the rest of the stream network (Ohio Environmental Protection Agency 2002).

Selecting study units that incorporate the range of hydrologic permanence (i.e., from ephemeral to perennial) was critical to meet the goals of the study. No data for annual hydrologic patterns were available prior to sampling, so the general positive relationship between drainage area and flow permanence was used to select sites (i.e., drainage area was used as a surrogate for flow permanence). As drainage area increases, groundwater storage increases and approaches the level of the streambed. Exceptions to this general pattern include perched aquifers and artesian springs in upper reaches that sustain year-round surface flow (Dunne and Leopold 1978) or substantial storage in swale soils above the channel head that sustains patches with perennial surface water (Hunter et al. 2005). These characteristics can result in fragmented longitudinal patterns of flow permanence along headwater streams (Lake 2003). Likewise, local changes in streambed topography along a stream influence the spatial pattern of hydrologic permanence. Sediments and woody debris originating from landslides, debris flows, and windthrow are transported downstream and deposited in reaches with lower gradient (Benda and Dunne 1987, Grizzel and Wolff 1998, Montgomery 1999). These deposits (i.e., sediment wedges) locally elevate the streambed above the dry season water table, causing reaches with such deposits to seasonally dry (May and Lee 2004, Harvey et al. 2005). Recognizing this, the study design incorporated multiple study units along multiple headwater streams. This design included a broad range of hydrologic regimes and capable of detecting repeating

associations between stream features (biological and physical) and hydrology.

The study units were 30-m long reaches of stream channel (land form with bed and bank features). This length is on average 40X the headwater channel width and is consistent with study units used by USEPA in the Environmental Monitoring and Assessment Program (EMAP). Adjustment of the reach length may be needed to incorporate repeating geomorphic channel units. Three or four 30-m study reaches were selected within each stream. The aim was to include 1 reach with perennial flow, 2 reaches with varying degrees of intermittent flow and 1 reach with ephemeral flow. This design ensures sampling across a sufficient range of hydrologic conditions within a stream, while also allowing for multiple streams to be assessed. This sampling regime of the study required at least 2 sampling periods for each site within a year. These periods included visits in spring (wet season) and late summer (dry season), but not necessarily in that order. An initial visit to the streams during the dry season helps ensure that a perennial site is sampled, but it may be difficult to determine if a dry reach is intermittent or ephemeral at that time. Field visits during wet and dry seasons prior to selecting sites, where possible, may provide greater confidence in the distribution of sites across the flow permanence gradient.

Desktop selection procedure

In most cases the upstream study reaches along the streams were not marked with “blue lines”, but appeared as “depressions” on 1:24 000 scale topographic maps (Figure 2-2). Red lines have been added to Figure 2-2 to show a more realistic and complete network of stream channels within the Falling Rock Branch watershed. The yellow line represents the approximate watershed boundary. Maps (typically 1:15 840 scale) published by USDA

NRCS (formerly Soil Conservation Service) provided better resolution of the headwater channel network, but still underestimated the extent of channels. Likewise, orthophotos (e.g., 1:12 000 scale) aided planning, but the ability to discern headwater channels varied with photo resolution and vegetation cover.

Both types of maps and photos were used in the planning stage, but the topographic maps were more useful while in the field. The definition of the upstream extent of headwater channels is discussed in detail in Section 3.3.

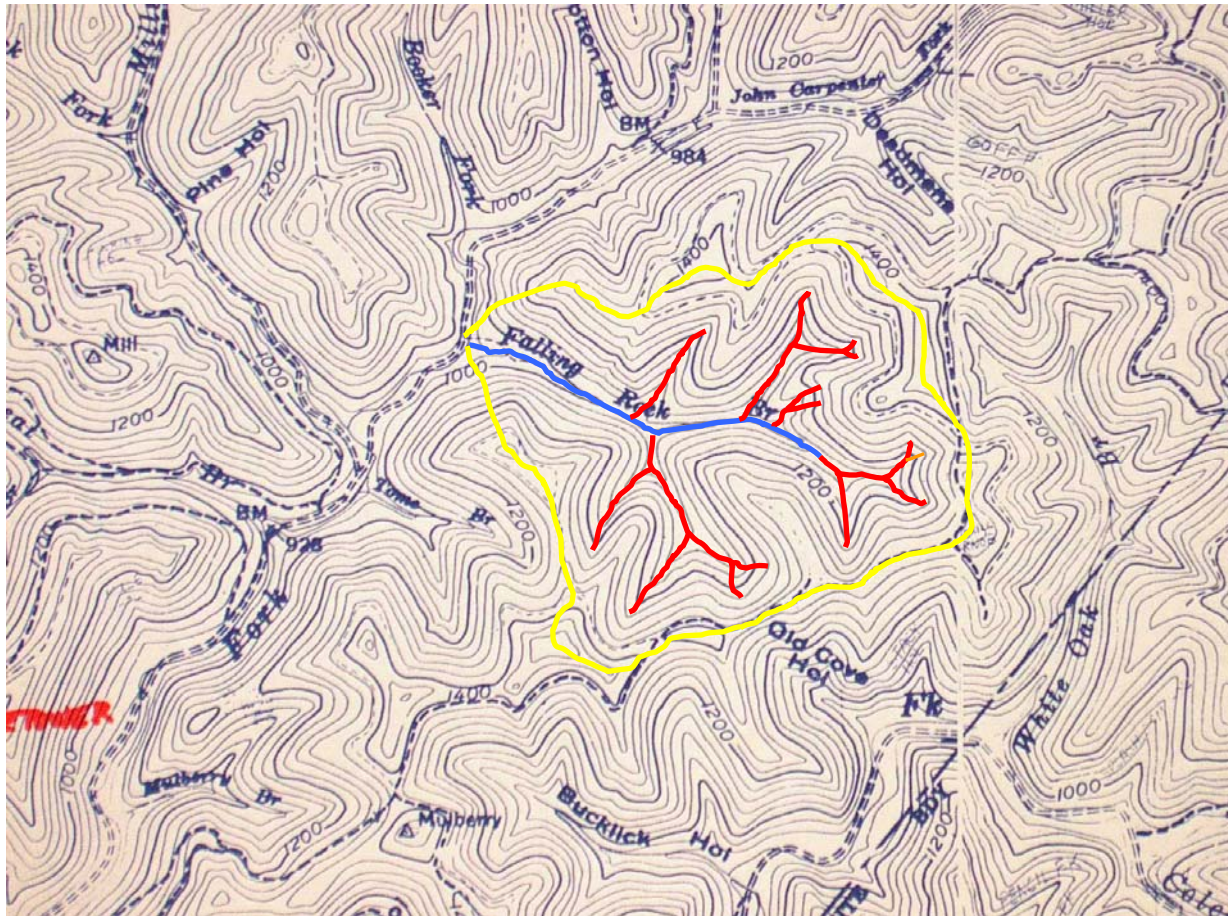


Figure 2-3 Map highlighting position of headwater channels within the watershed of Falling Rock Branch, KY. Yellow represents boundary of watershed, blue represents “blue line” designation on the 1:24 000 USGS topographic map (Noble 7.5 minute quadrangle, Breathitt County, KY), and red represent headwater channels not shown on the topographic map.

The channel network configuration, particularly dendritic or reticulate networks, creates a nonlinear relationship between distance downstream and drainage area. Drainage area does not increase gradually downstream, but rather increases in steps with each tributary confluence. This is an

important consideration when selecting study reaches to maximize the range of hydrologic permanence. Tributary confluences are also useful landmarks for returning to study reaches for subsequent visits. Where possible, a more gradual drainage area transition between study reaches is preferred (Figure 2-

3). In some cases the entire drainage area of headwater stream may not be sufficient to supply perennial flow. In this situation a

stream may need to be paired with an adjacent, larger tributary so there is a shared perennial site (Figure 2-4).

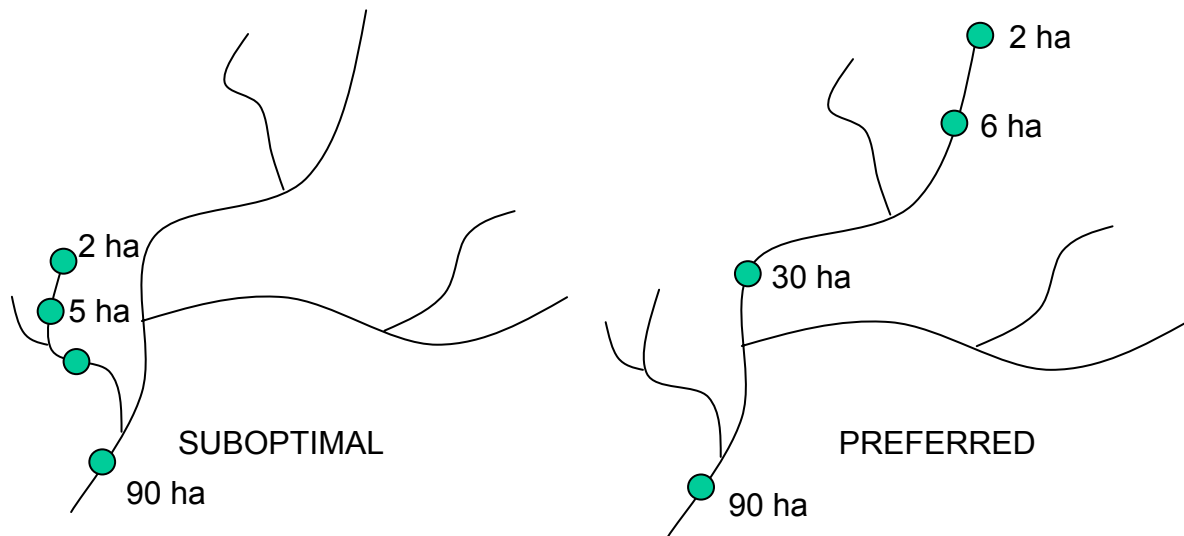


Figure 2-4 Schematic showing suboptimal and preferred longitudinal positioning of sites along headwater channels to maximize the range of hydrologic permanence across study sites. Hypothetical drainage areas are shown to further illustrate spatial hierarchy.

Properties that contained intact forest were identified and we obtained USGS 7.5 minute quadrangle maps (1:24 000 scale) for the selected areas. Land owners or managers of the properties were contacted. We described to them the objectives and design of the study and provided background material that they may need (e.g. Quality Assurance Project Plan, research proposal). We also inquired about headwater streams draining the property, especially pertaining to their flow permanence, current land use, ongoing or previous research downstream, and accessibility by roads and hiking trails. Being able to quickly travel between study reaches helped ensure that a sufficient number of study units were assessed and that sampling was done over a reasonable timeframe (i.e., within the same index period).

Field study reach – general selection guidelines

In the field we located the stream reaches preliminarily selected from the map. Final selection was adjusted to ensure that the study reaches were entirely upstream or downstream of tributary confluences. We also selected reaches having multiple habitat units (erosional and depositional habitats). Although large woody debris dams are characteristic features of intact forested streams, reaches with excessively large woody debris dams (prevented access to >50% of the wetted channel in the study reach) were avoided when possible. These structures are likely to 1) complicate the association between reach properties (physical and biological) and hydrology and 2) impede data collection. With this in mind, debris dams are common in some regions and will be unavoidable when designating study reaches.

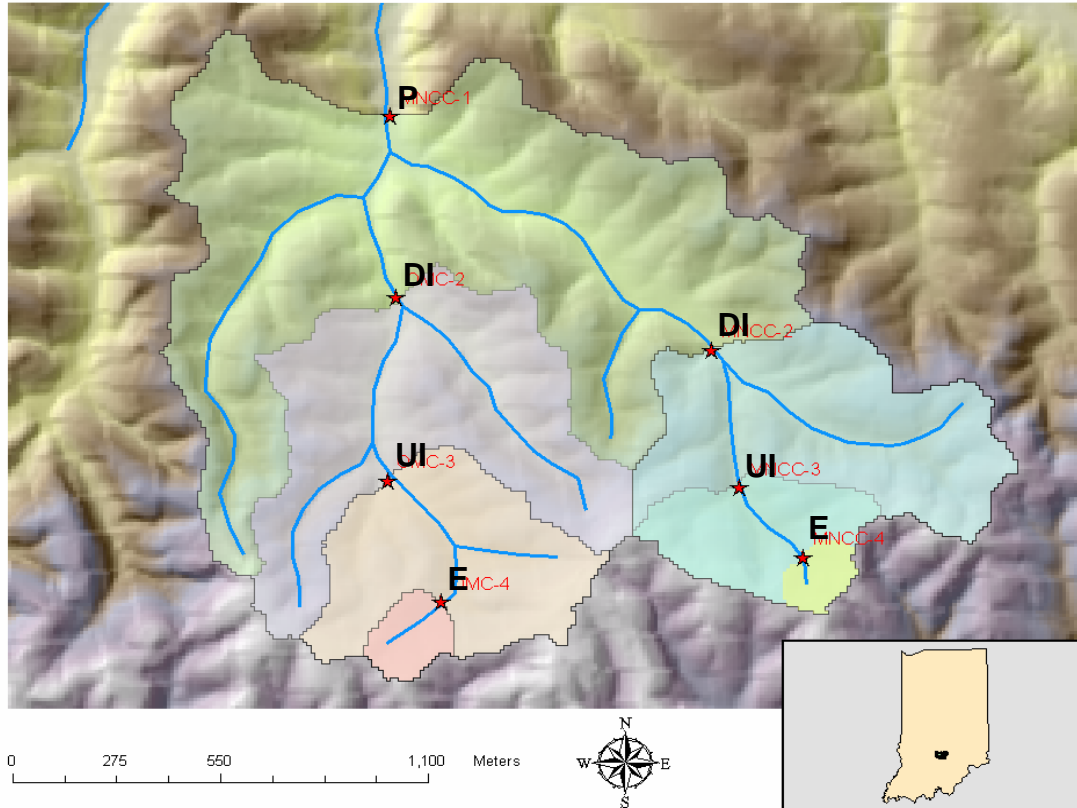


Figure 2-5 Map showing positioning of sites along two Indiana headwater streams where the downstream perennial site (P) is “shared” between two tributaries. DI = downstream intermittent; UI = upstream intermittent; and E = ephemeral. Shading shows cumulative drainage area in downstream direction.

Using a measuring tape, we marked the 30-m study reach from the downstream boundary (located at 0 m) to the upstream boundary (located at 30 m). The tape was positioned to follow the thalweg. The thalweg is the deepest flow path in a channel. The study reaches were designated using flagging tape or other clearly visible markers attached to trees near each boundary. The location of the study reach was identified on the topographic map or a PDA with electronic topographic maps, and a written description of the study reach location and appropriate locality information (e.g., topographic map, county, state) was

entered on the field forms. Photographs of the study reach were taken and coordinates from a GPS unit were recorded. Study reaches were consistently identified by site numbers that increased in an upstream direction starting with 1 at the downstream-most reach (Figure 2-5).

Field selection – initial visit in spring (wet season)

When the initial field visit to a study region was in the spring (wet season), then sites were located as follows. The ephemeral site for each headwater stream studied was designated

just upstream of the origin of intermittent flow (Paybins 2003; upstream-most location of spatially-continuous surface flow in the spring or wet season; Figure 2-5). The upstream intermittent site was positioned downstream of the origin of intermittent flow. The drainage

area of the downstream intermittent site often incorporated at least an additional ephemeral drainage. Similarly, the perennial site frequently incorporated at least twice the drainage area of the downstream intermittent site (Figure 2-5).

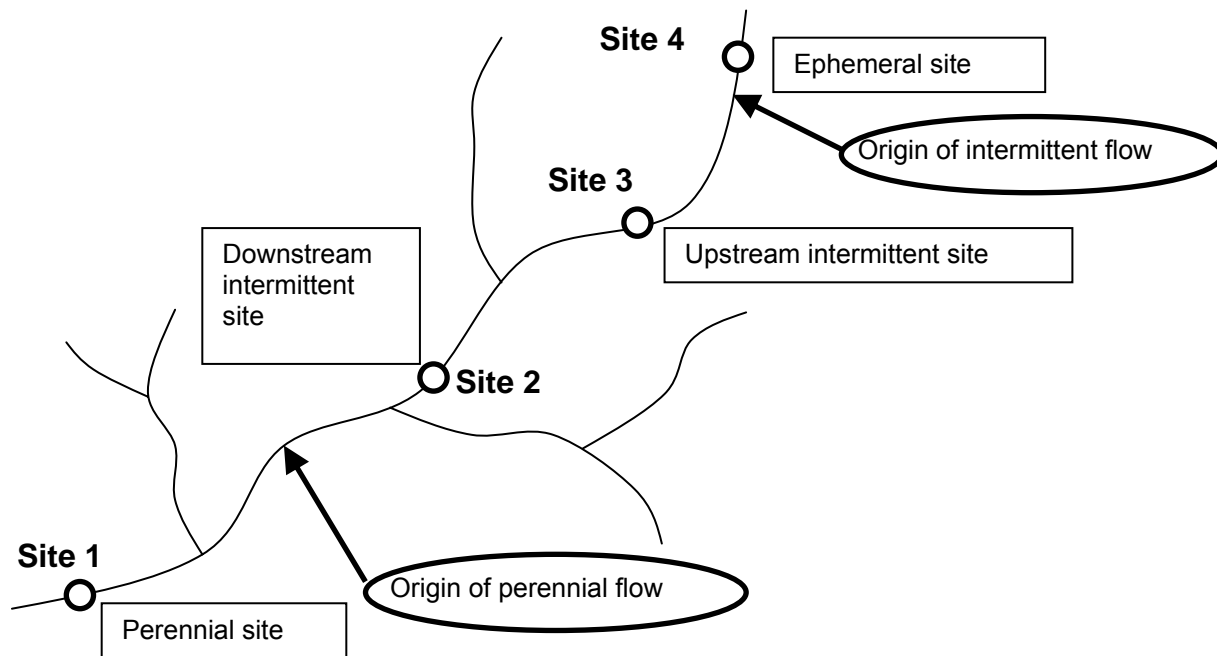


Figure 2-6 Schematic of headwater channels showing numerical designation and position of study sites relative to origins of intermittent and perennial flow.

The spatial pattern of hydrologic permanence may not reflect a downstream progression from ephemeral to intermittent to perennial reaches along headwater channels for reasons discussed earlier (e.g., perched aquifers, artesian springs). Incorporating multiple streams into the design may provide support for alternative longitudinal patterns of flow permanence within headwater drainages. Depending upon the precipitation and geographic setting the prevalence of some permanence categories and therefore variation in flow permanence among study sites is more subtle.

Field selection – initial visit in summer (dry season)

When the initial field visit to a study region was in the summer (dry season), then sites were located as follows. The origin of perennial flow (Paybins 2003; upstream-most location of spatially continuous surface flow in the summer or dry season; Figure 2-5) was located. The perennial site was positioned just downstream of the origin of perennial flow. The other three study reaches often did not have continuous surface flow during the summer. The next site upstream frequently

drained approximately half the drainage area of the perennial site. The upstream intermittent site (Site 3) was often positioned at least one confluence upstream of Site 2. The ephemeral site was designated near the top of the watershed, but where there was a defined streambed and banks. Terrestrial herbaceous vegetation was common within the channel of the ephemeral study reach.

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Equipment and supplies

USGS 7.5 minute quadrangle map(s)
 NRCS soil survey map(s)
 Measuring tape (50 m)
 Flagging or other marker
 Camera
 GPS unit or Handheld personal computer or Personal Digital Assistant (PDA) with digital maps and GPS card

3 PHYSICAL HABITAT CHARACTERIZATION

Physical habitat, typically refers to the structural attributes of the stream channel. For convenience of organization, we also discuss the measurement of physicochemical attributes of the stream water in this section. Habitat degradation from land-use change is the greatest threat to streams and their inhabitants (Allen and Flecker 1993, Sala et al. 2000, USEPA 2001). Although stream scientists generally agree that habitat degradation is a serious threat, no universally accepted index or procedure exists to rate physical habitat condition for streams. The complexity and natural variation of stream habitat, the need for rapid field protocols, and objectivity must be balanced before such a measure is accepted. While the development of such a universal tool is beyond the scope of this document, this work has modified existing procedures and developing new ones specifically for headwater streams. We believe that these procedures will contribute toward effectively quantifying condition, identifying causes of degradation, and restoring stream habitat.

Hierarchical classification across spatial and temporal scales is useful for delimiting sources of natural variability within and among complex systems and provides a framework for integrating information from different levels of resolution (O'Neill et al. 1986). Such a framework for streams ranges spatially from whole drainage networks down to microhabitats (Frissell et al. 1986). Implicit in this framework is an understanding that absolute linear dimensions for spatial scales across all streams (Brussock et al. 1985) or even longitudinally within a stream (Vannote et al. 1980, Montgomery 1999) are unattainable due to variation in geology, climate, and topography.

The stream reach is the most commonly used and practical spatial scale for study units. The spacing of distinctive features (e.g., pools, riffles) within streams is partly driven by channel width. The length of study reaches should be sufficient to incorporate multiple features of the same type to prevent evaluations based solely on potentially anomalous features. As discussed in Section 2.1, study reaches that are 30-m long are sufficient in most cases where streams are 1-to 2-m wide.

Transect sampling (i.e., line-intercept technique) is a commonly used method to quantify physical habitat at the reach scale (e.g., Platts et al. 1983, Fitzpatrick et al. 1998, Lazorchak et al. 1998). Transect sampling uses a series of lines (transects) that are positioned perpendicular to flow and cross the channel. Measurements are taken along these transects to characterize the stream reach, and thus, provide the investigator with mean estimations and a degree of variation along stream reaches. Transects can continue beyond the stream channel where measurements of the adjacent riparian zone, floodplain, and terraces are of interest. The number and positioning of transects should be sufficient to characterize the spatial scale of interest. Physical parameters that vary little along a stream reach will require fewer measurements (e.g., discharge) to arrive at representative values than those that can vary substantially (e.g., water depth). The positioning of transects can be done systematically (e.g., every meter), randomly or stratified random (e.g., stratified by habitat type). Systematic selection ensures that the measurements span the entire study reach and may be logistically easier; however, random

selection may be preferred because all cross-sections have an equal chance of being measured (see Section 2, *Field sampling designs*).

As streams dry, surface water will gradually become constricted to the channel thalweg. Therefore, the thalweg will often be the last area to dry for a given channel cross-section. The thalweg is an important location for measuring many physical parameters because this can be a consistent and conservative target when comparing across sites with varying hydrologic permanence and ecological condition. Where transect sampling is used, the thalweg (rather than the banks) is the central axis along the stream where the transects should be perpendicularly spaced. Many of the measures described in the following sections are centered on the thalweg at sampling transects. Because of the narrow widths of headwater channels, these sampling points represent most of the channel width and the portion of the channel width that is inundated longest.

Characterization of physical habitat is widely used in stream assessments (see Somerville and Pruitt 2004); however, assessment protocols vary in purpose, breadth, and targeted stream type (Montgomery and McDonald 2002). Few protocols specifically target headwater streams, but several region-specific assessment protocols are potentially available. The associated objectives of these protocols vary somewhat. For instance, the Ohio Environmental Protection Agency's Primary Headwater Habitat Evaluation Form (Ohio EPA 2002) was developed to differentiate among 1) coldwater perennial streams, 2) warmwater perennial and intermittent streams, and 3) ephemeral streams. The North Carolina Division of Water Quality's Classification Method (NCDWQ 2005) and Fairfax County (VA)

Stormwater Planning Division's Perennial Streams Field Identification Protocol (FCSPD 2003), were designed to classify streams based on hydrologic permanence (i.e., ephemeral, intermittent and perennial flow). Some agencies like the Louisville District of the U. S. Army Corps of Engineers (Sparks et al. 2003a, b) have adopted protocols developed for wadable streams (USEPA Rapid Habitat Assessment Form (RHAF); Barbour et al. 1999). The Louisville District uses RHAF, in conjunction with specific conductivity and macroinvertebrate bioassessment index scores (Pond and McMurray 2002), to assess the ecological integrity of headwater streams in the Eastern Coalbelt Region of Kentucky. Ideally, all three components are then used by district personnel when reviewing Clean Water Act Section 404 permit applications for dredging and filling headwater streams and determining appropriate mitigation or in lieu of fees for impacted streams.

Habitat assessment protocols vary in level of subjectivity; some use visually-based qualitative attributes across categories such as absent, weak, and strong, whereas others rely on quantitative measures. Qualitative protocols are advantageous under high workloads with limited resources and training because they often require less expertise and time to complete than quantitative assessments. However, the versatility, applicability, and rigor of qualitative assessments are more limited. For instance, the attribute scoring of individual measures or questions in qualitative assessments are weighted based on regionally derived investigations that may not be applicable outside the original region. The data for qualitative assessments are often categorical or discrete (i.e., integer values) over a limited range, whereas quantitative data can be distributed continuously or categorized for analyses. Lastly, data sets from sources that

use the same quantitative measures are more feasible to combine for broader assessments than qualitatively collected parameters. Many habitat characteristics, however, are currently limited to only qualitative or semi-quantitative methods for assessments (e.g., habitat unit designations, substrate embeddedness, instream fish cover). Wang et al. (1996) noted that among 27 habitat characteristics evaluated for among-observer precision, those that were scored quantitatively (directly measured, rather than visually scored across categories) were more precise than qualitatively scored characteristics. In their review of physical stream protocols used by regulatory agencies, Somerville and Pruitt (2004) recommended the use of quantitative measures in physical habitat assessments, where practicable, to limit observer bias as much as possible.

The following subsections provide methods for measuring physical habitat parameters in headwater streams. We have attempted to explain the ecological relevance of each parameter and keep the methods as straightforward as possible. Headwater streams may be remote from roads or even hiking trails, so many of the methods described in the following sections use minimal equipment. Rather than providing a single method for measuring a parameter, we have attempted to include multiple methods from which the reader can choose based on her/his particular needs and situations.

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3.1 Designating hydrologic condition for stream reaches

General

This subsection provides guidance for rapidly designating hydrologic condition in headwater stream reaches. The categories of hydrologic condition (discussed in detail below) represent the degree of departure from a spatially-continuous flow (or conversely, a completely dry condition) at a given point in time and space. These designations describe the level of connectivity or fragmentation of the aquatic phase in headwater streams (Boulton 2003). The degree of hydrologic connectivity is fundamental in controlling the structure and function of headwater streams because it affects physicochemical properties, biotic dispersal, and refuge availability (e.g., Boulton and Lake 1990, Dietrich and Anderson 1998, Maltchik et al. 1994).

Hydrology of headwater stream reaches may follow a predictable sequence of hydrologic conditions related to seasonal (and/or greater time frames) fluctuations in precipitation and evapotranspiration. Shannon et al. (2002) described hydrologic conditions in arid ephemeral channels that occur at lower frequencies than would occur in more humid regions. At a given time, the hydrologic condition also varies spatially within and among headwater streams associated with differences in distance to the groundwater table, watershed vegetation, groundwater storage capacity, etc.

The hydrologic designations discussed here differ from those that represent general flow regimes over time (e.g., perennial, intermittent and ephemeral hydrology, Uys and O’Keeffe 1997). However, in the absence of continuous monitoring of hydrologic condition,

designation of hydrologic conditions at least once during wet and dry seasons may provide a simple method for identifying flow regime types.

Procedure

Hydrologic condition is determined by visually assessing surface water connectivity and water velocity within the thalweg of the study reach. Designation should be based upon the predominant hydrologic condition within the study reach. Mark the appropriate box on the field forms for the hydrologic condition identified (Figure 3-1).

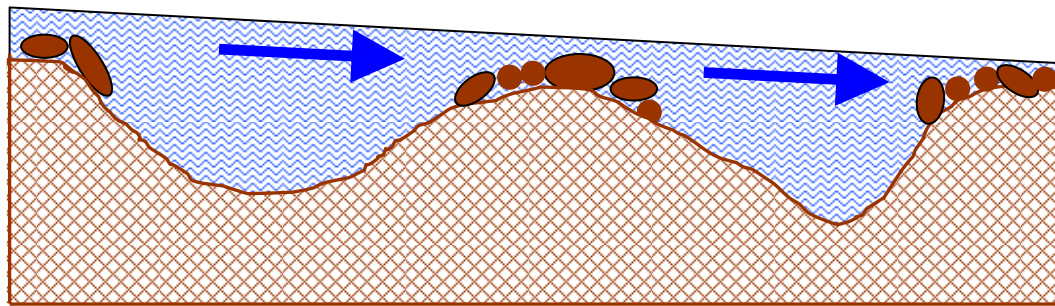
STUDY REACH HYDROLOGIC CONDITION
<input type="checkbox"/> Surface flow continuous (4)
<input type="checkbox"/> Flow only interstitial (3)
<input type="checkbox"/> Surface water present but no visible flow (2)
<input type="checkbox"/> Surface water in pools only (1)
<input type="checkbox"/> No surface water (0)

Figure 3-1 Appropriate location for recording hydrologic condition on page 1 of field forms.

