West Virginia Department of Environmental Protection

West Virginia Save Our Streams Program's Advanced Standard Operating Procedures Manual

The mission of West Virginia Save Our Stream's is to promote the preservation and restoration of our state's waters by providing a better understanding of their ecological integrity.

Introduction

The standard operating procedures (SOPs) are designed and intended for the assessment of wadeable streams. The SOPs consist of three basic elements: (**1**) **water chemistry analysis**; (**2**) **physical evaluations** mostly by observations using the characteristics and conditions described on the survey data sheet; and (**3**) **assessment** of the **benthic macroinvertebrate** community. For more information about general hydrologic and ecological concepts visit the Volunteer Monitoring Manual section of the West Virginia Save Our Streams website.

Prior to your site visit you should prepare for your survey making sure you have all the necessary equipment; the equipment should be in working condition and replaced or repaired if needed, check batteries if electronic equipment such as cameras or GPS units are used, check the expiration dates of chemicals, make sure all meters all calibrated, all nets are to be secured to their poles and have no tears or holes, all containers should be clean and dry; review the appropriate topographic maps and/or aerial photos prior to the site visit and possibly have copies on-hand during your survey; make sure all sites are accessible and that permission to cross private property is acquired prior to your site visit; at the site always wear the correct footwear (waders, boots, closed toed shoes) and cloths for working in and around the stream. Your footwear should be cleaned prior to visiting the next site to avoid possible transfer of any biological or chemical components. You should have a thorough knowledge of the risks associated with stream survey work in your area and have a corresponding safety plan to reduce those risks. The flow chart below provides the general steps of the stream survey procedure.

Clean your equipment: Felt glued to the bottom of the wading shoe will improve your traction for walking in the stream; however felt will also easily transfer certain biological components (i.e. algae, bacteria etc.) if they are not properly cleaned. Equipment and wading apparel should be cleaned between uses and especially if multiple streams are visited.

Reach delineation

Water chemistry analysis	Physical characterizations
Habitat conditions assessment	Macroinvertebrate collections
Discharge measurement	Pebble counts
Additional procedures	

The steps of the procedures can vary slightly, depending upon the number of volunteers available to perform the survey. However most of the physical observations and water quality analysis should occur prior to any sediment disturbance that may be caused by walking in the stream while performing pebble counts or habitat observations, or by the macroinvertebrate collection procedures. A wadeable stream reach is defined as a section of stream no deeper than waist deep (except pools); most of the reach are at depths between the thigh and the waist or shallower. Streams from first through fourth orders most commonly fall into this category. Some higher order streams may have wadeable sections.

Before deciding to begin a monitoring and/or restoration project it is very important for you to describe your study design. Think carefully about the why, what, where, when and how questions, and consider the quality assurance and quality control (QAQC) measures that are necessary to insure accuracy and precision. Your approach should be similar to the scientific method. The questions you ask, the methods you choose, and the way the data is analyzed and checked should be written into your study design. It's worth taking the time to figure out what you want to do. Your monitoring is much more likely to be successful and sustainable over a longer time, with the right plan. Click-here to learn more about quality assurance project plans (QAPPs).

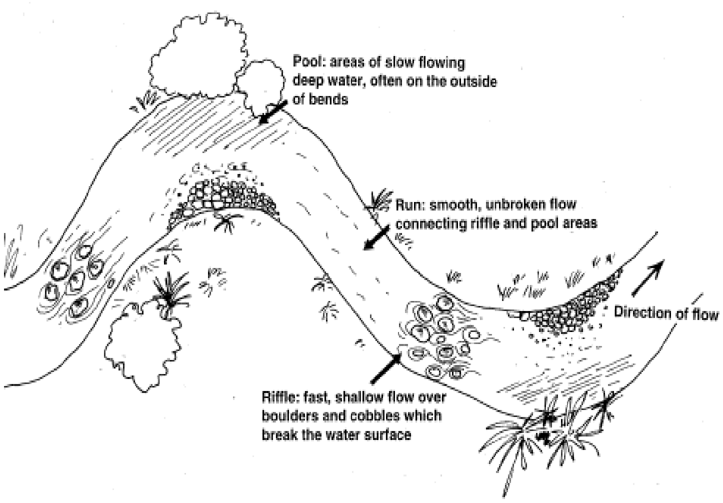
Table of Contents

Appendices

Reach delineation

The sampling locations on a stream should be a minimum of 50-yards upstream from a road or bridge crossing. Being upstream of these or other types of human encroachments minimizes the effects on stream velocity, channel shape and size, and overall habitat quality. In other words your reach should be as representative as possible of the natural characteristics of the stream. Additionally, no major tributaries (2nd order or higher) should be discharging within the reach.

Once the station is established the length of the reach is determined and set. The WV Department of Environmental Protection's (WVDEP), Watershed Assessment Branch (WAB) uses 100-meters. Volunteer monitors are encouraged to use the same length but other lengths are acceptable. Most hardware or home stores sell open-reel tape measures of up to 300-feet (100-meter open-reel tape measures are usually available from engineering supply companies). The 300-foot distance is allowable for the maximum reach length. In some cases younger volunteers may be monitoring so it is best to keep them within your line-of-sight. Certain stream-types may meander and have thick vegetation so the entire length of the reach may not be visible. Under these circumstances the length of the reach can be reduced as a safety precaution. The recommended minimum length is 150-feet. The reach should have at least one or more of the following channel features:



- Riffles have shallow, fast moving water broken on the surface by the presence of coarse substrate such as stacked gravel, cobble and boulders. Its channel shape is variable and often has portions of incline and decline. This feature is the best place from which to collect benthic macroinvertebrates.
- Runs are deeper than a riffle with a fast to moderate current and usually no breaks in the surface. The channel shape is relatively consistent with only a slight incline or decline. The substrate is variable but is mostly coarser materials. Your width, depth and velocity measurements should be completed within a run.
- Pools have deep, slow moving water. The channel shape is generally bowl like and often some of the bottom substrates consist of finer sediments such as sand and silt. In steep-gradient mountain streams, pools are often deep but may have many areas of very fast velocity and larger substrate. These types of pools are often referred to as steps.

Once the length is determined and set it should remain the same throughout the life of the station. Use flagging or natural features to mark the upper and lower boundaries of the reach. For the first few visits it is a good practice to measure the length of the reach by laying an open-reel tape measure along the banks (not in the stream). The tape measure provides you with many points of reference along the length and is very useful for marking the location of pebble count transects and other notable habitat features such as point bars, islands, or eroding banks that may occur within the reach.

The latitude and longitude (X-site) of your reach is determined at the downstream end of the reach. If you are using a GPS and cannot get a reading at the downstream end, do not move your tape measure once it is in place. Walk up stream until a signal is received and then indicate the location of the signal on your survey data sheet (i.e. middle reach, upper reach etc.). WV Save Our Streams prefers latitude and longitude readings in the degrees-minutes-seconds format.

Station locations

The number and location of your stations depends on what questions your monitoring study has been designed to answer. Table 2 provides some general considerations based upon two types of common monitoring criteria. Prior to selecting the site in the field use a topographic map to do preliminary selection of sites that meet your criteria. Always visit the site before making final determinations to make sure the site is easily and safely accessible and that it is on public access areas whenever possible. Site codes are important for keeping track of your stations and for reporting purposes. WV Save Our Streams recommends that you use a code consisting of series of numbers and letters that easily designate the site locations and allow for more sites to be added. For example, you have decided on three stations along Spruce Creek. The stations could be coded as follows:

SP-001: The most downstream site on the stream; **SP-010**: About 1-mile from the mouth; **SP-015**: About 1.5-miles from the mouth	The number to the far right is 0.1-mile, the next place to the left is 1.0-miles, the next is 10.0-miles etc.

(**SP**): First two-letters of the name	(**001**): Miles from the mouth

Choose the site that best fit the type of monitoring your group would like to perform. Most volunteer monitoring groups choose sites that determine baseline conditions and will be used to establish long-term or short-term trends. These sites are visited on a regular basis and the information collected is compared to determine if changes are occurring. The other type is for analysis of a particular impact or activity that is occurring on the stream. In this situation the stations are compared against a reference or control to determine the extent of the impact or activity. The table below provides more specific considerations.

Table 1 - Monitoring stations considerations based upon criteria

Characterization study (baseline/trends)	**Impact assessment study**
• Site is typical of the part of the river that interests your group. • Site has a variety of characteristics that represents those of the watershed. • Site may have some special natural or historical significance. • Site may be the location of previous monitoring activity.	• A reference or control station is established upstream of the potential impact. In some cases references may be in an adjacent watershed with similar characteristics, or they may be theoretical. • The impact station is at, or slightly downstream from the alteration or pollutant. • The recovery site is downstream of the impact where it is believed that the stream is beginning to recover from the insult.

How often should you monitor your stations?

Your study design should help you answer this question. But in general, typical surveys are performed two times within an index period. If there is a reason, volunteers may monitor water quality at a station more frequently such as seasonal or even monthly depending upon the situation. However, the other elements of the survey need only to be performed twice during the period. At a minimum the station can be monitored only once within the index period. The WV Save Our Streams index period is from April through October.

Water chemistry analysis

This section describes procedures and considerations for collecting water samples from wadeable stream reaches. For more specific considerations regarding the design of a water quality monitoring program refer to the program's Volunteer Monitoring Manual on the WV Save Our Streams website. This is the first task that should be performed following the delineation of your reach.

Where do I collect my samples?

Water samples should be collected from the most represented portion of the reach, which is usually the run, and as close to the downstream end (X-site) as possible. If the most downstream end of the reach is a riffle or pool, walk upstream until you encounter a run. Collect the water sample in the deepest section of the run. This may not be the center of the channel depending upon physical features or the curvature of the channel. For example a curved (meander) channel is usually deepest on the outside bend. When wading to your sample location, be careful not to disturb the bottom sediments (or at least keep the disturbance to a minimum). Once you have located the deepest section (thalweg) and have a clean sample container ready, follow the procedure described below. In most wadeable streams it is not absolutely necessary to locate the thalweg because the water is well mixed; however, you should never collect your samples from backwater areas.

- Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also tape your bottle to an extension pole to sample from deeper water if the area cannot be reached.
- Hold the bottle near its base and plunge it (opening downward) below the water surface. If you are using an extension pole, remove the cap, turn the bottle upside down, and plunge it into the water, facing upstream. Collect a water sample 8 to 12 inches beneath the surface or mid-way between the surface and the bottom if the stream reach is shallow.
- Turn the bottle underwater into the current and away from you. In slow moving stream reaches, push the bottle underneath the surface and away from you in an upstream direction.
- Leave about ½ inch air space (Except for DO and BOD samples). Do not fill the bottle completely, so that the sample can be shaken just before analysis. Recap the bottle carefully, remembering not to touch the inside.
- Fill in the bottle number and/or site number on the appropriate field data sheet. This is important because it tells the lab coordinator which bottle goes with which site.
- If the samples are to be analyzed in the lab, place them in the cooler for transport to the lab. Some types of samples may need to be preserved to reduce the possible changes that could occur during transport. Your local laboratory can provide you with the necessary preservatives and chain-of-custody forms.

Typically volunteer monitors focus on physical and biological assessments and collect minimal water quality analysis. WV Save Our Streams recommends that advanced monitors at a minimum, monitor for changes in **pH**, **temperature** and perhaps additional condition such as **dissolved oxygen** and nutrients (**nitrate**/**nitrite** and **phosphates**) depending upon the suspected insults. If water quality issues are suspected, chemical monitoring should occur more frequently. Your group should develop a regular schedule to monitor for the pollutants of concern.

1. Seasonal	Once in the winter, spring, summer and fall
2. Monthly	Once from January – December
3. Water conditions	Based on precipitation events (high, normal, low flows)

WV Save Our Streams does not usually provide chemical kits, nor can the program pay for laboratory analysis. However, the program has recently purchased several custom LaMotte kits and can work with the monitoring group to perform a more thorough chemical analysis at their stations. These kits are only available while working with the WV Save Our Streams Program Coordinator or with one of WVDEP's Nonpoint Source Program's Basin Coordinators.

Since there are a limited number of kits, they cannot be loaned or donated to the group unless there is a specific need such as a special study or project. In those cases the volunteer group must show reason for using the kit by providing a written description of the project proposal. The kits are provided at the discretion of the Citizen's Monitoring Coordinator.

Your water quality monitoring should be based upon the types of land uses common in your watershed. The table on the next page provides several examples of land use activities and the recommended water quality analysis.

Note: Because of the expense and difficulty involved, volunteers generally do not monitor for toxic substances such as heavy metals and organic chemicals such as pesticides, herbicides (agriculture/urban), solvents, and PCBs (industrial/urban). They might, however, collect water samples for analysis at accredited laboratories.

Filtering water samples - Filtering involves forcing water through a membrane filled with tiny holes, about 25 microns across (¼ of a human hair). The filter removes suspended solids from the water, and in fact, the weight of the solids caught in the filter is one common analysis parameter: total suspended solids (TSS). Such solids can be virtually invisible to the naked eye, but not dissolved, so they'll eventually settle out if the sample is left undisturbed.

Polluted coalmine water can have tiny bits of iron hydroxide (a.k.a. yellow boy) suspended in it. In a water sample fixed with acid but not filtered in the field, this iron hydroxide will dissolve into the water before it arrives at the laboratory, which will give incorrect test results for metals. Alternatively, in a sample that is not acidified, dissolved oxygen can cause dissolved ferrous iron to oxygenate into ferric iron en route to the lab. Unless the insoluble ferric iron is filtered out at sampling time, the lab can't determine how much ferrous vs. ferric iron the water actually contained when it was sampled. Ferrous iron oxygenation also consumes acid, so certain laboratory acidity or pH measurements will be wrong.

Although filtering water samples eliminates these errors to some extent, it requires special equipment and training. Many volunteer groups leave filtering to the analysis laboratory, accepting any inaccuracies that creep in beforehand. Ultimately, the decision to filter relies on a group's capacity and requirements, and it should be discussed with the laboratory that receives the water samples. The recommended preservative for various constituents related to water monitoring are based on a wide variety of references and information supplied by EPA Quality Assurance Coordinators. To learn more go to: http://www.uga.edu/sisbl/epatab1.html.

Table 2 - Water quality monitoring and land use

Land use practices	Recommended water quality analysis
Active construction	DO and BOD, Temperature, TDS and TSS, Turbidity
Forestry harvest	Temperature, TSS, Turbidity
Industrial discharges	Conductivity, pH, Temperature, TDS and TSS, Toxics
Mining	Acidity/Alkalinity, Conductivity, Metals, pH, TDS
Pastureland/Cropland	Bacteria, (Nitrates and Phosphates), Temperature, TDS and TSS, Turbidity
Septic systems	Bacteria, Conductivity, DO and BOD, Nutrients, Temperature
Sewage plants	Bacteria, DO and BOD, Conductivity, Nutrients, pH, Temperature, TDS and TSS
Urban run-off	DO and BOD, Conductivity, Nutrients, Temperature

Physical evaluation

This portion of the survey includes a wide variety of observations using all senses, making several judgments based on established rating descriptions, as well as collection and measurement procedures. In this section we discuss general physical observations. These observations should be completed prior to or immediately following the water quality analysis. Table 3 provides a list of the conditions that are assessed as wells as some general guidelines regarding what certain characteristics indicate. Water observations are made in a run or riffles, sediment observations are also made in a run or riffle and benthic algae observations are made in a riffle. These observations should be made multiple times throughout the reach to make sure conditions are consistent. You should write comments on the survey data sheet if any notable differences within the reach are observed.

