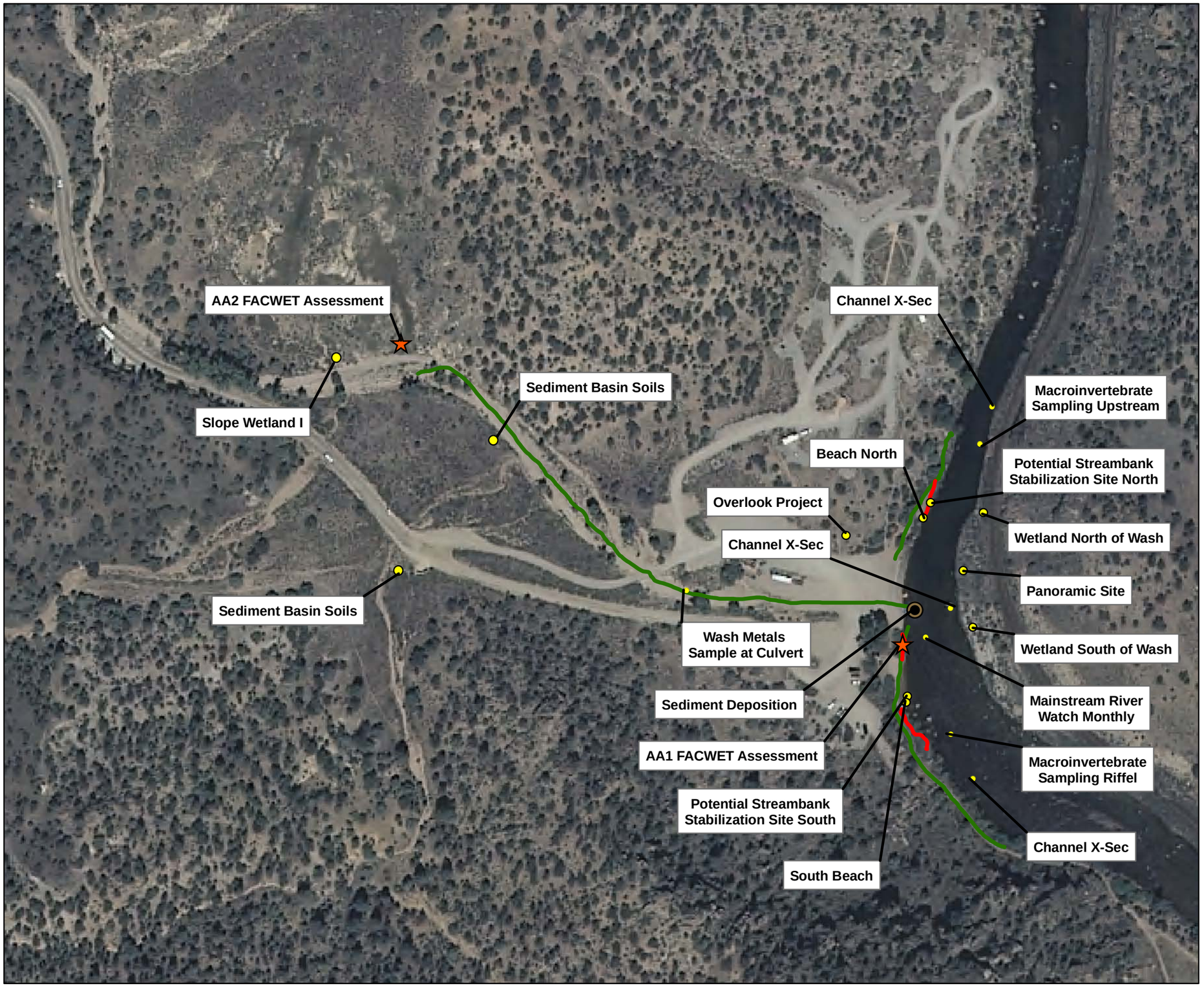
Hecla Jct. Stream Restoration & Sediment Reduction Project Baseline Monitoring Figure 1



Legend

- PFC Assessment Reach 2010
- Potential Stream Bank Restoration Site
- Sediment Deposition Extent 2010
- Photo Point
- FACWET Assessment 2010

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Feet

ICF INTERNATIONAL

COLORADO

Macroinvertebrate Collection and Physical Habitat Assessment Instructions

Overview

1. The primary objective for collecting macroinvertebrate data is to compile a species list over time and space to identify missing, additional and indicator species that might signify changes in community structure or function. One macroinvertebrate sample will be collected at a minimum of ONE station per group within a contract year. Your responsibility is for collection only. A Colorado Department of Public Health and Environment accepted taxonomist will complete identification to a species level.