The text below describes five categories of hydrologic condition seen in headwater streams. Each category is represented by photos and a diagram showing a longitudinal section along the channel thalweg. Blue shading indicates surface water, arrows indicate presence and direction of visible flow, coarse stone substrate on the streambed is represented by solid brown, and the hatched brown areas indicated finer streambed substrate and underlying geology. The term “habitat units” refers to riffles and pools, the dominant habitat types in headwater streams. The five hydrologic categories and their numerical descriptors are as follows:

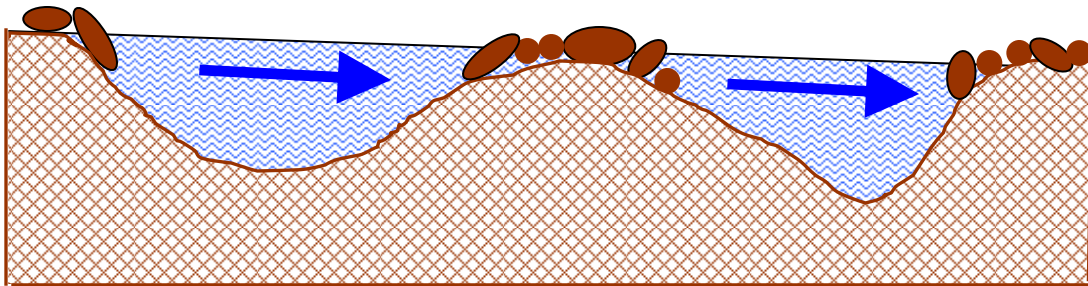
- **Visible surface flow continuous (4):**
Surface water is flowing and uninterrupted between habitat units

and flowing. Most of the streambed stones within the thalweg are submerged.



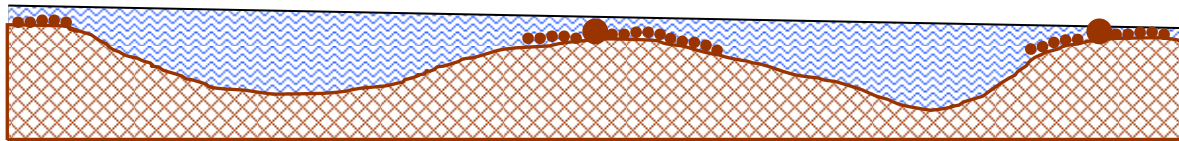
- **Visible flow interstitial (3):** Surface water is interrupted between habitat units, such that the majority of streambed stones in shallow habitat units (i.e., riffles) are exposed. However, interstitial flow connecting habitat units is evident as trickles or rivulets flowing between stones or visible

at the tail and heads of pools. Soluble tracers, such as fluorescein dye or NaCl solution may be added at the upstream end of a study reach and monitored downstream to determine if interstitial flow connects pools within a reach.



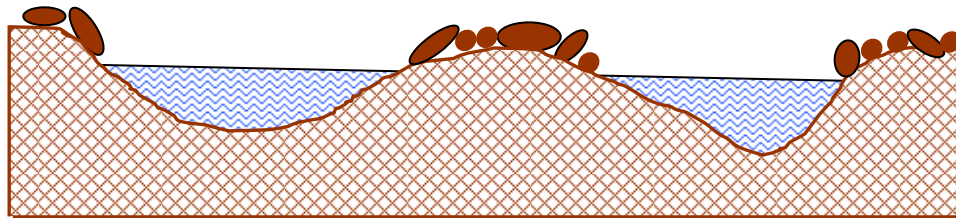
- **Surface water present but no visible flow (2):** Surface water is uninterrupted between habitat units, however there is no evidence that the water is flowing throughout study

reach. Water standing in pools may appear stagnant. This condition is likely to occur in low gradient headwater streams rather than in high gradient streams.

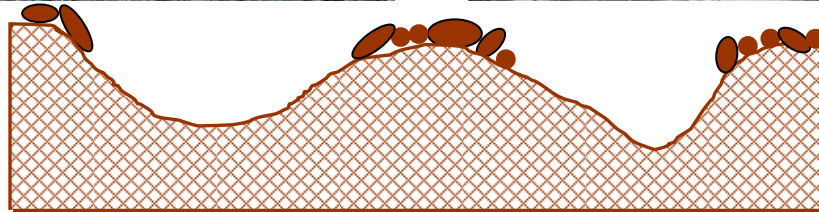


- **Surface water present in pools only (1):** Surface water is found only in pools and there is no visible water or

flow connecting pools. Stream bed sediments between pools may be moist.



No surface water (0): Surface water is absent from the channel thalweg.



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Equipment and supplies

Measuring tape (50 m)

Field forms

3.2 Continuous monitoring of hydrologic condition

General

This subsection describes a water sensor for continuously monitoring hydrologic condition (i.e., presence or absence of water) that is economical, light weight, and easy to install. The sensor described provides information regarding the timing, duration, and frequency of channel drying. Other methods such as float gages and pressure transducers with data loggers, which are widely used to continuously measure stream stage and subsequently, discharge (Rantz et al. 1982), also provide flow permanence data, but can be more costly and require more channel modification and maintenance.

Water sensors may be assembled by Intermountain Environmental, Inc (IEI)¹¹. The components of the water sensor include an Onset Hobo® state data logger, Onset submersible case, and an encased cable (see Figure. 3-2, pen shown for scale). The state data logger was designed to continuously record binary changes (i.e., open vs. closed; on vs. off). The modification by IEI has allowed this data logger to record the timing and frequency of changes in hydrology (in terms of presence and absence of water). When present, water completes the circuit between the two exposed copper wires on the contact end of the cable and sends a “closed” signal to the sensor. When a stream dries and water no longer is present to complete the circuit, the data logger records an “open” signal. The datalogger does not record on time intervals, and only records the time when a change of state occurs. The data logger can

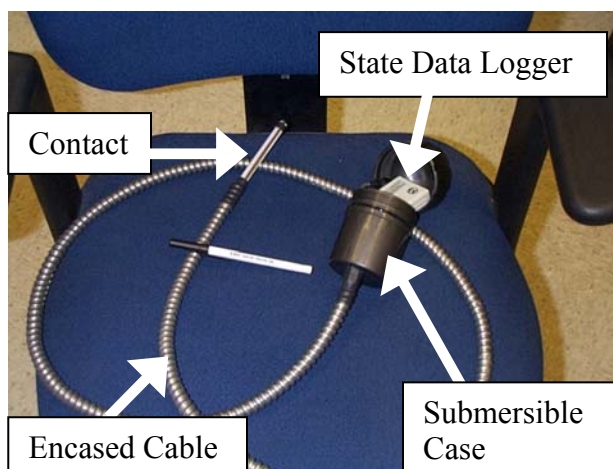


Figure 3-2 Primary components of a water sensor used to continuously monitor hydrologic condition.

record up to 2000 state changes (checking for changes of state every 0.5 seconds) and the battery will last approximately 1 year.

3.2.1 Launching and preparing for deployment

Procedure

Install the appropriate Onset software onto a personal computer. (Note that to launch data loggers via personal laptop computers Onset Boxcar Pro 4.3® or higher may be required.) Connect the PC interface cable to an open Com Port or serial port of the computer and the 3.5 mm jack of the data logger. Open the Onset Boxcar® program and either select Launch from the Logger menu or select the icon for launching on the tool bar. A launch dialog box should appear with setting options (Figure. 3-3). Note the condition of the battery; if it does not indicate that the battery is “good” then close the launch dialog box and change the battery (CR-2032 lithium). Under description, type locality information (e.g., Hoosier Natl. For.) and change text for “close” and “open” to “wet” and “dry”, respectively. Do not select “wrap around” (this overwrites data already stored when >2000 state changes occur) or “stealth mode” (this turns off

¹¹ Intermountain Environmental, Inc.
601 W. 1700 S. Suite B., Logan, UT 84321
(800) 948-6236¹

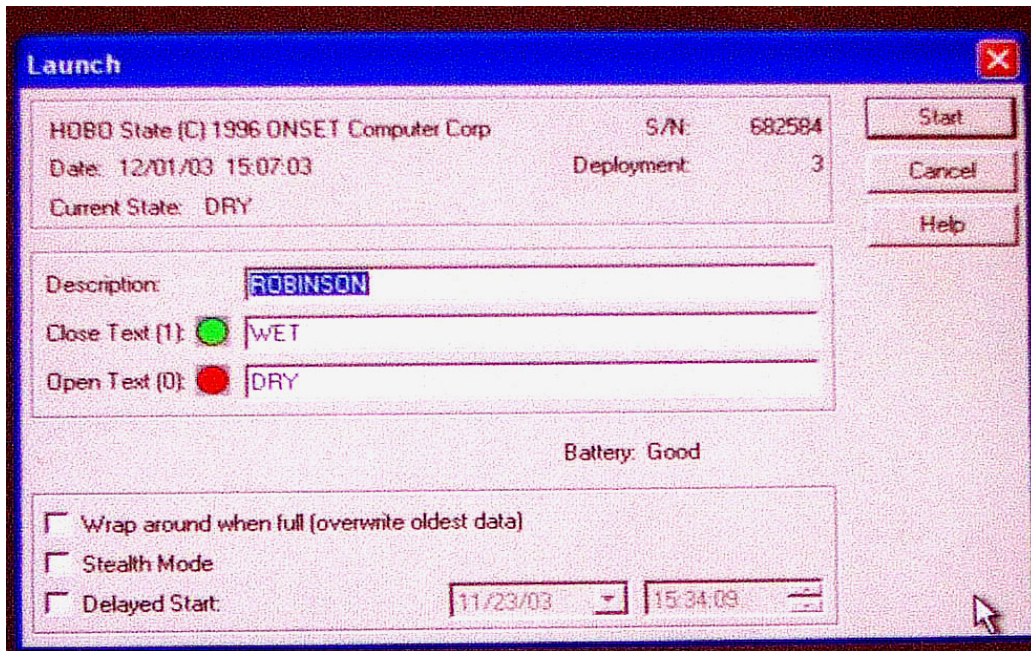


Figure 3-3 Launch dialog box for Onset Boxcar Pro.

indicator lights while data loggers are launched). Delayed setting may be selected, particularly when personal computers are not accessible near field sites and travel time to field sites may affect the battery life of data loggers. Select "start" and allow the launch progress bar to completely extend before disconnecting the data logger from the PC interface cable.

Note that the 6-digit serial number displayed by the BoxCar software matches the serial number printed on the data logger. Write this number on the outside of the submersible case using a permanent marker.

Note that the red LED (indicating "open" or "dry" state, see Figure 3-4) should be blinking if the data logger is properly launched. Place the data logger in its respective submersible case. Next connect the 2.5 mm cable to the 2.5 mm jack on side of data logger and place two desiccant packs inside the submersible case (Figure 3-4).

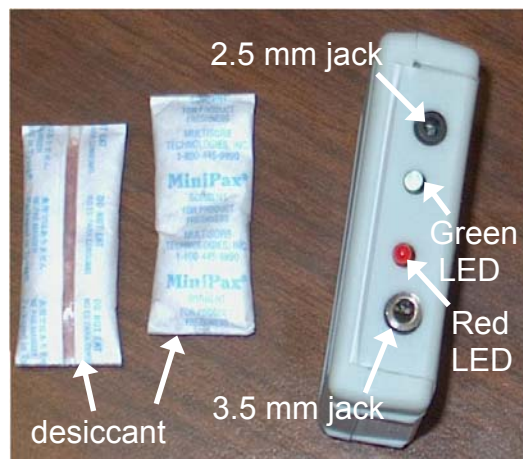


Figure 3-4 Desiccant packs and Onset Hobo® State data logger with jacks and LEDs shown.

Inspect the rubber O-ring and its seating on the submersible case (below the threads, Figure 3-5), making sure that the surfaces are clean and there are no cracks or damage to the O-ring. The O-ring should be replaced if there is cracking or damage. Lubricate the entire surface of the O-ring using the silicone compound by applying a thin, even coat.

Place the O-ring in its seating on the submersible case and screw on the submersible case cap. Ensure that the O-ring seats properly and does not extrude when screwing cap in place.

3.2.2 Deployment

Always make certain that the data logger stays dry. Record the location of the site using preferably GPS coordinates (e.g., latitude and longitude, UTM) or precise directions. These directions should include road names, compass headings, turn directions and distances.

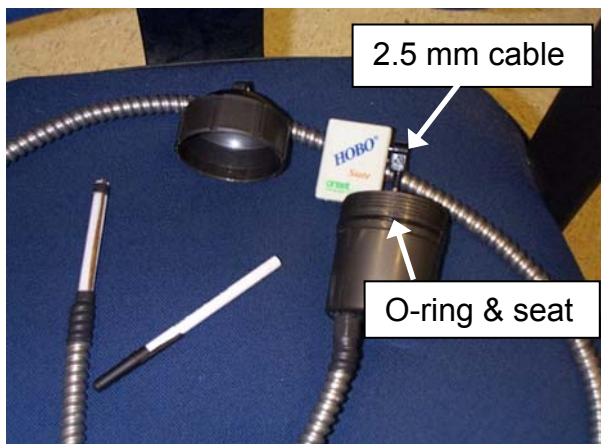


Figure 3-5 Water sensor with 2.5 mm cable, O-ring and seat shown.

Select a location within the channel thalweg that is the approximate average water depth of the thalweg for the entire 30 m reach. Where available, select a location that also has a steep bank; this will help to keep the data logger end of the water sensor dry during high flows. Use a small sledge hammer to drive a section of rebar or a stake into the stream bed. Make certain that the rebar is firmly embedded.

Select a water sensor with the appropriate encased cable length to extend from the streambed stake to a safe bank location. Assemble a stilling well by attaching PVC

pipe to a PVC cap (Figure. 3-6). Three to four holes should be drilled into the bottom of the cap to allow the water level within the stilling well to fluctuate with the stream water level (Figure. 3-6). This stilling well will prevent false readings associated with debris accumulating on the contact wires. The bottom of the stilling well is positioned so it is flush with the stream bed. An O-ring (#10, 1/2" inner diameter) may be used to seal out rain from the stilling well opening around the flexible cable housing. Place the contact end of the sensor inside the stilling well so that the contact wires are a few millimeters above the PVC cap.

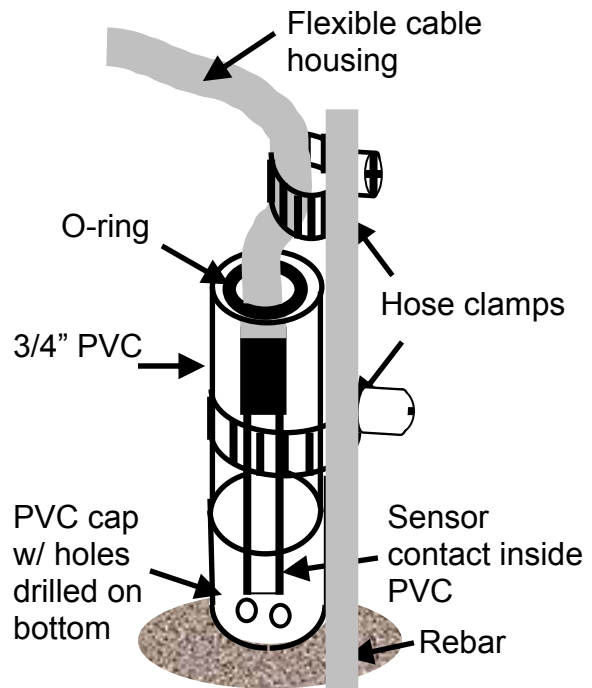


Figure 3-6 Schematic showing assembly of stilling well and contact end of water sensor.

Attach the contact end of the water sensor to the stake with 2 hose clamps or cable ties (above and around stilling well), making certain that they are tight (Figure 3-7). Extend the water sensor cable laterally to the bank, allowing the cable to conform to the contour of the channel and bank. Insert the second piece of rebar or stake by pounding it into the soil adjacent to the stream channel, making certain that it is firmly embedded.

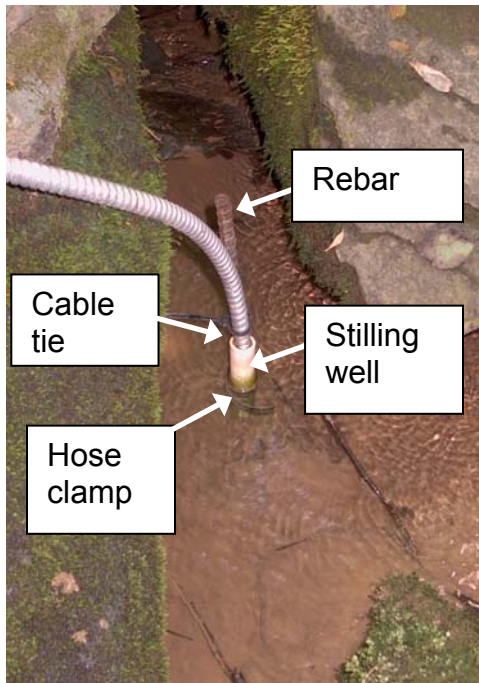


Figure 3-7 Water sensor securely attached to rebar above and below stilling well.

Attach the data logger end of the water sensor to the rebar with 2 cable ties. Gently place large cobble on top of cable to stabilize and camouflage the water sensor (Figure 3-8).

Unscrew the cap of the submersible case and note which light is blinking. Where the contact end is submersed in water the green LED (“closed” or “wet” state) should be blinking. If the contact end is not submersed in water the red LED (“open” or “dry” state)

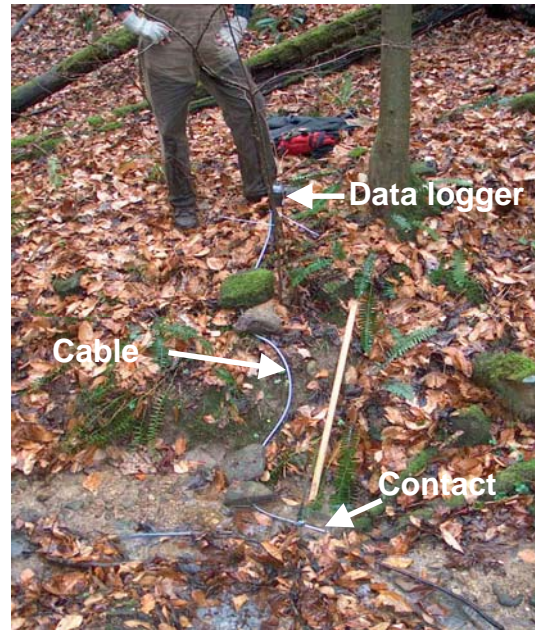


Figure 3-8 Water sensor positioned for continuous monitoring of hydrologic condition. Meter stick shown for scale.

should be blinking. If this is not occurring then check the connection of the 2.5 mm cable and jack. If this does not remedy the situation then replace the data logger or water sensor with another.

Record the data logger serial number, location, date, time, water depth (using a meter stick) and hydrologic condition for each sensor deployed. The predominant hydrologic condition of the study reach is categorized as: 1) visible surface flow continuous, 2) visible flow interstitial, 3) surface water present but no visible flow, 4) surface water in pools only and 5) no surface water.

3.2.3 Retrieval of the water sensor

Using the field sheets or notebook completed during deployment, return to the study reach within 1 year after the water sensor was installed. For each water sensor retrieved record the data logger serial number, location, date, time, water depth (using a meter stick) and hydrologic condition. Remove the rebar

and water sensor from the water. Use clippers to cut the cable ties to disconnect rebar pieces from the data logger and contact ends.

3.2.4 Transferring data

Remove data logger from submersible case and connect PC interface cable to 3.5 mm jack on data logger. Open the Onset Boxcar® program and either select “readout” from the Logger menu or select the icon for readout on the tool bar. You will then be asked to save the logger datafile (*.dtf). The serial number of the data logger and year should be used to name the file. For example, if the data logger serial number is 682537 and data were collected from 15 April to 23 September 2004 then the file is named “682537_04.dtf”. This will be a unique file name that can then be linked to field sheets or notebook for further site description. These files can then be saved within folders representing each stream.

From the File menu select “export” and the desired spreadsheet program (e.g., Microsoft Excel®, Lotus 1-2-3). You will then be asked to save the text file (*.txt). Use the same name given to the *.dtf file, but with the *.txt suffix.

Open the spreadsheet program and open a file containing water sensor data. The Text Import Wizard window should open. Select file type marked “delimited” and select “next”. In the next window select “tab” as the delimiter and select “finish”. This should then separate date + time, and hydrologic state into 2 separate columns. The number of data rows minus 1 should indicate the number of hydrologic state changes occurring over the period between launching and readout. Some of the state changes at the beginning and end of the data set may not represent the hydrologic changes at the study site. Using the date, time and hydrologic condition data from field sheets or notebook, the actual

starting and ending time is entered into the columns. When entering the starting and ending date and time, enter each into the spreadsheet as single cells with a space between the date and time. For example, if the water sensor was actually deployed at 1:24 pm on 15 April, enter the date & time as follows: 4/15/03 1:24 PM. Then highlight the cell and change its cell format to the “custom” category and the “mm:ss.0” type. Be sure to also enter the hydrologic state (“wet” or “dry”) for the starting and ending periods in the appropriate column. Below the cell that identifies the data logger serial number, enter the site name including stream name and site number.

To calculate the number of hours (duration) that had occurred between each state change use the following function in the column adjacent to the column containing hydrologic state labels. Type “= (A4-A5)*24. This example subtracts the date+time in cell A5 from a previous date+time in cell A4. Continue this down the column until the all durations are calculated. These can then be easily converted from hours to days by dividing the number of hours by 24. The total duration of dry or wetted condition can then be determined by summing every other cell within the column.

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Equipment and supplies

Water sensor
 Field notebook or field forms
 Pencil
 Map of area
 Metal stakes or rebar (2 per sensor)
 Mallet or small sledge hammer
 Watch
 Hose clamps or cable ties (4 per sensor)
 Personal computer (PC) with operating system that can support data logger software
 Onset software Boxcar® 3.0+ or any version of Boxcar® Pro
 PC interface cable (w/ 3.5 mm jack and serial port)
 Submersible case kit (rubber O-ring, 2 dessicant packs, and tube of silicone compound)
 Global Positioning System (GPS) unit
 Meter stick

3.3 Identifying the channel head

General

This subsection provides instructions for identifying and recording the location of the channel head or channel origin of streams. Headwater streams link valley hillslopes to downstream water bodies through the downstream transfer of sediment and organic matter (Gomi et al. 2002, Hutchens and Wallace 2002). The channel head or origin is the upstream boundary between hillslopes and channels in the landscape, specifically between the valley head and channel. Characteristics of the surrounding valley (e.g., slope, geology and land use) determine the evolution of channels and therefore the location of channel heads (Dietrich and Dunne 1993, Montgomery 1999). The channel head rarely extends to the valley divide, so the valley network envelopes the channel network. Swales, hollows, and zero-order basins are other names used to describe hillslope landforms that drain into channel heads. These are located upslope from

channel heads (Dietrich and Dunne 1993). The transition from hillslope to channel may be abrupt, in the form of headcut or step, or gradual (Figure 3-9). The channels emerging from zero-order basins have been called transitional channels and are often ephemeral or intermittent (Gomi et al. 2002).

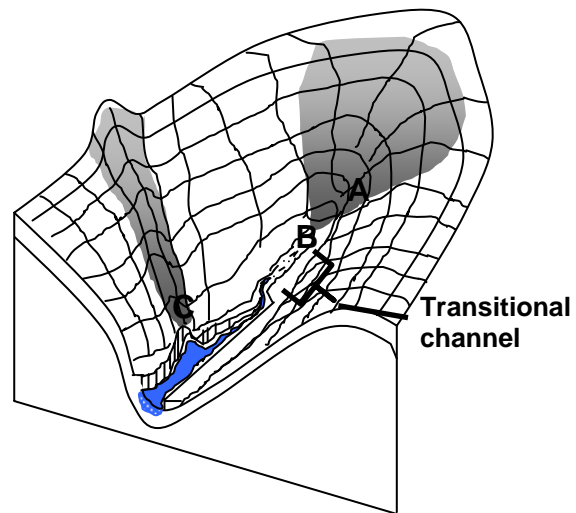


Figure 3-9 Drawing showing a valley hillslope (swale or hollow) relative to channel. Valley head (A), gradual (B) and abrupt (C) channel heads are identified. Gray areas indicate zero-order basins draining into channel heads. Redrawn from Dietrich and Dunne (1993).

The following procedure will provide field characteristics that can be used regardless of hydrologic status of a site. The observation of surface flow may not be the best indicator when defining whether a landform is or is not a channel. Surface runoff (Horton overland flow) and throughflow-return flow may be apparent on hillslopes, and are thus, not restricted by channel formation (Fetter 1988, Dietrich and Dunne 1993). Additionally, the distribution of surface flow in stream networks expands and contracts with water table fluctuations (Blythe and Rodda 1973, Stanley et al. 1997). Hydrologic permanence at the channel head may be dependent upon

underlying geology and connectivity to groundwater. Springs or seeps originating from contact zones, faults, joints and fractures in the underlying geology can coincide with and/or control channel head location (Higgins and Coates 1990). The flow of these springs may be continuous or discontinuous over time.

The resolution of most topographic maps is too low to reveal the extent of headwater channels (e.g., Hansen 2001). Therefore, the terminations of blue lines (e.g. on USGS 1:24 000 quads) do not accurately represent channel heads (Mark 1983). Typically channel heads are located upslope from blue line terminations, extending into the contour line crenulations (see Figure 2-2).

The location of the channel head is recorded once for a given stream during the study because it is unlikely to change significantly over the timeframe of most monitoring studies (1-2 years). However, channel head location can shift depending upon characteristics of the surrounding hillslope (e.g., gradient, soil cohesiveness, land use) and stochastic events (e.g., mass failures). The channel head is a particularly sensitive feature in arid and semi-arid landscapes, where gully erosion caused by unstable channel heads is a serious socio-economic and environmental problem (Bull and Kirkby 2002). Infilling by debris flows and landslides can move the channel head downslope, whereas gully erosion or headcutting moves the channel head upslope (Benda and Dunne 1987, Miller et al. 2003). Therefore, over long time frames (10s to 100s of years), the position of the channel head may fluctuate in response to these processes.

Procedure

Hike the channel upstream of the “ephemeral” or upstream-most study reach (see Section 2.1 for description of study reach selection). You should focus on characteristics of the

streambed and banks relative to the adjacent hillslope. The phrase “definable bed and banks” is often used to determine if a land form is a stream channel. Problematically, this phrase is not easily defined in objective terms although along larger streams and rivers it is visibly obvious. A channel is a landform that conveys water and sediment between banks. Banks are relatively narrow zones that have steeper gradients than adjacent hillslopes and the transverse slope of the channel bed (Dietrich and Dunne 1993).

Characteristics of abrupt channel heads

Abrupt channel heads appear as steep vertical steps from the valley head down to the channel (Figure 3-10). These abrupt steps are also known as “knickpoints” or “headcuts”. No evidence of bank or channel forms is usually visible above abrupt channel heads.



Figure 3-10 An abrupt channel head in Wayne National Forest, OH.

Thus, the abrupt channel head represents a distinct start of continuous streambed and banks in the downstream direction. These abrupt changes often correspond to differences in surface sediment between the valley head and channel. Surface sediments above the valley head will be of colluvial origin (e.g., transported by gravity from adjacent hillslopes) and/or have soil nature (e.g., humus layer). In contrast surface sediment in the channel will be of a mixture of recently deposited colluvium and weathered material exposed from surface flow (e.g., bedrock and boulders). Vegetation type and density may also differ up- and downslope of the channel head. Terrestrial vegetation may be sparse or absent in the channel below the channel head compared to the upstream valley head. Be aware that steep vertical steps and headcuts are not restricted to channel heads and may occur within continuous channels. In this case, definable bed and banks are clearly evident upstream of the headcut (see Section 3.4). Record coordinates (latitude & longitude) and description of the hydrologic condition at the channel head in the Notes section for the datasheet of the nearest study

reach.

Characteristics of gradual channel heads

Gradual channel heads are less distinct than abrupt channel heads. These are characterized by a more gradual or discontinuous transition in bank and bed features, rather than the obvious boundary of a step or headcut. As you approach the channel head, the height and angle of the banks decline. The defined bed and banks are often discontinuous and may be interrupted by debris dams, tree roots, or bedrock outcropping. For the purposes of this study, we define the channel head as the point where the channel no longer has continuous defined bed and banks. Be aware that steep channels can have a step-pool or cascade structure and appear less continuous than riffle-pool reaches (Church 1992). Banks typically are less well defined at the “steps” compared with “pools” in these streams. However, the channels should be considered continuous if the steps are composed of visibly eroded material exposed from surface flow (e.g., bedrock and boulder) that may or may not be covered with moss and organic debris piles (Figure 3-11).

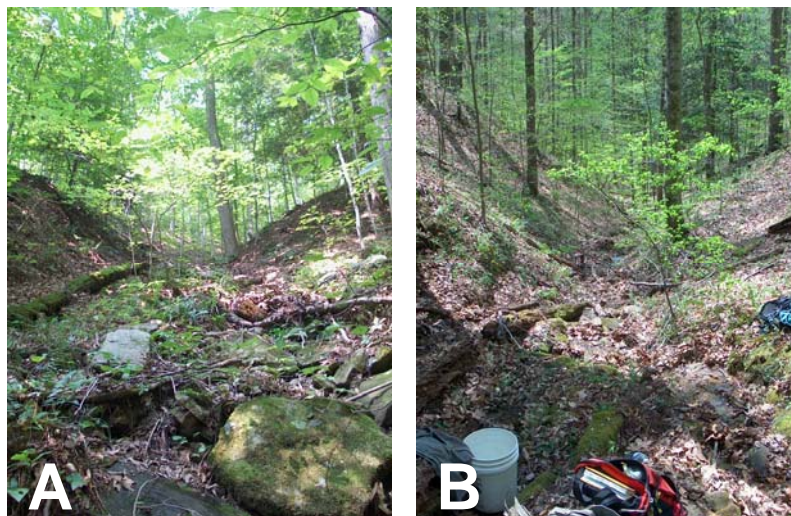


Figure 3-11 Views from gradual channel heads in east-central Kentucky. A) Looking upslope toward the valley head from the channel head position. B) Looking downslope at the cascade structure of the transitional channel.