Table 3 - Stream physical condition characteristics

Water conditions

Colors

Brown: Usually caused by sediment in the water. Some muddiness (brown color) is natural after storms, but if the condition persists look for an activity upstream that has disturbed the soil such as construction sites, logging, storm water runoff from roads or urban areas, or agricultural activities such as cattle in the stream.
Black: Usually caused by coalmine drainage, tar or sometimes waste material from road construction.
Green: Usually due to an algae bloom caused by excessive nutrients in the water. The source could be sewage, fertilizers

Substrate conditions

Colors

Brown: An indication of silt deposits from sediment sources. Most stream bottoms are normally brown in color.
Black: This deposit can occur naturally in heavy organic soils but can also be due to fine coal particles, tars, ashes, sludge etc.
Green: Possible indication of excessive algae growth from organic (nutrient) enrichment sources.
Orange, **yellow** or **red**: A coating of flocculates on the sediments is usually due to polluted coalmine drainage.
White or **gray**: A white cottony mass is a sewage fungus common to organic

Algae conditions

Color

Algae color varies from brown to dark green in most streams and rivers; although color is a noticeable condition of the algae it is not a particular indicator of the types or of the condition represented by the algal community.

Abundance

Coverage in a riffle is estimated based upon the following: none, scattered, moderate or heavy. A heavy coating of matted and floating algae is often an indication of nutrient rich conditions caused by excess nitrogen and phosphorous.

from farms, homes or golf courses or waste from animal feedlots.
Multi-colored sheen: Can occur naturally in stagnant waters, but a sheen that is moving or does not break up easily may be an indication of oil pollution. The source could be runoff from streets or parking areas or illegal dumping. In some areas the use of all-terrain (ATV's) vehicles may contribute to stream oil pollution.
Orange or **red**: Usually associated with mine drainage.
Tea colored: Usually associated with wetlands.
White or **gray**: Can be caused by runoff from landfills, dumps or sewage.

Odors

Rotten eggs: This strong sulfur-like odor can be an indication of sewage pollution or polluted coalmine drainage.
Musky: This slight organic odor is often natural, but in some cases may indicate nutrient enrichment from organic waste products or sewage contamination.
Oily: This odor may indicate pollution from oil and gas wells.
Chemical: There are a wide variety of chemical odors usually the result of industrial discharges, solvents and detergents.

polluted waters. An even coating of white or gray flocculates may be metals (aluminum) precipitated out of solution from contamination due to mine drainage.

Sediment odors

Monitors do not assess sediment odors; but in some cases it is good practice to compare sediment odors to odors in the water column. Stirring the bottom sediments and collecting a sample of water and sediment near the area that was disturbed assess sediment odors. The odors in the sediments should be similar to those described for water.

Streambed composition is either estimated or measured using a pebble count procedure. The major size categories are silt/clay (mud), sand, fine gravel, coarse gravel, cobble, boulder, bedrock and woody debris. See the pebble count section for more details.

The University of Maine's field guide to aquatic phenomenon provides a good overview (with pictures) of many conditions described here.

Growth habit

The growth habit characteristics are critical to understating the algae. Most stream algae will be evenly coated on the rocks and have a smooth or slimy texture; other types will be filamentous and have a hairy texture; and others will be matted. Matted algae are easily removed from the surfaces by slowly scraping with your fingers. If the algal community is mostly matted pieces will come off in junks like carpet when it has been removed from flooring.

Foam occurs naturally due to the decomposition of leaves (this foam is generally less than three inches high and cream colored). Excessive white foam may be due to detergent pollution.

Habitat assessment

The habitat assessment process involves rating many different **habitat conditions** as optimal, suboptimal, marginal or poor based upon criteria (descriptions and a rating scale) included on the survey data sheets. The optimal category is a description of conditions that meet natural expectations; suboptimal includes descriptions of criteria that are less than desirable, but satisfies expectations under most circumstances; marginal is a description of moderate levels of degradation with severity at frequent intervals throughout the reach; and poor are descriptions of criteria for streams that have been substantially altered with severe degradation characteristics.

Descriptions of the habitat conditions are provided below followed by a brief description of each condition. For additional information go to Chapter 5 of the US EPA's Rapid Bioassessment Protocols for use in Wadeable Streams and Rivers Manual. The conditions here should be applied when describing the habitats of rocky-bottom streams and rivers. Level two volunteer monitors assess five conditions: (1) sediment deposition, (2) embeddedness, (3) bank protection, (4) bank stability and (5) riparian buffer width.

1. **Embeddedness** refers to the extent to which rocks (gravel, cobble, and boulders) are surrounded by, covered, or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, fewer living spaces are available to macroinvertebrates and fish for shelter, spawning and egg incubation. To estimate the percent of embeddedness, observe the amount of silt and sand sediments overlying and surrounding the larger gavel and cobble size particles. You should base your embeddedness assessment on the composition of the materials that you observe. Embeddedness is always evaluated in the riffles used for your macroinvertebrate collections. In most cases the best persons to comment about this condition is the person(s) collecting macroinvertebrates. If cobble and gravel are easy to remove from the riffle and there is little sand or silt either in the net or suspended during collections, embeddedness is minimal. In some cases chemicals can cement (armoring) the substrate together and cause severe embeddedness.

2. **Sediment deposition** is an estimate of the amount of sediment that has accumulated and the changes that have occurred to the stream channel as a result of deposition. Deposition occurs from large-scale movement of sediment. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms. Sediment deposition should be rated throughout your reach and should not be confused with embeddedness. Sediment deposition is probably the most difficult condition to assess. It is a natural process and bars often form in streams that are very stable and have little sediment from the surrounding land or few problems with erosion. When assessing this condition look for indicators that are unusual or beyond what is expected to be normal for the stream.

The most effective way to learn is to view many different stream types representing both degraded and natural conditions. In most cases island formation, especially in small streams (1st through 3rd order), is an indication of excessive deposition. The most common cause for unusual or un-natural deposition in most streams is human encroachment (i.e. structures such as bridges, roads, culverts etc. to close to the stream or built so that the stream is narrowed) and bank erosion. Steep sloping banks with exposed surfaces are more likely to erode. Undercut banks can often erode but are sometimes very stable if covered with vegetation, tree roots and rocks. Look for deposition around eroding banks, especially if they show bare soils consisting mostly of fine materials (fine gravel, sand and silt). Hard surfaces no matter how steep or undercut are less likely to erode.

3. **Riffle frequency** is a way to measure the sequence of riffles and thus the heterogeneity occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna; therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. For high gradient streams where distinct riffles are uncommon, a run/bend ratio can be used as a measure of meandering or sinuosity. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refuge for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in some streams, a longer segment or reach than that designated for sampling should be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In headwaters, riffles are usually continuous and the presence of cascades or boulders provides a form of sinuosity and enhances the structure of the stream. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods.

4. **Attachment sites** includes the relative quantity and variety of natural structures in the stream, such as cobble (riffles), large rocks, fallen trees, logs and branches, and undercut banks, available as refuge, feeding, or sites for spawning and nursery functions of aquatic macro-fauna. A wide variety and/or abundance of submerged structures in the stream provide macroinvertebrates and fish with a large number of niches, thus increasing habitat diversity. As variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases. Riffles and runs are critical for maintaining a variety and abundance of insects in most high-gradient streams and serving as spawning and feeding refuge for certain fish. The extent and quality of the riffle is an important factor in the support of a healthy biological condition in high-gradient streams. Riffles and runs offer a diversity of habitat through variety of particle size, and, in many small high-gradient streams, will provide the most stable habitat. Snags and submerged logs are among the most productive habitat structure for macroinvertebrate colonization and fish refuge in low-gradient streams. However, "new fall" will not yet be suitable for colonization.

5. **Patterns of velocity and depth** are included for high-gradient streams under this parameter as an important feature of habitat diversity. The best streams in most high-gradient regions will have all four patterns present: slow-deep (pools); slow-shallow (glides); fast-deep (runs); and fast-shallow (riffles).

6. **Channel alteration** is a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened, or diverted into concrete channels, often for flood control or irrigation purposes. Such streams have far fewer natural habitats for fish, macro-invertebrates, and plants than do naturally meandering streams. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; and when other such changes have occurred. Scouring is often associated with channel alteration.

7. The degree to which the channel is filled with water is the **channel flow status**. The flow status will change as the channel enlarges (e.g., aggrading stream beds with actively widening channels) or as flow decreases as a result of dams and other obstructions, diversions for irrigation, or drought. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is limited. In high-gradient streams, riffles and cobble substrate are exposed; in low-gradient streams, the decrease in water level exposes logs and snags, thereby reducing the areas of good habitat. Channel flow is especially useful for interpreting biological condition under abnormal or lowered flow conditions. This parameter becomes important when more than one biological index period is used for surveys or the timing of sampling is inconsistent among sites or annual periodicity.

The remaining conditions are assessed on the left and right sides.

8. The **bank stability** parameter evaluates whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered to be unstable. Signs of erosion include crumbling, un-vegetated banks, exposed tree roots, and exposed soil. Eroded banks indicate a problem of sediment movement and deposition, and suggest a scarcity of cover and organic input to streams.

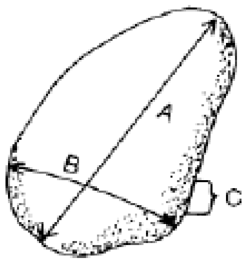
9. **Bank vegetative protection** parameter estimates the amount of vegetative protection afforded to the stream bank and the near-stream portion of the riparian zone. The root systems of plants growing on stream banks help hold soil in place, thereby reducing the amount of erosion that is likely to occur. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of in stream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetative protection or those shored up with concrete or riprap. This parameter is made more effective by defining the native vegetation for the region and stream type (i.e., shrubs, trees, etc.). In some regions, the introduction of exotics has virtually replaced all native vegetation.

10. **Riparian buffer width** is an estimate of the width of natural vegetation from the edge of the stream bank out through the riparian zone. The vegetative zone serves as a buffer to pollutants entering a stream from runoff, controls erosion, and provides habitat and nutrient input into the stream. A relatively undisturbed riparian zone supports a robust stream system; narrow riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic degradation of the riparian zones. Conversely, the presence of old fields, paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. Riparian buffers are the most valuable protection a stream system has against outside influences. In most cases healthy riparian directly reflects upon the condition of the stream unless the source of the insult is a specific pollutant. Enhancement of the riparian buffer by re-planting native grasses, forbs, shrubs and trees is the first step in the recovery of the stream back to a more natural condition.
Below are just a few of the benefits of a healthy riparian buffer.

- Provides organic material as food for invertebrate, fish and wildlife.
- Supplies large and small pieces of woody debris that provide habitat for fish, invertebrates and amphibians.

- Alters how sunlight reaches the stream and is an important temperature moderator.
- Stabilizes stream banks and reduces erosion.
- Filters sediment and materials from overland runoff and roots of many plants traps and holds the sediments.
- Absorbs nutrients from overland and sub-surface flows.
- Reduces the impacts of flooding through temporary storage, interception/diversion and slow releases.

Pebble count



A – Long axis
B – Intermediate axis
C – Short axis

The composition of the streambed and banks is an important facet of stream character, influencing channel form and hydraulics, erosion rates, sediment supply, and other parameters. Observations tell us that steep mountain streams with beds of boulders and cobbles act differently from low-gradient streams with beds of sand or silt. You can document this difference by collecting representative samples of the bed materials using a procedure called a pebble count.

The most efficient basic technique is the Wolman pebble count. This requires a person with a metric ruler who walks through the stream, and a note taker who remains on the bank with the field book or survey data sheet. The note taker records the count is recorded by size classes or categories similar to those shown in Table 4.

Pebble counts can be collected using grids, transects, or a random step-toe procedure. Select the portion of the reach that you wish to measure (this may be the entire reach or riffles only). For stream characterization, sample pools, runs and riffles in the same proportions as they occur in the study reach. For other purposes, it may be more appropriate to use a more random method. Measure a minimum of 100 particles to obtain a valid count. Use a data sheet to record the count.

Table 4 - Pebble count size classes (modified)

Size categories	Size range (mm)
Silt	Determined by feel
Sand	< 2
Fine gravel	2 - 8
Medium gravel	9 - 16
Coarse gravel	17 - 32
Very coarse gravel	33 - 64
Small cobble	65 - 90
Medium cobble	91 - 128
Large cobble	129 - 256
Small boulder	257 - 512
Medium boulder	513 - 1024
Large boulder	> 1025
Bedrock	Large solid surface
Woody debris	Sticks, leaf packs etc.

Start at a randomly selected point near the downstream end of the reach. Start on the shoreline and take steps across the stream. Averting your gaze, pick up the first particle touched by the tip of your index finger at the toe of your wader. Measure the intermediate axis (neither the longest nor shortest of the three mutually perpendicular sides of each particle picked up). Measure embedded particles or those too large to be moved in place. For these, measure the smaller of the two exposed axes. Call out the measurement. The note taker tallies it by size class and repeats it back for confirmation. Continue across the channel slightly upstream in the direction of the opposite bank and repeat the process, continuing to pick up particles until you have the requisite number (100 or more) of measurements. The note taker keeps count. Traverse across the stream perpendicular to the flow or in a zigzag pattern.

Pebble counts should be completed at least once during the index period and is much easier during low-water conditions that are common in the fall season of the index period. They can be incorporated into your bioassessment survey or they can be completed as a separate survey.

Macroinvertebrate collection and assessment

The groups of animals found in leaf packs, rocks, woody debris and other areas of streams, rivers, ponds and wetlands are collectively called benthic macroinvertebrates. Benthic refers to the ability to cling to bottom surfaces such as rocks, leaves or roots. Macro-invertebrates are animals without a backbone that can be seen with the naked eye. These bottom-dwelling animals include crustaceans, mollusks and annelids but in many aquatic environments, most are larvae of aquatic insects. Macroinvertebrates are an important link in the food web between the producers (leaves, algae) and higher consumers such as fish. They are the key indicators of biological integrity in a wide variety of aquatic environments. Depending upon the stream environment a variety of methods and equipment are used to collect macro-invertebrates from wadeable streams. In rocky-bottom streams, WV Save Our Streams recommends using a two-pole screen-barrier net or a single pole rectangular style kick-net. Both types should be equipped with 500-micron mesh netting.

WV Save Our Streams provides advanced volunteers monitors with the professional two-pole kick-net at no charge and offers the rectangular version for special projects or studies. Groups may purchase other nets as needed but only the two-types discussed here are recommended for rocky-bottom collections. Ultimately, the types of nets chosen depend upon the goals and objectives of the volunteer monitoring program and the stream types they plan to monitor.

Collecting benthic macroinvertebrates

Benthic macroinvertebrates live in a wide variety of aquatic environments. In lakes, wetlands and large river systems they are common in shallow edge microhabitats along shorelines in tangles of vegetation, roots, and leafs, in gravel shoals or along rocky and undercut banks; some kinds burry themselves in mud and sand of in shallow flowing water. In streams and swift-flowing rivers they are more common and diverse in rocky areas, especially riffles, but are also found in runs, which are sampled when riffles are not present. The collection procedure described here is designed for rock-bottom streams from riffle habitats. For information about the muddy-bottom bioassessment procedure visit WV Save Our Streams Volunteer Monitoring Manual web page. The procedure is described in Chapter 4: Macro-invertebrate and Habitat beginning on page seven. The number of samples collected depends upon the type and size of the net. Standard two-pole kick-nets are approximately 3-feet to 1-meter^2 wide so three-samples are adequate. These types of nets can only be used in riffles and runs. Other nets such as the rectangular kick-net or D-nets are much smaller but more versatile than the two-pole net. However, more samples are necessary in order to collect and adequate representation of benthic macroinvertebrates. For example if you use the rectangular kick-net, six to eight samples should be collected.

1. **Choose the best habitats**: Your goal is to collect macroinvertebrates from three different riffle areas. (If the riffles are as wide as the stream then multiple samples could be collected within the same riffle.) The riffles should have different characteristics (i.e. different composition but mostly cobble and gravel and different velocities). Often different types of riffles hold different varieties of macroinvertebrates, so to properly assess the biological conditions you need to collect a representative sample. Your choices of habitats will ultimately depend upon what your reach provides. Once you've chosen your location always approach from the downstream end, sampling the site farthest downstream first. This approach insures that the sample is representative of its location and reduces the chances of biasing your second and third sample.

2. **Get into position**: Select an area approximately the same width (or slightly less) than the width of your kick-net. Most two-pole screen-barrier kick-nets have about 1-meter^2 width. The width of the single-pole rectangular kick-net is about ¼ to ½ of a meter. So if you are using a smaller diameter-net you will need to collect more samples (two-three samples using the rectangular kick-net is equivalent to one sample with the screen-barrier kick-net). The net holder will place the net snugly against the bottom of the streambed; rocks can be removed if necessary to make sure you have a close-fit. Once you are satisfied with the position, line the front of the net with rocks heavy enough to hold the net in-place. However, be careful to choose rocks that are not too heavy or too wide/high. Large rocks will damage the net and will influence how the macroinvertebrates flow into the net thus making capture less successful.

   The net holder now tips the kick-net backward at about a 45-degree angle from the water's surface. This provides greater surface area and more even flow into the net. If the net is held to high some of the macro-invertebrates will wash around the sides and not be captured in the net. While holding the kick-net backwards the net-holder must make sure that water does not wash over the top of the net. Have one 3 ½ to 5-gallon bucket ready before you begin collecting the sample.

3. **Begin disturbing the streambed**: The second person approaches the sample area from upstream and determines the approximate sample-size. Once the area is delineated, the sampler begins disturbing the streambed directly in front of the net. The process starts with rock rubbing. First, pick-up all large rocks (cobble size and larger) and inspects them. You are looking for snails, clams and caddisfly cases. These animals often cling very tightly to the rocks and are not removed by just a simple rub with the hands or a small brush. If the rocks have any of these animals, remove them from the rocks and place them inside the bucket. Move the rocks you picked-up towards front and slightly into the net; brush all sides of the rocks with your hands or a small vegetable-brush to dislodge other clinging macroinvertebrates.

   Continue the rock rubbing process until the larger sized stones have been thoroughly cleaned. If the rocks cannot be lifted from the streambed, simply rub them where they lay. As much as possible the rock rubbing should proceed from the upstream portion of the sample area towards the front of the net. After the rocks are rubbed they should be placed aside (outside of the sample area) so they are not rubbed a second time. Some volunteer programs choose a timed approach to rock rubbing since this is a rather intensive and somewhat time consuming step. The recommended time frame for adequate rock rubbing is four minutes (or less) depending upon the abundance of cobbles and boulders within your sample area. If you choose a timed approach you should make a note on the survey data sheet and record the time frame you use. After you are satisfied that all or most of the larger rocks have been cleaned you will disturb the remainder of the streambed using a kicking method. Position yourself upstream from the net inside your sample area and begin shuffling your feet back and forth from one side of the sample area to the other. Slowly move towards the net while kicking from side to side. The action dislodges macroinvertebrates from smaller size gravels and also disturbs those that might be burrow themselves down into the soft bottom sediments. While you're moving from side to side and forward you are also pressing your feet downward in an attempt to make a depression in front of the kick-net. If you have chosen a timed approach then you should limit your kick-time as well. The recommended kick-time is two minutes, so your entire sample should take about six minutes to collect.

4. **Remove the kick-net from the streambed and capture the collection**: This is a very important step; since the sample collection is laborious you do not want to lose any of the macroinvertebrates collected by sloppy procedures here. Very slowly remove the rocks that have acted as anchors to hold the kick-net in place, rub them off while you remove the rocks, or you may choose to rub them before using them to anchor the kick-net in place. While the net-holder grabs and holds the top of the net in-place the kicker grabs the bottom edge of the net near the handles. The net is removed with a scooping motion, the kicker moved slightly forward and upward while the holder keeps the net steady so that no macroinvertebrates are lost from washing over the top of the kick-net. Both persons then pick-up the net and roll it into a loose cylinder, securing the ends and taking it to the shoreline. The bucket that was used earlier in the procedure should be at the ready to accept the contents of the kick-net.

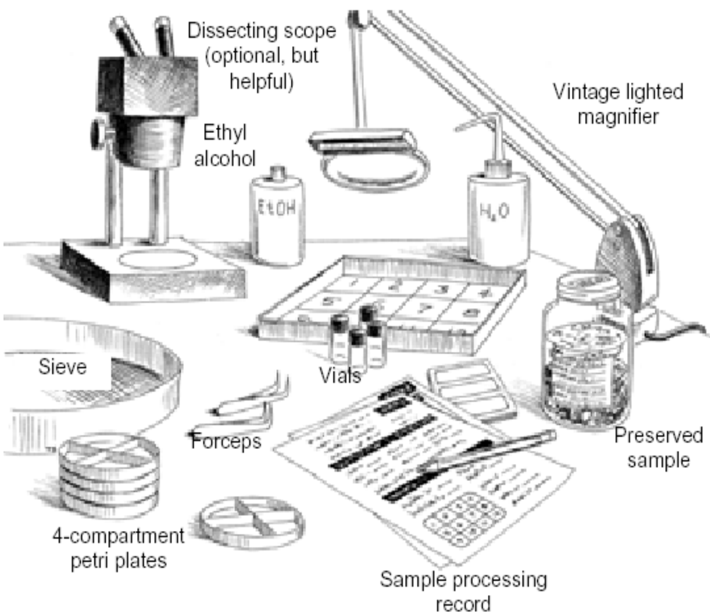
5. **Place the net into the bucket**: Slightly unwind the net so that it fits inside the bucket. With a smaller bucket or a spray bottle, wash the contents of the kick-net into the bucket. It will take several minutes and several washes to knock loose most of the macroinvertebrates. Between each attempt, remove the net and check for macroinvertebrates that have not been dislodged. Often these hardy clingers are found near the edges of the kick-net along the bottom-side and in the seams of the net. Be sure to check the opposite side for macroinvertebrates that may have crawled in an attempt to escape. You must be very careful not to overfill the bucket. If the bucket begins to fill with stream water more than about two-thirds its heights, remove some of the water by seining it through the kick-net (hold the net tightly on the bucket and pour off the water) so that the water is poured off and the macroinvertebrates remain in the bucket. The process is complete when you are satisfied that the kick-net has been thoroughly washed and most of the macroinvertebrates are now in the bucket.

6. **Remove the captured macroinvertebrates from the bucket and begin sorting**: The goal of this step is to remove all captured macroinvertebrates so that they can be observed, identified and counted. The best way to start is by trapping the macroinvertebrates as they are poured from the bucket. Before starting the steps below, remove all larger materials that may have been collected with your sample from the bucket. Make sure to check these for macroinvertebrates before they are discarded. At certain times of the year leaves and other debris are very plentiful in the stream and this material must be sorted. (It is common to find many kinds of macroinvertebrates in leaf-packs; this material is one of their favorite places to live.) The best way to deal with the leaves is to remove as many as possible, place them in smaller bucket or container and wash them to remove the macroinvertebrates. Pay close attention to the leaves that appear chewed and have begun to decay. Newly fallen leaves are less likely to have many macroinvertebrates.

   There are several ways to complete this step: You can use a second bucket with your kick-net on top, and then pour the captured organisms over the net so that they are trapped against the net. The pouring is stopped periodically so that the macroinvertebrates can be removed from the net and placed into the collection trays. Small forceps are the best tool for this job; however the macroinvertebrates can also be removed by hand. Another method is to use a sieve bucket (wash bucket) or EZ-strainer. The EZ-strainer is recommended, and is available in limited-quantity from the WV Save Our Streams Program. The EZ-strainer comes in a variety of mesh sizes (some even finer than 500-microns) and fits nicely inside a 3 ½ or 5-gallon bucket. The bucket used to capture your collections can easily be poured into the second bucket and the macroinvertebrates are removed from the EZ-strainer.

For advanced groups, WV Save Our Streams recommends that volunteer groups sort, count and identify the samples after they are preserved. Complete family-level identification can be difficult, but not impossible for experienced volunteer monitors. The preservative should be at least 90% denatured ethyl alcohol, which is the percentage after the collection (debris and macro-invertebrates) has been added. Rubbing alcohol works very well as a temporary preservative. If you decide to preserve your collection, you must apply for and receive a Scientific Collection Permit from the WV Division of Natural Resources. The preservation and laboratory sub-sampling procedures are reviewed here. Level two monitors' count their collections streamside and determine the number of families.

Important Note: The collection procedures are repeated three or more times, one for each sample collected. Volunteers may choose to sort each sample separately or combine all three samples and sort them at one-time. The latter is highly recommended since the bioassessment procedure is based on a composite sample. In certain (rare) circumstances samples may need to be counted, sorted and identified separately to compare different portions of the reach. These comparisons are done only when there is a suspected insult to the macroinvertebrates such as a habitat alteration or a suspected chemical that may not be affecting the entire reach. The collection, sorting, counting and identification of macro-invertebrates are the most intensive tasks of the survey procedures. The Globe Program provides some easy to understand macroinvertebrate protocols, click-here to learn more.



Sample preservation and bug picking

Fill jar nearly full with 95% ethanol so that the concentration of ethanol is at least 70%. If there is a small amount of water in the sample, it may not be necessary to fill the jar entirely to reach a 70% concentration. It is very important that sufficient ethanol be used to reach 70% concentration. In addition, enough alcohol should be added to at least immerse all of the material in the jar. Make sure that a label for the inside of the jar is written in pencil (ink will run); include stream name, code, and date. Place the label inside of the sample jar. Place the jar in a cooler or other container designated for the storage of macro-invertebrates. Try to keep from shaking the jars as much as possible. Never invert the jars. All macro-invertebrate samples are brought to the lab/home for in-house sorting and identification or shipped to a contractor for sorting and identification (this is a substantial cost). Sorting is done utilizing a modification of the standard sub-sampling method.

Macroinvertebrates are identified to the family level and the functional feeding group and tolerance values are determined. WV Save Our Streams suggest using a 300-organism sub sample or a one-quarter sub-sample procedure with an additional scan for rare taxa in certain circumstances.

Sorting macroinvertebrates from survey samples (a procedure often referred to as "bug picking") is an extremely important step in the biological research. The quality of the work performed by the "picker" influences the quality of subsequent processes, such as identification and data analysis. A competent "picker" must be able to recognize the morphological diversity of aquatic organisms, as well as the various methods these organisms may use to hide themselves from predators. The outcome of the final study may be affected, even if only a few organisms are overlooked during the picking process. Table 5 provides a list of materials and supplies needed for this work.

Table 5 - Recommended supplies for bug picking and identification

1. Sample jar: contains the unprocessed sample.
2. Sample bottle: for storage of processed sample.
3. Pans (larval trays): contains sample during the sorting process.
4. Denatured alcohol: preservative used in unprocessed and processed samples; 75% ethanol or isopropanol is used to preserve the samples and to prevent desiccation during identification.
5. Sieves: #30 sieve or EZ-strainers are used to separate alcohol and fine debris from the sample prior to picking.
6. Gridded tray: a white enamel or plastic tray used to evenly distribute the sieved sample for randomly selecting the sub sample. The tray is marked into equal size grids ranging in size from 1 x 1 to 3 x 3 inches. The grids define the sub-sample to be picked.
7. Cookie cutter: a homemade cookie cutter, used in conjunction with the gridded tray to isolate each of the sub samples.
8. Labels: Self-adhesive labels are used to identify the contents of the sample bottle (i.e., the picked sample).
9. Scotch tape: Used on label as additional adhesive.
10. Pencil: used to label sample bottle.
11. Crucible or other small container: used for short-term storage of the sample during the picking process.
12. Forceps: fine tipped forceps are used to remove the organisms from the debris and to manipulating specimens during examinations to determine identification.
13. Illuminated magnifier: an optical aid to illuminate and magnify the sample during the picking process. Alternatively, a desk lamp can be used.
14. Squirt bottle: filled with alcohol, used to rinse organisms into sample bottle.
15. Plexiglas: used to cover sample overnight to prevent evaporation.
16. Dissecting microscope: for examination of gross features.
17. Macroinvertebrate lab sheet: for recording results of identification and enumeration.
18. Taxonomic keys: a variety of dichotomous keys, pictures, and guides etc. that provide family-level descriptions.

Most volunteer monitors use a catch-and-release approach to identifying, sorting, and counting. In other words the macroinvertebrates are returned to the stream after the samples are sorted, counted and identified. This process can be very tedious, but it is an effective method for general assessment purposes. To complete the task you will need several large white pans, gridded pans or ice cube trays, forceps and adequate magnification (box magnifiers and 10x loupes). Even if you choose a streamside approach, preserve one example of each family for reference.

After the counting, sorting and identifications is complete, the macroinvertebrate collections are assessed using five or more metrics. An overall stream index score is determined using reference formulas or best standard values (BSV). These formulas are based upon hundreds of collections from all across West Virginia. Point values are determined for each metric and these values are averaged to calculate the stream index score.

Note: WV Save Our Streams provides on-line spreadsheets for calculating these metrics. If a monitoring group prefers not to calculate the streams score, the coordinator will gladly calculate the scores when the data is evaluated. The Level-two and three metrics are shown below.

			Level-2 (5-metrics)				**Level-3** (6-metrics)	
Metrics	Results	Points	8	6	4	2	(X) Value	Reference formulas
Total Taxa			> 18	18 - 13	12 - 8	< 8	22	= 100 x (X ÷ 22)
EPT Taxa			> 10	10 - 7	6 - 4	< 4	13	= 100 x (X ÷ 13)
% EPT			> 80	80 – 60	59.9 – 40	< 40	90	= 100 x (X ÷ 90)
Biotic Index			< 4.0	4.0 – 5.0	5.1 – 6.0	> 6.0	3	= 100 x [(10 – X) ÷ 7.0]
% Tolerant			< 2	2 – 10	10.1 – 30	> 30	2	= 100 x [(100 – X) ÷ 98]
% Dominance							20	= 100 x [(100 – X) ÷ 80]
Stream score			Optimal	Suboptimal	Marginal	Poor	**Stream index is the average of the above**	
			> 32	32 – 24	23 – 16	< 16	Integrity rating (Level 2)	
			> 80	80 – 65	64.9 – 50	< 50	Integrity rating (Level 3)	

Measuring velocity

The stream's velocity (commonly called flow) is modified by conditions along and around the stream, such as:

- Structures, such as dams and weirs, in the waterway
- Removal (diversion) of water for use in irrigation, industry and households
- Rainfall, snow melt, and water releases from dams, power stations and industry
- Entry of groundwater
- Evaporation (evapotranspiration)
- Leakiness of the river bed and banks

The size of a waterway and its flow rate affect its water quality. For example, discharges containing contaminants will have less effect on large swiftly flowing rivers than on small slow streams. This is one reason for measuring flow - to work out the load of contaminants and sediment the waterway is carrying. Because discharge can have a significant effect on water quality, it is important that it is recorded at the time of sampling and, if possible, during the previous few days. It is particularly valuable to know if flows are at low, moderate or high level and if the level is rising or falling. This is because the concentrations of nutrients, turbidity and contaminants tend to be higher when the stream level is rising than when it is falling. There are three ways to measure discharge. A simple method is to see how fast a floating object travels downstream over a chosen distance. This is called the float method. Secondly, flow data can be obtained from US Geological Service or the State Emergency Management Office, if your site is near a gauging station. Thirdly, the velocity head rod (VHR) method can be used.

The float method is easy to understand and something most of us have done as children. You simply float an object on the water and measure the time it takes to travel a set distance. The equipment you will need for this method includes:

- Foam golf ball (preferred), tennis ball, apple or orange peel
- Net to catch the ball
- Open-reel tape or survey rod
- Stopwatch

Procedure: Mark off a 20-foor length of the stream. Choose a section that is relatively straight and free of vegetation or obstacles. Avoid areas with a culvert or bridge because those structures will modify the true flow. If the flow is very slow, mark out a shorter distance. Position a person at each end of the section. Place the ball on the surface near the middle of the stream at least two-feet or more upstream of the end of the tape so it has time to come up to water speed. When the ball is in line with the beginning of the tape, start the stopwatch. Stop the watch when the ball gets to the end of the section. Repeat the procedure at least three times at this site and average the results. To calculate the water velocity, divide the distance travelled in feet by the time taken in seconds. Then multiply by a correction factor of 0.8 to compensate for the variability in velocity with depth and across the channel, i.e. water will move slower at the edges than in the middle and at different speeds within the water-column. See the example below.