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Equipment and supplies

- GPS unit
- Topographic map
- Field forms

3.4 Identifying channel headcuts

General

This section provides instructions for the identification of channel headcuts in

headwater streams. Headcuts are abrupt changes in streambed elevation (i.e., knickpoint) that migrate in an upstream direction (Leopold et al. 1964). This migration is a natural geomorphic process that is often accelerated due to human modification of the channel and/or surrounding watershed (Patrick et al. 1994, Montgomery 1999). The upstream migration of headcuts results in downcutting (i.e., degradation) of the streambed and incised channel morphology (Galay 1983, Simon 1989). Among the ecological effects downstream of headcuts may be loss of streamside vegetation, scoured streambeds, decreased sinuosity, and temporary increase in downstream sedimentation (Patrick et al. 1994). Headcuts can also influence the connectivity along headwater streams by steep changes in streambed elevation and hydrology. Abrupt changes in summer baseflow hydrology (and water temperature) occur at headcuts and are related to differences in distance from the groundwater table. As the summer groundwater table lowers (lower precipitation, higher evapotranspiration), it falls below the

streambed upstream of the headcut before dropping below the stream bed downstream of the headcut. This causes flow to remain for longer periods downstream (often perennially) than upstream of headcuts. The presence of headcuts is determined once for a given reach during the study because their presence is unlikely to change significantly over short time periods (e.g., 1-2 y), however any upstream advance should be noted.

Procedure

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. Survey the study reach for abrupt changes in streambed elevation. If a knickpoint is located, determine first whether the formation is simply a natural grade control point (e.g., large boulders, bedrock outcrops, or large woody debris). If it is not, then look for the following: 1) undercutting beneath the headcut face or headwall (Figure 3-12), 2) seepage or piping from the headwall, and 3) alluvial fan or deposits in the channel downstream of the headcut. Be aware that headcuts may stall their upstream migration at grade control features between large floods.

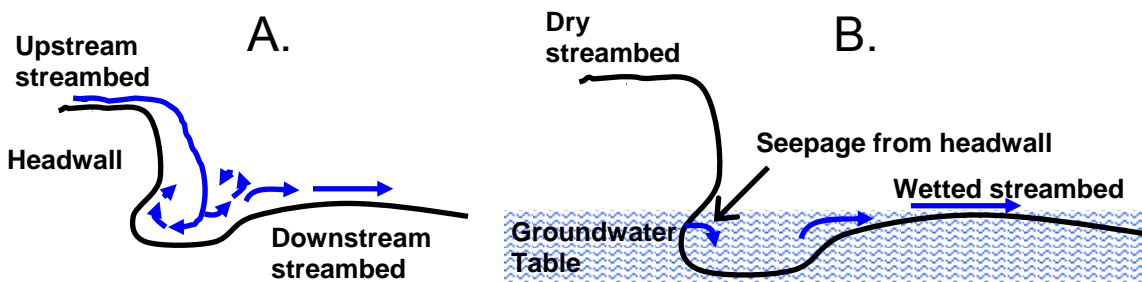


Figure 3-12 Longitudinal view of a headcut, (A.) Blue arrows illustrate flowpaths that lead to undercutting, failure of the headwall, and eventually upstream migration of the headcut; (B.) Abrupt change in summer baseflow hydrology at a headcut.

Indicate on the field form (Figure. 3-13) the presence or absence of a headcut within the study reach. Note location of headcut on study reach drawing and make notes characterizing the formation. Photographs of

headwater streams with headcut formations are shown in Figures 3-14, 3-15 and 3-16.

PRESENCE OF HEADCUT IN REACH		ALGAL COVER INDEX					# CORES FOR SUBSTRATE MOISTURE (depositional)	
Y	N	1	1 ^{1/2}	2	3	4	5	

Figure 3-13 Portion of page 1 of field forms showing the cell for recording presence of channel headcuts.



Figure 3-14 Subtle headcut in Falling Rock Creek in east-central KY (looking upstream).



Figure 3-15 Huge headcut (~2 m change in bed elevation) in an unnamed stream in Athens, GA (looking upstream).



Figure 3-16 Headcut in Taylor Branch in south-central IN (looking downstream), where streambed elevation at the arrow was ~ 1 m higher than streambed below headcut at the yellow circle.

References

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- Patrick, D.M., S.T. Ross, and P.D. Hartfield. 1994. Fluvial geomorphic considerations in the management and stewardship of fluvial ecosystems. Pages 90-99 in *Riparian ecosystems in the humid U.S.: Functions, Values and Management*. National Association of Conservation Districts, Washington, D.C.
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Equipment and supplies

Measuring tape
Field form

3.5 Measuring channel sinuosity

General

This section provides instructions for rapidly scoring channel sinuosity of headwater streams. This procedure is similar to that used by the Ohio Environmental Protection Agency (Ohio EPA 2002), where sinuosity is described as the number of well-defined bends or meanders over a distance of stream channel. This differs from the more quantitative measure, sinuosity index, which is the ratio of the channel thalweg distance to the downvalley distance (Gordon et al. 1992, Platts et al. 1983, Rosgen 1996). In association with other measures (e.g., channel slope, substrate particle size), sinuosity provides useful information regarding the degree of channel modification to headwater streams. Retention of nutrients and organic matter increases with increasing sinuosity, ensuring transformations that may be beneficial for downstream waters (e.g., Gücker and Boëchat 2004, Muotka and Laasonen 2002). Sinuosity is measured once for a given reach during the study because it is unlikely to change significantly over short time periods (e.g., 1–2 years).

Procedure

Delineate the 30-m study reach so the Measuring tape is positioned along the thalweg. Sinuosity is based on the number of well-defined bends over the 30-m study reach (approximately 20X the bankfull width of most headwater streams). Examples showing various degrees of sinuosity are shown in Figure 3-17. On the first page of the field form indicate the sinuosity in the appropriate cell (Figure 3-18).

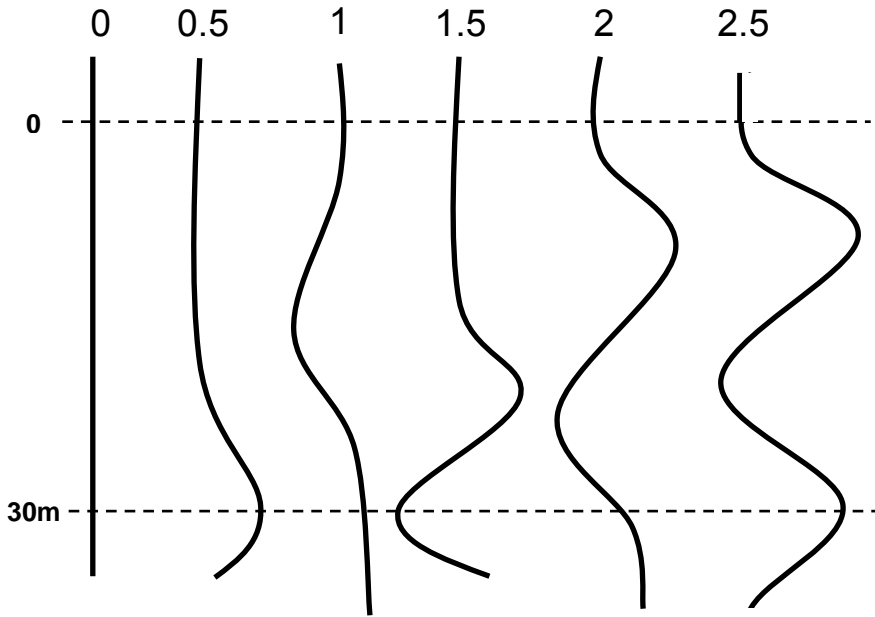


Figure 3-17 Examples of stream channels varying in sinuosity (number of bends) along 30-m study reaches.

MAX. POOL DEPTH (cm)	DEPTH TO BEDROCK / GROUNDWATER (m) (3 measures in depositional habitat)	SINUOSITY (number of bends)

Figure 3-18 Portion of page 1 of field forms showing the cell for recording channel sinuosity.

References

Gordon, N.D., T.A. McMahon, and B.L. Finlayson. 1992. *Stream Hydrology: An Introduction for Ecologists*. John Wiley & Sons, Chichester, United Kingdom.

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http://www.epa.state.oh.us/dsw/wqs/headwaters/PHWHManual_2002_102402.pdf

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Equipment and supplies

Measuring tape

Field forms

3.6 Designating habitat units

General

This subsection provides instructions for identifying habitat or channel units within headwater stream reaches. Habitat units (or “meso-habitats”) are distinct channel units having characteristic physical properties. They are smaller than stream reaches and larger than microhabitats, according to the hierarchical levels used to describe the physical template of streams (Frissell et al. 1986). Within headwater streams with moderate to high gradient (slope $\geq 2\%$), habitat units can range from <1 to 10 m in linear stream length (K. M. Fritz, personal observation). Habitat units in sandy, low-gradient or bedrock-dominated channels may be > 10 m long. These units are found longitudinally along the channel and may be spaced at fairly regular intervals along a stream reach (Leopold et al. 1964, Beschta and Platts 1986). Habitat units are delimited by elevational and lateral changes of the streambed (Hawkins et al. 1993). This is particularly evident in streams where the streambed particles are not primarily sand or silt (Leopold et al. 1964). Associated with these distinct channel units are characteristic water flow and depth regimes. Therefore, physical variation within a study reach can be accounted by the proportions of these habitat

types. In many instances these characteristics lead to differences in the dominant streambed particle sizes among types of habitat units.

Assessment and restoration of streams are typically limited to the reach scale. However, for logistical reasons, biological communities are often sampled at spatial scales below the reach level (Cuffney et al. 1993, Lazorchak et al. 1998, Barbour et al. 1999), often stratified by habitat type. Inter-habitat variability in ecological measures can exceed variation seen among reaches or streams (e.g., Angradi 1996, Rabeni et al. 2002). Therefore, quantifying the extent of habitat types within stream reaches is fundamental to understanding the ecological status of water resources at larger spatial scales, not because of the inherent measurement of habitat units (Poole et al. 1997) but to put other measures in context for comparison.

The number of the physical parameters needed for designating habitat types increases as classification become more complex. The utility of a complex classification becomes limited because the variety of habitat types that can be identified within stream reaches can vary greatly among regions. To be useful, the categories of habitat type need to be applicable for all reaches examined in a study. In addition, as the specificity of habitat types increases there is typically a greater level of subjectivity involved in their designation (Roper and Scarnecchia 1995). The following procedure provides guidance to delimit the most basic categories of habitats within headwater streams (see Hawkins et al. 1993 and Lazorchak et al. 1998 for descriptions of finer levels of habitat types). These include erosional and depositional habitats (Moon 1939). Erosional habitats are identified as shallow areas with rapid flow and typically coarse streambed substrate. They include such habitats as riffles, fast runs, sheets,

cascades and steps (in step-pool reaches). In contrast, depositional habitats are deeper areas with little or no visible flow and typically have fine streambed substrates but may also be bedrock. They include such habitats as pools and slow runs. Because water flow and depth are primary parameters used to designate habitat type and these can vary seasonally, this procedure should be carried out during each sampling period.

Procedure

Delineate the 30-m study reach so that the measuring tape is marking transects along the thalweg from downstream to upstream. At each meter mark along the thalweg of the study reach (0, 1, 2,...30m) assess water flow, water depth and substrate type to designate whether the habitat is erosional or depositional. The dotted line represents the study reach thalweg and the black arrow is pointing in the direction of flow in Figure 3-19.

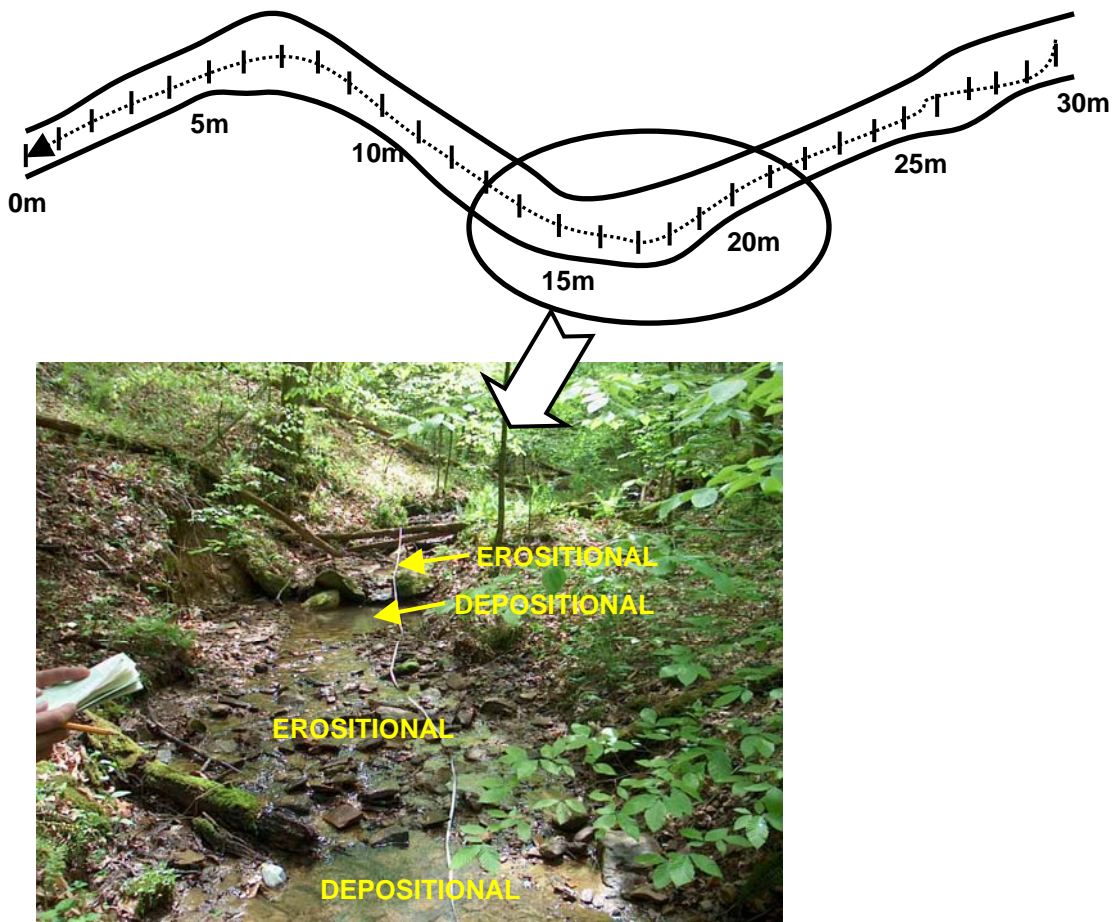


Figure 3-19 Plan view of study reach (top) and picture showing series of alternating erosional and depositional habitats along a headwater stream.

On the second page of the field form mark the habitat type for each transect (Figure 3-20). There is also a column on the field form for notes concerning the presence of large woody

debris (LWD, diameter ≥ 10 cm), leaf packs, bryophytes, herbaceous vegetation, etc. within the thalweg at that meter mark (Figure 3-20).

The designation of habitat type relies more on the streambed characteristics where the stream is dry. Substrate size, streambed elevation and

the distribution of organic matter are useful in determining habitat type at locations along dry channels.

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF Page 2 of 4									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0			E	Leafpack					
1			E						
2			D	LWD					
3			D						

Figure 3-20 Appropriate location for recording habitat units and notes on Page 2 of the Field Forms.

References

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Cuffney, T.F., M.E. Gurtz, and M.R. Meador. 1993. *Methods for Collecting Benthic Invertebrate Samples as Part of the National Water Quality Assessment Program*. U.S. Geological Survey Open File Report 93-406.

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Frissell, C.R., W.J. Liss, C.E. Warren, and M.D. Hurley. A hierarchical framework for stream habitat classification: viewing streams in a watershed context. *Environmental Management* 10:199-214.

Hawkins, C.P., J.L. Kershner, P.A. Bisson, M.D. Bryant, L.M. Decker, S.V. Gregory, D.A. McCullough, C.K. Overton, G.H. Reeves, R.J. Steedman, and M.K. Young. 1993. A hierarchical approach to classifying stream habitat features. *Fisheries* 18:3-12.

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Leopold, L.B., M.G. Wolman, and J.P. Miller. 1964. *Fluvial Processes in Geomorphology*. W.H. Freeman & Co. San Francisco, CA, USA.

Moon, H. P. 1939. Aspect of the ecology of aquatic insects. *Transactions of the Society for British Entomology* 6:39-49.

Poole, G.C., C.A. Frissell, and S.C. Ralph. 1997. In-stream habitat unit classification: inadequacies for monitoring and some consequences for management. *Journal of the American Water Resources Association* 33:879-896.

Rabeni, C.F., K. E. Doisy, and D.L. Galat. 2002. Testing the biological basis of a stream habitat classification using benthic invertebrates. *Ecological Applications* 12:782-796.

Roper, B.B. and D.L. Scarnecchia. 1995. Observer variability in classifying habitat types in stream surveys. *North American Journal of Fisheries Management* 15:49-53.

Equipment and supplies

Measuring tape
Field forms

3.7 Measuring channel slope

General

The following subsection provides methods for measuring channel slope or gradient in headwater streams. Channel slope is the drop in elevation per unit length of channel (“rise-over-run”, Figure 3-21). Slope is an important variable because it determines the velocity, stream power, and tractive forces which shape channel morphology and control export of sediment and organic matter. Measurement of slope can range in spatial scale, generally losing resolution with increasing spatial extent. Slope can be determined either at the streambed or water surface. The following procedure describes the estimation of slope for the streambed along the study reach thalweg. Slope is measured once for a given reach during the study because it is unlikely to change significantly over short time periods (e.g., 1–2 years). This procedure will require 1–2 field crew members to perform depending on the method chosen.

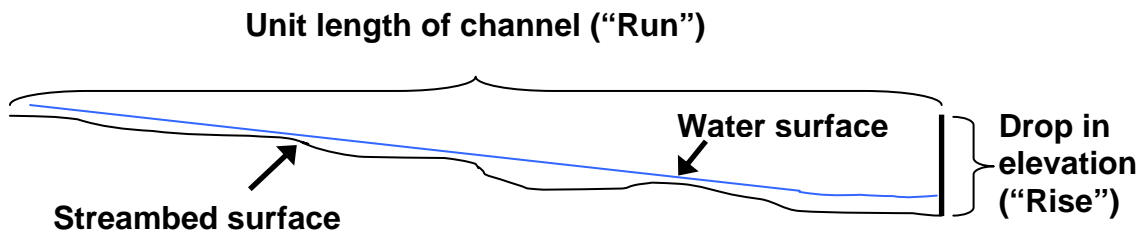


Figure 3-21 Longitudinal section of channel.

Procedure

Delineate the 30-m study reach so that a measuring tape is marking locations along the

thalweg. Slope is measured at 10-m intervals (at 0-10, 10-20, and 20-30 m marks) along the study reach (Figure 3-22).

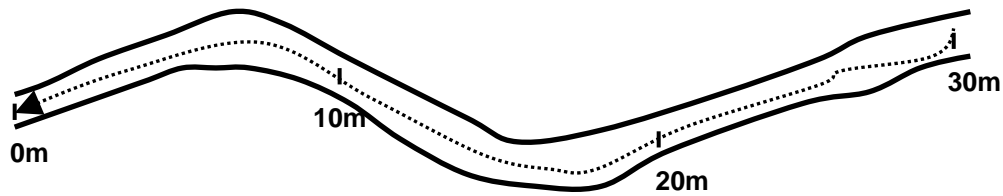


Figure 3-22 Plan view of study reach showing measurement locations (vertical black tick marks) for channel slope. Flow is from right to left and the dotted line represents the thalweg.

3.7.1 Measuring slope with a clinometer and stadia rod

The procedure requires one person holding the stadia rod and another person (“viewer”) viewing the stadia rod through the clinometer. While standing on level ground, mark the stadia rod at the viewer’s eye level with brightly colored flagging. This will be the target for the viewer when measuring slope. Make sure the viewer’s posture is the same (stand-up straight and flat footed) when marking the stadia rod and when taking measurements. Alternatively, the clinometer may be positioned at a set height (top of meter stick or hiking pole), rather than held by an observer. The target height on stadia rod would then be flagged at the same set height.

The viewer stands at the 0-m mark in the thalweg, whereas the person holding the stadia rod stands at the 10-m mark in the thalweg (Figure 3-23). The stadia rod should be held perpendicular to the streambed at the 10-m mark. To standardize for differences in thalweg depth the viewer and the stadia rod should be positioned at the same water depth (e.g., level with surface of water; see Kaufmann and Robison 1998). However, this difference is often negligible when all three slope measurements along the reach are averaged.

The viewer looks through the clinometer with one eye and at the stadia rod with the other



Figure 3-23 Crew members measuring slope of intermittent stream.

eye. Allow the images to appear to be superimposed on each other and position the horizontal center line of the clinometer level with the marking on the stadia rod (Figure 3-24). Avoid covering side window of clinometer with your hand while viewing. This window allows light through, enabling you to read values. There are two scales along the measurement wheel: degrees and percentages. The percentage scale is on the right side of the measurement wheel of most

clinometers. Tip your head up while viewing through the clinometer to see unit markings (e.g., %) and determine which side is the percentage scale. Slope measurements are recorded in percentages (to the nearest 0.5%) on the datasheet (Figure 3-25). Repeat the procedure for 10-20 and 20-30 m intervals along the reach thalweg.

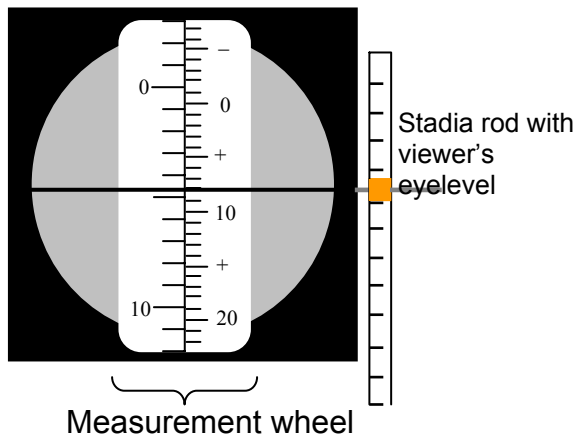


Figure 3-24 Superimposed views through clinometer and at stadia rod. Example shows percent scale on right side and degrees scale on left side of measurement wheel.

CHANNEL SLOPE (%) (for three 10 m sections of study reach)		
4.5	10	8

Figure 3-25 Portion of page 1 of field forms showing cells for percent slope values.

Conversion between percent and degrees can be done using:

$$\text{degree slope} = \tan^{-1} (\text{percent slope} / 100)$$

$$\text{percent slope} = (\tan (\text{degree slope})) \times 100$$

Modifications to the procedure can accommodate the use of alternatives to a

clinometer for measuring slope (e.g., Abney level, theodolite, total station; see Gordon et al. 1992). This procedure can be modified to measure water slope by simply accounting for differences in water depth (or ensuring equal water depth) at the stadia rod and where the viewer is standing.

3.7.2 Hydrostatic (manometer) measurement of slope

Position stakes at the 0 and 10-m marks along the thalweg. Fill vinyl tube with water and ensure no air bubbles are trapped. Attach the ends of the vinyl tubing to the stakes and position the tubing along the thalweg of the streambed (Figure 3-26). Allow water level within the vinyl tubing to equilibrate. Using the meter stick, measure (in meters) the distance between the streambed and the water level (bottom of meniscus) within the vinyl tubing at both ends. Streambed slope (%) is $((h_2 - h_1) / L) \times 100$, where $L = 10$ m. Slope measurements are recorded in percentages on the datasheet (Figure 3-25). Repeat the procedure for 10-20 and 20-30-m intervals along the reach thalweg. An alternative to using rebar and clamps to hold the manometer in place is to have two people hold the ends of the manometer against meter sticks while taking measurements of h_1 and h_2 .

An advantage of this procedure is that it can be done without a clear line of view along the reach and it is more accurate than the clinometer method. A disadvantage is that water must be available for the manometer. Water slope can be determined by measuring the distance between the water level within the tube and the water surface (rather than the streambed surface) at both ends.

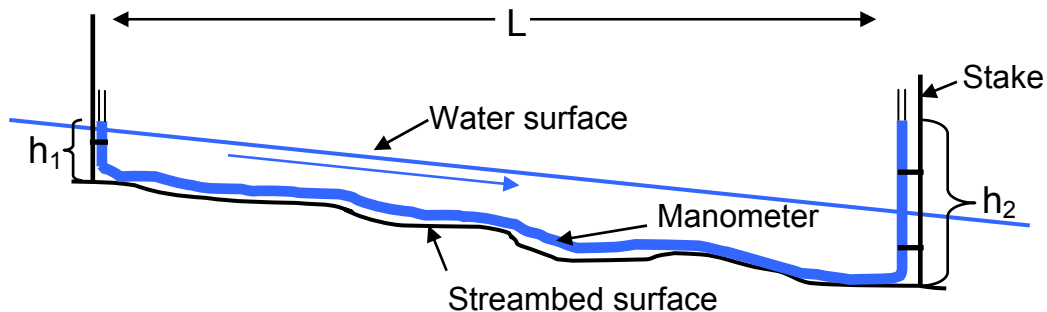


Figure 3-26 Longitudinal section of channel showing position of manometer and points of measurement to calculate slope (redrawn from Gordon et al. 1992). Blue arrow shows direction of flow. L = horizontal length, h_1 = height at the upstream end and h_2 = height at downstream end.

References

- Gordon, N.D., T.A. McMahon, and B.L. Finlayson. 1992. *Stream Hydrology: An Introduction for Ecologists*. John Wiley & Sons, Chichester, United Kingdom.
- Kaufmann, P. R. and E. G. Robison. 1998. Physical habitat characterization. Pages 77-118 in J.M. Lazorchak, D.J. Klemm, and D.V. Peck (editors). *Environmental Monitoring and Assessment Program – Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Wadeable Streams*. EPA/620/R-94/004F. U.S. Environmental Protection Agency, Washington, D.C., USA.

Equipment and supplies

- Measuring tape (50 m)
- Field forms
- A or B
- A. Stadia rod and clinometer – See Procedure 3.7.1
- B. Manometer (clear vinyl tubing, >10 m in length and ~10mm inner diameter), 2 survey

stakes or pieces of rebar, hose clamps, and meter stick – See Procedure 3.7.2

3.8 Measuring water depth

General

This subsection provides instructions for measuring water depth (including maximum) for reaches of headwater streams. Along with wetted width (next section), water depth is a critical measure of the extent of wetted habitat available and a measure of water persistence or susceptibility to terrestrial predators. Water depth is therefore important in governing the distribution of biota in headwater streams (e.g., Harvey and Stewart 1991, Taylor 1997). Because water depth can vary considerably over time, this procedure should be carried out during each sampling visit.

Procedure

3.8.1 Longitudinal thalweg measurements
A total of 31 measurements of water depth are taken along each study reach (Figure 3-27). Water depth is measured at the center of the thalweg (illustrated as dotted line in Figure 3-27) at meter intervals (i.e., 0, 1, 2...30 m).

The meter stick is positioned with zero-end down, side(s) with units facing perpendicular to the direction of flow and the stick held

perpendicular to the water level (Figure 3-27). Water depth measurements are recorded to the nearest 0.5 cm on the field form (Figure 3-28).

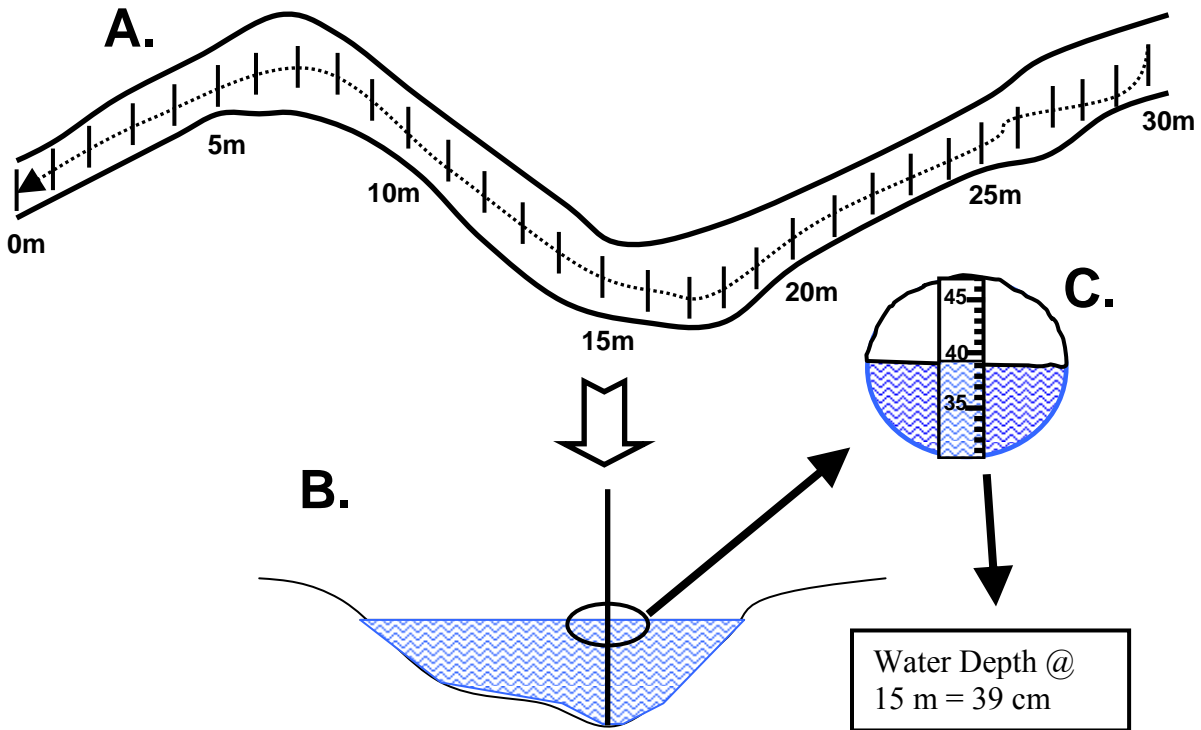


Figure 3-27 Overhead view of study reach showing locations for water depth measurement (vertical black tick marks) along the reach thalweg (dotted line). Water is flowing from right to left. (A.) overhead view of study reach (B.) channel cross-section, and (C.) lateral close-up of depth.