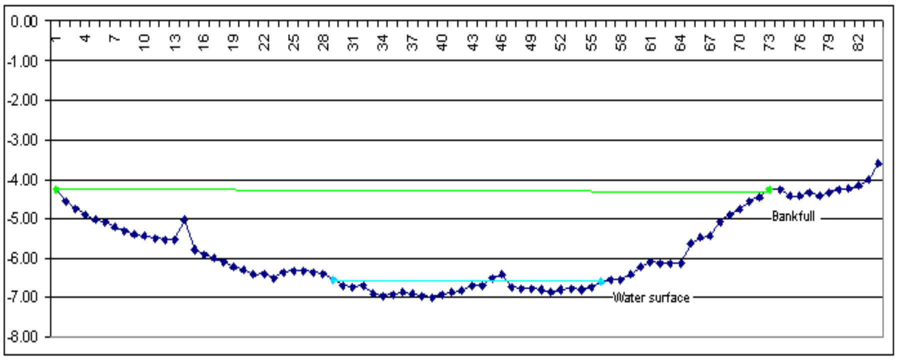
Distance travelled = 20 feet	Average time = 18 seconds
Velocity = 20 ÷ 18 = 1.11 feet/second	Correction factor = 0.8
Final velocity = 1.11 x 0.8 = 0.89 feet/second	

The velocity head rod (**VHR**) is a fast and inexpensive method of measuring velocity in a stream. The rod can be a yardstick or meter stick, or it can be made using a 3 - 6 foot long, very thin, piece of wood. A 26-gauge copper sheet is sometimes fastened to the cutting edge to protect it from abuse. Mark a scale in ½ inch increments on the rod, starting with zero at the bottom of the rod and stopping at 18 inches. Steps to find the flow rate with a velocity head rod are as follows:

- Place rod in the water with sharp edge upstream. Measure stream depth on scale.
- Place rod sideways in the water. This will create turbulence and the water will "jump" or rise above its normal depth; velocity is proportional to this jump.
- Measure depth of turbulent water next to rod. Subtract stream depth from the turbulent depth reading to obtain the "jump height," or velocity head in inches.
- Find the stream velocity in feet per second from the table provided on the survey data sheet.
- Determine the stream velocity at intervals across the stream and average them to obtain the average stream velocity in feet per second.

Cross sectional shape varies with position in the stream, and discharge. The deepest part of channel occurs where the stream velocity is the highest. Both width and depth increase downstream because discharge increases downstream. As discharge increases the cross sectional shape will change, with the stream becoming deeper and wider. The measurement of the cross section is necessary to determine the total discharge, which is the volume and velocity of the water.

In most cases the velocity and cross sectional area (width x depth) should be determined from a run. Each time these measurements are made they should be completed from the same section of the reach.



Graph of a Sleepy Creek cross-section measurement

Measuring the width and depth of the waterway, and multiplying these measurements together determine the cross section. The depth will vary across the stream and so the width and depth should be measured in small intervals and aggregated to determine the total area. To determine average discharge, measure the depth in at least five positions across the stream, one of these positions being the deepest portion of the channel (thalweg).

Additional survey procedures

Sketch the reach (the site map): In addition to the observations above, volunteer monitors are asked to sketch a site map of the reach. Draw the site map from direct observation. It should show the main features of the site and their relationship as accurately as possible. As fieldwork continues, modify the map with features such as important habitat conditions (i.e. eroding banks, bars etc.) and indicate the approximate location of water sample and macroinvertebrate sample collection locations. Scale the map to show the entire reach surveyed.

Photo documentation: Photographs provide a qualitative and potentially semi-quantitative record of conditions in a watershed, or within a stream reach. Photographs can be used to document general conditions, pollution events or other impacts, and document temporal progress for restoration efforts or other projects designed to benefit water quality. Use the same camera to the extent possible for each photo throughout the life of the station. From the inception of any photo documentation until it is completed, always take each photo from the same position (photo point), and at the same bearing and vertical angle. For general reach photo documentation, take at least two photos that show the entire reach. More specific photos can be taken if needed. Try to include landscape features that are unlikely to change over several years (i.e. rocky outcrops, cliffs, large trees, buildings or other permanent structure), so that repeat photos will be easy to position. It is often important to include a ruler, stadia rod or person to convey the scale of the image. Often times an overhead or elevated shot from a bridge, cliff, peak, tree, etc. will be important in conveying the full dimensions.

Land use assessment: The purpose of this portion of the survey is to get an overall picture of the land surrounding and draining the stream reach. A basic assessment will help you better understand what problems to expect and where to look for those problems. The first step is to review your topographic map and aerial photographs that include your stream stations. Prior to or after completing a stream survey, drive or walk portions of the watershed upstream from your stations to locate any possible activity that may threaten your stream reach. Keep in mind that this rating is simply your judgments of the level of impacts; it is not an actual assessment of the real impacts. The only way to assess a specific impact or activity is to set-up an impact assessment study.

All surveys are mailed to the coordinator so that a proper quality assurance review can occur. After the review is complete the survey data sheets are returned to the volunteer monitoring groups along with a summary of the results, and including other comments or questions the coordinator may have. The data is then entered into the Volunteer Assessment Database (VAD).

References

1. Bouchard R.W. 2002. Guide to Aquatic Invertebrates of the Upper Midwest. University of Minnesota's Water Resources Center
2. Harrelson, C.C et al. 1994. Stream Channel Reference Sites: An Illustrated Guide to Field Techniques. USDA, Forest Service Technical Report RM-245
3. Jessup, B.K., A. Markowitz and J.B. Stribling. 2002. Family-Level Key to Stream Invertebrates of Maryland and Surrounding Areas. Tetra Tech, Inc.
4. U.S. EPA, Office of Wetland, Oceans and Watersheds. 1999. Rapid Bioassessment Protocols for use in Wadeable Streams and Rivers. EPA 841-B-99-002
5. U.S. EPA, Office of Wetlands, Oceans and Watershed. 1996. The Volunteer Monitor's Guide to Quality Assurance Project Plans. EPA 841-B96-003
6. U.S. EPA, Office of Wetlands, Oceans and Watershed. 1997. Volunteer Monitoring: A Methods Manual. EPA 841-B97-003
7. Voshell, J.R. 2000. A Guide to Common Freshwater Invertebrates of North America. McDonald & Woodward Publishing Company

Appendix A - Safety Precautions

WV Save Our Streams does not require signatures on, or provide any type of liability or hold-harmless waiver forms for volunteer monitors. The primary objectives of the program are to provide education; training and resources volunteers need to carry out a water monitoring survey. The safety at the site is the responsibility of the volunteer monitoring group. If the volunteer group fills the need for such forms, they should seek the advice of a trusted legal advisor. However, the program strongly recommends that volunteers develop specific safety preparedness plans and attend safety and outdoor first-aid training on an annual basis. At least one of the persons present during the survey should have up-to-date safety and first-aid training.

1. **Develop a safety plan**: Find out the location and telephone number of the nearest telephone and write it down. Locate the nearest medical center and write down directions on how to get between the center and your site(s) so that you can direct emergency personnel. Have each member of the sampling team complete a medical form that includes emergency contacts, insurance information, and pertinent health information such as allergies, diabetes, epilepsy etc.
2. **Never drink the water in a stream**: Assume it is unsafe to drink, and bring your own water from home. After monitoring, wash your hands with antibacterial soap.
3. **Always monitor with partner**(**s**): Use a minimum of 2 persons; teams of 3-4 or more people are best; always let someone else know where you are, when you intend to return and what to do if you don't return at the appropriate time.
4. **Have first aid kits handy**: Know any important medical conditions of team members (e.g., heart conditions or allergic reactions to bee stings). It is best if at least one team member has first-aid and CPR certification.
5. **Listen to weather reports**: Never go sampling if severe weather is predicted or if a storm occurs while at the site.
6. **Never wade high water**: Do not monitor if the stream is very swift or at flood stage; adult volunteers should not enter swift-flowing water above waist-deep, unless absolutely necessary, and young volunteers should not enter swift-flowing water just above knee-deep.
7. **Park in a safe location**: If you drive, be sure your car doesn't pose a hazard to other drivers and that you don't block traffic.
8. **Put your wallet and keys in a safe place**: Use a watertight bag you keep in a pouch strapped to your waist. Without proper precautions, wallet and keys might end up downstream.
9. **Never cross private property without the permission of the landowner**: Better yet, sample only at public access points such as bridge or road crossings or public parks. Take along a card identifying you as a volunteer monitor.
10. **Confirm your location**: Prior to visiting your site(s) check maps, and make sure all volunteers are aware using site descriptions and specific directions.
11. **Know what to do if you get bitten or stung**: Watch for irate dogs, wildlife (particularly snakes), and insects such as ticks, hornets, and wasps.
12. **Watch for vegetation in your area that can cause rashes and irritation**: Learn to identify (in all seasons) poison ivy, poison oak, sumac and other plants that may cause irritation; be aware of briers and thorny plants as well.
13. **Do not monitor if the stream is posted as unsafe:** Do not monitor if the water appears to be severely polluted. No matter what the water conditions are; always remove wet shoes and clothes as soon as possible after leaving the stream; use anti-bacterial soap and shower soon after the stream survey is completed.
14. **Do not walk on unstable stream banks**: Disturbing these banks can accelerate erosion and might prove dangerous if a bank collapses. Disturb streamside vegetation as little as possible.

15. **Be very careful when walking in the stream itself**: Rocky-bottom streams can be very slippery and can contain deep pools; muddy-bottom streams might also prove treacherous in areas where mud, silt, or sand has accumulated. If you must cross the stream, use a walking stick to steady yourself and to probe for deep water or muck. Your partner(s) should wait on dry land ready to assist you if you fall. Do not attempt to cross a stream that is swift.
16. **Come prepared for outside work**: Wear clothes such as a hat, loose fitting clothes (especially during warmer weather), closed toed shoes such as sneakers, boots or waders. Felt glued to the bottom of the shoe will improve your traction for walking in the stream. The outside conditions should determine your overall manner of dress. You should also have plenty of drinking water, sunscreen and insect repellent.

A standard well-equipped first aid kit should suffice for most of your medical situations. At a minimum your kit should contain the following items:

1. Telephone numbers of emergency personnel such as the police and ambulance (Know the location of the nearest medical facility and the nearest cell-phone signal.)
2. Several different size band-aids for minor cuts.
3. Antibacterial and/or alcohol wipes.
4. First-aid crème or ointments.
5. Triangular bandages and several gauze-pads 3-4 inches square for deeper cuts.
6. Acetaminophen for minor pain relief.
7. A needle and tweezers for removing splinters.
8. Small scissors or a single-edge razor blade for cutting tape to size.
9. A two-inch roll of gauze for large cuts (tunicates).
10. A large compress bandage to hold dressings in place.
11. A three-inch wide elastic bandage for sprains and to aid in applying pressure when necessary to slow bleeding.
12. If participants are allergic to bee stings, include a doctor prescribed antihistamine; make sure all volunteers have their necessary allergy medications prescribed for their specific condition.
13. Always have emergency telephone numbers, contact person(s) and medical information for all participants in case of emergency.

Appendix B – Level two survey data sheet (next page)

Level-two survey data sheet

Save Our Streams

A citizens volunteer stream monitoring program

(**1**) Determine your stream-reach boundary; this is a stream length up to 100-meters, which may be more or less under certain circumstances. (**2**) Near the lower end of the reach (in the deepest portion of the run), collect water samples and analyze using the chemical tests you have available. You may use your collection container to observe watercolor and clarity and to determine water odors. (**3**) Measure the width-depth and velocity, and estimate the water level. (**4**) If you use a two-pole **kick-net**, collect a minimum of three benthic macro-invertebrate samples from the best riffle or runs within your stream reach. Use the table on page five to record information about your collections. (**5**) Evaluate the physical and habitat conditions; record information about known land use activities. (**6**) Sketch your reach or submit photographs with the survey, and add any other comments that you feel are important for evaluating the conditions of your stream study site.

Stream name: SPRING RUN Survey date: 10/23/2009

Watershed: SOUTH BRANCH POTOMAC Station code: SR (2.1)

Latitude: 38-55-15 Longitude: 79-05-12 Directions to site: FROM DORCAS TRAVEL SPRING RUN RD FOR ABOUT 1 ½ MILES, SITE IS ABOUT ¼ MILE DOWNSTREAM FROM HATCHERY

Survey completed by: N. GILLIES, B. KEPLINGER, C. RETTENBURG AND T. CRADDOCK

Current weather conditions: PARTLY CLOUDY AND COOL

Past weather conditions (last 3-days): SCATTERED SHOWERS, SOME HEAVY AT TIMES

Affiliation: SPRING RUN PROJECT TEAM Email:

Mailing address: Phone number:

Water chemistry: Use the spaces below to record the results of your water chemistry analysis; attach additional sheets if necessary.

	Result	units		Result	units		Result	units
Temperature (C/F)	15.6	C	Conductivity	296	µ/CM	Alkalinity		
Dissolved oxygen	9.1	PPM	Nitrate/Nitrite			Metals (describe)		
pH	8.4		Phosphate			Fecal/E-coli		

Additional tests (describe and record results)

Physical conditions: Use the check boxes below to describe the conditions that closely resemble those of your stream. The extra lines are provided to write in any additional comments. You may see more than one type of condition; if so, be sure to indicate these on your survey (check all that apply). If multiple conditions are observed, always indicate the most dominant condition. If the condition you observe is not listed, describe it in the comment section.

Water clarity		Water color		Water odor		Surface foam	
Clear	X	None	X	None		None	
Murky		Brown		Fishy		Slight	X
Milky		Black		Musky	X	Moderate	
Muddy		Orange/red		Rotten egg		High	
Other (describe)		Gray/White		Sewage			
		Green		Chemical			

Algae color		Algae abundance		Algae growth habit		Streambed color	
Light green		None		Even coating		Brown	X
Dark green	X	Scattered		**Hairy**	X	Black	
Brown		Moderate		Matted	X	Green	
Other (describe)		Heavy	X	Floating		White/gray	
						Orange/red	

Physical condition comments: ALGAE EXTREMLY ABUNDANT, EVEN ENTRAINED IN THE SEDIMENT

Level-two survey data sheet

Estimate the percentage of your reach that is shaded.

> 80	80-60	60-40	< 40
Excellent	Good	Marginal	Poor

Circle your estimate

Width and depth: Record the wetted width and depth of the channel's habitats (riffles, runs or pools). Choose one or more features to measure. Record the average depth from a minimum of four measure-ments (one of these should be from the deepest part of the habitat). The width should be measured from the widest section of the feature. Be sure to indicate the type(s) of habitat that you have chosen. **It is best to measure the width and depth when you determine the discharge**.

	Width (feet)	Depth (feet)	Riffle	Run	Pool
1.	13	0.7			
2.				X	

Channel profiles: Width and depth measurements can be used to create a cross section profile within your reach. Choose a location in your reach across one of the channel types above. Stretch a tape from bank to bank and anchor it at both ends. Move from left to right facing in an upstream direction; measure the distance from the stream bottom to the top of the tape at selected intervals (i.e. every foot). Record your measurements in the table below. The table provides enough spaces for 20 measurements; if more are necessary you can create your own table on a separate piece of paper. Your tape measure will probably not start at zero so make sure to record the actual position of the tape as you measure across the channel.

Width intervals

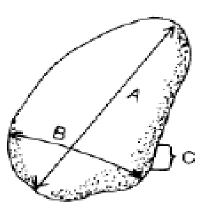
1	2	3	4	5	6	7	8	9	10
11	12	13	14	15	16	17	18	19	20

Depth measurements

1	2	3	4	5	6	7	8	9	10
11	12	13	14	15	16	17	18	19	20

Pebble count: Collect a minimum of 100-particles from your reach using a Zigzag method, percent habitat method or specific transects (e.g. every 10-meter). If you do not complete a pebble count, **always estimate** streambed composition from the riffles/runs chosen for your macroinvertebrate sample collections.

Indicate your method from the choices below.	Size Classes (Intermediate axis in millimeters)						
	Silt/clay < 0.06	Sand 0.06 – 2	Fine Gravel 2 – 24	Coarse Gravel 25 – 64	Cobble 65 – 255	Boulder 256 – 1096	Bedrock > 1096
Zigzag: X % Habitat: 10-m Transects: Woody Debris (Includes sticks, roots, leaves etc.): IIII	IIIII IIIII IIIII III	III	IIIII IIIII IIIII IIIII IIIII IIIII III	IIIII IIIII IIIII IIIII IIIII IIIII II	IIIII IIIII III	III	
Totals	18	3	33	32	13	3	



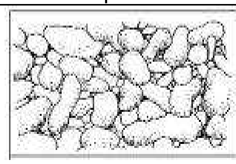
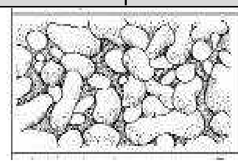
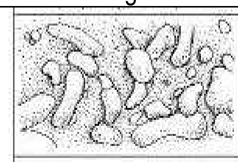
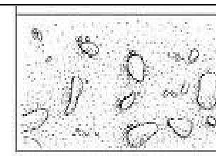
(A) Long axis (**Length**)
(B) Intermediate axis (**Width**)
(C) Short axis (**Height**)

Pebble counts require two people, one in the stream and one on shore. The person in the stream slowly walks upstream from bank to bank using one of the methods above. After each step the person reaches down without looking, picks up the first particle touched, and measures the intermediate axis with a ruler. The on-shore partner records the measurement. The process continues until 100 pebbles have been measured or the reach has been walked.

Station SR (2.1) Date(s) 10/23/09

Level-two survey data sheet

Habitat conditions: Score each habitat condition using the scales provided. Add all of the scores to determine your overall habitat score and integrity rating. Feel free to describe additional features that you feel are important. See the next page for more information about sediment deposition.

	Point values	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1
Sediment deposition		Little or no formation of depositional features; < 20% of the reach affected.	Some increase in depositional features; 20-40% of the reach affected.	Moderate amounts of depositional features; 40-60% of the reach affected.	Heavy amounts of deposition; > 60% of the reach affected.
Rating	12	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Embeddedness		 Fine sediments surrounds <10% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 10-30% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 30-60% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds > 60% of the spaces between the gravel, cobble and boulders.
Rating	9	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor

Embeddedness should be evaluated in riffles, prior to or during your macroinvertebrate collections.

	Point values	10 9 8	8 7 6	5 4 3	3 2 1
Bank vegetative protection		> 90% of the banks are covered by natural vegetation; all levels (trees, shrubs and herbs) represented; disruption from grazing, mowing etc. minimal or absent; all plants allowed to grow naturally.	70-90% of the banks covered by natural vegetation; one level of plants may be missing or not well represented; some disruption of vegetation evident; > 50% of the potential plant height remains.	50-70% of the banks covered by natural vegetation; patches of bare soil may be present and closely cropped vegetation is common; < 50% of the potential plant heights remains.	< 50% of the banks covered by natural vegetation; disruption is high; vegetation has been removed or the potential plant heights are greatly reduced.
Left / Right	4 / 7	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Bank stability		Banks are stable; no evidence of erosion or bank failure; little or no potential for future problems.	Banks are moderately stable; infrequent areas of erosion occur, mostly shown by banks healed over.	Banks are moderately unstable; 60% of the reach has some areas of erosion; high potential for erosion during flooding events.	Banks are unstable; many have eroded areas (bare soils) along straight sections or bends; obvious bank collapse or failure; > 60% of the reach has erosion scars.
Left / Right	8 / 4	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Riparian buffer width		Mainly undisturbed vegetation > 60 ft; no evidence of human impacts such as parking lots, road beds, clear-cuts, mowed areas, crops, lawns etc.	Zone of undisturbed vegetation 40-60 ft; some areas of disturbance evident.	Zone of undisturbed vegetation 20-40 ft; disturbed areas common throughout the reach.	Zone of undisturbed vegetation < 20 ft; disturbed areas common throughout the entire reach.
Left / Right	7 / 2	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor

Totals	53	> 80	80 - 60	59 - 40	< 40
		Optimal	**S**uboptimal	**M**arginal	**P**oor

Habitat comments: SITE WAS ADJACENT TO A RESIDENCE

 Station SR (2.1) Date(s) 10/23/09

Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends.

Land use: Indicate the land uses that you believe may be having an impact on your stream station. Use the letters (**S**) streamside, (**M**) within ¼ mile and (**W**) somewhere in the watershed, to indicate the approximate location of the disturbance and the numbers (**1**) slight, (**2**) moderate or (**3**) high, to represent the level of disturbance.

Active Construction	1	W	Pastureland	1	W	Single-family residences	2	S
Mountaintop mining			Cropland			Sub-urban developments		
Deep mining			Intensive feedlots	3	M	Parking lots, strip-malls etc.		
Abandoned mining			Unpaved Roads			Paved Roads	2	S
Logging			Trash dumps			Bridges	1	M
Oil and gas wells			Landfills			Other (describe)		
Recreation (parks, trails etc.)			Industrial areas					

Land use comments: THE HATCERY IS THE INTENSIVE FEEDLOT

Pipes? Yes No

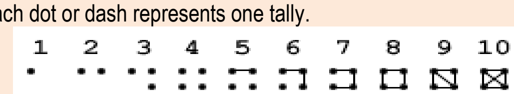
Describe the types of pipes observed and indicate if there is any discharge from the pipes. Also describe the colors and odors of the discharge.

Photograph and **sketch your reach**: Use the space below or a separate piece of paper to draw your study reach. Indicate the direction of flow, north, sample locations and important features of the reach. Photographs are an excellent method for tracking changes, especially changes related to the condition of the habitat. Choose a minimum of two permanent locations from which to take your photos. Submit your photos with your survey data sheet.

Benthic macroinvertebrates: Assess your macroinvertebrate collections by counting and identifying to the family-level if possible. Use the table on the **next two pages** to record your collections data.

Note: Although streamside identification is possible, WV Save Our Streams Coordinator recommends preserving your samples using a full count or standard sub-sampling procedure in a well-lit and more comfortable setting.

The dot-dash tally method is a convenient way to record your data. Each dot or dash represents one tally.



Level-two survey data sheet

Insect Groups

Patterned stoneflies Taxa Total	Winter stoneflies Taxa Total	Roach-like stonefly Total
Giant stonefly Total	Brown stonefly Total	Spiny crawler mayfly Total
Square-gilled mayfly Total	Minnow mayflies IIIII Taxa 1 Total 5	Flatheaded mayfly Total
Brush-legged mayfly Total	Burrowing mayflies Taxa Total	Net-spinning caddisflies III Taxa 1 Total 3
Case-building caddisflies Taxa Total	Free-living caddisfly IIIII IIIII IIIII IIIII Total 20	Common netspinner IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII Total 45
Dragonflies Taxa Total	Damselflies Taxa Total	Riffle beetle IIIII IIIII IIII Total 14
Long-toed beetle Total	Water penny Total	Other beetles (true bugs) Taxa Total
Hellgrammite/Fishfly Total	Alderfly Total	Aquatic moth Total
Non-biting midge IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII II Total 77	Black fly IIIII I Total 6	Crane fly IIII Total 4
Watersnipe fly Total	Dance fly I Total 1	Dixid midge Total

 Station SR (2.1) Date(s) 10/23/09

Level-two survey data sheet

Net-wing midge Total:	Horse fly Total:	Other fly larva I Taxa: 1 Total: 1

Non-Insect Groups

Crayfish Total:	Scud/Sideswimmer IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII IIIII II Total: 62	Aquatic sowbug Total:
Water mite Total:	Operculate snails Taxa: Total:	Non-operculate snails I Taxa: 1 Total: 1
Pea clam Total:	Asian clam Total:	Mussel Total:
Flatworms II Total: 2	Aquatic worms IIIII IIIII IIIII IIIII IIIII IIIII I Total: 31	Leeches Total:
Other aquatic invertebrates Taxa: Total:	Comments:	

Total Taxa	Total Number
14	272

Describe other aquatic life (e.g. fish, amphibians) collected or observed, as well as other indications that the reach is being used by other animals (i.e. birds, mammals, reptiles). RAINBOW TROUT

Discharge

Determine the discharge by using a flow meter (if available) or other methods such as the **float method** or a **velocity head rod** (VHR). Discharge should be measured from a run (area of the channel with fast moving water with no breaks in the surface such as protruding rocks). The more measurements collected the more accurate your discharge results will be. To convert inches into feet divide by 12. For example, if your depth measurement was 6-inches the result in feet would be 0.5. Indicate the methods chosen to measure the discharge and use the tables to record your results. Use the table on the next page to record your measurements.

Discharge method used

Float	Velocity Head Rod	Flow meter
	X	

Water Level

Low	Normal	High	Dry
	X		

Channel width 13 feet

Use the table on the next page to record your velocity data

Level-two survey data sheet

	Distance (ft)	Depth (ft)	Velocity (ft/sec)	VHR (Rise-inches)	Float (sec)	Discharge (cfs)
1	0.6	0.3	-	-		0
2	2	0.4	1.2	¼		0.96
3	2	0.6	1.2	¼		1.44
4	1.5	0.9	2.3	1		3.11
5	1.5	1.3	3.1	1 ¾		6.01
6	1.5	1.1	3.3	2		5.45
7	1.5	1.2	3.1	1 ¾		5.58
8	1	0.8	2.3	1		1.84
9	0.5	0.4	1.2	¼		0.24
10	0.3	0.3	-	-		0
11						
12						
13						
14						
15						
16						
17						
18						
19						
20						
	12.4	0.73	1.77			24.13

Average Depth 0.73 feet

Cross Sectional Area (CSA) 9.05 ft^2
(CSA = Average Depth x Width)

Discharge = CSA x Velocity
= ______ x ______
= 24.13 cfs (ft^3/sec)

If you use a float record your distance below and the number of seconds it took to travel the distance in the column indicated.

Float distance (feet) ______

Use the table below to determine **VHR velocity** from the rises recorded above. The rises below are in inches.

Rise (R)	Velocity	Rise (R)	Velocity
¼	1.2	3 ¼	4.2
½	1.6	3 ½	4.3
¾	2.0	3 ¾	4.5
1	2.3	4	4.6
1 ¼	2.6	4 ¼	4.8
1 ½	2.8	4 ½	4.9
1 ¾	3.1	4 ¾	5.0
2	3.3	5	5.2
2 ¼	3.5	5 ¼	5.3
2 ½	3.7	5 ½	5.4
2 ¾	3.8	5 ¾	5.5
3	4.0	6	5.7

Additional comments: THIS STATION WAS ADDED TO THE PROJECT IN ORDER TO GATHER ADDITIONAL INFORMATION ABOUT THE RECOVERY OF SPRING RUN

Submit an original or clear copy of your survey to the coordinator at the address provided below.

WV Department of Environmental Protection
Save Our Streams Program
601 57th Street, SE
Charleston, WV 25304

Office: (304) 926-0499 (1040); Mobile: (304) 289-7630
E-mail: timothy.d.craddock@wv.gov
Web page: http://www.dep.wv.gov/sos

 Station SR (2.1) Date(s) 10/23/09

Level-two survey data sheet

Level-two assessment

The **light blue** shaded boxes indicate that multiple families (**kinds**) are possible; tolerance values are provided.

Macroinvertebrates		Totals	Tolerance score	Number of kinds	Macroinvertebrates		Totals	Tolerance score	Number of kinds
1	Patterned stoneflies				6	Aquatic moth			
2	Winter stoneflies				4	Riffle beetle	14	56	1
1	Roach-like stonefly				5	Long-toed beetle			
1	Giant stonefly				3	Water penny			
2	Little brown stonefly				5	Whirligig beetle			
3	Spiny crawler mayfly				7	Other beetles/bugs			
5	Square-gilled mayflies				3	Hellgrammite/Fishfly			
4	Minnow mayflies	5	20	1	6	Alderfly			
3	Flatheaded mayfly				9	Non-biting midge	77	693	1
3	Brush-legged mayfly				6	Black fly	6	36	1
5	Burrowing mayflies				4	Crane fly	4	16	1
4	Net-spinning caddisflies	3	12	1	3	Watersnipe fly			
3	Case-building caddisflies				6	Dance fly	1	6	1
5	Common netspinner	45	225	1	5	Dixid midge			
3	Free-living caddisfly	20	60	1	2	Net-wing midge			
4	Dragonflies				7	Horse fly			
7	Damselflies				8	Other fly larva	1	8	1
Non-Insect Groups									
5	Crayfish				5	Pea clam			
5	Scud/Sideswimmer	62	310	1	6	Asian clam			
7	Aquatic sowbug				4	Mussel			
6	Water mite				5	Operculate snails			
10	Aquatic worms	31	310	1	7	Non-operculate snails	1	7	1
10	Leeches				Other invertebrates (**Describe**)				
7	Flatworms	2	14	1					

Complete your calculations using the metrics below. These metrics are combined to determine your overall score and integrity rating.	**Total Number**	**Total Tolerance**	**Total Kinds**
	272	1463	14

Comments: ______________________________

Metrics	Results	Points	**8**	**6**	**4**	**2**
1. Total Taxa	14	6	> 18	18 - 13	12 - 8	< 8
2. EPT Taxa	4	4	> 10	10 - 7	6 - 4	< 4
3. Biotic Index	5.38	4	< 4.0	4.0 – 5.0	5.1 – 6.0	> 6.0
4. % EPT Abundance	26.8	2	> 80	80 - 60	59.9 - 40	< 40
5. % Tolerant	41.2	2	< 2	2 - 10	10.1 - 30	> 30

Stream Score 18

Integrity Rating

> 32	32 - 24	23 - 16	< 16
Optimal	Suboptimal	Marginal	Poor

1. **Total Taxa** is simply the total number of families collected.
2. **EPT Taxa** is the total number of families within the orders of **Ephemeroptera**, **Plecoptera** and **Trichoptera**.
3. **Biotic Index** is calculated by multiplying the organism by its tolerance value to determine a tolerance score. The total tolerance is then divided by the total number of organisms collected.
4. **% EPT Abundance** is calculated by dividing total number of organisms within the orders Ephemeroptera, Plecoptera and Trichoptera by the total number collected. This result is multiplied by 100 to determine the percentage.
5. **% Tolerant** is calculated by dividing the number of tolerant organisms (≥ 7) by the total number collected. This result is multiplied by 100 to determine the percentage.

- The **Stream Score** is the sum of all five point values.

 Station SR (2.1) Date(s) 10/23/09

Appendix C – Benthic lab sheet

Stream ______________________ Watershed ______________________
Date ____________ Code ____________ Latitude ____________ Longitude ____________
Location ______________________ County ____________

	COUNT		COUNT
ANNELIDA /TURBELLARIA		**TRICHOPTERA**	
BIVALVIA			
GASTROPODA			
		PLECOPTERA	
CRUSTACEA			
EPHEMEROPTERA			
		ODONATA	
		COLEOPTERA /HEMIPTERA	
MEGALOPTERA		**DIPTERA**	
MISC. MACROINVERTEBRATES			
TOTAL FAMILIES		**TOTAL NUMBER**	

Sub-Sample Type ____________ Collected by ____________ Identified by ____________ ID Date ____________
Project Description ______________________________
Number of grids picked: ____________

Appendix D - Equipment list

In many cases not all of the equipment listed is necessary for each monitoring session; however, you should carefully plan your fieldwork and make a checklist of equipment and other items needed prior to your excursions to the stream.