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF Page 2 of 4									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0		22							
1		18							
2		5.5							
3		1							
4		2.5							
5		3							
6		50							
7		30							
8		35.5							
9		0							
10		0							
11		3							
12		34							
13		12							
14		8							
15		39							

Figure 3-28 Appropriate location for recording longitudinal water depth measurements on page 2 of the field forms.

The water level on the meter stick is usually not perpendicular to the unit markings where the water velocity is fast (Figure. 3-29). Measurements should be taken at the middle of the meter stick, rather than at the upstream

or downstream-facing edges. Where there is no surface water present, zero water depth is recorded. Where there is surface water present, but it is less than 0.5 cm deep, “< 0.5 cm” should be recorded.

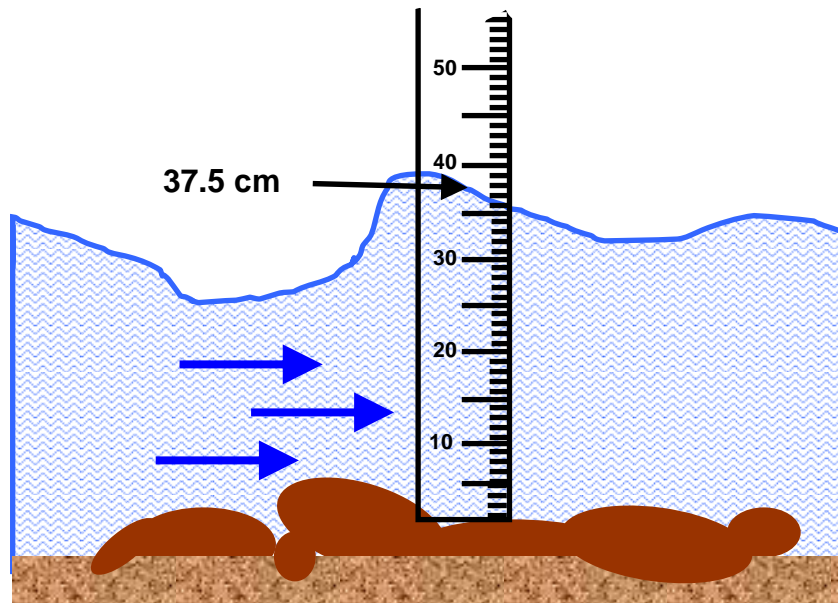


Figure 3-29 Schematic showing appropriate reading of water depth where water surface is turbulent.

3.8.2 Maximum water depth in study reach
 A single measurement is recorded for the greatest water depth within the study reach. This measurement is not restricted to the 31 (1-m interval) thalweg measurements.

Maximum water depth is recorded to the nearest 0.5 cm on the field form (Figure 3-30).

MAX. POOL DEPTH (cm)	DEPTH TO BEDROCK / GROUNDWATER (m) (3 measures in depositional habitat)		SINUOSITY (number of bends)
54	/	/	

Figure 3-30 Appropriate location for recording maximum pool depth measurement on page 1 of the field forms.

References

Harvey, B.C. and A.J. Stewart. 1991. Fish size and habitat depth relationships in headwater streams. *Oecologia* 87:336-342.

Taylor, C. M. 1997. Fish species richness and incidence patterns in isolated and connected stream pools: effects of pool volume and spatial position. *Oecologia* 110:560-566.

Equipment and supplies

- Meter stick (with at least 0.5cm increments)
- Measuring tape (50m)
- Field forms

3.9 Measuring wetted width

General

This subsection provides instructions for measuring wetted width in headwater streams. Wetted width (or top width) is the stream width at the surface water level (Figure 3-31) and is perpendicular to the channel direction. This measure (and water depth) describes the

extent of surface water habitat available within a study reach. Because wetted width can vary considerably over time, this procedure should be carried out during each sampling period.

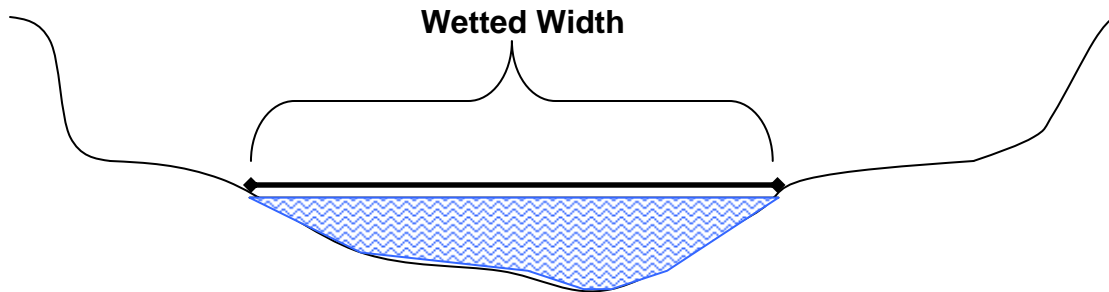


Figure 3-31 Channel cross-section illustrating wetted width.

Procedure

Delineate the 30-m study reach so that a measuring tape is marking locations along the

thalweg. Wetted width is measured at 5-m intervals (at 0, 5, 10, 15, 20, 25 and 30-m marks) along the study reach (Figure 3-32).

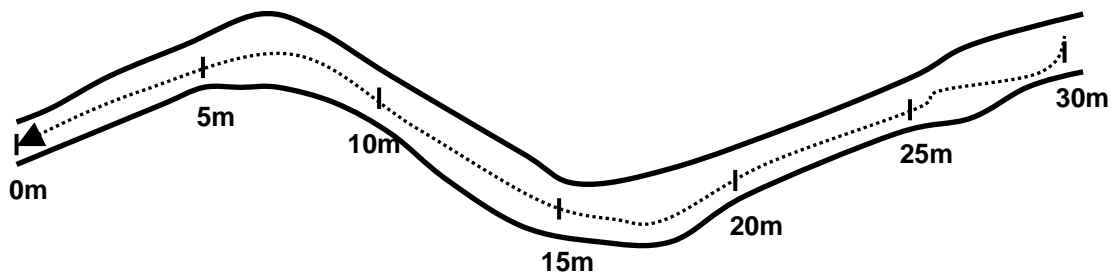


Figure 3-32 Overhead view of study reach showing measurement locations (vertical black tick marks for wetted width). Flow is from right to left and the dotted line represents the thalweg.

The meter stick can be used to measure wetted widths ≤ 1 m, whereas wider channels may require using a measuring tape (and a survey stake if done by one individual). At each location place the zero-end of the meter stick or tape at the water's edge on one side of the channel, position the measuring device

perpendicular to channel direction, and determine the distance to the water's edge on the other side of the channel. Record the distance to the nearest 0.01m in the appropriate cell on the field form (Figure 3-33).

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0						0.76			

Figure 3-33 Appropriate location for recording wetted width measurement on page 2 of the field forms.

If there is no surface water at a measurement location, indicate on the field form that the wetted width is 0 m. Where there are individual boulders or cobbles interrupting the surface water along the wetted width or there is visible interstitial flow (see Section 3.1), include the emergent particles in the measurement (Figure 3-34A). If there are

isolated pools along the channel edge (no surface connection to main channel) or the channel is braided (where there are vegetated islands or patches of emergent substrate) do not include width of isolated side-pools and islands in the wetted width measurement (Figure.3-34B, C).

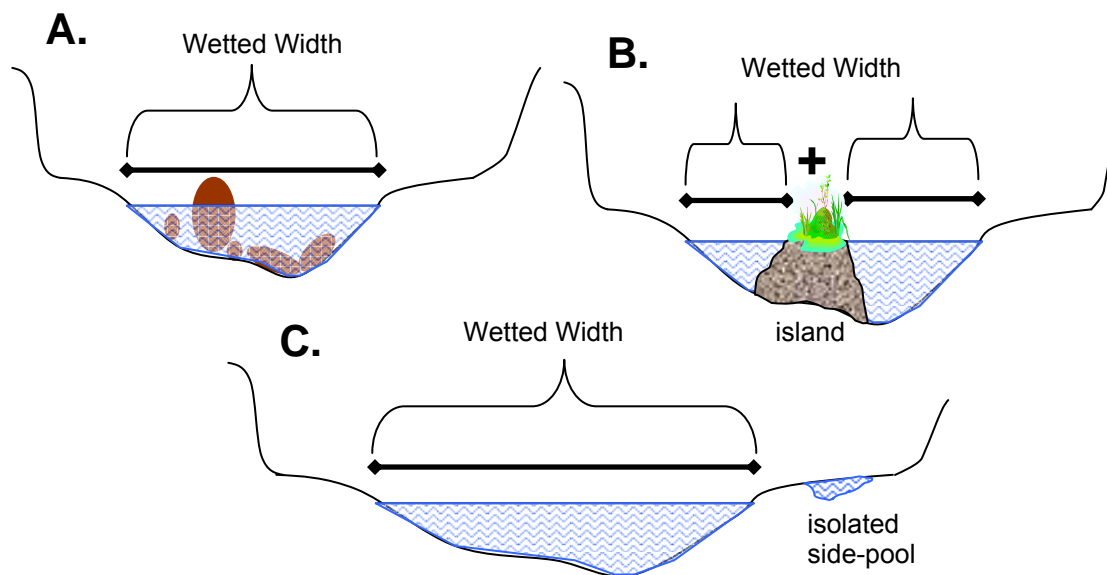


Figure 3-34 Channel cross-sections showing wetted width measurements where there is emergent cobble (A.), island (B.), and side-pool (C.).

Equipment and supplies
 2 Measuring tapes (50m)
 Meter stick

Survey stake (optional)
 Field forms

3.10 Measuring basic channel geomorphology

General

This section provides instructions for rapidly measuring basic channel form of headwater streams. Specifically, this section provides directions for measuring three channel parameters: bankfull width, bankfull depth, and flood-prone area width. The stream channel is composed of the banks and the streambed. The banks often have steeper gradient (in cross-section) and are often composed of finer sediments than the streambed (Figure 3-35). Bankfull discharge occurs when there is sufficient flow to fill the entire channel. This level is called bankfull stage and typically occurs once every 1-2 years. Bankfull width is the horizontal distance between the banks (perpendicular to flow) at bankfull stage. Bankfull depth is the

vertical distance between the streambed and the bankfull stage height at the thalweg. Flood-prone area width is the distance across the channel at a vertical level equaling 2X the bankfull depth. Entrenchment ratio is the ratio of the flood-prone area width to the bankfull width and is used to describe the degree of channel incision or “down-cutting” (Rosgen 1994, 1996). Channel dimensions vary with flow, the sediment being transported, and the material composition of the bed and banks. Channel geomorphology influences many structural and functional aspects in streams, including streambed substrates, organic matter retention, and biotic response to floods. The scouring forces of floods are dissipated on the banks to greater extent in wide, shallow channels, whereas these forces are focused on the streambed in constrained or incised



Figure 3-35 Headwater stream channel showing the location of the streambed and the banks (white arrows).

channels (Carling 1983). Geomorphology also governs the distribution of water as streams dry. Wetted widths will contract faster in wide, shallow channels than in incised channels. Wide, shallow channels may be more prone to surface water drying than incised channels because the summer groundwater table is more likely to be above the streambed (Stanley et al. 1997). However, where drying is severe, incised channels offer less interstitial refugia because the substrate layer above underlying bedrock may be thin. Habitat simplification reduces the biotic diversity directly, but also affects diversity indirectly through loss of refugia (Lake 2003).

Channel geomorphology is measured once for a given reach during the study because they are unlikely to change significantly over short time periods (e.g., 1-2 years). However, floods can significantly reshape channel geometry over short periods of time and should be taken into account when investigators need fine temporal resolution data. The following procedure will require

2-3 field crew members, depending upon the channel width.

3.10.1 Bankfull width (BF width)

Field determination of bankfull stage is particularly difficult for small channels where the floodplain may not be well-developed or may be absent. Useful indicators of bankfull stage include breaks in sediment particle size and bank vegetation. Swift and Ledford (1994) identifies the following characteristics for estimating bankfull stage in small southern Appalachian streams:

1. Topographic break from vertical bank to floodplain
2. Topographic break from steep to gentle slope
3. Top of point bar
4. Change in vegetation from temporary to permanent
5. Upper elevation of fine debris deposition
6. Rocks and/or roots exposed in banks
7. Change in size distribution of deposits
8. Change in texture of fines lodged between rocks

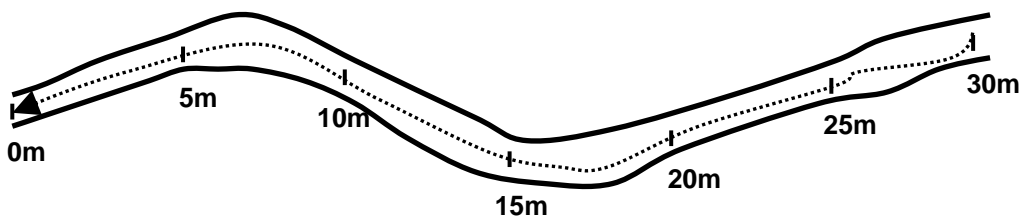


Figure 3-36 Plan view of study reach showing 5-m intervals. Direction of arrows shows direction of flow, and the dotted line represents the thalweg.

Procedure

Delineate the 30-m study reach so that a measuring tape is marking locations along the thalweg. Bankfull width and depth are measured at 5-m intervals (at 0, 5, 10, 15, 20, 25 and 30-m marks) along the study reach, whereas flood-prone area width is measured at

15-m intervals (at 0, 15, and 30-m marks; Figure 3-36). Measurements should be taken at the next meter mark (upstream or downstream) along the study reach where obstacles (e.g., large woody debris) or certain channel features (e.g., meanders, knickpoints) are present at original measurement locations

(0, 5, 10, 15, 20, 25, and 30 m). Note on the field form where measurements were taken.

It is useful to look upstream and downstream along both banks of measurement location to identify appropriate bankfull stage. When a consensus among crew members is made about the appropriate bankfull stage, the end of a measuring tape is staked at bankfull stage. The tape is pulled across the channel (perpendicular to direction of flow) to the other bank to determine bankfull width (Figure 3-37). A second crew member, standing downstream, provides instruction for adjusting the tape position so that it is horizontally level at the bankfull stage. This

can be done more accurately if a laser level is used to adjust the tape position. Ensure that the tape is taut and record the distance (to the nearest 0.01 m) in the appropriate cell on the second page of the field form (Figure 3-38).

3.10.2 Bankfull depth (BF depth)

While the tape is still positioned for measuring bankfull width, a crew member uses the meter stick to measure bankfull depth (Figure 3-37). The meter stick (zero-end down) is positioned perpendicular to the tape measuring bankfull width at the center of the thalweg. Record the distance (to the nearest 0.01 m) between the



Figure 3-37 Photograph shows measurement of bankfull (BF) width and bankfull depth.

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF Page 2 of 4									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0							1.43	0.2	3.02

Figure 3-38 Appropriate location for recording bankfull (BF) width (red), bankfull depth (blue), and flood prone area (FPA) width (black) measurements on page 2 of the field forms.

streambed and the tape in the appropriate cell on the second page of the field form (Figure 3-38).

3.10.3 Flood-prone area (FPA) width and entrenchment ratio

At the 0, 15 and 30-m locations the crew members then locate 2X the bankfull depth and raise the tape to that level for measuring

the width of the flood-prone area (FPA width, Figure 3-39). The crew member with the tape adjusts ends of the tape so that it is horizontally level and extended tautly across the channel to touch soil at both ends. Where the distance of the flood-prone area width is >2.2X the bankfull width, record “>2.2X BFW”, otherwise record to the nearest 0.01 m



Figure 3-39 Photograph illustrating flood-prone area (FPA) width.

in the appropriate cell (Figure 3-38). The significance of the 2.2X bankfull width is

based on Rosgen (1994, 1996) channel classification, where entrenchment ratios >2.2

(FPA \geq 2.2X BFW) are classified as slightly entrenched (stream types C, D, or E). As was done when measuring the bankfull width, a crewmember provides instruction for adjusting the tape position so that it is horizontally level at the 2X bankfull depth. This can be done more accurately if a laser level is used to adjust the tape position.

References

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Equipment and supplies

- 2 Measuring tapes (50m)
- Meter stick
- Field forms
- Survey stakes
- Laser level (optional)

3.11 Measuring water velocity

General

The following subsection provides methods for measuring water velocity in headwater streams. Water velocity is the rate of water moving through a point and represents one aspect of stream flow. Hydraulics is among the more complex and dynamic characteristics of the stream environment (Statzner et al. 1988, Vogel 1994). For example, the relevancy of a velocity is dependent on organism size. Under the same velocity, smaller organisms may experience the near-bed velocity as laminar syrup, whereas larger organisms would experience a turbulent maelstrom. Although water velocity is just one aspect of stream hydraulics, it provides ecologically-relevant information. The following methods will offer coarse estimates that are useful in for making relative comparisons. For fine-scale and less-invasive measurements, alternative methods such as acoustic Doppler velocimeter (ADV, Bouckaert and Davis 1998, Finelli et al. 1999) and thermistor probes (LaBarbera and Vogel 1976, Dodds and Biggs 2002) are more suitable. As already discussed in Subsection 3.6, water velocity is useful for designating habitat units and can directly (e.g., food availability, dispersal) and indirectly (e.g., refuge from predators) affect the distribution of organisms (Hart and Finelli 1999). Mean water velocity for a stream reach may not necessarily decline as streams first begin to dry, but it will drop dramatically when streambed materials such as cobbles and boulders become emergent and flow becomes mostly interstitial. Because water velocity can vary considerably over time, measurements should be taken during each sampling visit. Below we detail four simple procedures for measuring water velocity along a stream reach; additional procedures are discussed by John (1978), Newbury (1984), and Ciborowski (1991).

Procedure

Delineate the 30-m study reach so that the measuring tape is marking locations along the thalweg. Point measurements of water velocity (Procedures 3.11.1, 3.11.2 and 3.11.3) at the streambed are taken at 5-m

intervals (at 0, 5, 10, 15, 20, 25 and 30-m marks) along the study reach thalweg (Figure 3-40). Below are four procedures that can be used. In most cases (and when available) the velocity meter procedure is preferred;

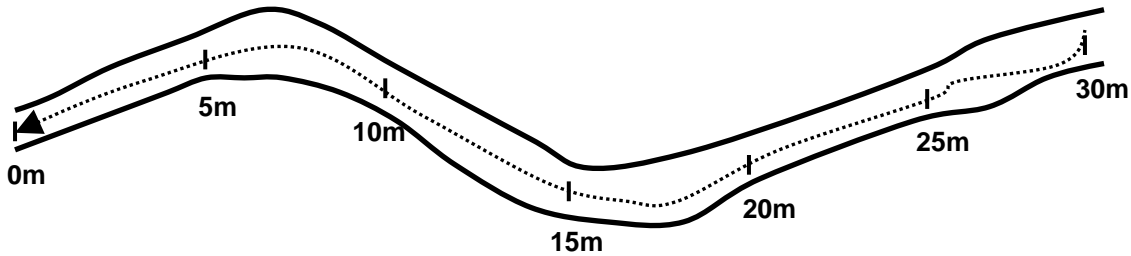


Figure 3-40 Plan view of study reach showing measurement locations (vertical black tick marks) for current velocity measurements. Flow is from right to left and the dotted line represents the thalweg.

however under some circumstances the other three procedures may be more suitable.

3.11.1 Velocity meter procedure

Before arriving at the field site read the instruction manual for the velocity meter (e.g., electromagnetic, propeller). Attach the wading rod to the velocity meter probe. Check to see that the meter is functioning properly and is calibrated. Set the selector switch to m/sec and the time constant switch to the lowest setting that gives stable readings. Stand downstream and to the side of each of the measurement locations when taking velocity readings. Hold the rod perpendicular

to the water surface with the front of the velocity probe facing upstream, perpendicular to the channel cross-section (Figure 3-41). Set the bottom of probe ~ 0.5 cm off the streambed and take flow reading. Write the water velocity in the appropriate cell on the second page of the field forms (Figure 3-42). If no surface water is found at a measurement location, indicate on the field form that the water velocity is 0. If there is flowing surface water at a location but it is too shallow to measure with a velocity meter, then indicate that the water velocity is “>0”.

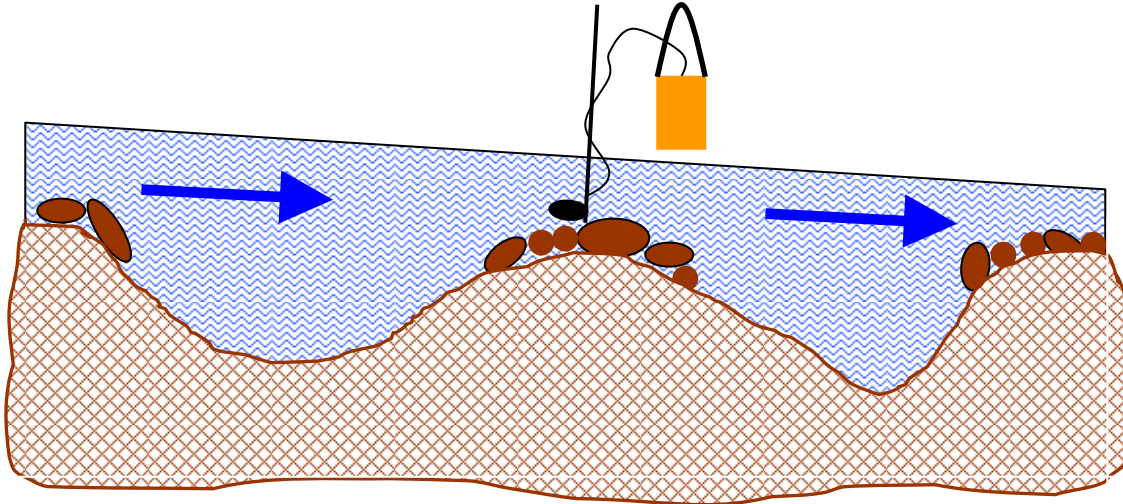


Figure 3-41 Longitudinal section across the channel thalweg showing orientation of the velocity probe for measurements.

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF Page 2 of 4									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0					0.02				

Figure 3-42 Appropriate location for recording water velocity on page 2 of the field forms.

3.11.2 Velocity-area procedure using a bag meter (Gessner meter)
 A simple alternative to electromagnetic or propeller meters is the bag meter or Gessner meter (Gessner 1950). To assemble the bag meter: tape a plastic bag (e.g., small plastic grocery or bread bag) over the larger opening of a small plastic funnel with duct tape. Make sure that it is completely sealed and there are

no holes in the plastic bag. Then tape a cylinder (e.g., plastic cup with bottom cut out, PVC pipe) that has a diameter slightly larger than the large opening of the funnel), to the outside of the large funnel opening and over the plastic bag (Figure 3-43). Calculate the area of the small funnel opening (i.e., $A = \pi r^2$).

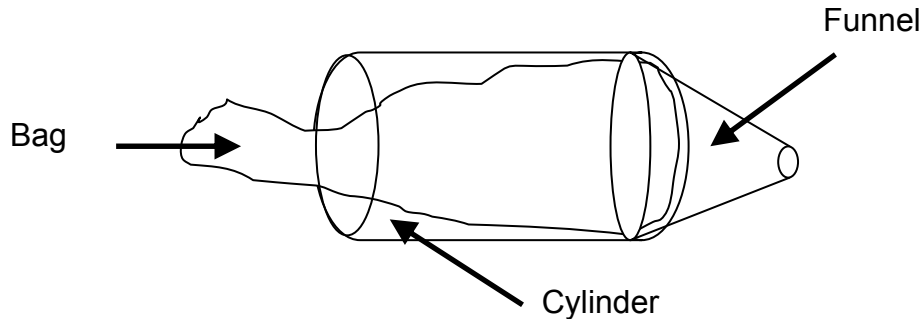


Figure 3-43 Bag meter used to measure water velocity.

Stand downstream and to the side of each of the measurement locations when measuring velocity. Before taking a measurement, empty and deflate the bag as much as possible. While holding the bag meter by the cylinder in one hand, use your other to cover the small funnel opening. Submerge and hold the bag meter near (will depend on diameter of large funnel opening) and parallel to the stream bed, so that the small opening is facing into the current. Simultaneously note the second hand position on your wristwatch (alternatively signal “start” to another crew member with a stopwatch) and uncover the small funnel opening. Let the bag fill with water for 10 seconds (or shorter time in very fast current) and recover the funnel opening. Carefully pour the water from the bag into the calibrated container. Determine the volume to the nearest 0.005 liter. In the cells for water velocity on the field form (Figure 3-42) write the volume and fill time (e.g., 0.25 L / 10 sec). Indicate on page 3 of the field form that the bag meter was used to measure discharge and the area of the small funnel opening.

After returning from the field, the water velocities can be calculated by first converting the volumes from liters to m^3 (i.e., divide by 1000). The volume is divided by the filling time (e.g., 10 sec) and then the resulting value is divided by the area of the small funnel

opening (in meters). Repeat this for all measurement locations. If there is no surface water at a measurement location, indicate on the field form that the water velocity is 0. If there is flowing surface water at a location but it is too shallow for this method indicate that the water velocity is >0 .

3.11.3 Neutrally-buoyant object procedure

This procedure can be used when a velocity meter is not available or if flow is too shallow for accurate meter readings. Indicate on page 3 of the field form that this procedure was used. Designate the upstream and downstream boundaries of 2-m segments that are centered on each of the measurement locations (1 m upstream and downstream of the 0, 5, 10, ... 30-m locations, Figure 3-44). While standing downstream of the release point and outside the thalweg, hold the neutrally-buoyant object (see Equipment and supplies for examples; consistently use the same object across all measurements) in the thalweg (at 0.4X the water depth). In unison, gently release the neutrally-buoyant object and start the stop watch. Note the time required for the object to travel the 2-m segment. If the object becomes stuck or drags along the bottom repeat the release and/or slide the segment position upstream or downstream to avoid areas where the object sticks or drags. In fast segments 2 people may be required to

accurately measure segment travel time. One person at the upstream boundary simultaneously releases the object and signals “start”. This indicates to the second person who is standing at the downstream boundary to start the stopwatch. The second person then stops the watch when the object crosses the downstream boundary. Divide the segment

length by the travel time and write this in the appropriate cell on the field form. If there is no surface water at a measurement location, indicate on the field form that the water velocity is 0. If there is flowing surface water at a location but it is too shallow for this method indicate that the water velocity is >0 .

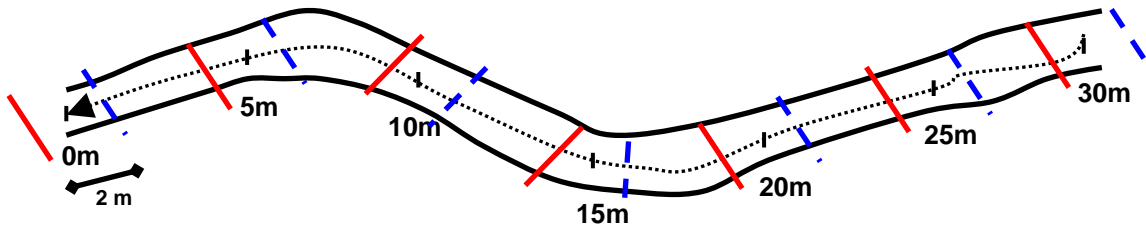


Figure 3-44 Overview of study reach showing measurement locations (black tick marks crossing the thalweg, shown as dotted line), upstream (dashed blue lines) and downstream segment boundaries (solid red lines) for the neutrally-buoyant procedure to measure water velocity.

3.11.4 Fluorescent dye procedure

This procedure can be used when a velocity meter is not available or if flow is too shallow for accurate meter readings. Indicate on page 3 of the field form that this procedure was used. This procedure provides only a general measure of water velocity for the entire reach, in contrast to the methods described above which provide estimates for average and variation of water velocity. Pour ~ 1 ml fluorescein dye (or rhodamine WT) into a 1 L plastic bottle and add 500 ml of stream water. Cap and shake bottle until dye is thoroughly dissolved. In fast-flowing reaches 2 people may be required for this method, one person with the dye at the 30-m location (upstream boundary of study reach) and the other person with a stopwatch at the 0-m location (downstream boundary of study reach).

Before starting, make sure that other field personnel are outside of the study reach. The person at the upstream boundary will simultaneously release the dye (gently pouring bottle contents from ~ 5 cm above the water level) into the thalweg at the 30-m location and signal “Start”. This indicates to the person at the downstream boundary to start the stopwatch. The downstream person records the time when the “leading” and “trailing” edges of the dye plume cross the downstream boundary (Figure 3-45). The trailing edge is identified as the last visible portion of the plume in the thalweg. Ignore any dye that may have gotten caught in backwater pockets. On the field form write the distance of the dye release (should be 30 m if entire study reach is flowing) and travel times for leading and trailing edges in seconds.

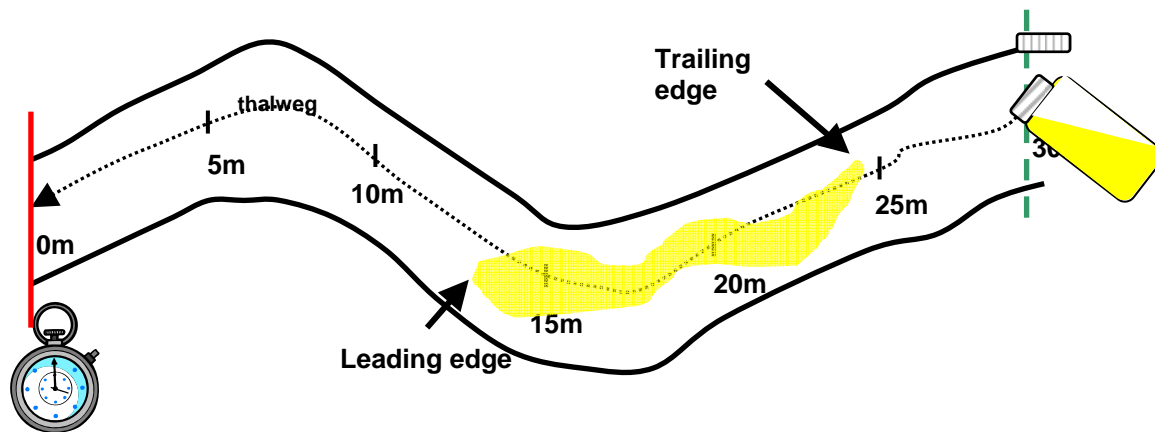


Figure 3-45 Overhead view of study reach showing leading and trailing edges of fluorescein plume.

References

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Vogel, S. 1994. *Life in Moving Fluids, 2nd edition*. Princeton University Press, Princeton, New Jersey.

Equipment and supplies

Measuring tape (50 m)

Field forms

A, B, C or D

- A. Velocity meter (electromagnetic, propeller, or cup) and spare batteries – see Procedure 3.11.1
- B. Stopwatch and bag meter – see Procedure 3.11.2
- C. Stopwatch and neutrally buoyant object (e.g., piece of orange peel, film canister partially filled with stream water, small stick) – see Procedure 3.11.3
- D. Stopwatch, 1L plastic bottle, fluorescein dye (1 ml per 500 ml streamwater) – see Procedure 3.11.4

3.12 Measuring discharge

General

This subsection provides methods for measuring discharge (Q) or flow rate of water in headwater streams. Discharge (in conjunction with stream size or drainage area) is a quantitative measure for describing the hydrologic condition. This measure of flow is useful in following and describing temporal patterns in water chemistry. When conditions are allowable, discharge should be measured during each sampling visit. The methods described in this subsection are modified from those described by John (1978), Platts et al. (1983), Kilpatrick and Cobb (1985), Gordon et al. (1992), Gore (1996), and Kaufmann (1998). For long-term studies continuous discharge monitoring may be considered. The simplest method is a staff gauge, where discharge can be determined by monitoring the stage (or water depth) at a permanent

location. Stage-discharge relationships (rating curves) are plotted by measurements of stage against discharge over a range of flows (Gordon et al. 1992). Peak flow between field visits can be determined from crest gauges (Gordon et al. 1992, Harrelson et al. 1994). A simple crest gauge consists of stilling well, a meter stick, and ground cork. The stilling well can be a length of plastic pipe (3 to 4 cm diameter) with caps on both ends. Holes are drilled in the bottom cap so the water level within the stilling well represents the stage. The top cap of the well should be loose fitting or vented. Finely ground cork and the meter stick are placed in the well. After a peak flow the cork will adhere to the meter stick at the crest or peak stage. The gauge is then easily reset by washing the cork off the meter stick and back into the well. The design and equipment for gauging stations can vary from a simple staff gauges to more permanent flumes and weirs. Gauging station design and data storage are discussed in John (1978), Herschy (1995), Clemmons et al. (2001), and Bureau of Reclamation (2001).

Procedure

Delineate the 30-m study reach so that a measuring tape is positioned along the thalweg.

3.12.1 Velocity-area procedure using a velocity meter

Before arriving at the field site read the instruction manual for the velocity meter. Attach the wading rod to the velocity meter probe. Check to see that the meter is functioning properly and is calibrated. Set the selector switch to m/sec and the time constant switch to the lowest setting that gives stable readings (unit setting may be switched to ft/s under extremely low flow conditions). The location for discharge

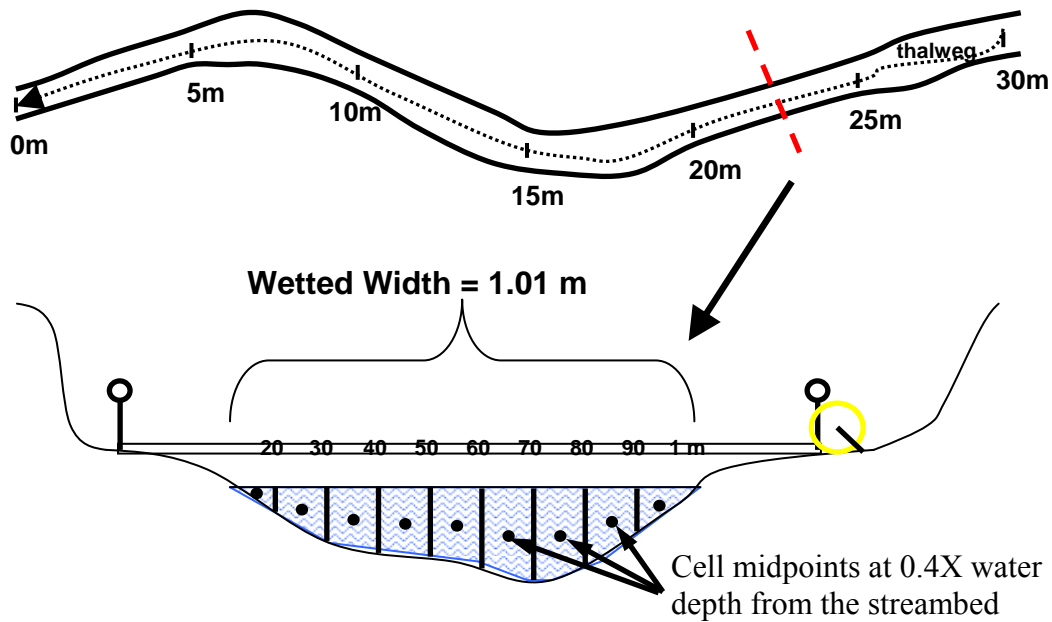


Figure 3-46 Plan view of study reach (top) showing discharge measurement cross-section (red dashed line). Cross-section for discharge measurement (bottom) showing measurement cells.

measurement is not restricted to the 30-m study reach; however, the discharge at the measurement location should be representative of the discharge seen in the study reach. Locate a channel cross-section that has the following characteristics (or can be modified to have these characteristics*): 1) channel immediately upstream and downstream is straight (~ 3 m in both directions of discharge transect), 2) free of obstructions (e.g., woody debris, macrophytes, emergent stones, braided channel), 3) “U” shaped so that $\geq 90\%$ of the cross-section has water depths sufficiently deep for accurately measuring water velocity with the velocity meter, and 4) water velocity across the channel is relatively uniform and $\geq 90\%$ of the cross-

section has water velocities $>0.01 \text{ ms}^{-1}$. Runs and glides are typically good habitat units for measuring discharge.

At the measurement cross-section, stretch the second measuring tape taut across the channel so that it is perpendicular to flow and $\geq 5 \text{ cm}$ above the stream surface (Figure 3-46). Determine the wetted width of the channel to the nearest 0.01 m. Divide the wetted width into 6 to 12 equally sized intervals or cells. Cells should be $\geq 5 \text{ cm}$ wide.

Write the wetted and cell widths in the appropriate blanks on the field form (Figure 3-47). Water depth and water velocity are measured midway across each cell or cell midpoint (Figure 3-46).

* The channel can be modified (e.g., remove rocks, obstructions) prior to taking any discharge measurements. Once measurements have begun however, do not modify the channel.

Start measurements from one bank and move across. Stand downstream and to the side of each depth and velocity measurement. Use the meter stick to measure water depth (to the nearest 0.5 cm). Water velocity is then measured at ~ 0.4X water depth from the streambed for each cell. If this depth is too shallow to submerge the velocity meter probe

or propeller, measure velocity closer to the streambed. Write the water depth and its associated water velocity measurement in the cells on the field form (Figure 3-47). Discharge is calculated by multiplying the cell width * water depth * water velocity of each cell then summing across all cells.

STREAM DISCHARGE										Page 3 of 4	
Wetted Width (m) : <u>1.01</u>		CELL WIDTH (m) : <u>0.1</u>									
Depth(cm)	<u>4</u>	<u>7</u>	<u>11</u>	<u>16.5</u>	<u>25</u>	<u>28</u>	<u>27</u>	<u>10.5</u>	<u>5.5</u>		
Velocity(m/s)	<u>0</u>	<u>0.05</u>	<u>0.09</u>	<u>0.12</u>	<u>0.22</u>	<u>0.25</u>	<u>0.18</u>	<u>0.08</u>	<u>0</u>		
$Q \text{ (m}^3\text{s}^{-1}\text{)} = \underline{0.02908}$		Discharge procedure: <u>Velocity-area</u>									
		Velocity procedure/meter model: <u>Marsh-McBirney Flowmate</u>									

Figure 3-47 Appropriate location for recording discharge and procedures used on page 3 of field forms. Example values shown in red.

3.12.2 Velocity-area procedure using a bag meter

To assemble the bag meter: tape a plastic bag (e.g., small plastic grocery or bread bag) over the larger opening of a small plastic funnel with duct tape. Make sure that it is completely sealed and there are no holes in the

plastic bag. Then tape a cylinder (e.g., plastic cup with bottom cut out, PVC pipe that has a diameter slightly larger than the large opening of the funnel), to the outside of the large funnel opening and over the plastic bag (Figure 3-48). Calculate the area of the small funnel opening (i.e., $A = \pi r^2$).

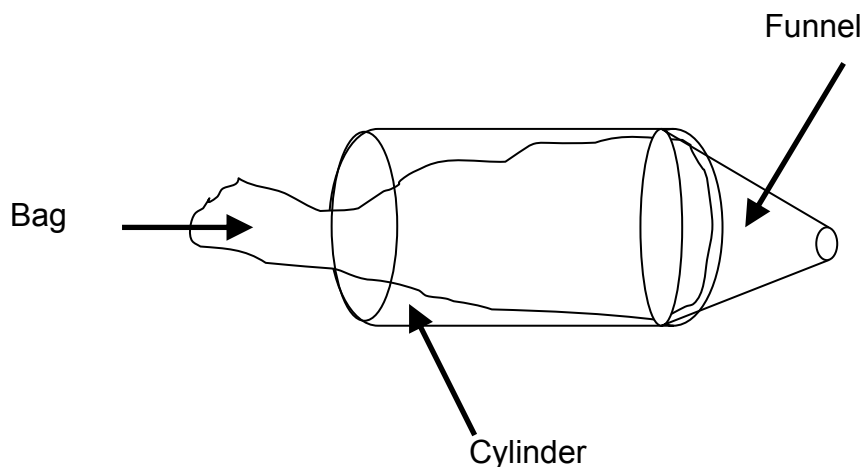


Figure 3-48 Bag meter used to measure discharge.

Select and delimit a measurement cross-section as described in **3.12.1**. Follow the same procedures except use the bag meter to measure water velocity at 0.4X the water depth from the stream bed. Before taking a measurement completely empty the bag of water and deflate the bag of air as much as possible. While holding the bag meter by the cylinder in one hand use your other to cover the small funnel opening. Submerge and hold the bag meter at the appropriate measuring depth, so that the small opening is facing into the current and the bag meter is perpendicular to the measurement cross-section.

Simultaneously note the second hand position on your wristwatch (alternatively shout “start” to another crew member with a stopwatch) and uncover the small funnel opening. Let the bag fill with water for 10 seconds (or shorter time in very fast current) and recover the funnel opening. Carefully pour the water from the bag into the calibrated container. Determine the volume to the nearest 0.005 liter. In the cells for water velocity on the field form write the volume and fill time (e.g., 0.25 L / 10 sec). Indicate on the field form that the bag meter was used to measure discharge and the area of the small funnel opening.

After returning from the field, the cell water velocities can be calculated by first converting the volumes from liters to m^3 (i.e., divide by 1000). The volume is divided by the filling time (e.g., 10 s) and then the resulting value is divided by the area of the small funnel opening (in meters). Repeat this for all cells of the measurement cross-section and determine discharge as instructed in **3.12.1**.

3.12.3 Timed filling procedure

This method can be used where the channel is small and there are one or more natural spillways or plunges along the reach where the entire stream flow can be captured (the

channel can be modified to ensure that all the flow is funneled). Simultaneously start the stopwatch and position the wide-mouth container (i.e., bucket or basin) under the spillway to collect the entire flow. Collect water for 10–30 seconds, depending upon the level of discharge. Transfer the water from the wide-mouth container to a calibrated one and determine the volume (to the nearest 0.005 liter). Alternatively, one may simply record the time required to fill a bucket or basin to a known volume (e.g., 2 L). Repeat this procedure 3 times at given spillway. Indicate that the timed filling procedure was used to measure discharge and write the volume and respective filling time for each trial on the field form.

3.12.4 Dilution gauging procedures

These methods use dilution over time of biologically inert substances introduced into a stream reach. Commonly used substances (tracers) included salt solutions (NaCl, KBr) and dyes (e.g. fluorescein, rhodamine WT). Tracers should be readily detectable at low concentrations (low or no background concentrations), and soluble in water at stream conditions (Gordon et al. 1992). Depending upon the tracer used, general (electrical conductivity meter, fluorometer) or tracer-specific probes can be used for *in situ* measurements. Alternatively, samples can be collected in bottles and returned to the laboratory for analysis. An estimate of discharge is needed to determine the initial tracer concentration so that the measured concentration is easily detectable (5 to 10 times background). The two general methods for dilution gauging are the slug injection and constant injection. The slug injection method involves releasing a known volume and concentration of a tracer as a single pulse. Background measurement for the tracer should be measured before beginning the injection. The point of injection should be a

zone with turbulent mixing. Tracer concentration is measured at regular intervals at a downstream station from the start of the injection until concentrations reach background levels. The measurement interval will depend upon the level of discharge and the size of the study reach. Discharge (Q) is determined from using the area under the concentration curve (Figure 3-49). The following equation from Gordon et al. (1992) is used:

$$Q = 1000 \frac{Vc_t}{\int_{t_1}^{t_f} (c - c_0) dt}$$

Where V is the slug volume (in liters), c_t is the initial tracer concentration, c_0 is the background concentration in the stream water, c is the concentration at time t .

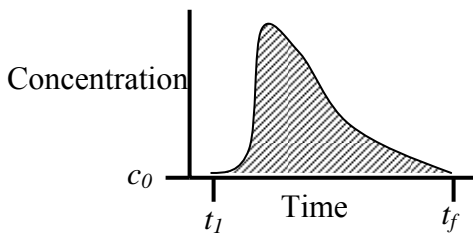


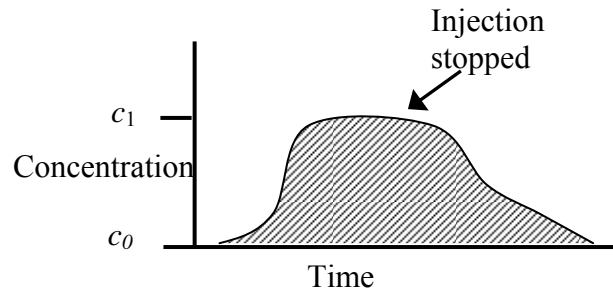
Figure 3-49 Example of a concentration curve from a slug injection. Discharge ($\text{m}^3 \text{s}^{-1}$) is the hatched area under the curve.

The constant injection method also uses a known concentration of the tracer, but the rate of injection is constant over the duration of the measurement rather than as a slug. Tracer concentration will increase and then stabilize at the downstream station (Figure 3-50). Constant injection can be done using a peristaltic pump or a Mariotte bottle (see Webster and Ehman 1996). Discharge using this method is calculated using the equation from Gordon et al. (1992):

$$Q = 1000 \frac{(c_t - c_1)}{(c_1 - c_0)} Q_t$$

Where c_1 is the stabilized concentration, Q_t is the tracer injection rate (l s^{-1}), and the other variables are the same as shown in the previous equation.

Figure 3-50 Example of a concentration curve from a continuous injection. Discharge ($\text{m}^3 \text{s}^{-1}$) is the hatched area under



the curve.

Although these methods may be more accurate and feasible during low flows than previously described methods, insufficient mixing and anastomosing flow through reaches may also limit discharge measurement using dilution gauging methods. Some disadvantages of dilution methods compared to other methods include need for prior knowledge of approximate discharge level, additional equipment bulk, and drift response by biota (Wood and Dykes 2002).

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- Equipment and supplies*
 Measuring tape (50 m)
 Field forms
 A, B, C, or D
- A.** Second measuring tape, survey stakes, meter stick, velocity meter (electromagnetic, propeller, or cup) and spare batteries – see **Procedure 3.12.1**
- B.** Second measuring tape, meter stick, wristwatch with second hand, bagmeter (small funnel taped to plastic bag enclosed in plastic pipe), and calibrated container (e.g., volumetric cylinder) – see **Procedure 3.12.2**
- C.** Stopwatch, wide-mouth container (e.g., bucket, wash basin), and calibrated container (e.g., volumetric cylinder) – see **Procedure 3.12.3**
- D.** Stopwatch, tracer substance (stock solution), calibrated pipette and tips, volumetric cylinder, mixing container, tracer probe or fluorometer, peristaltic pump or Mariotte bottle – see **Procedure 3.12.4**
- 3.13 Measuring depth to bedrock and groundwater table**
General

This section provides instructions for measuring depth to underlying bedrock and groundwater table in headwater streams. The hyporheic zone is the interface between the surface stream and the underlying groundwater (Boulton et al. 1998, Jones and Mulholland 2000). The importance of the hyporheic zone to the structure and function of streams depends upon the permeability and discharge through the hyporheic zone to the overlying surface water (Brunke and Gonser 1997). Because the subsurface environment (e.g., temperature, flow) is relatively more stable than the overlying streambed surface, the hyporheic zone may serve as a refuge for stream organisms from disturbances such as floods and drying (e.g., Clinton et al. 1996, Dole-Olivier et al. 1997). This rapid method provides an estimate of the extent and hydrologic status of the hyporheic zone, and therefore the potential for it to serve as refuge.

Depth to groundwater table can vary with intra- and interannual differences in catchment precipitation and evapotranspiration, and it is important to measure whenever surface water is absent. Depth to bedrock is unlikely to change significantly over short time periods (e.g., 1-2 years), and therefore only needs to be measured once during the study period. Because these measurements use the same procedure (e.g., sounding rod, Valett 1993), we recommend taking both measurements during drier periods (when more than one sampling visit is planned).

The development of ground-penetrating radar (GPR) offers an alternative, non-invasive method to describe subsurface features of streambeds, including the depth to bedrock and groundwater (Naegeli et al. 1996, Huguenberger et al. 1998). However, the utility of GPR can be limited where interfaces are not clearly defined (e.g., saturated fine sediments) or below dense layers, such as

clays (Poole et al. 1997). The cost and bulk of equipment are other considerations that may limit the application of GPR in large scale assessments of headwater streams.

Procedure

Delineate the 30-m study reach so that a measuring tape is marking locations along the thalweg. Locate 3 depositional habitat units near the 0, 15, and 30-m marks of the study reach. Depth measures are taken in the thalweg at these 3 locations.

3.13.1 Depth to bedrock

Hammer the sounding rod or “T”-bar vertically into the stream bed at intervals of 5-10 cm with the hand sledge (Figure 3-51). Wiggle the upper end of the sounding rod in circular motion by hand (Figure 3-52). This will prevent the rod from becoming stuck within the stream bed. Continue tapping the rod until it strikes bedrock (or large boulder). This will be evident from the “pinging” sound the rod makes when hammered (and resistance to further downward movement). Some stream beds have cobble deposition that may impede the rod’s downward progress. You can penetrate through cobble layers by rotating the rod tip in a circular motion while continuing to hammer (Figure 3-52). This process will often allow the rod to pass through interstitial spaces between the cobbles. If not, simply shift the rod location slightly and repeat the process. When you have struck bedrock, use your forefinger and thumb (or cable tie) to mark the point on the rod where it is even with the stream bed surface. Pull the rod out of the stream bed and measure the distance with the meter stick (to the nearest 1 cm) between the lower end of the rod and your finger. Write this measurement in the appropriate cell on the field form (Figure 3-53). If the depth to bedrock appears to exceed the length of the sounding rod (> 85 cm for 91 cm sounding rod) then indicate

“>85 cm” on the field form. Where the stream bed surface is bedrock then indicate “0 cm” on the field form.



Figure 3-51 Using sounding rod and hand sledge hammer to estimate depth to bedrock and the groundwater table.

3.13.2 Depth to groundwater table

Where the stream contains surface water the depth to the groundwater table will equal the water depth at the measurement location. Indicate this on the form by writing "+" and the water depth. Where the stream bed is dry begin by following the same procedure used to measure depth to bedrock. After the groundwater table is reached, water seeping into the hole will create resistance on the rod.

Moving the top of the rod in a circular motion or gently lifting the rod a few centimeters will help you determine if you have entered the groundwater table. If the rod has entered the water table, you may either hear a “slurping” sound or feel suction resistance when the rod is lifted. Before fully removing the sounding rod from the streambed, mark the point (with a finger or cable tie) on the sounding rod where it is even with the stream bed surface.

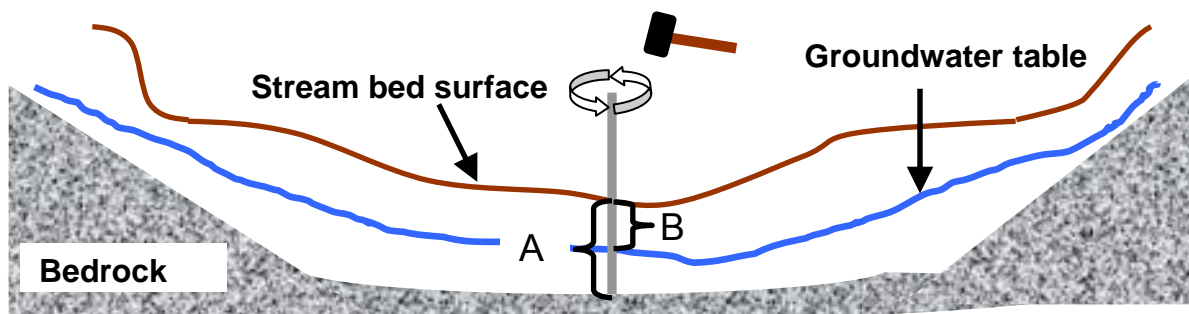


Figure 3-52 Cross-section of a dry channel illustrating depth to underlying bedrock (A) and depth to the groundwater table (B).

Immediately after removing the sounding rod from the stream bed identify the highest point along the rod where there is water (wet enough to drip). Measure the distance between stream bed level and the highest wetted point on the rod with the meter stick (to the nearest 1 cm). Write this measurement in the appropriate cell on the field form

(Figure 3-53). Indicate that this represents a measurement below the stream bed surface by writing a “-“ before the distance. If the depth to the groundwater table appears to exceed the length of sounding rod (>85 cm for 91 cm sounding rod) then indicate “>-85 cm” on the field form.

MAX. POOL DEPTH (cm)	DEPTH TO BEDROCK / GROUNDWATER (cm) (3 measures in depositional habitat)	SINUOSITY (number of bends)
	54 -24 34 +8 0 -14	

Figure 3-53 Appropriate location for recording depth to bedrock (example values in blue) and depth to groundwater (example values in red) on page 1 of field forms.

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Equipment and supplies

Measuring tape (50 m)
Sounding rod (steel rod, ≥ 3 ft or 91 cm) or "T"-bar
Hand sledge hammer
Meter stick
Field forms

3.14 Gravimetrically measuring streambed sediment moisture

General

This subsection provides instructions for measuring the relative moisture of streambed sediments in dry headwater channels. In other words, this procedure quantifies the degree of "dryness" or desiccation of the benthic habitat in streams when visible surface water is absent. This is especially relevant to organisms that inhabit intermittent or ephemeral streams and possess life histories or physiological traits (i.e., diapause, quiescence, and aestivation) for surviving the dry periods (Davis 1972, McKee and Mackie 1983, Danks 1987, Williams 1998, Dunphy et al. 2001). Mortality during such periods can depend on the desiccation level of the surrounding sediments, and therefore can influence the spatial distribution of organisms (Suemoto et al. 2005). Soil moisture is measured during the summer sampling period (when sediment moisture is expected to be lowest) within study reaches that have no visible surface water.

Procedure

Delineate the 30-m study reach so that a measuring tape is positioned along the thalweg. Mark three 15-cm pieces of 3/4" PVC with a line 10 cm from one end using a permanent marker (volume ~ 28.5 cm³).

3.14.1 Sediment collection

Three individual sediment cores are taken along study reaches lacking visible surface water. Cores are extracted from the streambed in depositional habitat units with fine sediments (e.g., sand, silt, fine gravel). In addition to being more feasible to collect, moisture content is expected to be relatively high in thick patches of fine sediment (i.e., capillary fringe) because of greater capillary tension compared to levels associated with coarser particles (Dunne and Leopold 1978). Where possible, cores from each study reach

should be taken from separate depositional units within the thalweg.

Find a suitable location and brush aside detritus (i.e., leaf litter) from the streambed surface. Position the core vertically so that the 10-cm mark is away from the streambed (Figure 3-54). Tap the core vertically into the streambed with the hand sledge until the

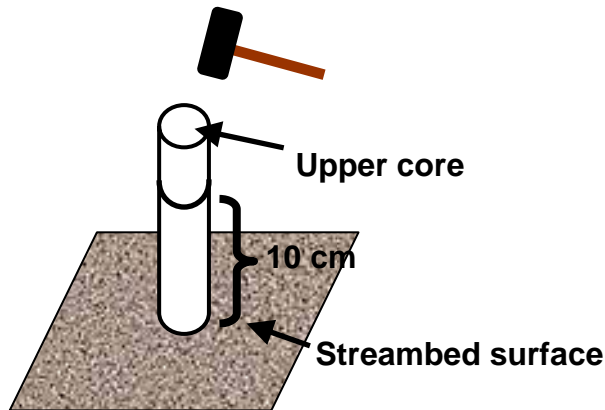


Figure 3-54 Sampling sediment moisture.

10-cm mark is flush with the streambed (Figure 3-55). Place a rubber stopper into the upper core opening. Carefully pull the core out of the streambed and place a second rubber stopper into the lower core opening.

Place core in a resealable plastic bag. Label the bag and/or core using a permanent

marker with relevant information (e.g., locality, date, collector’s initials). Remove excess air within the bag when sealing. Log the number of sediment core samples taken at each site on the field forms (Figure 3-56). Store samples in a cooler with ice or in a refrigerator until the samples can be measured in the laboratory. Measure moisture of the sample within 4 days of collection. A soil borer or auger can be used collect samples rather than PVC cores. Care must be taken to keep sediment samples airtight (e.g., Shelby tube) to maintain soil moisture levels.



Figure 3-55 Tapping core vertically into streambed.

PRESENCE OF HEADCUT IN REACH		ALGAL COVER INDEX					# CORES FOR SUBSTRATE MOISTURE (depositional)	
Y	N	1	1 ^{1/2}	2	3	4	5	3

Figure 3-56 Appropriate location for recording the number of sediment moisture cores collected on page 1 of field forms.

3.14.2 Laboratory measurement

In the laboratory, use a weighing spatula or thin metal rod to transfer sediment from cores into separate evaporating dishes or crucibles. Be sure that the dishes are uniquely identified (e.g., dish #s), so that results can be associated

with specific samples. Measure the wet weight of the sediment samples with an analytical balance to the nearest 0.01g. Record dish identification, sample abbreviation, and wet weight on the data sheets (Figure 3-57).

HISS Sediment Moisture Data Sheet			DATE: 9/23/03	Page 1 of 1
Dish #	Study Site Abbrev.	Sediment Core	Wet Weight (g)	Dry Weight (g)
123	Four-FC-3	A	39.36	
144	Four FC-3	B	38.47	
135	Four FC-3	C	38.95	
142	Four-FC-4	A	29.29	

Figure 3-57 Example of the sediment moisture data sheet.

Place samples into the drying oven for 24 h with temperature set at 90° C. Remove samples from the oven using tongs and allow them to cool to room temperature. If a desiccator is available, the samples can be

directly placed into the desiccator to cool. Measure the dry weight of the sediment samples with the balance to the nearest 0.01g and record on the data sheet. Percent moisture is calculated using the following equation:

$$\text{Percent Moisture} = \frac{\text{Wet Weight} - \text{Dry Weight}}{\text{Wet Weight}} \times 100$$

Cores can be collected from locations where/when surface water is present to provide a relative comparison of sediment moisture. Alternatively, water can be added to previously dried samples until visibly saturated. The cores are then weighed to determine percent moisture at saturation. The amount of organic matter within the sediments can be determined by ashing the core contents in a muffle furnace at 550° C for two hours and then reweighing to determine ash-free dry mass (AFDM).