1. Appropriate clothes and foot wear (i.e. hats, loose-fitting clothes for warm weather, layered clothes for cooler weather; raingear for wet-weather; waders, boots, close-toed shoes all with felt bottoms)
2. Carrying case (i.e. backpacks, plastic containers etc.)
3. Chemical kits and meters (pH meters, dissolved oxygen kit and other chemical monitoring kits as needed)
4. Collection bottles for water samples and vials for macroinvertebrate preservation
5. Collection Nets (usually a two-pole or single-pole kick-net)
6. First aid kit
7. Flow meter or equipment that can be adapted to measure flow such as, an aluminum straight-rod or a float
8. Forceps (tweezers)
9. Gloves (non-allergenic latex or rubber)
10. Open-reel tape measure (100 feet and 100 meters)
11. Plastic (flexible) mm ruler or gravelometer for pebble counts
12. Preservatives (i.e. alcohol or formalin)
13. Sample size delineator and embeddedness survey ring (optional)
14. Scrub brush (for rock rubbing)
15. Several sizes of buckets: (1) 2 to 3 ½ gallon and at least (1) 5-gallon
16. Sieve such as a wash bucket or 400-micron EZ-strainer, to aid in sorting, counting and separating
17. Small folding table and chairs
18. Sorting trays (white bottom and durable; divided plastic inserts***;*** ice cube trays or plastic craft organizers also work well)
19. Survey instruments (stadia rod etc.) for measuring channel profiles
20. Submersible thermometer
21. Various types of magnifying lenses (box magnifiers, 10x hand-lens, etc.)
22. Wash bottles

Appendix E - Guide to Aquatic Invertebrates

This section is designed to assist volunteer monitors with the identification of many, but not all, aquatic invertebrates found in our rivers, streams and wetlands. General information is included about the distinguishing features of the aquatic stage that aid in identification. Also included are the organisms, habit, feeding group, tolerance rating, size range, and habitat preferences. For images or more information, simply click-on the links provided; [Adult] images are provided for many families. Most of the images are courtesy of the Cacapon Institute; Jennifer Gillies (JG) artist.

Note: Many of the words that may be unfamiliar are defined using on-line dictionaries. The credibility of the web pages included here has not been thoroughly investigated. Apply the normal standards of Internet research to your investigation of each website in order to determine its veracity.

Habitat preferences

- (**F**) **Fast-moving** waters with rocky substrate (i.e. riffles and runs of streams and rivers)
- (**S**) **Slow-moving** or still waters with soft substrate and vegetation (i.e. pools and backwater areas of streams and rivers; wetlands and ponds)

Size range categories (mm)

> 50 (VL)	50 – 30 (L)	29 – 15 (M)	14 – 5 (S)	< 5 (VS)

General	1. Kingdom	
	2. Phylum	
	3. Class	
	4. Order	
	5. Family	
	6. Genus	
Specific	7. Species	

Classification: Plants and animals are classified according to a hierarchal system that arranges the organisms into groups based upon their similarities. These groups are arranged from general to very specific. The science of classification is known as taxonomy. The table provides the basic taxonomic groups. In certain situations these major groups are sub-divided. This occurs when a group of organism is different enough to be noted but not different enough to place them in a separate classification.

Stress (pollution) tolerance is the organism's ability to withstand a certain amount of anthropogenic influences. The general stress categories of low, moderate and high are described below. The index range for each category is based upon a (0-10) scale, which is based mostly on the invertebrate's ability to withstand varying levels of dissolved oxygen and other chemical and physical disturbances. For example, invertebrates with a low tolerance need adequate dissolved oxygen and chemical and physical stability, while those with a high tolerance can survive for a period of time when dissolved oxygen levels are less than adequate or other disturbances may be present. In some cases tolerance values are undetermined (**U**).

Low (**L**)			Moderate (**M**)				High (**H**)		
1	2	3	4	5	6	7	8	9	10

Insect groups

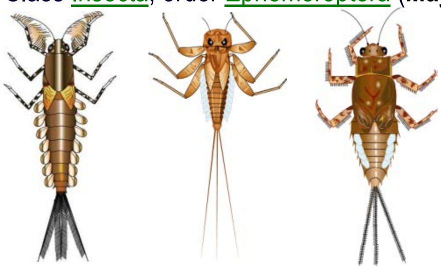
Order Collembola (Springtails)
Order Coleoptera (Beetles)
Order Diptera (True flies)
Order Ephemeroptera (Mayflies)
Order Hemiptera (True bugs)
Order Lepidoptera (Aquatic moth)
Order Megaloptera (Hellgrammites and Alderflies)
Order Neuroptera (Spongillaflies)
Order Odonata (Dragonflies and Damselflies)
Order Plecoptera (Stoneflies)
Order Trichoptera (Caddisflies)

Non-insect groups

Phylum Annelida (Leeches and Worms)
Class Bivalvia (Clams and Mussels)
Sub-phylum Crustacea (Crayfish, Scuds and Sowbugs)
Class Gastropoda (Snails)
Class Hydrozoa (Freshwater jellyfish)
Class Spongilla (Freshwater sponge)
Order Trombidiformes (Water mites)
Class Turbellaria (Flatworms)

Arthropod Groups

Class Insecta; order Ephemeroptera (**Mayflies**) JG



Wing pads may be present on the thorax; three pairs of segmented legs attach to the thorax; one claw occurs on the end of the segmented legs; gills occur on the abdominal segments and are attached mainly to the sides of the abdomen, but sometimes extend over the top and bottom of the abdomen; gills consist of either flat plates or filaments; three long thin caudal (tails filaments) usually occur at the end of the abdomen, but there may only be two in some kinds. (Hemimetabolism)

Swimming mayflies

1. Ameletidae (Comb-mouth minnow mayfly): Comb of stiff spines on the mouthparts; gills have a dark, sclerotized (hard) band along the outside edge; antennae are shorter than twice the width of the head; usually have dark bands on the tail and alternating dark and light on the abdomen. Swimmer, clinger; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
2. Baetidae (Small minnow mayfly): Antennae two times longer than the width of the head; gills variable in shape and attached at abdominal segments one through seven; two or three caudal (tail) filaments. Swimmer; Collector, gatherer; (**M**); VS-M (**F**/**S**) [Adult]
3. Isonychiidae (Brush-legged mayfly): Forelegs have a double row of setae (hairs); gills oval shaped and present on abdominal segments one through seven; long hairs on the margins of the caudal filaments. Swimmer, crawler; Collector, gatherer; (**L**); S-M (**F**) [Adult]
4. Siphlonuridae (Primitive minnow mayfly): Antennae less than two-time the width of the head; gills usually oval shaped and present on abdominal segments one through seven; long setae on the caudal filaments. Swimmer; Collector, gatherer; (**L**); S-M (**F**/**S**)

Clinging/crawling mayflies

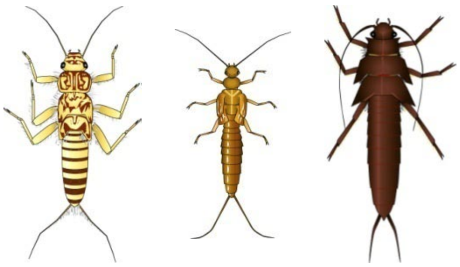
5. Caenidae (Square-gilled mayfly): Gills on the first abdominal segment very small; gills on the second segment operculate (plate-like) covering much of the remaining gills. Clinger, crawler, burrower; Collector, gatherer; (**M**); VS-S (**F**/**S**)
6. Ephemerellidae (Spiny-crawler mayfly): Gills present of the first abdominal segment but absent from the second; gills usually present on the remaining segments; two or three caudal filaments. Clinger, crawler; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
7. Heptageniidae (Flatheaded mayfly): Body, head and legs are flattened (femora); gills present on abdominal segments one through seven; usually three caudal filaments, but some may have two. Clinger; Scraper; (**L**); S-M (**F**) [Adult]

8. Leptophlebiidae (Prong-gilled mayfly): Gills on abdominal segments two through seven forked and variable in shape; gills on the first segment finger-like; short hairs usually cover the caudal filaments. Clinger, crawler; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
9. Tricorythidae (Stout-crawler mayfly): Gills absent from abdominal segment one; gills on segment two are (operculate), plate-like triangular or oval shaped and conceals gills on segments three through six. This family is similar in appearance to Caenidae. Clinger, crawler, burrower; Collector, gatherer; (**M**); VS-M (**F**/**S**) [Adult]

Burrowing mayflies

10. Beatiscidae (Armored mayfly): Top portion of the thorax is fused and coves most of the abdomen concealing the gills; caudal filaments are short and fringed with hairs. Burrower, crawler; Collector, gatherer; (**M**); VS-M (**F**) [Adult]
11. Ephemeridae (Burrowing mayfly): Has upturned mandibular tusks; head and front legs slightly widened and are used for burrowing; gills on the upper abdominal segments are small and the remaining gills are forked with fringed margins (feathered) and held over the top and sides of the abdomen. Burrower; Collector, gatherer; (**M**); M-L (**S**/**F**) [Adult]
12. Potamanthidae (Hackle-gilled mayfly): Mandibular tusks present; front legs slender, not modified for burrowing; gills on segment one small, gills on remaining abdominal segments are feathery. Burrower; Collector, gatherer; (**M**); S-M (**S**/**F**)

Class Insecta; order Plecoptera (**Stoneflies**) JG



Long thin antenna project in front of the head; wing pads usually present on the thorax but may only be visible in older larvae; three pairs of segmented legs attach to the thorax; two claws are located at the end of the segmented legs; gills occur on the thorax region, usually on the legs or bottom of the thorax, or there may be no visible gills (usually there are none or very few gills on the abdomen); gills are either single or branched filaments; two long thin tails project from the rear of the abdomen. Stoneflies have very low tolerance to many insults; however, several families are tolerant of slightly acid conditions. (Hemimetabolism)

Winter stoneflies

1. Capniidae (Small winter stonefly): Slender elongated body; front of thorax slightly wider than the abdomen; wing pads not divergent from the midline; abdominal segments separated by a membranous fold. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
2. Leuctridae (Rolled-wing stonefly): Slender elongated body; front of thorax slightly wider than the abdomen; wing pads not divergent from the midline; abdominal segments not separated by a membranous fold. Very similar characteristics to Capniidae. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
3. Taeniopterygidae (Large winter stonefly): Stout bodies with pronotum much wider than the abdomen; wing pads greatly divergent from the midline. Clinger, crawler; Shredder, scraper; (**L**); S-M (**F**) [Adult]

Patterned stoneflies

4. Chloroperlidae (Green stonefly) Body elongated, front of the thorax slightly wider than the abdomen; wing pads not divergent from the midline; tails (cerci) shorter than the abdomen. Will sometimes have patterns similar to Perlodidae. Clinger, crawler; Shredder, predator; (**L**); M (**F**) [Adult]
5. Perlidae (Common stonefly): Usually a large strikingly patterned and often having a golden color; finely branched gills present on all thoracic segments; wing pads diverge slightly from the midline. Sometimes called the golden stonefly. Clinger, crawler; Predator; (**L**); M-L (**F**) [Adult]
6. Perlodidae (Patterned stonefly): Strikingly patterned and colored similar in appearance to Perlidae; hind wing pads divergent; no gills on the thoracic segments. Clinger, crawler; Shredder; (**L**); M-L (**F**) [Adult]

Other stoneflies

7. Nemouridae (Little brown stonefly): Very small, often hairy appearance; wing pads diverge greatly from the midline; hind legs as long as the abdomen; gills often present between the head and thorax. Clinger, crawler; Shredder; (**L**); S-M (**F**/**S**) [Adult]
8. Peltoperlidae (Roach-like stonefly): Small stout body; rear divergent wing pads; thoracic segments are oval or triangular shaped and cover much of the upper body; some have fine gills on the front legs. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
9. Pteronarcyidae (Giant stonefly): Very large and usually dark brown in color; finely branched gills on all thoracic segments plus the first two abdominal segments. Clinger, crawler; shredder; (**L**); M-VL (**F**) [Adult]

Class Insecta; order Trichoptera (**Caddisflies**) JG

Head has a thick hardened skin; antennae are very short, usually not visible; no wing pads occur on the thorax; top of the first thorax always has a hardened plate and in several families the second and third section of the thorax have a hardened plate; three pairs of segmented legs attach to the thorax; abdomen has a thin soft skin; single or branched gills on the abdomen in many families, but some have no visible gills; pair of prolegs with one claw on each, is situated at the end of the abdomen; most families construct various kinds of retreats(also called cases or nets) consisting of a wide variety of materials collected from the streambed. (Holometabolism)

Net-spinning caddisflies

1. Hydropsychidae (Common netspinner): Top of all thoracic segments hardened; most abdominal segments have tufts of finely branched gills; anal prolegs terminate into a brush of hairs. Do not make cases but instead creates a retreat (net) made of a variety of materials held together by fine strands of silk. Clinger, crawler; Collector, filterer; (**M**); M-L (**F**/**S**) [Adult]
2. Philopotamidae (Finger-net caddisfly): Labrum (structure between the mouthparts) is t-shaped and membranous; head capsule large usually orange in color; only first thoracic segment is hardened; abdominal gills usually absent. Builds a long tube or finger-like net. Clinger, crawler; Collector, filterer; (**L**); M (**F**) [Adult]
3. Polycentropodidae (Tube-net caddisfly): Labrum is rounded and hardened; only first thoracic segment is hardened; no plates or gills on the abdominal segments. Does not build cases but instead constructs a net that is often in the shape of a long tube. Clinger, crawler; Collector, filterer, predator; (**M**); S-L (**F**/**S**) [Adult]
4. Psychomiidae (Trumpet-net caddisfly): Bottom of thorax is hardened with black edges; middle thoracic segment is swollen and usually larger than the others; has an enlarged hatchet shaped leg segment on the upper legs. Clinger, crawler; Collector; (**M**); M (**F**)

Free-living caddisfly

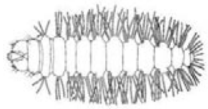
5. Rhyacophilidae (Free-living caddisfly): First thoracic segment is hardened; abdominal gills variable; hardened plate on top of abdominal segment nine; distinctive anal prolegs with large claws; is often green in color; referred to by trout fisherpersons as the green sedge. This family does not build a case or net, but often uses silk strands to attach itself to substrates. Clinger, crawler; Predator; (**L**); M-L (**F**) [Adult]

Case-building caddisflies

6. Brachycentridae (Humpless-case caddisfly): Antennae close to the margins of the head capsule; first two thoracic segments with hardened plates; no humps on abdominal segments; gills simple or lacking. Case is elongated and made of strips of materials, resembles a log cabin. Clinger; Collector, gatherer, shredder; (**L**); M (**F**) [Adult]
7. Glossosomatidae (Saddle-case caddisfly): First thoracic segment is hardened; hardened plate on top of abdominal segments nine. Case resembles a tortoise shell or saddle. Clinger; Scraper, shredder; (**L**); VS-M (**F**) [Adult]
8. Helicopsychidae (Snail-case caddisfly): Body is curled; all three thoracic segments are hardened; stout hairs at the end of the third thoracic segment; gills present on anterior abdominal segments. Case resembles a snail shell. Clinger; Scraper; (**L**); VS-S (**F**)
9. Hydroptilidae (Purse-case caddisfly): All three of their thoracic segments have sclerotized dorsal plates; no gills on the abdomen; most commonly build cases with sand, algae, silk or detritus, but the shapes vary considerably. Clinger, crawler; Scraper; (**M**); VS-S (**F**/**S**) [Adult]
10. Lepidostomatidae (Lepidostomid-case caddisfly): Antennae located close to the eyes; lateral hump on abdominal segment one; first two thoracic segments hardened; gills simple or lacking; hardened plate on top of abdominal segment nine. Case is usually four-sided built with square pieces of barks and leaves. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
11. Leptoceridae (Longhorn-case caddisfly): Antennae prominent; first two thoracic segments hardened; hind legs are usually longer than the front legs; abdominal gills variable. Cases are built from a variety of materials and vary considerably; the most common is a stone/sand case resembling a long tube. Clinger, crawler; Collector, predator; (**L**); S-M (**F**/**S**) [Adult]
12. Limnephilidae (Northern-case caddisfly): Antennae between the eyes and the mouth; first two thoracic segments hardened; dorsal and lateral humps on first abdominal segment; hardened plate on the top of abdominal segment nine; abdominal gills variable. The cases are built from many kinds of bottom materials and exhibit a wide variety of shapes and sizes. Clinger, crawler; Shredder; (**L**); S-L (**F**) [Adult]
13. Molannidae (Hooded-case caddisfly): Two-thirds of the top of the thorax is hardened; tarsal claws on the hind legs smaller than the rest and are covered with fine hairs; gills along the abdomen are simple or branched; a hardened plate sits atop abdominal segment nine. The cases are constructed mostly with sand and are shaped like a flattened tube with a hood that extends over the opening of the case. Clinger, crawler; Collector, shredder; (**M**); S-M (**S**) [Adult]