Alternative means for measuring soil moisture include the use of soil moisture probes (e.g., tensiometers, capacitance sensors; see Miller et al. 1997); but these are not commonly used in the relatively coarse sediments of intermittent streambeds. A procedure described by Greacen et al. (1989) indirectly measures sediment moisture by way of water absorption onto filter paper and then gravimetric determination of water content. Techniques that have been used to extract water from soil cores include centrifugation, squeezing, and vacuum extraction (e.g., Adams et al. 1980).

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water invertebrate communities. *Archiv für
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Equipment and supplies

Measuring tape (50 m)
Cores (15 cm long, 3/4 inch inner diameter
PVC pipe)
Hand sledge hammer
Rubber stoppers (2 per core, No.1 or 2)
Resealable plastic bags (1 per core)
Cooler
Ice or ice packs
Permanent marker
Field forms
Weighing spatula or metal rod (laboratory)

Evaporating dishes or crucibles (laboratory)
Drying oven (laboratory)
Analytical balance (laboratory)
Desiccator (laboratory)
Lab notebook or bench sheets (laboratory)

3.15 Characterizing the size distribution of streambed sediments

General

This subsection provides a simple method for
characterizing the size structure of streambed
sediments within headwater stream reaches.
Sediment characteristics influence many other
physical properties, including habitat stability,
interstitial habitat volume, nearbed velocities,
organic matter retention, and re-aeration.
Consequently, streambed sediments directly
and indirectly influence community structure
and stream processes. The characteristics of
the streambed are expected to influence
stream processes to a greater degree in
headwater streams than in larger rivers
because headwaters have a higher ratio of
streambed surface area to instantaneous flow
volume (m^2/m^3) than larger streams and rivers.
Geology, climate, topography, and drainage
area are factors that naturally govern the
natural composition of stream sediments.
Land-use changes can cause deleterious
alteration to streambed properties (e.g.,
siltation) and subsequent shifts in biological
integrity.

Particle size is the most common measure
used to characterize streambed sediments,
mainly because of the ease to which it can be
objectively quantified compared to other
characteristics (e.g., sphericity, specific
density). A frequently used method to
characterize sediments on streambed surfaces
is the Wolman pebble count procedure
(Wolman 1954, Leopold 1970, Kondolf & Li
1992), where the sizes of individual stones are
randomly selected and measured along a

reach. Vertical characterization can be done by coring (Cummins 1962, Everest et al. 1980, Wesche et al. 1989) and ground-penetrating radar (Naegeli et al. 1996, Huggenberger et al. 1998). Other aspects of the streambed sediments that have been measured include texture (Downes et al. 1998, Bergey 1999), porosity (Maridet et al. 1992), bed roughness (Statzner 1981, Ziser 1985), topographic complexity or fractal geometry (Schmid 2000, Robson et al. 2002, Stewart and Garcia 2002) and stability (Biggs et al. 1997, Duncan et al. 1999). The composition of streambed sediments influences aspects related to the rate of stream drying (i.e., permeability), wetted surface area as stream levels decline (boulder-dominated reaches will have more emergent sediments at low flows than gravel reaches), and the availability of interstitial refugia when streams are dry.

The protocol below is based on methods described in Walters et al. (2003) for particle size characterization by patches rather than individual grains or stones. Streambed sediment characterization is measured once for a given reach during the study because reach-level particle size measures are unlikely to change significantly over the timeframe of most ecological studies (1-2 years).

Procedure

Streambed surface sediments are measured at 31 locations, longitudinally at every meter mark along the thalweg of each 30-m study reach (Figure 3-58). Each particle size measurement is based upon 0.25 m² patches of particles, rather than a single particle measurement. The patches are centered around each meter mark (0, 1, 2, ... 30 m) along the study reach thalweg. The modal particle size class or the size class with the greatest patch coverage is estimated for each patch location. Once the patch is located, visually assess the size classes within each patch, determine which size class has the greatest coverage, and select a representative particle of that size class. The dimension used to determine particle size is the intermediate axis (i.e., β -axis) or the median value among the length, width, and height of the particle. Exact measurement of the intermediate axis is not needed because size classes are used. Particle size classes are based upon the Wentworth size classification or phi (Φ) scale (Cummins 1962, Table 3-1). The value for sediment particle size to be entered in the field form (Figure 3-59) is the upper bound value of the size class (bold-faced values in Table 3-1). The particle size classes are also listed on the bottom of page 3 of the field forms.

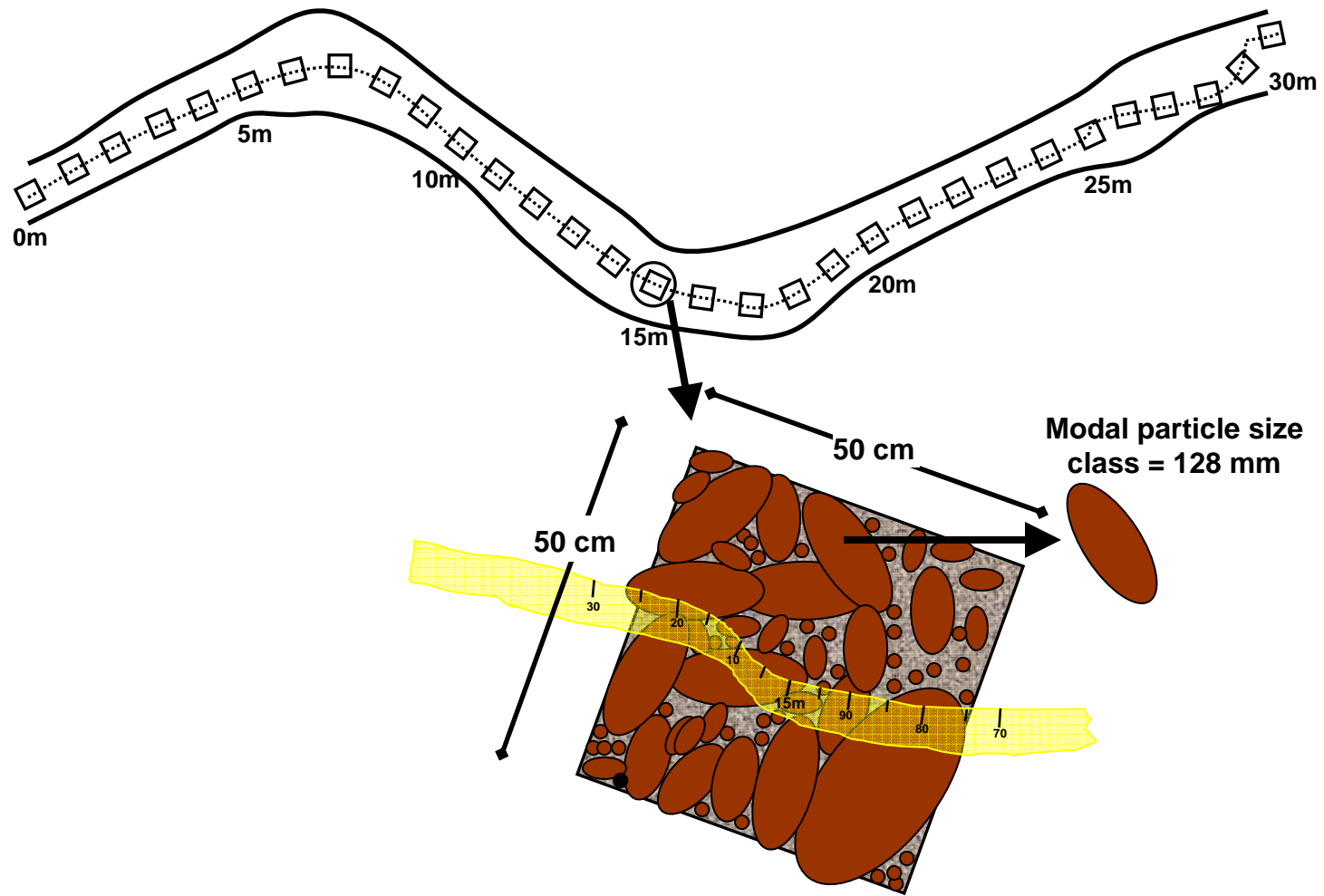


Figure 3-58 Schematic of study reach illustrating thalweg (dotted line) and patch locations for determining modal sediment particle size class. Inset provides a close-up of a patch (overlaid) with measuring tape used in designating patch locations longitudinally along the study reach).

Table 3-1 Modified Wentworth scale for sediment particle size classes. Bold-faced numbers indicate values to be entered on field forms

Class	Size range (mm)	Phi (Φ)
Sand, silt, and clay	\leq 2	\geq 0
Fine gravel	>2 to 4	-1 to -2
Medium gravel	>4 to 8	-2 to -3
Coarse gravel	>8 to 16	-3 to -4
Small pebble	>16 to 32	-4 to -5
Large pebble	>32 to 64	-5 to -6
Small cobble	>64 to 128	-6 to -7
Large cobble	>128 to 256	-7 to -8
Boulder	>256 to 512	-8 to -9
Bedrock and hardpan	> 512	\leq -9

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- Equipment and supplies*
Meter stick or ruler
Measuring tape (50m)
Field forms

* MEASURES TAKEN IN THALWEG § Where FPA Width > 2.2X BF Width then indicate: >2.2BF									
Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m) *	FPA width (m) §
0	< 2								
1	< 2								
2	4								
3	16								
4	> 512								
5	> 512								
6	16								
7	64								
8	64								
9	< 2								
10	< 2								
11	32								
12	8								
13	> 512								
14	256								
15	128								

Figure 3-59 Appropriate location for recording modal particle size data on page 2 of field forms (example from Figure 3-58 highlighted).

3.16 *In situ* water chemistry measurements

General

This subsection provides procedures for measuring *in situ* water chemistry of headwater streams. The basic water chemistry measurements discussed in this section are: 1) temperature, 2) conductivity, 3) pH, and 4) dissolved oxygen. Instructions for collecting water samples and measuring additional chemical parameters (i.e., nutrients, cations, anions) can be found in Wetzel and Likens (1991), Herlihy (1998), and APHA (2005). Because characteristics of water change with residence time, these measurements may be useful in distinguishing between groundwater

and throughflow (i.e., water in unsaturated soil zones during and immediately after precipitation). Physicochemical amplitudes (seasonal and diel) are typically greater in temporary waterbodies than in perennial counterparts (Zale et al. 1989, Boulton et al. 2000). As flow begins to decline, deeper groundwater inputs may represent the dominant source of surface flows, resulting in relatively subtle physicochemical shifts (Dahm et al. 2003). More dramatic changes in water physiochemistry can occur as waterbodies dry, and such changes can have equally dramatic effects on the inhabiting biota (Moore and Burn 1968, Magoulick and

Kobza 2003). Maximum diel variation and absolute extremes are commonly measured when surface water becomes limited to disconnected pools (Stehr and Branson 1938, Boulton and Lake 1990) and depending upon the pool depth, vertical stratification can occur (e.g., Neel 1951, Wood et al. 1992). Conductivity and water temperature typically increases as streams dry (e.g., Baron et al. 1998), whereas dissolved oxygen tends to decrease (e.g., Slack and Feltz 1968, Chapman and Kramer 1991). Declines in water volume from evaporation and evapotranspiration lead to greater water surface area to volume ratios that subsequently cause water temperatures to rise from rapid solar heating. Warmer water and contraction of surface water intensifies community respiration that can lead to declines in dissolved oxygen. Evaporation, increased soil residence time, and organic matter breakdown elevates stream water concentrations of dissolved ions and alters pH (Williams and Melack 1997, Hamilton et al. 2005). The buffering capacity (or acid-neutralizing capacity, ANC) of stream water will determine the direction of pH change during drying. In some streams, high leachate concentrations from organic matter may decrease pH (Slack and Feltz 1968). Increases in pH during dry seasons can occur where ANC is strongly influenced by acid rain or snowmelt during wet seasons (Wigington et al. 1996) or where stream water is naturally low in base cations (e.g., Ca^{++} , K^+ , Mg^{++}) and drying concentrates strong acid anions (e.g.,

SO_4^- , Cl^- , NO_3^- , Bayley et al. 1992). Although water quality can decline with drying, these changes may be mitigated where there is intact forest to buffer the stream environment (e.g., Feminella 1996). Conversely, reduced flows and drying exacerbates water quality problems in areas with nutrient input and removal of riparian canopy (Casey and Ladle 1976, Chessman and Robinson 1987), particularly if remaining flow is effluent-dominated (e.g., Lewis and Burraychak 1979, Jennings and Gasith 1993, Suren et al. 2003, Brooks et al. 2006). Because *in situ* water chemistry can vary considerably over time, measurements should be taken during each sampling visit. Note that the following procedure is for taking point measurements rather than measuring diel variation or extremes.

Procedure

Before arriving at the field sites read the instruction manual for portable meters and check batteries. Check to see that the meters are functioning properly and are calibrated. Use standards to calibrate meters at least daily. Record pre- and post-calibration values on the instrument log sheet (Figure 3-60). Calibrate the dissolved oxygen meter for the appropriate elevation for each study site (elevation can be read from the 7.5 min. topographic maps, or GPS units). Suggested data quality objectives (DQO) for *in situ* water chemistry are shown in Table 3.16.1.

Date	Instrument	Inspected by	Pass inspection (Y or N)	Degree deviated from calibration standard (+ or - include units)	Recalibrated (Y or N)
9/22/03	Hydrolab Quanta (#2)	KMF	Y	pH @ 3: +0.1 @ 7: +0.2	Y
9/22/03	Hydrolab Quanta (#2)	KMF	Y	Cond @ 45: +7 μ S @ 147: -12 μ S	Y

Figure 3-60 An example of an instrument inspection and calibration log sheet.

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. *In situ* water chemistry measurements should be taken before all other measurements. Note the location and time of measurements on the field form (Figure 3-61). If the pH, conductivity, and dissolved oxygen meters also measure temperature, consistently use one of these to measure temperature. When available, submerge probes in the area of flowing water (note that some probes cannot be completely submerged) and monitor the readout until values stabilize. Where hydrologic condition is “surface water in

pools only” (see Section 3.1 for designation of hydrologic condition), *in situ* water chemistry should be measured in all pools where biological samples are taken. Write values for measurements in the appropriate cells on the field form (Figure 3.16.2). Record time of day when measurements were taken in "comments" section. If additional space is needed use space on page 3 of the field form. Turn off meters and then repeat measurements to meet DQO in Table 3.16.1. If repeat measurements do not meet DQO standards then flag those values on the field forms to indicate that they are suspect.

Table 3-2 Data Quality Objectives (DQO) for in situ water chemistry measurements

Measurement	Data Quality Required
Temperature	two measurements taken with less than 5% deviation.
pH	two measurements with less than 10% deviation
Dissolved Oxygen (DO)	two measurements with less than 10% deviation
Conductivity	two measurements with less than 10% deviation

IN SITU WATER QUALITY MEASUREMENTS					
Location of Measurements	Cond ($\mu\text{S/cm}$)	Temp ($^{\circ}\text{C}$)	DO (mg/l)	pH	Comments
10 m	24	10	1.23	6.3	@ 9:30 am

Figure 3-61 Appropriate locations for recording in situ water quality measurements on page 1 of field forms, example values shown in red.

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- Equipment and supplies*
Measuring tape (50 m)
pH meter

Conductivity meter
Dissolved oxygen meter
Thermometer
Associated calibration standards for meters
Field form
Spare batteries

3.17 Measuring riparian canopy cover

General

This subsection provides instructions for measuring riparian canopy cover for headwater streams. Canopy cover is a useful measure of riparian condition that can strongly influence the structure (e.g. organic substrate, algal biomass) and function (e.g., primary production) of streams (Gregory et al. 1991, Naiman and Decamps 1997). This procedure is a modification of the original method described by Lemmon (1957) for use with a convex spherical densiometer. Measurements of irradiance with pyrhelimeters or photosynthetically active radiation with quanta sensors provide quantitative measures of incoming solar energy (Moulton et al 2002, also see reviews by Hauer and Hill 1996, Jennings et al. 1999). A disadvantage of these measures is their sensitivity to cloud cover and angle of the sun. Another method for estimating canopy cover is the use of fisheye or hemispheric photography (Davies-Colley and Payne 1998, Ringold et al. 2003, Kelly and Krueger 2005). Especially with the advent of digital photography and analytical software this method offers short processing times, consistency, and precision. One limitation of photographic methods is ensuring proper lighting conditions. Direct overhead

sunlight, reflection on vegetation, and dark clouds can lead to data misinterpretation (Kelly and Krueger 2005). Measurements of canopy cover are taken during each season (spring and summer) because this will likely change through time.

Procedure

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. Canopy cover is measured while facing upstream, downstream, left bank, and right bank at the 15-m mark of the study reach.

Canopy measurements are taken by holding the densitometer about 0.3 m above the stream surface at the thalweg. Level the densitometer using the bubble level and position it so that your reflection is just below the mirror grid (Figure 3-62). Calculate the percent cover by first identifying the grid intersections (of 37 total intersections) that are covered by vegetation (e.g., leaves, branches, trunks) or stream banks. Percent cover values for intersections are equivalent to the number of squares meeting at an intersection (Figure 3-62), ranging from 1% to 4%. For example, an intersection where 4 squares meet and is covered by vegetation is equivalent to 4% cover (Note that based on this value system, the total percentage is 98% and therefore approximates 100%). Sum percent cover and record in the appropriate cell on the field form (Figure 3-63).

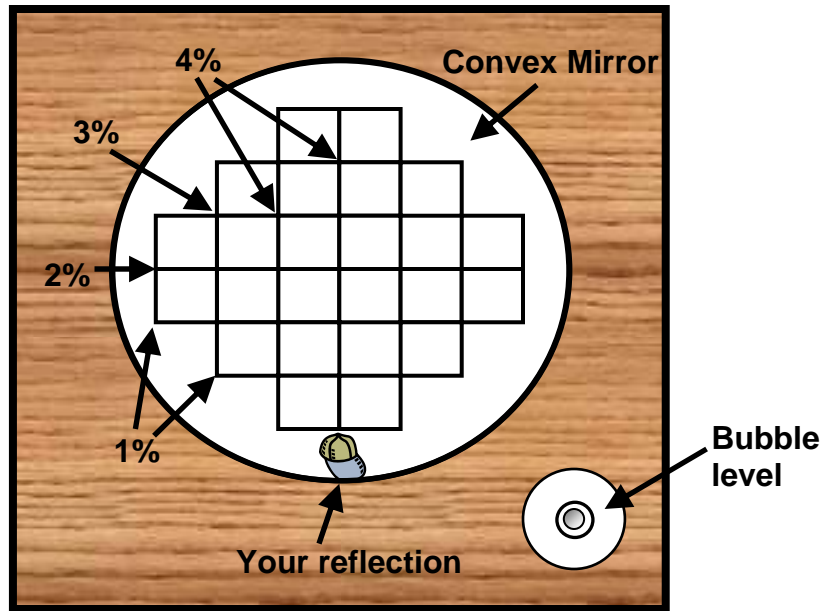


Figure 3-62 Plan view of a convex spherical densitometer, showing percent cover values associated with intersections. Values are equivalent to the number of squares meeting at each intersection.

Where canopy over the stream channel is heavy, it is more efficient to measure the percentage of open by identifying and summing grid intersections values that are not

covered by vegetation, etc. Percent canopy cover is then simply calculated by subtracting total percent open canopy from 100%.

DISTANCE TO NEAREST SURFACE WATER (m)				CHANNEL SLOPE (%) (for three 10 m sections of study reach)			% CANOPY COVER (facing upstream, downstream, right & left banks)			
0	<100	100-500	>500							

Figure 3-63 Appropriate location for recording percent canopy cover on page 1 of field forms.

The methods used by USEPA’s EMAP and USGS’s National Water-Quality Monitoring Program (NAWQA) differ slightly from the method discussed above. Rather than using all 37 intersections on the convex mirror for measurements, only 17 intersections are evaluated (Figure. 3-64, Fitzpatrick et al. 1998, Kaufmann and Robison 1998). A “V” is taped on the mirror surface to delimit the 17 intersections. This modification is intended to minimize repeated observations of cover

structures during multiple readings from the same position (e.g., facing upstream, downstream, left and right bank) and reduces measurement time (Strichler 1959). Each intersection is weighted equally, rather than by the number of squares meeting at the intersections. The number of covered intersections is recorded for measurements facing upstream, downstream, left and right banks (standing at mid-channel) for the 11 transects (per study reach) in the EMAP

protocol (Kaufmann and Robison 1998), whereas two measurements, facing the each bank at the water's edge, are taken at the 11 transects (per study reach) in the National Water Quality Assessment (NAQWA) protocol (Fitzpatrick et al. 1998). The NAQWA protocol measures canopy closure, rather than canopy cover (also called canopy density). Canopy closure includes the

overhead area bracketed by vegetation, whereas canopy density includes only area of sky completely blocked by vegetation. Canopy closure is intended to be less influenced by season (i.e., leaf abscission) than canopy density (Strichler 1959). For both protocols, percent cover or closure is calculated as the ratio of covered to total intersections.

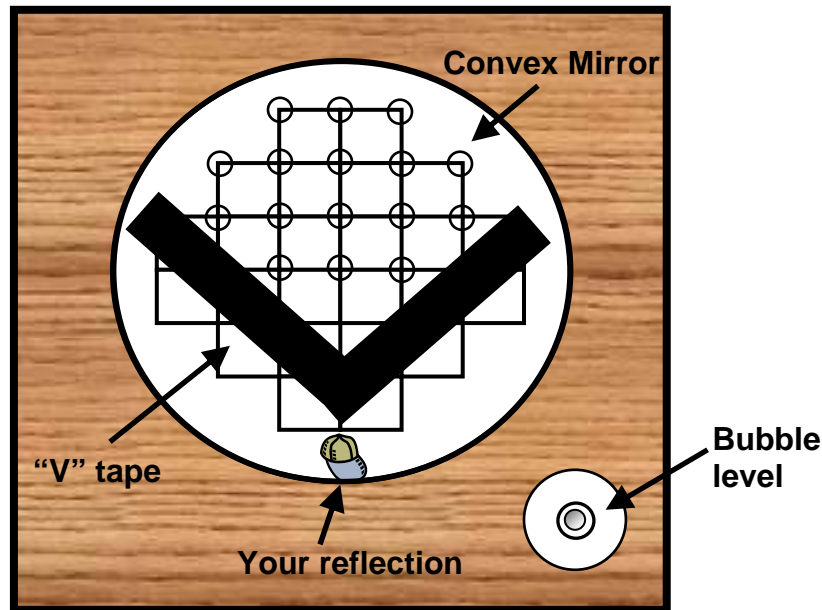


Figure 3-64 Plan view of a convex spherical densitometer, modified for measuring over 17 intersections (open circles) that are delimited by a “V” taped to the convex mirror.

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- Equipment and supplies*
 Measuring tape (50 m)
 Convex spherical densiometer
 Field forms

4 BIOLOGICAL SAMPLING

This section provides background material related to biological sampling in headwater streams. The incorporation of biomonitoring (i.e., the use of organisms to evaluate changes in an environment) into assessment programs is advocated because biota are ubiquitous across the landscape, represent a diversity of responses, integrate stressor effects over time, and are relevant to societal needs (Rosenberg and Resh 1993, Whitton and Kelly 1995). The primary biological levels used in biomonitoring are community and population levels, although biological measurements can range from molecules to ecosystems. The response measures in bioassessment are typically abundance, biomass, and diversity; however, there is a trend toward quantifying characteristics (i.e., species traits) and functional roles of biota for predicting biological responses to specific disturbances or stressors. Some of these traits include life span, maximum size, phenology, and physiology (Muotka and Virtanen 1995, Biggs et al. 1998, Charvet et al. 1998, Usseglio-Polatera et al. 2000). Species traits may provide important insight in understanding stressor-specific responses and have a place in bioassessment, as do tolerance values (e.g., Hilsenhoff 1987, Van Dam et al. 1994) and functional feeding groups.

The choice of biological level and group should match the study objectives. Be aware of attributes and limitations of particular taxonomic groups. For example, primary producers will respond immediately to changes in light and nutrients, whereas a lag-response is expected for consumers. Long-term monitoring of individuals is possible for most bryophytes, but not so for invertebrates with relatively short life spans. In addition, ethical and legal considerations (e.g.,

sampling permits) are more prevalent for some biota than others. Particular sampling regimes may also be more conducive to some groups than others. For instance, organisms with a patchy distribution may require larger sample areas (or more samples) than those with a uniform distribution.

As discussed for physical habitat assessments, methods of biological sampling can range from qualitative to quantitative. Sampling methods should match the investigators' study objectives. Objectivity, comparability and precision of the methods increase as the level of quantification increases. For example, quantitative methods measure biota over a specified area or volume (e.g., Hess sampler) with greater precision and repeatability than semi-quantitative (e.g., kick nets) or qualitative methods (e.g., dip net jabs). However, the level of effort (especially time) and training may increase with more quantitative methods. Therefore, when deciding on a sampling method, one should consider the purpose of the resulting data (e.g., species list for the area, statistical comparison among treatments) and the resources available to accomplish the study objectives. Although not discussed in detail in this manual, post-sampling procedures are equally important to consider and should match the study objectives (see Klemm et al. 1990 and Charles et al. 2002).

Their small size and likelihood of drying make headwater intermittent streams unique habitats for sampling biota. Many methods developed for perennial streams may not be as effective or consistent in headwater streams. Water depth and flow may not always be sufficient for some sampling methods. In addition, the sampling area may need to be reduced to

minimize damage to streams and populations (or even to logistically collect a sample). The fluctuation of flow affects the wetted surface area to a greater extent in headwater streams than in larger, downstream water bodies. Therefore, it is critical to monitor wetted surface area when sampling biota. As streams dry, surface water contracts and organisms may track surface water and concentrate (e.g., Stanley et al. 1994). A density increase may be misinterpreted as increased abundance in response to drying if the context of wetted surface area is ignored. Studies that compare biological responses across time periods and/or among habitat types should use sampling methods that are equally efficient across the range of associated hydrologic conditions (Resh 1979, Boulton 1985)

The study objectives and the spatial distribution of the fauna should determine the number of samples or total sample area. Where diversity (or richness) is of interest, species-area relationships should be assessed to determine the appropriate sample area. Under ideal circumstances the number of species collected over area sampled should level-off. Therefore, the appropriate sampling area should coincide with the asymptote (where slope ≈ 0) of the species-area curve. However, because of the diverse and patchy, but numerically skewed nature of aquatic assemblages, the effort needed to reach the asymptote is typically enormous and logistically unattainable (Figure 4-1A). In addition to the preponderance of rare taxa, the limitations associated with fine-scale sample stratification among habitat patches contribute

to the inability to attain the species-area asymptote. An alternative goal that is more feasible to achieve (in time and effort) is the asymptote of the relationship between species gained and sample area (Figure 4-1B). This relationship measures the amount of information gained per unit effort. Similar considerations should be applied where laboratory subsampling is done prior to enumerating and identifying organisms (Vinson and Hawkins 1996, Larsen and Herlihy 1998). Because taxa richness increases with the number of organisms sampled, another consideration when comparing among sites or treatments is to standardize for the number of individuals or rarefy the data (e.g., Downes et al. 1998, McCabe and Gotelli 2000).

The following subsections are organized according to biological group. Included are more traditionally used communities of algae and invertebrates as well as less commonly used bryophytes and amphibians. Sampling methods used in Headwater Intermittent Streams Study are detailed. The identification of indicators of flow permanence was the objective of this study, sampling commonly was restricted to the thalweg. This was done because it was a consistent and conservative target when comparing across sites with varying hydrologic permanence and ecological condition. Other spatial configurations of field samples may be more suitable depending upon the study objectives. Alternative sampling methods are briefly discussed at the end of each subsection.

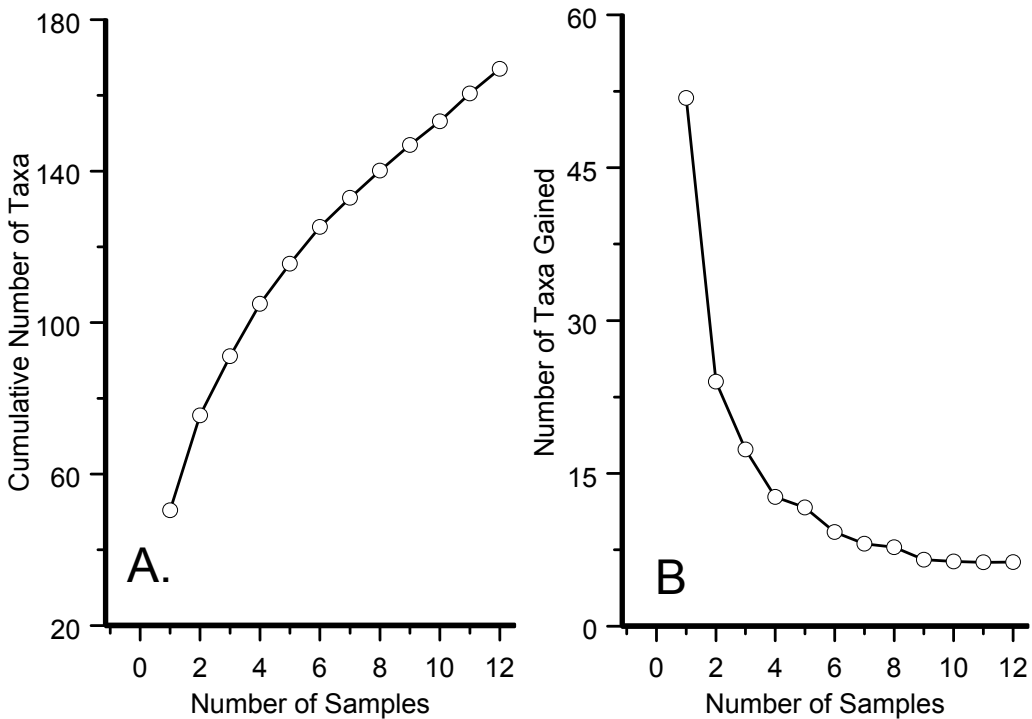


Figure 4-1 Examples of a species-area curve (A) and a species gained-area curve (B) for benthic invertebrates samples (sample area = 0.053 m²) collected from a perennial site on Falling Rock Branch, Robinson Forest, KY. Each point represents the mean (± 1 SE) of 100 permutations.

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4.1 Sampling the bryophyte assemblage *General*

This section describes sampling methods for bryophyte assemblages (mosses and liverworts) in headwater streams. Bryophytes include the nonvascular, seedless plants belonging to the classes Musci (mosses, Figure 4-2) and Hepaticae (liverworts, Figure 4-3). Both groups share a life cycle composed of two generations, the sporophyte (spore-producing) and gametophyte (gamete-producing). The sporophyte is directly attached to and nutritionally dependent upon the larger and longer-lived gametophyte.

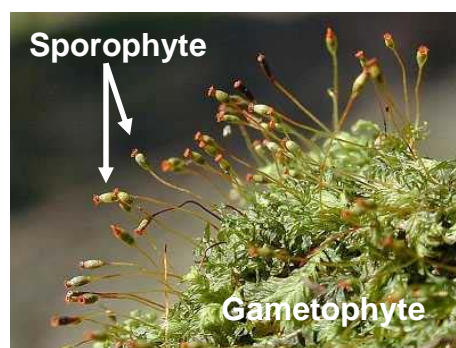


Figure 4-2 Sporophyte and gametophyte generations of a moss. (Photo by Michael Lüth)



Figure 4-3 An epilithic moss (Musci) growing in a headwater stream.



Figure 4-4 An epilithic liverwort (Hepaticae) growing in a headwater stream.

A fundamental gradient that governs the spatial distribution of bryophytes is moisture (Craw 1976, Glime and Vitt 1979). Bryophytes range from being xerophytes (adapted to dry habitats) to obligate hydrophytes (requiring water). This range enables mosses and liverworts to be potentially useful indicators of headwater stream hydrology. Additionally, because bryophytes are sessile and relatively long-lived compared to other stream-dwelling organisms, their distributions may be useful descriptors of hydrologic and ecological conditions over several years. Bryophytes have been used to monitor heavy metals and other pollutants through accumulation in tissue (e.g., Glime 1992, Engleman and McDiffett 1996), biochemical change (Lopez and Carbeilleira 1989), and species composition (Vrhovšek et al. 1984, Stephenson et al. 1995). Shifts in biomass, species dominance and composition of bryophyte assemblages have been linked to changes in water chemistry (Omerod et al. 1987, Bowden et al. 1994), sediment particle size (Vuori and Joensuu 1996), and hydrology (Englund et al. 1997, Downes et al. 2003). Texts for taxonomic identification of bryophytes are referenced at the end of this subsection.

Procedure

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg.

4.1.1 Qualitative sampling

Bryophyte sampling is confined to the thalweg (deepest flow path) of the 30-m study reach. Avoid unattached specimens or specimens growing on loose woody debris because these may have recently been deposited from adjacent forest or upstream. Most specimens suitable for sampling will be growing on stone substrate and submerged in water (see Figures 4-2 and 4-3). In dry channels be careful to select only specimens growing within the thalweg. If no specimens are found note this on the field form (Figure 4-4). Scrape small samples (~ 10 cm²) of all representative species seen in the thalweg using a scoopula or similar tool. Collect specimens with the sporophyte generation whenever possible, because the sporophyte characteristics are often critical for species-level identification. Place all collected specimens from a study site into a single 24-oz Whirl-Pak® bag with a sample label that includes relevant information (e.g., locality, date, collector’s initials). Keep samples cool (cooler with ice bags or ice packs) while transporting them to the laboratory and until the sample can be air-dried in the laboratory. Protect samples from ice meltwater. In the laboratory remove samples and associated labels from Whirl-Pak® bags and place them into paper bags or envelopes for air drying. Write the label information on the outside of the envelopes with a permanent marker.

BRYOPHYTES SAMPLED: Y N	
SAMPLE ID _____	COMMENTS

Figure 4-5 Appropriate location for recording bryophyte sample information on page 1 of field forms.

4.1.2 Quantitative sampling

As opposed to the qualitative sampling described above, quantitative sampling of

bryophyte assemblages requires field identification (or at least recognition of distinct taxa) and therefore some expertise on

the flora (Slack 1984, Bowden et al. 2006). There are three primary measures used to quantify bryophytes by taxa: frequency, percent cover, and standing crop. Frequency measures the proportion of the samples collected that contains a taxon, whereas percent cover measures the proportion of the sample area that is covered by a taxon. Standing crop biomass is a measure of the biomass of a taxon within a sample and is usually reported as g m⁻² dry weight or ash-free dry mass. The advantage of the percent cover and frequency over standing crop biomass is that they are non-destructive, enabling subsequent measurements. Percent cover is usually considered an estimate and therefore more subjective than frequency or standing crop biomass. Some investigators have subdivided the sample area using plexiglass grids or other viewing devices to improve repeatability. In addition, rather than using absolute percentages, percentage categories are often used when estimating cover (e.g., Braun-Blanquet cover scale, see Mueller-Dombois and Ellenberg 1974, Bowden et al. 2006). Voucher specimens for each taxon are collected for later identification or confirmation in the laboratory.

Three sampling methods commonly used and are listed by increasing level of effort: point-intercept, transect, and quadrat (i.e., plot). Points, transects and quadrats should be randomly or haphazardly selected across the study reach to avoid samples biasing their reach representation. Depending upon the study objectives, it may be useful to stratify sampling within a study reach (e.g., habitat type, stream margin versus mid-channel, height relative to water surface). Stratified sampling (or floristic habitat sampling) has been advocated when compiling comprehensive surveys of bryophyte diversity (Newmaster et al. 2005). The point-intercept method uses a grid or coordinate system.

Each randomly or haphazardly selected coordinate (point) is sampled by simply recording the species present at the point, on the nearest substrate (e.g., cobble), or a surrounding area (making it similar to quadrat sampling). The transect method uses randomly or haphazardly placed transects (measuring tape or string) typically positioned perpendicular to the direction of flow. Sampling along transects may span only the wetted width, entire active, or into the adjacent riparian zone. Percent cover for each species is determined by the percent of the transect length that is intercepted by each species. Frequency may be assessed among transects or within transects by recording individual species-patches along each transects. Some investigators treat transects as belts, where bryophytes are sampled within a set distance (e.g., 0.1 m) upstream and downstream of each transect (e.g., Steinman and Boston 1993, Suren and Duncan 1999). The quadrat method uses circular, square, or rectangular plots of known area that are randomly or haphazardly positioned in the study area. The percent cover of each species within the quadrat is recorded and frequency is typically assessed across replicate quadrats. Some investigators have sampled quadrats along transects to quantify assemblage shifts across geomorphic units (e.g., Jonsson 1996). Potential edge effects (perimeter:area) are lower for circular plots than for square or rectangular plots (Krebs 1999). The number and size of replicate sampling units depends on the patchiness of bryophytes with the study reaches and the resources available for the study. Studies comparing transect, point intercept, and quadrat methods have generally found similar estimates of bryophyte and macroalgae abundance (Rout and Gaur 1990, Neechi et al. 1995). However, because the quadrat method usually covers a larger sampling area, this method will include more

rare taxa and tend to have higher estimates of taxa richness.

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Equipment and supplies

Measuring tape (50 m)
Metal scoopula or spatula
24 oz Whirl-Pak® bags
Pencils
Permanent marker
Label paper
Field forms
Cooler
Ice or ice packs
Paper bags or envelopes

4.2 Sampling the epilithic algal assemblage *General*

This subsection describes methods for sampling the algal assemblage in headwater streams. The particular algae sampled in these procedures are epilithic algae (or algae associated with stone surfaces) and includes diatoms (Bacillariophyta) and “soft” algae (i.e., Chlorophyta, Cyanophyta, Rhodophyta, and Chrysophyta). Epilithic algae are associated with fungi, bacteria, heterotrophic protists, and organic matter and together they form a matrix called periphyton, biofilm, or aufwuchs. The target organisms for laboratory identification are the algae within the periphyton, but because algae are difficult to exclusively collect, the periphyton is sampled. Algal assemblages have been shown to be useful indicators of ecological condition in wadable streams (e.g., Pan et al. 1996, Hill

et al. 2000). The ubiquity, diversity, sampling efficiency, and responsiveness to physical and chemical stressors are all attributes for the use of algae in bioassessment (Patrick 1973, Stevenson and Lowe 1986). Despite being ubiquitous, algae have received less attention than invertebrates in temporary streams research (see review by Stanley et al. 2004). Algae are potentially useful indicators of hydrologic permanence because algae inhabit a wide range of habitats (terrestrial to aquatic) and varying in desiccation tolerance and presence of resistant structures (e.g., akinetes, cysts, zygotes, mucilage) among taxonomic groups (Davis 1972).

4.2.1. Quantitative sampling of epilithic algae

This first procedure is modified from the procedure described by Hill (1998) and focuses only on the collection of periphyton on natural substrates to determine the taxonomic composition of the algal assemblage (by abundance and biovolume). The algae assemblage is sampled during each season (spring and summer) because it is likely to vary with season.

Procedure

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. Identify erosional and depositional habitats with the study reach. Two separate algae samples are taken from each study reach, one from each habitat type (erosional and depositional). Sampling is confined to the

thalweg of the study reach and is done regardless of hydrologic condition. Each sample is a composite of 12 cm² areas from upper surface of 6 individual stones.

4.2.1.1. Substrate collection

Begin by haphazardly collecting 6 stones (>12 cm² upper surface area) from the thalweg of a habitat type and placing them in the large basin with the upper surface facing upward. Avoid disturbing the streambed as much as possible when collecting stones and make sure that the stones have not been disturbed by other sampling activities (communicate with fellow crewmembers). Spread the sampling across multiple units of each habitat type along the study reach. However, where hydrologic conditions vary among units of (or stones from) a habitat type in a study reach (i.e., there are pools with and without surface water), restrict sampling to the dominant hydrologic condition represented by the habitat units within the study reach. For example, if a study reach has 5 depositional habitat units and 4 had surface water and one was dry, collect the 6 stones from the 4 wet units. Indicate on the field form the number of stones collected and whether the stones collected were wet or dry (Figure 4-5). Stones can be randomly selected within available habitat units in the reach. Numbers ranging from 1 to 100 can be drawn and the nearest stone along the thalweg coinciding proportionally with the unit length is selected.

ALGAE				
# STONES SAMPLED: Depositional Habitats <u>6</u> Erosional Habitats <u>6</u> TOTAL AREA SAMPLED: <u>144</u> cm ² (each 12 cm ²)				
SAMPLE ID	HABITAT TYPE	NUMBER OF BAGS	COLLECTED BY:	COMMENTS
Four - JC - 1 - D	Depositional	1 of 1	KMF	Wet
Four - JC - 1 - E	Erosional	1 of 1	KMF	Wet

Figure 4-6 Appropriate location for recording algal sample information on page 1 of field forms. Example information is shown in red.

4.2.1.2. Compositing and preserving sample
Fill the wash bottle to the 50-ml mark with stream water. Place a 1.5" diameter PVC circle (i.e., delimiter) on the upper surface of a stone to define a 12 cm² area. Use the metal spatula and a firm-bristle toothbrush (trim bristles to half original length) to dislodge algae from the stone surface within the delimiter (Figure 4-6). Rigorously scrape and brush the surface for 30 seconds. Be aware that because clay particles have similar density (mass per unit volume), excess clay particles in the sample may hamper identification and enumeration of algae. Using the wash bottle, sparingly wash the dislodged algae from the delimited area into the small plastic container. Repeat this step with the 5 remaining stones to make a composite sample. Use the remaining water in the wash bottle to rinse off tooth brush and spatula into the small basin. Pour the composite sample from the small container through the small funnel and into a 50-ml centrifuge tube. Use a syringe or bulb pipette to add 2 ml of 10% formalin to preserve the sample*.

If formalin is not taken to the field, keep sample in the dark and on ice until it is preserved. Tightly cap the centrifuge tube and seal with electrical tape. Gently shake the tube to distribute the formalin throughout the sample. Make a label (waterproof paper and pencil) that includes relevant information (e.g., locality, habitat type, date, collector's initials). Attach the sample label securely to the outside of the centrifuge tube using packing tape or clear tape strips. Also write

* Wear gloves and safety eyeglasses when using formalin and work in a well-ventilated area. Formalin is extremely caustic and potentially carcinogenic. It may cause severe irritation on contact with skin and eyes. Rinse immediately with water in case of contact with skin or eyes.

label information on the field form (Figure 4-5). Thoroughly rinse all sampling equipment with clean stream water to prevent cross-contamination between samples. If additional measures of biomass and/or pigment are needed, the sample can be split volumetrically (see Hill 1998).

Repeat procedures outlined in 4.2.1.1 and 4.2.1.2 for the remaining habitat type.



Figure 4-7 Collecting epilithic algae from a stone within the sample delimiter.

4.2.1.3. Sample transport and shipping
Before leaving a site, check that all samples are labeled properly and safely stowed. At the vehicle, consolidate all algae samples in one location. All the samples can then be placed in a crush-resistant and leak-proof container and transported to the laboratory for processing. If samples need to be shipped, include any special shipping forms that may be required for the formalin-preserved samples.

4.2.2 Alternative methods

Here we briefly describe various techniques used to sample epilithic algae in streams. More details are available in reviews by Stevenson and Lowe (1986) and Aloi (1990). Algal sampling methods can be separated into

two broad categories: natural substrates and artificial substrates. As previously described, natural substrate sampling involves quantitatively collecting epilithon found growing naturally on substrates in streams. In contrast, artificial substrate sampling involves placing substrates (e.g., glass slides, ceramic tiles, bricks) into streams for periphyton colonization. Because the exposure time is known, artificial substrates provide the investigator with more control and perhaps less variability among sampling units than natural substrates. However, assemblages colonizing artificial substrates may not provide a realistic characterization of the algal assemblage in streams. Taxonomic composition and measures of biomass (chlorophyll *a* and AFDM) on artificial substrates can differ from these algal measures on natural substrates (Lay and Ward 1987, Cattaneo and Amireault 1992). Homogeneous substrate texture and short incubation times of artificial substrate procedures have been identified as the likely causes for lower algal biomass and lower representation by green and blue-green algae than seen in adjacent natural substrates. Both methods have benefits and drawbacks for monitoring algal assemblages and these should be weighed carefully when designing monitoring studies.

Algae in headwater streams are often logistically easier to collect than from deeper rivers and lakes. Many substrate types are easily removed from the stream for subsequent collection of algae. Rather than subsampling periphyton on a substrate particle, several investigators have used the entire substrate as a sampling unit (e.g., Biggs and Close 1989, Dodds et al. 1999, Mosisch 2001). Surface area of substrates can be estimated using substrate dimensions and geometric equations (e.g., Graham et al. 1988). Others have determined stone surface area by covering stones with aluminum foil,

plastic wrap, or ink stamps (Doeg and Lake 1981, Lay and Ward 1987). Surface area – weight relationships are then used to determine surface area of substrates. Large boulders and bedrock common to steep headwater streams can pose a problem in retrieving samples and quantifying surface area. Syringe type samplers (e.g., Loeb 1981, Flower 1981, Peters et al. 2005) offer a solution, where a sample can be collected *in situ*. Syringe samplers use brushes to remove attached periphyton within an enclosed area; then the sample is suctioned and transferred onto a filter or into a sample container. One drawback noted about syringe samplers is an underestimation of chlorophyll *a* concentrations from stream samples but not from lake samples (Cattaneo and Roberge 1991). Sampler brushes are likely ineffective at removing tightly attached members of the periphyton assemblage in streams. Davies and Gee (1993) developed a scouring disc for periphyton removal and reported higher concentrations of chlorophyll *a* were obtained from the scouring disc than from either brushing or scraping. Algae associated with fine particles (epipsammon and epipelon) can be sampled simply by using an area delimiter (e.g., inverted petri dish) and spatula or trowel. The delimiter is positioned into the upper sediment layers and the spatula is positioned beneath. The spatula is carefully lifted and the sample is transferred to a sample container using a funnel. Because algae may be firmly attached to sand grains, additional laboratory steps (i.e., sonication) are needed prior to microscopic or fluorometric measurement (Miller et al. 1987, Romaní and Sabater 2001). Further details on sampling algae from various substrates are discussed in Moulton et al. 2002.

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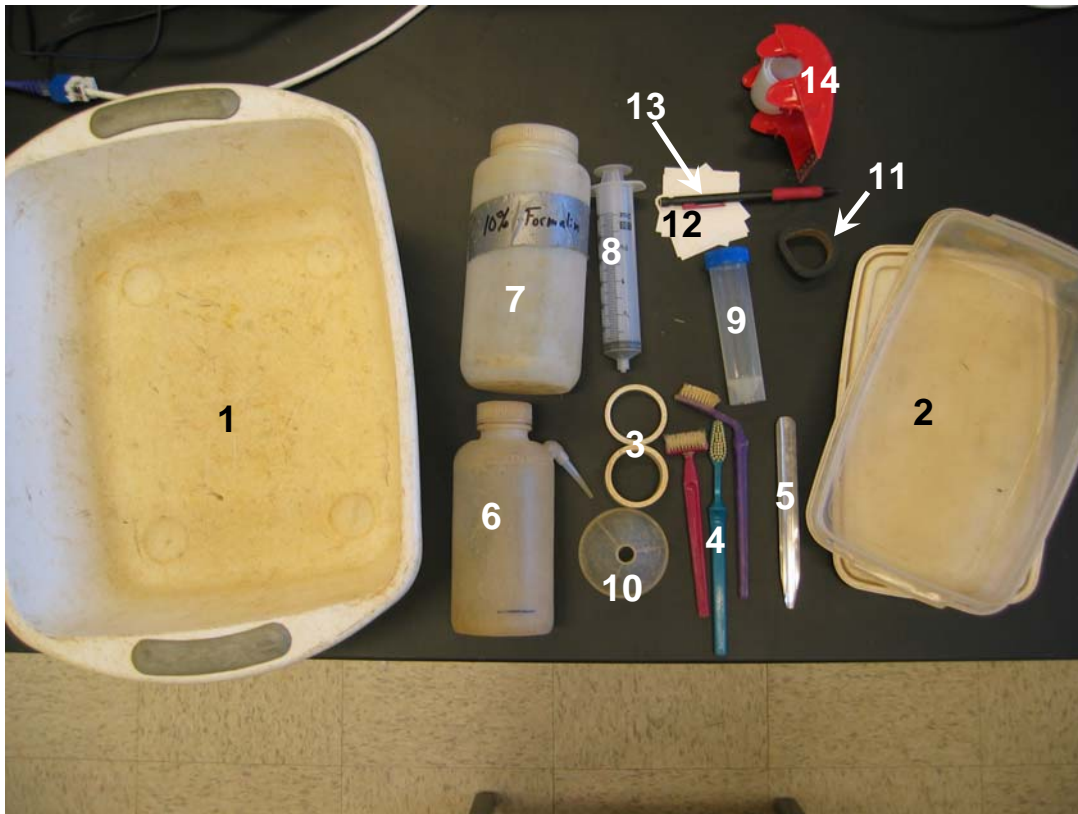


Figure 4-8 Equipment used to collect and preserve algal assemblage samples. Numbers correspond to the Equipment and Supplies list.

Equipment and supplies (Numbers correspond to items in Figure 4-7)

Measuring tape (50 m)

1. Plastic wash basin (approximately 35 cm x 29 cm x 14 cm)

2. Small plastic container or basin (e.g., Tupperware® or Rubbermaid® container) (approximately 25 cm x 16 cm x 6 cm, large enough to contain a cobble and large gravel particle and can be used to store items listed below)

3. PVC ring delimiter (1.5 in or 3.8 cm diameter pipe cut 2 to 3 cm in length)

4. Firm-bristle toothbrush (2) – trim bristles to half their original length

5. Spatula or scoopula

6. Water squirt bottle (with 50 ml volume marked)

7. Buffered formalin (10%)

8. Small syringe or bulb pipette

9. 50 ml centrifuge tubes

10. Small funnel

11. Electric tape

12. Label paper

13. Pencils

14. Packing tape or clear tape strips

4.3 Visual and tactile assessment of algal cover

General

This subsection provides instructions for rapidly assessing algal cover in headwater streams. The method uses a categorical index, Algal Cover Index (ACI) that is based on visual and tactile characteristics of periphyton (and associated algae). The ACI scores and associated characteristics are shown in Table

1. The ACI has been field tested and ACI scores have explained 68-85% of the variation in measured levels of algal (chlorophyll a) and periphyton (AFDM) biomass in streams (Feminella and Hawkins 2000). The protocol described here is modified from Hawkins et al. (2001). Another field-based rapid periphyton method that separately characterizes macroalgal and microalgal cover is described in Stevenson and Bahls (1999).

EMAP protocols include percent classes for filamentous algae (Kaufmann and Robison 1998). The NAQWA qualitative algae sampling protocol includes designating the abundance classes (dense to none) for periphyton at a site (Moulton et al. 2002). The ACI is measured during each season (spring and summer) because it is likely to vary with season.

Table 4-1 Algal Cover Index (ACI) scores and their associated characteristics

ACI Score	Visual and Tactile Characteristics
1	substrate is rough with no apparent growth
1.5	substrate is slimy, but biofilm not visible (i.e., tracks from scratching rock with back of fingernail is not visible)
2	thin layer visible (0.5-1 mm thick, i.e., tracks from scratching rock with back of fingernail is visible)
3	algal mat thickness ranges from 1-5 mm thick and filamentous algae is rare
4	algal mat thickness ranges from 5-20 mm thick and filamentous algae common
5	algal mat thickness >2 cm and/or filamentous algae dominates

Procedure

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. Algal cover assessment is based on ≥ 25 substrate particles on the streambed surface. Particles assessed should be spread along the thalweg of the entire study reach. Final ACI score for the study reach is based on the dominant score of the assessed particles. Where there is a clear discrepancy between habitat types (i.e., depositional and

erosional), note ACI scores for both habitat types. Algal cover can be assessed while sampling benthic invertebrates (assessing surface cobble and gravel while scrubbing attached invertebrates). Photographic examples of ACI scores are shown in Figure 4-8. The ACI score is circled on the field form (Figure 4-9). The ACI scores and associated characteristics are also listed on the bottom of page 3 of the field forms.

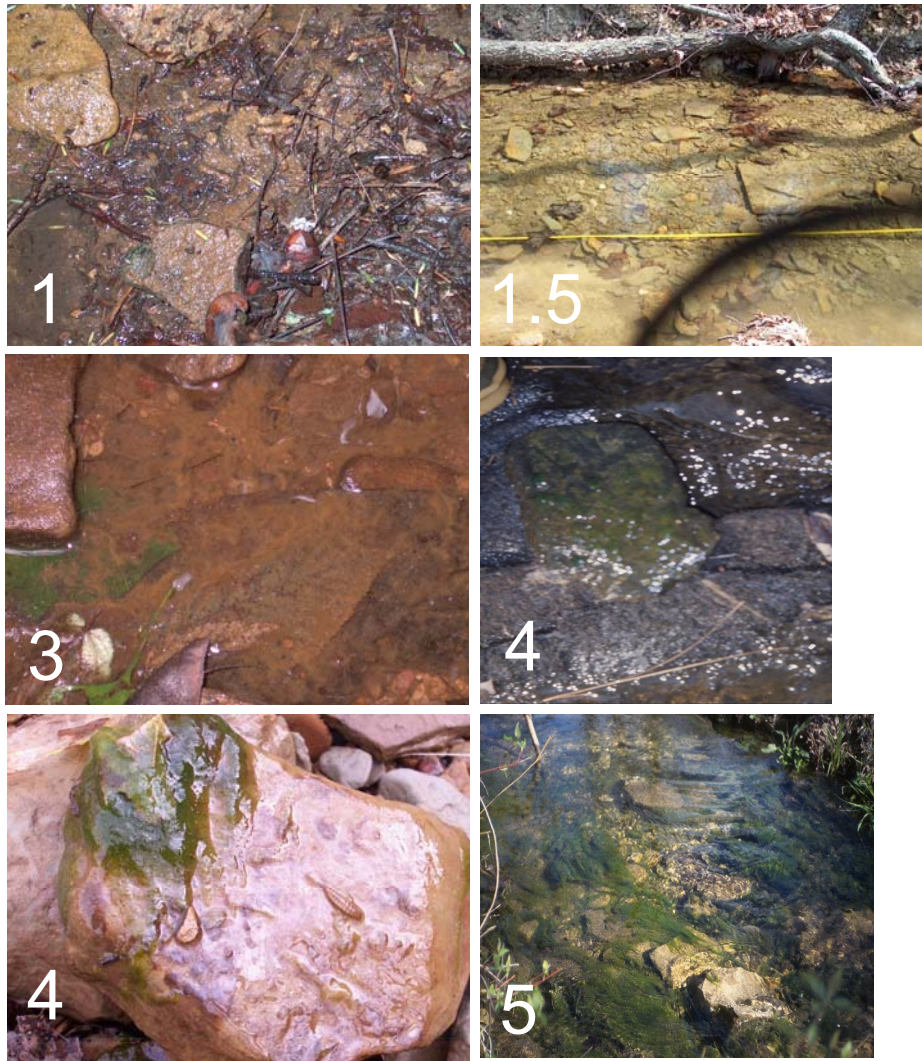


Figure 4-9 Categorical examples of algal cover based on visual and tactile characteristics. Numbers represent Algal Cover Index (ACI) scores associated with periphyton on stones in the photographs.

PRESENCE OF HEADCUT IN REACH		ALGAL COVER INDEX						# CORES FOR SUBSTRATE MOISTURE (depositional)
Y	N	1	1 ^{1/2}	2	3	4	5	

Figure 4-10 Appropriate location for recording the dominant reach score for the Algal Cover Index on page 1 of field forms.

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Equipment and supplies

Measuring tape (50 m)
Field forms

4.4 Sampling the benthic invertebrate assemblage

General

This subsection provides methods for quantitatively sampling the benthic invertebrate assemblage in headwater streams. Benthic invertebrate surveys are widely used to evaluate the condition or health of water bodies (Hellawell 1986, Rosenberg and Resh 1993, Rader et al. 2001). Invertebrate assemblages are composed of a wide range of taxonomic and functional groups, many of which can be found in headwater streams. Furthermore, a diversity of life histories (e.g., voltinism, cohort production interval, dormancy stages) and physiological tolerances are found among aquatic invertebrates (Williams 1996, Frouz et al. 2003). Habitat characteristics (e.g., predictability, disturbance intensity, productivity) set the template governing the evolution of life histories and therefore the composition of assemblages (Southwood 1977, Townsend and Hildrew 1994). Flow is considered one of the ultimate drivers of lotic systems (Lytle and Poff 2004), and may be even more critical to temporary water bodies (Walker et al. 1995, Schwartz and Jenkins 2000). Thus, the composition of invertebrate assemblages should reflect the flow permanence in headwater streams. However, among past investigations there is no consensus regarding the distinctiveness of invertebrate communities among stream reaches of different flow permanence (Deluchi 1988, Feminella 1996, Dietrich and Anderson 2000, Fritz and Dodds 2002, Price et al. 2004). As is often the case in ecological systems, this disparity suggests that the relationship between flow permanence and assemblage organization may be complex.

Plasticity of life histories, subtle variation of drying intensity, degree of connectivity to refugia, physiographic variation, anthropogenic impacts, and other factors may influence assemblage structure.

4.4.1 Quantitative bucket sampling for invertebrate assemblage

The surface water conditions of many headwater streams fluctuate from continuous flow to only standing water in pools to complete absence. Therefore, the method used to collect benthic invertebrates needs to be effective and consistent across the range of hydrological conditions seen in headwater streams. Many existing sampling methods take advantage of flowing conditions to trap invertebrates in nets positioned downstream of the sampling area (e.g., Surber sampler, kick net); however, flow in headwater streams is often too low (sometimes absent) to effectively use these methods. The quantitative bucket sampling method described here is: 1) not dependent upon flow conditions, 2) performed by a single operator, 3) light weight, and 4) inexpensive. The bucket sampling method is modified from methods described by Wilding (1940) and Statzner (1981). The sample area of a 5 gallon bucket sampler is 0.053 m² (26-cm diameter). A smaller sample area (e.g., coffee can) may be required to collect benthos from step-pool streams dominated by boulders and large woody debris. The benthic invertebrate assemblage is sampled during each season (spring and summer) because it is likely to vary with season.