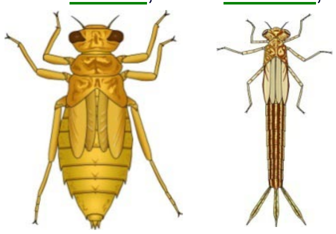
14. Phryganeidae (Giant-case caddisfly): Head and portions of the thorax marked with prominent stripes; front part of the thorax hardened; dorsal and lateral humps on abdominal segment one; hardened plate on top of abdominal segment nine. Builds elongated cases out of plant fragments. Clinger, crawler; Collector, predator; (**M**); M-L (S) [Adult]
15. Uenoidae (Uenoid-case caddisfly): The first two thoracic segments are hardened and there are some small plates present on the third; abdominal segment one has a hump, and the anterior margin of their mesonotum is notched on either side of the midline; cases are variable but usually always constructed with small stones and sand. Clinger, crawler; Scraper; (**L**); S-M (**F**) [Adult]

Class Insecta; order Lepidoptera (**Aquatic moth**)



Head hardened; a few families have elongated lateral gills; three pairs of segmented legs attach to the thorax; abdomen with prolegs that end in tiny hooks. They often look very similar to terrestrial caterpillars; closely related to Trichoptera. (Holometabolism) Crawler, burrower; Shredder; (**M**); 3-35 (**S**/**F**) [Adult]

Class Insecta; order Odonata; sub-orders Anisoptera (**Dragonflies**) and Zygoptera (**Damselflies**) JG



Dragonflies: Lower lip (labium) is long and elbowed to fold back against the head when not feeding, thus concealing other mouthparts; wing pads are present on the thorax; three pairs of segmented legs attach to the thorax; no gills on the sides of the abdomen; Dragonflies have three pointed structures may occur at the end of the abdomen forming a pyramid shaped opening; bodies are long and stout or some- what oval. Damselflies have three flat gills at the end of the abdomen forming a tail-like structure and their bodies are long and slender. (Hemimetabolism)

Dragonflies

1. Aeshnidae (Darner dragonfly): Prementum and papal lobes are flattened; six or seven antennal segments present all of a similar size. Clinger, crawler; Predator; (**L**); M-VL (**S**) [Adult]
2. Cordulegastridae (Spiketail dragonfly): Often appear hairy; prementum large, covering much of the underside of the head, usually triangular shaped. Clinger, crawler; Predator; (**L**); M-L (**S**/**F**) [Adult]
3. Gomphidae (Clubtail dragonfly): Body shape variable from long cylindrical to oval and flattened; prementum flattened; third antennal segment large and different from the rest. Clinger, crawler; Predator; (**M**); M-L (**S**/**F**) [Adult]
4. Libellulidae (Skimmer dragonfly): Antennal segments similar in shape and size; prementum and palpal lobes spoon shaped or small and rounded. Crawler, burrower; Predator; (**H**); M-L (**S**) [Adult]

Damselflies

5. Calopterygidae (Broad-wing damselfly): Lower portion of labium is diamond shaped; first antennal segment longer than all the others together; middle gills shorter than the lateral two; no visible veins on the gills. Clinger, crawler; Predator; (**M**); M-L (**S**/**F**) [Adult]
6. Coenagrionidae (Narrow-wing damselfly): Slender but slightly more stout bodied than most damselflies; labium triangular shaped; antennal segments same length; gills same length, veins radiate diagonally. Clinger, crawler; Predator; (**H**); M-L (**S**) [Adult]
7. Lestidae (Spread-wing damselfly): Long and slender bodied; labium stalked and spoon shaped; all gills similar in shape with perpendicular veins. Clinger, crawler; Predator; (**H**); M-L (**S**) [Adult]

Class Insecta; order Coleoptera (**Beetles**) JG

Head has thick hardened skin; thorax and abdomen of most adult families have moderately hardened skin, several larvae have a soft-skinned abdomen; no wing pads on the thorax in most larvae, but wing pads are usually visible on adults; three pairs of segmented legs attach to the thorax; no structures or projections extent from the sides of the abdomen in most adult families, but some larval stages have flat plates or filaments; no prolegs or long tapering filaments at the end of the abdomen. Beetles are one of the most diverse the insect groups, but are not as common in aquatic environments. Most of the adult stages of the families listed here are aquatic or semi-aquatic. (Holometabolism)

1. Chrysomelidae (Reed beetle): The body is soft; three-pairs of segmented legs attached to the thorax and two hooks on the lower end of the abdomen. Crawler; Collector, gatherer; (**H**); S-M (**S**) [Adult]
2. Dryopidae (Long-toed beetle): Adults are hard bodied with very short comb-like antennae. The family is similar in appearance to the adult riffle beetle. The larva of this beetle is not aquatic but may be found in the splash zone. Clinger, crawler; Shredder; (**M**); VS-S (**S**)

3. Dytiscidae (Predacious diving beetle): Legs have five-segments and two-claws on the end; abdomen terminates into a pair of filaments. [Adult] slender antennae; hind coxae extends posterior dividing the first abdominal segment into two sections. Swimmer, crawler; Predator; (**M**); VS-VL (**S**)
4. Elmidae (Riffle beetle): Legs with four segments and a single claw; nine abdominal segments some with a cavity that protect the hind gills. [Adult] hard bodied, slender sometimes clubbed antennae; the forewings have numerous rows of indentations; legs long compared to body. Clinger, crawler; Scraper, shredder; (**M**); VS-S (**F**)
5. Gyrinidae (Whirligig beetle): Two claws of each leg, legs with five segments; ten abdominal segments with pairs of lateral filaments. [Adult] compound eyes, which appear divided into pairs; antennae clubbed; mid and hind legs paddle-like. Swimmer, crawler; Predator; (**M**); S-L (**S/F**)
6. Haliplidae (Crawling water beetle): Legs with five segments and a single claw; abdomen terminates into long filaments; some have many long slender filaments along the entire length of the body. [Adult] antennae long and slender; forewings have many indentations; legs lined with small hairs for swimming. Swimmer, crawler; Shredder; (**H**); S-M (**S**)
7. Hydrophilidae (Water scavenger beetle): Large mandibles; legs with four segments and a single claw; end of the abdomen usually blunt. [Adult] antennae clubbed with cup-like segments at the base; hind coxae (joined base) do not extend or divide the abdomen. Swimmer, crawler; Predator; (**H**); VS-VL (**S**)
8. Ptilodactylidae (Toe-wing beetle): Legs with four segments and a single claw; abdomen has ventral gills. Very similar in appearance to the riffle beetle larva. Clinger, crawler; Scraper, shredder; (**M**); VS-S (**F**) [Adult]
9. Psephenidae (Water penny): Body flattened with thoracic and abdominal segments expanded so that the legs and head are obscured from above; legs terminate into a single claw. The adult is semi-aquatic, sometimes encountered near the stream. Clinger, crawler; Scraper; (**L**); VS-M (**F**)

Class Insecta; order Hemiptera (**True bugs**)

The most distinguishing characteristic of the order is the mouthparts that are modified into an elongated, sucking beak. Most adults have hemelytra, which are modified leathery forewings. Some adults and all larvae lack wings; both most mature larvae posses wing pads. Both adults and larvae have three-pairs of segmented legs with two tarsal claws at the end of each leg. Many families are able to also utilize atmospheric oxygen. This order is generally not used to assess the biology of flowing waters, due to their ability to use atmospheric oxygen. Several families are described below. (Hemimetabolism)

Surface

1. Corixidae (Water boatman): Broad triangular beak; forelegs are scoop-like and fringed with hairs; antennae are short and concealed beneath the eyes. Swimmer; Predator; (**H**); VS-M (**S**)
2. Gerridae/Veliidae (Water striders): Variable body shape; cylindrical beak; rear legs extend well beyond the tip of the abdomen. Swimmer; Predator; (**H**); VS-M (**S**)
3. Notonectidae (Backswimmer): Body cylindrical; antennae are short and concealed beneath the eyes; hind legs are oar-like; hind tarsal claws inconspicuous. Swimmer; Predator; (**H**); M (**S**)

Sub-surface

4. Belostomatidae (Giant water bug): Large oval body; antennae are short and concealed beneath the eyes; raptorial front legs. Swimmer, clinger; Predator; (**H**); M-VL (**S**)
5. Hydrometridae (Water measurer): Body slender and elongated; antennae longer than the head; head usually longer than the thorax; legs are long and slender, each with two claws. Clinger, crawler; Predator; (**H**); S-M (**S**)
6. Nepidae (Water scorpion): Body long and cylindrical; antennae are inconspicuous and concealed beneath the eyes; forelegs are raptorial other legs are long and slender; abdomen terminates into a long breathing appendage. Clinger, crawler; Predator; (**H**); M-L (**S**)

Class Insecta; order Megaloptera (**Hellgrammites** and **Alderflies**) JG

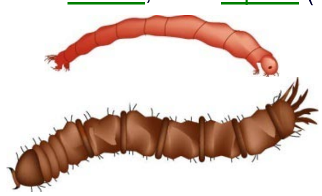
Head and thorax has thick hardened skin, while the abdomen has thin soft skin; prominent chewing mouthparts project in front of the head; no wing pads on the thorax; three pairs of segmented legs attach to the thorax; seven or eight pairs of stout tapering filaments extend from the abdomen; end of the abdomen has either a pair of prolegs with two claws on each proleg, or a single long tapering filament with no prolegs. (Holometabolism)

1. Corydalidae (Hellgrammite/Fishfly): Elongate dorsally flattened body; large jaws on the head, projecting forward; first eight abdominal segments and segment ten with paired lateral filaments; abdomen terminates in fleshy appendages bearing hooks. Clinger, crawler; Predator; (**L**); M-VL (**F**) [Adult]
2. Sialidae (Alderfly): Elongate dorsally flattened body; large jaws on the head, projecting forward; first seven abdominal segments and segment ten with paired lateral filaments; abdomen terminates into a single long hairy filament. Crawler, burrower; Predator; (**M**); S-L (**S**/**F**) [Adult]

Class Insecta; order Collembola (**Springtails**)

Abdomen consisting of six segments, the first having collophores; abdomen terminates into a forked appendage. Has a habit of jumping on the surface of the water. Swimmer, crawler; Collector, gatherer; (**M**); VS (**S**) (Holometabolism)

Class Insecta; order Diptera (**True flies**) JG



Head may be a capsule-like structure with thick hard skin; head may be partially reduced so that it appears to be part of the thorax, or it may be greatly reduced with only the mouthparts visible; no wing pads occur on the thorax; false-legs (pseudo-legs) may extend from various sections of the thorax and abdomen in some families; no segmented legs in the larval forms; thorax and abdomen composed of entirely soft skin, but some families have hardened plates scattered on various body features. (Holometabolism); the larval stages do not have segmented legs.

Midges/mosquitoes

1. Blephariceridae (Net-wing midge): Head fused with thorax and first abdominal segment; six abdominal segments with deep constrictions between segments; gill tufts present ventrally. Clinger; Scraper; (**L**); VS-S (**F**) [Adult]
2. Ceratopogonidae (Biting midge): Variable characteristics occur in this family, often similar in appearance to Chironomidae; usually a distinct head is visible with small mandibles. Crawler, burrower; Predator; (**H**); VS-M (**F**/**S**) [Adult]
3. Chironomidae (Non-biting midge); Hardened clearly visible head; long worm-like body; two pairs of prolegs with terminal hooks. Some families may be red in color due to a hemoglobin-like compound. Crawler, burrower; Collector, gatherer; (**H**); VS-L (F/S) [Adult]
4. Culicidae (Mosquito): Head hardened and separate from the thorax; brush-like setae near the labrum (upper-lip); thorax is fused and swollen and wider than the abdomen. Swimmer; Collector, gatherer, filterer; (**H**); VS-M (**S**) [Adult]
5. Dixidae (Dixid midge): Head hardened and rounded; prolegs terminate in hooks on abdominal segment one and two; abdomen terminates into two lobes fringed with hairs. Crawler, burrower; Collector, gatherer; (**M**); VS-M (**S**/**F**) [Adult]
6. Syrphidae (Rat-tailed maggot): Head blunt or reduced and withdrawn into the thorax; 7 prolegs; abdomen terminates into a very long respiratory tube. Crawler, burrower; Collector, gatherer; (**H**); VS-M (**S**) [Adult]

Flies

7. Athericidae (Watersnipe fly): Body long (caterpillar-like); head reduced but may be visible; prolegs on most abdominal segments; abdomen ends in a fringed tail. The family is often green in color. Clinger, crawler; Predator; (**L**); S-M (**F**) [Adult]
8. Empididae (Dance fly): Body elongated; head reduced or pulled into the thorax; prolegs present on most abdominal segments; prolegs longer on segment eight; abdomen is blunt on the end or terminates in welts. Crawler, burrower; Predator; (**H**); VS-M (**S**/**F**) [Adult]
9. Muscidae (Muscid fly): Anterior portion of the body tapered, posterior is blunt; head reduced or withdrawn into thorax; whelps on abdominal segments; abdomen terminates into a pair of respiratory tubes. Predator; (**H**); (S-M); (**S**/**F**) [Adult]
10. Psychodidae (Moth fly): Head hardened, rounded and separate from the thorax; body segments with two to three secondary divisions (annuli) often having hardened plates. Burrower; Collector, gatherer; (**H**); VS-S (**S**) [Adult]
11. Ptychopteridae (Phantom cranefly): Head hardened and rounded; prolegs present on abdominal segments one through three, terminating with claws; abdomen terminates into a long respiratory tube. Crawler, burrower; Collector, gatherer; (**H**); M (**S**) [Adult]
12. Simuliidae (Black fly): Head hardened and rounded bearing a pair of labral fans (mouth brushes); prolegs on lower thorax; lower third of the abdomen is swollen (vase-like) and terminates in a ring of hooks. Clinger; Collector, filterer; (**M**); VS-M (**F**) [Adult]
13. Stratiomyidae (Solider fly): Body is flattened dorsally with a leathery feel; head is reduced but visible; thorax is broader than the head; spiracles at the end of the abdomen for breathing. Swimmer, burrower; Collector, gatherer; (**H**); S-L (**S**) [Adult]
14. Tabanidae (Horse fly): Body spindle shape both ends tapered; head reduced usually not visible; creeping welts with small hooks present on abdominal segments one through seven; no prolegs. Crawler, burrower; Predator; (**H**); M-VL (**S**/**F**) [Adult]

15. Tipulidae (Crane fly): Rounded head capsule, often reduced and barely visible; ventral welts on some abdominal segments; abdomen terminates into a disc surrounded by lobes or tentacle-like projections of varying shapes. Crawler, burrower; Shredder, predator; (**M**); VS-VL (**F**/**S**) [Adult]

Class Insecta; order Neuroptera (**Spongillaflies**)

Antennae long and multi-segmented; jaws long and needle-like; body covered in tiny hairs (seta); has a pair of hardened plates on the thorax and each abdominal segment. They are associated with freshwater sponges, found on the outside or in the canals of the sponge. (Holometabolism) Clinger; Piercer; (**U**); VS-S (**S**)

Class Arachnida; order Trombidiformes; family Hydrachnidae (**Water mites**)

Four-pairs of segmented legs; one-pair of pedipalps; body is rounded and appears to consist entirely of an abdomen without segments. When captured will move very rapidly in a circular pattern. Click-here to view the life cycle. Swimmer, crawler; Predator; (**M**); S-M (**F**)

Sub-phylum (class) Crustacea (**Crayfish**, **Shrimp**, **Scuds** and **Sowbugs**) JG

More than three pairs of legs (> 6) attached to the thorax; the first several pairs of legs may have a hinged claw, which is often enlarged as in the order Decapoda; bodies strongly flattened from top to bottom or from side to side; abdomen consists of individual segments or the segments may be fused to form a thoracic shield; some kinds have a broad flipper on the end of the abdomen.

Order **Amphipoda**

1. Gammaridae (Sideswimmer/Scud): Having a shrimp-like appearance; body flattened from side to side; one pairs of antennae of equal length; seven-pairs of walking legs, first two are claw-like the remaining legs are simple. Has a habit of swimming sideways. Crawler, swimmer; Collector, gatherer; (**M**); S-M (**F**)

Order **Decapoda**

2. Cambaridae (Crayfish): Body mostly dorsally flattened; two-pairs of antennae one longer than the other; five-pairs of legs, first three-pairs with hinged claws and the first pair of claws are greatly enlarged; abdomen terminates in a flipper-like structure. Crawler, burrower; Collector, gatherer; (**M**); M-VL (**S**/**F**)
3. Palaemonidae (Freshwater shrimp): Cephalothorax and abdomen cylindrical with some side-to-side flattening; 5-pairs of walking legs the first two have claws, which are not enlarged; abdomen terminates in a flipper-like structure. Crawler, swimmer; Scraper; (**M**); M-VL (**S**)

Order **Isopoda**

4. Asellidae (Aquatic sowbug): Body dorsally flattened; two-pairs of antennae one longer than the other; seven-pairs of legs, the first is claw-like and slightly enlarged, and the others have a simple pointed claw. Looks similar in appearance to its terrestrial cousin, the pill bug. Crawler, burrower; Collector, gatherer; (**H**); S-M (**S**/**F**)

Non-Arthropod Groups

Class Gastropoda; sub-classes Prosobranchia (**Operculate snails**) and Pulmonata (**Non-operculate snails**) JG

Operculate snails have a flat lid-like structure called an operculum that can seal the body of the snail inside the shell; the whorls of the shell bulge out distinctively to the sides (inflated); most have their opening on the right when the narrow (dextral) end is held up; shells often extended into a spiral shape. Non-operculate snails have no operculum; the whorls of the shell do not distinctly bulge out to the sides; often the shells of most kinds are shaped like a low flat cone or coiled flat instead of being extended in a spiral shape. Typical size range for most snails is VS-L, which includes the shell. For more information and images, visit Marshall University's Aquatic Snails of West Virginia.

Operculate snails

1. Hydrobiidae (Pebble snail): Shell is whorled and bulges out to the side (inflated); opens to the right when the narrow end is held up. The family is very diverse in shell size and shape; shell shape can range from conical (cone-like) to spherical (rounded). Clinger, crawler; Scraper; (**L**) (**F**)

2. Pleuroceridae (Rock snail): Shell is spiraled and whorled but does not budge out to the side (flattened); opens to the right when the narrow end is held up; operculum is smaller than most others and can be pulled into the shell. Clinger, crawler; Scraper; (**L**) (**F**)
3. Viviparidae (Viviparid snail): Shell is whorled and bulges out to the side (inflated); opens to the right when the narrow end is held up; operculum has concentric lines, which are slightly off-center. Clinger, crawler; Scraper; (**M**) (**S**)

Non-operculate snails

4. Ancylidae (Limpet): Shell shaped like a low flat cone; no operculum. Clinger, burrower; Scraper; (**H**) (**F**)
5. Planorbidae (Orb snail): Shell is coiled flat instead of extended in a spiral; no operculum. Clinger, burrower; Scraper; (**M**) (**S**/**F**)
6. Physidae (Left-handed snail): Shell is high, spiraled, with a slight bulge; opens to the left when the narrow end is held up; no operculum. Clinger, crawler; Scraper; (**H**) (**S**/**F**)

Class Bivalvia (**Clams** and **Mussels**) JG

Two shells opposite of each other and strongly connected by a hinged ligament; the shell is thick and strong or thin and fragile in some kinds; growth rings on the shell are either far apart and are distinctly raised, or very close together and hardly raised at all; the foot usually consists of two tubular structures that can often be seen protruding from the shell; the body is soft tissue, often pinkish or gray in color.

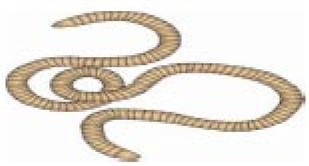
Clams

1. Corbiculidae (Asian clam): Shell is rounded; brown in color usually lighter than mussels; raised separated ridges along the top and sides of the shell. Clinger, burrower; Collector, filterer; (**M**); VS-VL (**S**/**F**)
2. Sphaeriidae (Pea clam): Shell is very small and rounded; light colored; ridges spaced close together, not raised. Clinger, burrower; Collector, filterer; (**M**); VS-M (**S**/**F**)

Mussels

3. Unionidae (Mussel): Largest of the bivalves; shell usually dark in color, variable in shape but maybe somewhat oblong; has many indentations and ridges on the tops and sides of the shell. Clinger, burrower; Collector, filterer; (**L**); VS-VL (**S**/**F**)

Phylum Annelida (**Leeches** and **Worms**) JG



Body is soft, muscular and cylindrical in shape; body consists of many similar, round ring-like segments arranged in rows; numerous segments along the entire length, number often depends upon the order or family. Leeches have distinct suckers situated on the bottom of the body, one at the front and one at the rear.

Leeches

1. Hirudinea (Leech): Body dorsally flattened with 34 segments, which are divided so there appears to be more; suction disks present on one or both ends; eyespots may be present. Crawler, burrower; Predator, parasite; (**H**); S-VL (**S**/**F**)

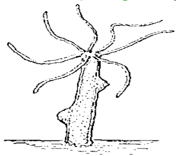
Worms

2. Oligochaeta (Aquatic worm): Body elongated (worm-like); divided into many segments most having bundles of small hairs; no eyespots or suckers present. Aquatic earthworms are common in riffles; however wetlands have a much wider variety from this group (i.e. Naidid worms, Tubiflex worms etc.) Crawler, burrower; Collector, gatherer; (**H**); VS-VL (**F**/**S**)
3. Nematoda (Round worm): Worm-like; no segmentation; body usually translucent or having a pale coloration. Crawler, burrower; Collector, gatherer; (**H**); VS-M (**S**)
4. Nematomorpha (Horsehair worm): Body very long and slender; no segments. Burrower; Parasite, predator; (**H**); VL (**S**)

Class Turbellaria (**Flatworms**) JG

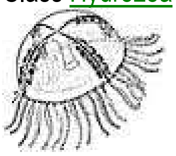
Soft-elongated body flattened from top to bottom; no individual segments; digestive track with only one opening which functions both as the mouth and anus; mouth usually on the bottom side positioned about one-fifth to the length of the body; sides of the body constricted towards the front forming a head that is somewhat triangular shaped; two eyespots situated on top of the head gives the animal a cross-eyed appearance. Most families can withstand high nutrient and organic enrichment, but some are very sensitive to toxics. Crawler, burrower; Collector, gatherer; (**H**); VS-L (**S**/**F**)

Class Spongilla (**Freshwater sponge**)



Sponges are delicate in structure, growing as encrusting or branching masses; usually appear greenish because of the algae that live on them. Freshwater sponges may attain a volume of more than 2,500 cubic centimeters (150 cubic inches). The larva of the Spongilla fly lives as a parasite on freshwater sponges. (**U**); S-L (**S**)

Class Hydrozoa (**Freshwater jellyfish**)



Usually small and bell-shaped; tentacles of varying lengths protrude from the margins of the velum; is often translucent with a whitish or greenish tinge. Swimmer, clinger; Collector, predator; (**U**); VS-S (**S**)

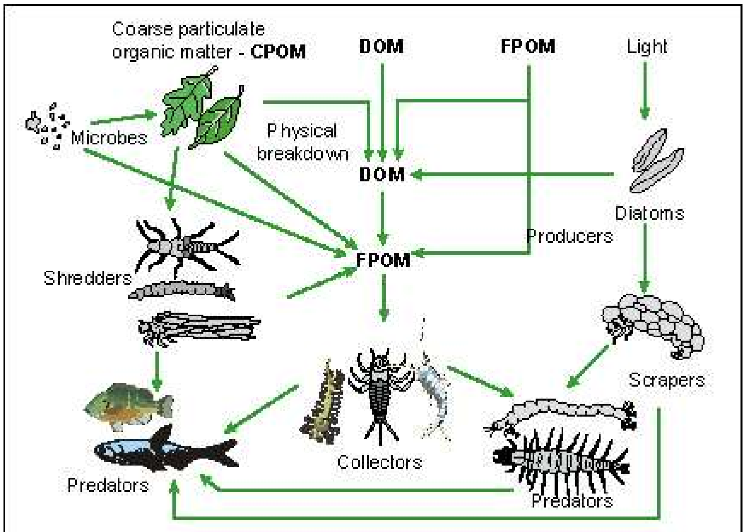


Figure: A simplified view of a food web in a woodland stream. Energy inputs include fallen leaves, subsequently colonized by microbes; small autotrophs, primarily diatoms; and DOM and FPOM, originating from external sources and upstream.

Functional feeding groups are a classification approach that is based on behavioral mechanisms of food acquisition rather than taxonomic group. The same general behavioral mechanisms in different species can result in the ingestion of a wide range of food items. The benefit of this method is that instead of hundreds of different taxa to be studied, a small number of groups of organisms can be studied collectively based on the way they function and process energy in the stream ecosystem. Individuals are categorized based on their mechanisms for obtaining food and the particle size of the food, and not specifically on what they are eating.

This method of analysis avoids the relatively non- informative necessity to classify the majority of aquatic insect taxa as omnivores and it establishes linkages to basic aquatic food resource categories, coarse particulate organic matter (CPOM), and fine particulate organic matter (FPOM), which require different adaptations for their exploitation. The major functional feeding groups are: scrapers (grazers), which consume algae and associated material; shredders, which consume leaf litter or other CPOM, including wood; collectors (gatherers), which collect FPOM from the stream bottom; filterers, which collect FPOM from the water column using a variety of filters; and predators, which feed on other consumers. A sixth category includes species that do not fit neatly into the other categories such as parasites. It is important to keep in mind, however, that many kinds of invertebrates use a variety of food acquisition methods.

Glossary: The glossary below includes select terminology used here, and it also includes other terms often associated with the description of aquatic invertebrates. The source of most of these definitions is the publications listed in the reference list below.

1. Abdomen: The third main division of the body; behind the head and thorax.
2. Anterior: In front (before).
3. Apical: Near or pertaining to the end of any structure, part of the structure that is farthest from the body.
4. Basal: Pertaining to the end of any structure that is nearest to the body.
5. Burrower: Animal that uses a variety of structures designed for moving and burrowing into sand and silt, or building tubes within loose substrate.
6. Carapace: The hardened part of some arthropods that spreads like a shield over several segments of the head and thorax.
7. Caudal filament: Threadlike projection at the end of the abdomen, like a tail.
8. Clinger: Animal that uses claws or hooks to cling to the surfaces or rocks, plants or other hard surfaces and often moves slowly along these surfaces.
9. Concentric: A growth pattern on the opercula of some gastropods, marked by a series of circles that lie entirely within each other; compare multi-spiral and pauci-spiral.
10. Crawler: An animal, whose main means of locomotion is moving slowly along the bottom, usually has some type of hooks, claws or specially designed feet to help hold them to surfaces.
11. Detritus: Disintegrated or broken up mineral or organic material.

12. Dextral: The curvature of a gastropod shell where the opening is visible on the right when the spire is pointed up.
13. Distal: Near or toward the free end of any appendage; that part farthest from the body.
14. Dorsal: Pertaining to, or situated on the back or top, especially of the thorax and abdomen.
15. Elytra: Hardened shell-like mesothoraic wings of adult beetles (Coleoptera).
16. Femur: The leg section between the tibia and coxa of Arthropoda, comparable to an upper arm or thigh.
17. Flagellum: A small fingerlike or whip-like projection.
18. Gill: Any structure especially adapted for the exchange of dissolved gases between animal and a surrounding liquid.
19. Glossae: A lobe or lobes front and center on the labium; in Plecoptera, the lobes are between the paraglossae.
20. Hemimetabolism: incomplete metamorphosis.
21. Holometabolism: complete metamorphosis.
22. Labium: Lower mouthpart of an arthropod, like a jaw or lip.
23. Labrum: Upper mouthpart of an arthropod consisting of a single usually hinged plate above the mandibles.
24. Lateral: Feature or marking located on the side of a body or other structure.
25. Ligula: Forming the ventral wall of an arthropod's oral cavity.
26. Lobe: A rounded projection or protuberance.
27. Mandibles: The first pair of jaws in insects.
28. Maxillae: The second pair of jaws in insects.
29. Multi spiral: A growth pattern on the opercula of some gastropods marked by several turns from the center to the edge.
30. Operculum: A lid or covering structure, like a door to an opening.
31. Palpal lobes: The grasping pinchers at the end of the Odonata lower jaw.
32. Pauci-spiral: A growth pattern on the opercula of some gastropods marked by few turns from the center to the edge.
33. Periphyton: Algae and associated organisms that live attached to underwater surfaces.
34. Posterior: Behind; opposite of anterior.
35. Proleg: Any projection appendage that serves for support locomotion or attachment.
36. Prothorax: The first thoracic segment closest to the head.
37. Rostrum: A beak or beak-like mouthpart.
38. Sclerite: A hardened area of an insect body wall, usually surrounded by softer membranes.
39. Seta (pl. setae): Hair like projection.
40. Sinistral: The curvature of a gastropod shell where the opening is seen on the left when the spire is pointed up.

References and recommended reading

1. B.K. Jessup, A. Markowitz and J.B. Stribling. 2002. Family-Level Key to Stream Invertebrates of Maryland and Surrounding Areas. Tetra Tech, Inc.
2. Bouchard, R.W. 2004. Guide to Aquatic Invertebrates of the Upper Midwest: Identification Manual for Students, Citizen Scientist's and Professionals. University of Minnesota
3. BVWM Biology Web - Using aquatic invertebrates to determine water quality
4. Cacapon Institute's – Benthic portal
5. Creek Connections – Aquatic Invertebrate Fact sheets. Allegheny College, PA
6. Introduction to aquatic invertebrate monitoring (Riverwatch)
7. Izaak Walton League of America. 2000. Field Guide to Aquatic Macroinvertebrates. J. Reese Voshell, Jr. 2002. A Guide to Common Freshwater Invertebrates of North America. McDonald and Woodward Publishing Company
8. J. Adams, M. Vaughan and S.H Black. 2004. The Xerces Society's CD-ROM: Stream Bugs as Biomonitors. Xerces Society.
9. McCafferty, W.P. 1981. Aquatic Entomology: The Fisherman's and Ecologist's Guide to Insects and their Relatives. Jones and Bartlett Publishers
10. R.W. Merritt and K.W. Cummins. 1996. An Introduction to the Aquatic Insects of North America 3rd Edition. Kendall/Hunt Publishing Company
11. Stroud Water Research Center's – Macroinvertebrate images and dichotomous key
12. University of Alberta, Department of Biological Sciences. 2007. Aquatic Invertebrates of Alberta Online Textbook
13. Valley City State University. Digital Key to Freshwater Invertebrates of North Dakota
14. Wilkes University Center for Environmental Quality – Macroinvertebrate metrics
15. WV Save Our Streams – Macroinvertebrate sub-sampling protocols

http://www.dep.wv.gov/WWE/getinvolved/sos/Documents/Macroinvertebrates/WVSOS_MacroinvertebrateGuide.htm

West Virginia Department of Environmental Protection

WV Save Our Streams Program
601 57th Street, SE
Charleston, WV 25304
http://www.dep.wv.gov/sos

February 2010