Procedure

Before hiking to the study reach(es) make sure all the equipment is stowed in the backpacks and there are ample Whirl-pak bags and ethanol. Transfer the 95% ethanol (usually from 5 gal. container) into transport jug(s) using a funnel. Typically 1 to 1.5 liters is

sufficient to preserve all the samples taken from a study reach.

Delineate the 30-m study reach so that the measuring tape is positioned along the thalweg. Identify erosional and depositional habitats within the study reach (see Subsection 3.4 for designating habitat units). Preliminary data from perennial and intermittent headwater streams in Kentucky, Indiana, and Ohio indicate that the number of additional taxa collected within a 30-m reaches an asymptote after 8 samples (0.42 m², see Figure 4.1B). We recommend that investigators independently assess species-area curves for study reaches, particularly if estimation of invertebrate diversity (a common metric used in ecological condition assessments) is an objective.

Eight separate invertebrate samples (each 0.053 m²) are haphazardly taken from each study reach, 4 from each habitat type (erosional and depositional). Samples are kept separate to 1) determine the sampling effort needed to sufficiently represent the invertebrate assemblage and 2) provide within-reach measures of variance. Where these objectives are not a concern, samples may be composited. Sampling is confined to the thalweg of the study reach and only where surface water is present (see Subsection 3.1 for designating hydrologic condition). For instance, if there is continuous surface flow throughout the study reach, 4 samples are taken from each habitat type. If there is surface water only in depositional units, only 4 depositional samples are taken. Samples should be spread across multiple units of each habitat type (i.e., erosional and depositional) along the study reach. Where the study reach has ≤ 1 habitat unit (e.g., a pool) with surface water and the other units of that type are dry, do not sample that habitat type. Do not sample areas where the water depth exceeds

the height of the bucket sampler. Samples can be randomly positioned within available habitat units in the reach. For example, numbers ranging from 1 to 100 can be drawn and the center of the bucket sampler is positioned along the thalweg coinciding proportionally along the unit length.

4.4.1.1. Sample collection and preservation
 Attach the canvas skirt to the bucket sampler by sliding the elastic band of the canvas skirt over and around the bottom edge of the bucket sampler (Figure 4-11). A foam ring, rather than the canvas skirt, should be fitted to the bucket where the streambed is bedrock. Sample collection should proceed in an

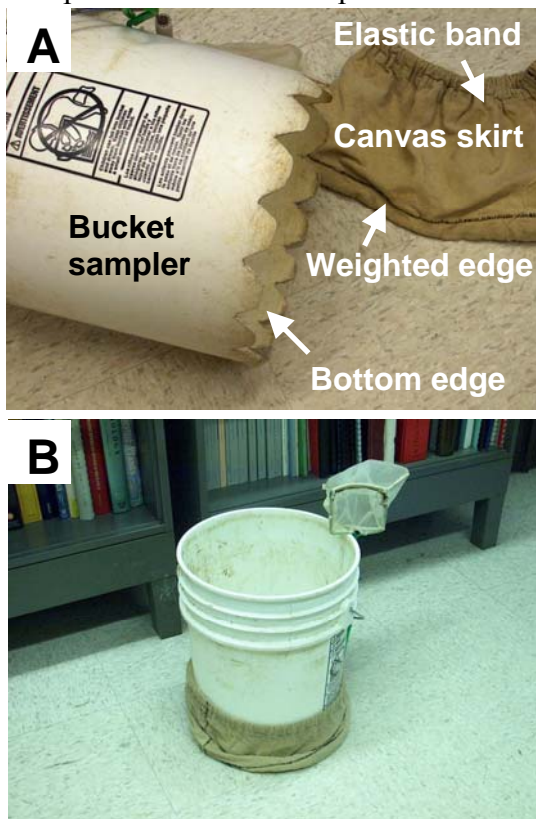


Figure 4-11 Photographs of bucket sampler and canvas skirt A) unassembled and B) assembled for sampling. upstream direction. Avoid disturbing the streambed outside of the sampling area as much as possible by walking along the banks rather than in the thalweg. Make sure that the

areas sampled have not been disturbed by other sampling activities. Begin by identifying a sampling location within the channel thalweg and place the wash basin, sieve, hand net, and trowel on a bank or gravel bar near the sampling location. With both hands lift the weighted edge of the canvas skirt above the bottom edge of the sampling bucket (to prevent the skirt from being inside the sampling area once the bucket encloses the sampling area). Push the bottom edge of the bucket 3 to 5 cm vertically into the streambed. Adjust the weighted edge of the skirt to seal the sampling area. By hand remove the coarse surface substrates (i.e., large gravel and cobble) from the enclosed sampling area and place them into the wash basin or sieve to be scrubbed (Figure 4-12). Stir by hand or trowel the remaining substrate within the bucket for 10 seconds to a depth of 10 cm (or bedrock, whichever is shallower). This step helps to suspend invertebrates from streambed interstices into the water column.



Figure 4-12 Coarse surface substrate set aside in basin for scrubbing.

Immediately sweep the hand net through the water column for 10 seconds to capture the suspended invertebrates (Figures 4-12 and 4-13). Repeat the substrate stirring and net sweeping steps 2 more times. After the sweeping is completed, empty the net contents into the wash basin. Look for any



Figure 4-13 Sweep the hand net through the water column to collect suspended invertebrates within the bucket area.

invertebrates that may be attached to the net and put these in the basin or sieve. (If the net becomes full before completing the three sets of sweepings, empty its contents into the basin and then continue sampling.) Where the water depth within the bucket area is too shallow to effectively sweep the water column, additional substrate will need to be excavated and placed into the wash basin.

Remove all invertebrates attached to coarse surface substrate by scrubbing them by hand or scrub brush into the sieve or basin (Figure 4-14). Carefully add stream water to the basin (Figure 4-15). Rinse invertebrates from large detritus (e.g., leaves and sticks) that may be in



Figure 4-14 Scrubbing attached invertebrates off the coarse surface substrate in the wash basin (or sieve).



Figure 4-15 Carefully adding water to the wash basin before sample elutriation.

the basin and discard the detritus. Elutriate the remaining contents by swirling the basin by hand and pouring the water and low-density contents (e.g., invertebrates and fine organic matter) into the sieve (Figure 4-16). This step will separate most of the fine sediment particles from the invertebrates. Repeat the elutriation until no organic matter is remaining in the basin. Carefully search the remaining basin contents for heavy-bodied invertebrates (i.e., mollusks, mineral-cased caddisflies, Figure 4-17). Place any heavy-bodied invertebrates into the sieve. Empty



Figure 4-16 Sample elutriation in the wash basin and pouring invertebrates and fine detritus into the sieve.



Figure 4-17 Carefully search the basin for heavy-bodied invertebrates that were not transferred to the sieve.

and rinse the wash basin. At the stream edge carefully wash the sieve contents to one side by gently agitating the sieve while the sieve mesh is partially submerged in water (Figure 4-18). With the wash basin positioned underneath, transfer the majority of the sieve contents by hand or a minimal amount of water using a wash bottle into a 24-oz Whirl-Pak® bag or other container. Agitate the sieve again if necessary to combine the remaining contents against one side of the sieve (Figure 4-19). Wash the remaining sieve contents into the bag with 95% ethanol using the wash bottle (Figure 4-20). Ensure



Figure 4-18 Washing sieve contents to one side by gentle agitation while sieve is partially submerged.



Figure 4-19 Sieve contents condensed for transfer to sample bag.

there is enough space in the bag to sufficiently preserve the sample with additional ethanol. Use more than one bag if necessary to contain a sample. Pour more ethanol into the bag until the sample is completely submerged and the final preservative concentration is $\geq 70\%$ ethanol. Note that the amount of ethanol needed for sufficient preservation increases with the amount of organic matter within a sample.



Figure 4-20 Sieve contents rinsed into sample bag (over basin) using ethanol squirt bottle.

4.4.1.2. Sample labeling

Make a label (waterproof paper and pencil) that includes relevant information (e.g., locality, habitat type, date, collector's initials). Where more than one bag is needed to contain

an entire sample, indicate this on the label by writing “1 of 2”, “2 of 2”, etc. Place label inside the Whirl-Pak ® with the sample and seal the bag. When sealing the bag, remove as much air space as possible from the bag (this will make samples more compact for transport). Seal the bag by folding the tab

over a couple of times then while holding the wire ends, whirl the bag 3-4 times, and lastly twist wire ends together. Write label information on the field form (Figure 4-21) and on the outside of the bag with a permanent marker.

INVERTEBRATES				
# BUCKET SAMPLES: Depositional Habitats <u>4</u> Erosional Habitats <u>4</u> TOTAL AREA SAMPLED: <u>0.4</u> m ² (each ~0.05 m ²)				
SAMPLE ID	HABITAT TYPE	NUMBER OF BAGS	COLLECTED BY:	COMMENTS
Four – JC – 1 – D (1-4)	Depositional	5	KMF	Rep #1 in 2 bags
Four – JC – 1 – E (1-4)	Erosional	6	KMF	Reps. 3 & 4 in 2 bags

Figure 4-21 Appropriate location for recording invertebrate sampling information on page 1 of field forms. Example sample information shown in red.

Repeat procedures outlined in 4.4.1.1 and 4.4.1.2 for the remaining sample replicates in each habitat type. Thoroughly rinse sampling equipment with stream water between sample replicates to prevent transporting any attached invertebrates.

4.4.1.3. Sample transport and shipping
 Before leaving a site, check that all samples are labeled properly and safely stowed in a backpack or container (e.g., 5-gallon bucket) for transport to the vehicle. At the vehicle consolidate all invertebrate samples in a crush-resistant and leak-proof container (e.g., cooler) for transport to the laboratory for processing. If samples need to be shipped, include any special shipping forms that may be required for the ethanol-preserved samples.

4.4.2 Alternative methods
 Here we will briefly identify other flow-independent methods for quantitatively

sampling macroinvertebrates. More detailed reviews of sampling methods for stream invertebrates can be found in Peckarsky (1984), Klemm et al. 1990, and Merritt et al. (1993). Among the simplest methods is stone sampling, where individual stones are used as the sampling units (Dall 1979, Doeg and Lake 1981, Wrona et al. 1986, Scrimgeor et al. 1993). Stone surface area is estimated as described in Subsection 4.2.2. Some advantages of this method include: 1) simplification of streambed heterogeneity, 2) represent natural sampling units, and 3) efficient, cost-effective method (samples contain little detritus from which to sort invertebrates). For small headwater streams this method also causes minimal degradation to the habitat (if stone area or dimensions are measured immediately after collection) and logistically feasible where channel width limits use of larger samplers. Some disadvantages of stone sampling are the

exclusion of some habitats (e.g., interstitial spaces, bedrock), overestimation of extrapolations to 1 m², and depending upon the typical stone surface area, this method may require large sample sizes to reduce sample variability (Morin 1985).

Like the bucket sampler, vacuum samplers (Boulton 1985, Brown et al. 1987, Brooks 1994) are flow-independent and can be used with equal efficiency across habitat types and flow conditions. Vacuum samplers are devices that enclose a sampling area and transfer sample material by bilge or peristaltic pump to a sieve or net. The major drawbacks to vacuum samplers are related to their overall size. Most of these samplers require more than one person for sample collection and are likely too heavy or unwieldy for long hikes often necessary to reach headwater study sites.

Artificial substrates, such as rock baskets and multi-plate samplers, are commonly used for comparing invertebrate assemblages among sites (e.g., Poulton et al. 2003, Rinella and Feminella 2005). Artificial substrate methods provide the investigator with control over the colonization or exposure time and the standardized size may reduce sample variability. Artificial substrates minimize streambed disturbance to small headwater streams. Some limitations of artificial substrates are differential colonization among taxa, the requirement of multiple visits for deployment and retrieval, and susceptibility to vandalism or natural disturbance.

Modifications to standardized substrates (e.g., Hester-Dendy multiplate samples) may be required to ensure complete submergence in shallow headwater streams (e.g., Winterbourn 1982). Variable submergence among units counters one of the advantages of artificial samplers.

Aquatic insects often represent a significant proportion of the invertebrate assemblage in headwater streams. A wide variety of traps have been used to capture the adult life stage of aquatic insects as they emerge from streams (Davies 1984). Emergence traps can be designed to collect and preserve emerging adults daily or for up to several weeks (LeSage and Harrison 1979, Whiles and Goldowitz 2001). This method can be used to easily sample across a wide range of hydrologic permanence because it is not dependent upon the presence of water (Progar and Moldenke 2002, Price et al. 2003). However, this method requires multiple visits, is susceptible to vandalism or natural disturbance, excludes invertebrate taxa that do not have a winged-adult stage and, depending upon trap design and position, may differentially capture taxa.

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- Equipment and supplies (Numbers correspond to items in Figure 4-22)*
- Measuring tape (50 m)
1. Bucket sampler = 5-gallon bucket with bottom cut out w/ bottom area ~ 0.053 m²
 2. Canvas skirt
 3. Hand-net (243 µm mesh)
 4. Plastic wash basins
 5. Hand trowel
 6. 250 µm sieves
 7. Funnel
 8. Ethanol transport jug(s)
 9. Squirt bottle (250 ml)
 10. 24 oz Whirl-pak bags or other sample containers
 11. Label paper
- Ethanol (95%)
Sharpies & Pencils
Field forms

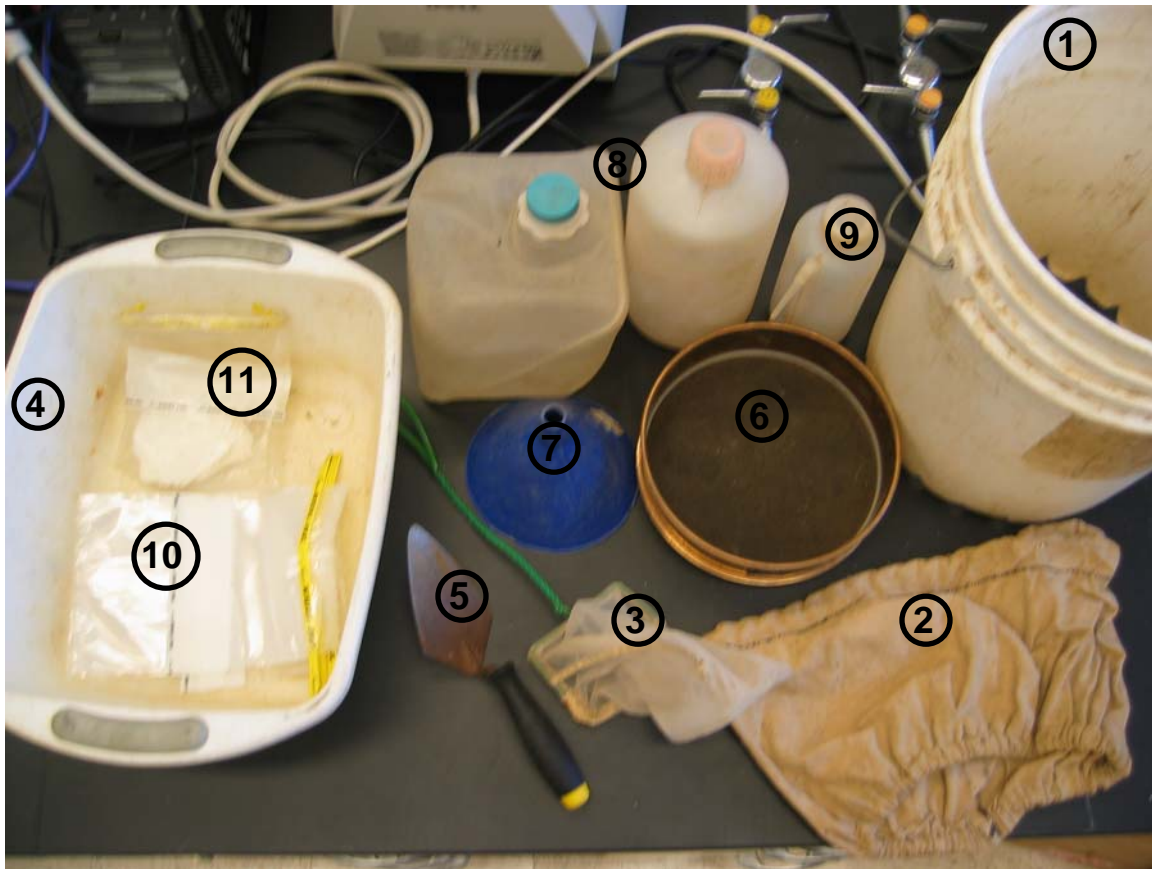


Figure 4-22 Equipment used to collect and preserve benthic invertebrate samples. Numbers correspond to the equipment and supplies list above.

4.5 Surveying the amphibian assemblage *General*

This subsection provides instructions for characterizing amphibian assemblages in headwater streams using a visual encounter survey. All amphibians are highly dependent on water and the amount of moisture in the environment influences their geographic range, life history characteristics, and behavior. With the exception of the tailed frog in the western United States, most headwater stream-dwelling amphibians are urodels (salamanders) rather than anurans (frogs and toads). This discussion therefore focuses on the use of salamanders as

indicators of hydroperiod, but the methods are similarly effective for anurans populations.

Following hatching, all stream salamanders go through a gilled larval stage during which they are obligate to the aquatic environment. The larval stage may last from months to several years depending on species and locality. At the end of the larval stage, most species metamorphose into juveniles and leave the stream to become semi-aquatic or terrestrial as adults. Adults subsequently return to streams for courtship and egg-laying. Some salamanders are permanently aquatic as adults (eg., *Cryptobranchus alleganiensis*, *Necturus* spp., etc.) and retain their gills. All stream

salamanders are predatory and they are often the top predators in high-gradient, fishless headwaters (Davic and Welch 2004, Johnson and Wallace 2005).

The fact that stream-dwelling salamander larvae are obligate to the aquatic environment and have larval periods that can vary greatly in length means they are potentially ideal indicators of stream hydroperiod. This protocol focuses on the larval stage because adult salamanders are less dependent on water and may move far from the stream channel. Unfortunately, identification of larval salamanders can be difficult and few good comprehensive larval keys are available. Field crews should therefore attempt to become familiar with the salamander species in their area prior to sampling. Larvae of the two-lined salamanders (*Eurycea bislineata*, *E. cirrigera*, and *E. wilderae*) are among the most commonly encountered salamanders in streams of the eastern United States. Larvae are dusky colored dorsally, have branched external gills, and have 6-9 pairs of light dorsolateral spots (Figure 4-23B). The Appalachians are home to the greatest salamander diversity, and larvae of *Desmognathus* spp., *Gyrinophilus* spp. (Figure 4-24), and *Pseudotriton* spp. are also frequently encountered in streams of this region. Amphibian diversity is lower in the western United States, but species often encountered in streams include the giant salamanders (*Dicamptodon* spp.) and tailed frogs (*Ascaphus* spp.). Petranka (1998) provides a larval key and distribution map for salamanders of the United States. Other regional keys and distribution maps may be available for your area (e.g., Green and Pauley 1987, Pfingsten and Downs 1989, Minton 2001) and many larval descriptions can be found in the primary literature.

4.5.1 Time-constrained sampling

Time-constrained sampling is an effective way of sampling salamander larvae from a variety of habitats, typically with minimal cost, effort, and stream disturbance. While the timed-search method makes density determinations difficult, it can be used to estimate relative abundance of species. Timed sampling has an additional advantage in that it increases the chance of collecting rare taxa, or rare individuals when salamanders are scarce (Crump and Scott 1994, Barr and Babbitt 2001). Chalmers and Droege (2002) additionally found that timed-search sampling was more effective than use of leaf litter bags in estimating abundance of larval *E. bislineata*. We chose the time-constrained approach for this protocol because it is robust and best suited for collection of larvae from streams across multiple regions where species composition and densities may be highly variable. The method is also effective over a wide range of stream size, hydroperiod, and condition.

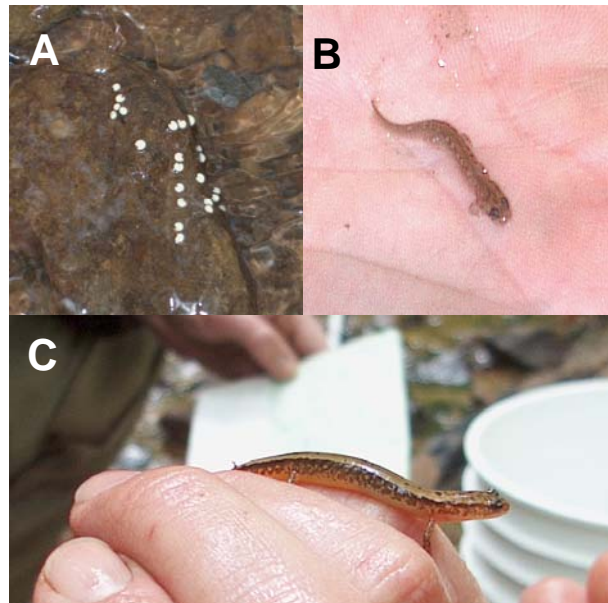


Figure 4-23 Northern two-lined salamander, *Eurycea cirrigera*, from Robinson Forest, KY: A) egg clutch; B) larva; and C) adult.



Figure 4-24 Larval spring salamander, *Gyrimophilus porphyriticus*, from Robinson Forest, KY.

Procedure

When possible, sampling should be conducted with clear skies to maximize visibility. The amphibian survey reach and primary sampling reach should have similar discharge and habitat characteristics. Sampling typically begins ca. 10-m upstream of the primary 30-m sampling reach and progresses in the upstream direction. If there is an obvious change in habitat or discharge (i.e., a tributary confluence or headcut) above the primary reach, then sampling should be conducted downstream of the primary sampling reach. Sampling begins after noting the starting time and then continues for exactly 30 minutes. Only one person should conduct the sampling to standardize the level of effort and, if possible, the same person should conduct the survey at each site to minimize sampling variability.

Sampling is confined to the wetted area of the stream because the survey focuses on the larval stage. One crew member moves carefully upstream (or downstream in some cases) turning loose cover objects (leaves,

cobble, woody debris, etc.) in all available habitat types (shallow, deep, fast, slow, etc.). Salamander larvae are often found in isolated backchannels or at the stream margins where there may only be a thin surface film. Cover objects are turned individually by hand rather than using kick nets or other more destructive sampling methods. This saves time in sorting through debris and allows for the survey to cover a greater stream area. The investigator should slowly and methodically turn random cover objects from bank to bank while moving along the stream. Salamanders encountered are carefully collected into the hand net for identification. It should not be the intent of the investigator to turn over every object in the stream and there is no distance objective. Surveys in larger perennial streams with a greater wetted area will therefore cover less stream length than surveys done in small intermittent stream reaches. The objective is to keep the level of effort the same in every study reach, regardless of stream size or habitat types. Approximate length (m) of stream surveyed should be noted at the end of the 30-minute survey.

Larval salamanders observed during the survey are identified in the field when possible and recorded on the amphibian survey field sheet along with corresponding life stage (larva, juvenile, adult) (Figure 4-25). Mean snout-vent length (SVL) for each cohort should be visually estimated (mm) for each species and recorded on the field form to help determine when larvae may have hatched. Presence of larger/older larvae of species with multi-year larval stages (e.g., *Desmognathus quadramaculatus*, *Gyrimophilus porphyriticus*, northern populations of *E. bislineata*) should be noted as this can be an important indicator of stream permanence. Unknown species and voucher specimens should be recorded photographically, showing top and side views at a minimum. Each species should be

vouchered for each area sampled (i.e., ecoregion, national forest, etc.). Salamanders should not be collected and returned to the laboratory without appropriate collection permits. Salamanders collected for vouchers or for species confirmation should be anaesthetized with 0.1% MS-222 (tricaine methylsulfonate) (Beachy 1994) and then preserved with a 10% formalin solution. Preserved specimens should be placed in vials labeled with the site name, date, and name of the collector. Photos and preserved specimens may be sent to regional experts for species confirmation. Though the survey is aimed at the larval stage, adults and egg clutches found during the survey should also be noted on the field form and photographed. Any fish observed during sampling should also be recorded.

4.5.2 Alternative methods

Salamanders have been collected from streams using a wide variety of sampling methods (reviewed by Heyer et al. 1994). Larvae may be qualitatively sampled using kick-nets or conventional dredge nets typically used in benthic macroinvertebrate sampling. Such collection methods; however, can be destructive and time-consuming and may not adequately represent rare taxa. Typical quantitative approaches include: other benthic sampling devices (e.g., benthic corers, Surber samplers), quadrats (e.g., Welsh and Lind 1996, Rocco and Brooks 2000), transects (e.g., Resetarits 1997, Welsh and Oliver 1998), and artificial habitats (Pauley 1998). Bury and Corn (1991) provide an example of a more intensive sampling method for western streams, whereby all moveable objects are removed from a 10-m stream section by a two people over ca. 5 hours.

AMPHIBIAN ASSESSMENT FORM					Page 4 of 4
SITE NAME: _____		DATE: / /			
COLLECTION TIME: _____ to _____		REACH LENGTH COVERED (m): _____			
SURVEYOR: _____					
SPECIES / LIFE STAGE					
SPECIES	#LARVAE	#JUVENILE	#ADULT	TOTAL	VOUCHER?
<i>Desmognathus fuscus</i>					
<i>Desmognathus monticola</i>					
<i>Desmognathus ochrophaeus</i>					
<i>Desmognathus walteri</i>					
<i>Eurycea bislineata</i>					
<i>Eurycea longicauda</i>					
<i>Gyrinophilus porphyriticus</i>					
<i>Pseudotriton montanus</i>					
<i>Pseudotriton ruber</i>					
OTHER					
NOTES ON AMPHIBIANS					
NOTES ON FISH (include species present)					

Figure 4-25 Amphibian survey field form.

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Equipment and supplies

Aquarium dip net (approximately 15.5 x 12 cm, 1-mm mesh)

Wristwatch or stopwatch

Specimen containers (optional)

0.1% MS-222 (optional)

10% formalin (optional)

Digital camera

Field forms

5 APPENDIX FIELD FORMS

HEADWATER STREAM ASSESSMENT FORM						Page 1 of 4
STREAM NAME (& ABBREV): _____ SITE # _____ DATE: / /						
COUNTY: _____ STATE: _____ USGS QUAD NAME: _____ REACH LENGTH (m): _____						
LATITUDE: _____ ° _____ ' _____ " LONGITUDE: _____ ° _____ ' _____ "						
DIRECTIONS TO STREAM SITE						
STUDY REACH HYDROLOGIC CONDITION						
<input type="checkbox"/> Visible surface flow continuous (4) <input type="checkbox"/> Visible flow interstitial (3) <input type="checkbox"/> Surface water present but no visible flow (2)						
<input type="checkbox"/> Surface water in pools only (1) <input type="checkbox"/> No surface water (0)						
MAX. POOL DEPTH (cm)	DEPTH TO BEDROCK / GROUNDWATER (m) (3 measures in depositional habitat)			SINUOSITY (number of bends)		
	/					
DISTANCE TO NEAREST SURFACE WATER (m)	CHANNEL SLOPE (%) (for three 10 m sections of study reach)			% CANOPY COVER (facing upstream, downstream, right & left banks)		
0 <100 100-500 >500						
PRESENCE OF HEADCUT IN REACH	ALGAL COVER INDEX			# CORES FOR SUBSTRATE MOISTURE (depositional)		
Y N	1	1 ^{1/2}	2	3	4	5
Terrestrial Herbaceous Vegetation in Active Channel? _____ Roots of Riparian Vegetation in Active Channel? _____ Base Flow Conditions?(Y/N) _____ Date of Last Precipitation? _____						
IN SITU WATER QUALITY MEASUREMENTS						
Location of Measurements	Cond (µS/cm)	Temp (°C)	DO (mg/l)	pH	Comments	
BIOTIC SAMPLES (ALL TAKEN IN THALWEG & LABEL SAMPLES COMPLETELY)						
INVERTEBRATE						
# BUCKET SAMPLES: Depositional Habitats _____ Erosional Habitats _____ TOTAL AREA SAMPLED: _____ m ² (each ~ 0.05 m ²)						
SAMPLE ID	HABITAT TYPE	NUMBER OF BAGS	COLLECTED BY:	COMMENTS		
ALGAE						
# STONES SAMPLED: Depositional Habitats _____ Erosional Habitats _____ TOTAL AREA SAMPLED: _____ cm ² (each 12)						
BRYOPHYTES SAMPLED: Y N						
SAMPLE ID _____						
			COMMENTS			

Meter #	Modal Sediment Particle Size (mm) *	Water Depth (cm)*	Habitat Type (E/D)	Notes (e.g., LWD, Leafpack)	Velocity (m/s)*	Wetted Width (m)	BF Width (m)	BF Depth (m)*	FPA width (m)§
0									
1									
2									
3									
4									
5									
6									
7									
8									
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10									
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AMPHIBIAN ASSESSMENT FORM

SITE NAME: _____ DATE: / /

COLLECTION TIME: _____ to _____ REACH LENGTH COVERED (m): _____

SURVEYOR: _____

SPECIES / LIFE STAGE

SPECIES	#LARVAE	#JUVENILE	#ADULT	TOTAL	VOUCHER?
<i>Desmognathus fuscus</i>					
<i>Desmognathus monticola</i>					
<i>Desmognathus ochrophaeus</i>					
<i>Desmognathus welteri</i>					
<i>Eurycea bislineata</i>					
<i>Eurycea longicauda</i>					
<i>Gyrinophilus porphyriticus</i>					
<i>Pseudotriton montanus</i>					
<i>Pseudotriton ruber</i>					
OTHER					

NOTES ON AMPHIBIANS

NOTES ON FISH (include species present)



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