2. RW staff will identify the station you are to sample and notify you if you are to sample this year via a sample calendar and send you supplies. Ten percent of participating groups will be chosen to provide a quality control sample.

3. A physical habitat and analysis must be completed with each macroinvertebrate sample. The habitat analysis describes the bug's residence and will record changes in aquatic environment over time. The macroinvertebrate and physical habitat data sheets must be submitted with each collection. A physical habitat analysis **can** be completed without a macroinvertebrate sample.

4. Your water quality sample should be collected at the same time/day the macroinvertebrate sample is collected. This tells us the "condition" of the river for the bugs at the time of collection. A water sample should include pH, Temperature, dissolved oxygen, alkalinity, hardness and both total/dissolved metals. If possible collect a nutrient sample as well (analyzed for total nitrogen, ammonia, total phosphorus, chloride, sulfate and total suspended solids).

5. Full instructions for both macroinvertebrate collection and physical habitat assessment are in this manual. A video /picture training is available that illustrates many of the steps and definitions (October 2006).

6. Each bug collection and/or habitat assessment is a sampling event, given a unique combination of station number, date and time. If water quality samples are collected at the same time, all these samples will have the same sample identifier.

7. Ship macroinvertebrates, data sheets and chain of custody within two weeks after collection. This will help to insure we can have the bugs identified prior to the end of each contract/school year.

Equipment for Macro Collection

Equipment provided by River Watch:

- A modified D-net (18" x 8"), The net is a 500-micron mesh net.
- Two forceps to pick organisms from net
- A 600-micron sieve (#36)
- One small brush
- Two 0.5 to 1.0 liter containers with alcohol preservative four jars if you are to collect a QA/QC sample

Additional equipment provided by you:

- A clean sample bucket (**not** your River Watch bucket)
- A squirt bottle (can be any water bottle with a squirt nozzle)
- A timing device that can time 60 seconds (a second hand on a watch)
- Waders
- A ruler to measure substrate
- A broom, pole or pipe with inch and foot marks on it to measure depths
- A tape measure (can be marked string or twine)
- Rubber gloves (optional) and magnifying glasses (optional)
- A large **white** enamel or plastic tray

Field Preparation Overview

1. Retrieve blank data sheets and complete the information above Part 1. Check all appropriate boxes. If you have been chosen to collect a QA/QC sample, check that box also. Be sure to check box for either Rocky or Sandy Substrate.

2. Using a permanent marker, label each macroinvertebrate sample bottle with river name, station name, station number, time and date.

3. Gather gear from list above.

4. If you are collecting water quality samples, prepare to do so BEFORE any macroinvertebrate sampling as this method involves disturbing the substrate and could contaminate a surface water sample.

Choosing and Recording Your Sampling Site

1. The first step is to determine if your segment is one classified as rocky or sandy. This delineation is based on whether your stream or river is covered by sand/silt greater than 50% (sandy bottomed) or less than 50% and dominated by hard bottom substrate (rocky).

2. Once you have made this identification, determine the general area to collect the sample by surveying a stream reach. Identify and measure a 200 foot segment. For ROCKY substrate samples, look for a segment that you can kick in two fast and two slow riffles. Riffles are the shallow fast moving sections of the river, not the slow deep pool areas (see the Rocky Substrate Collection section below for further discussion). For SANDY substrate sampling, look for a segment you can sample in multiple habitats (i.e. submerged vegetation, large woody debris, vegetated banks, water column and substrate) for 4 full minutes.

    - If possible, you want to be at least 100 feet upstream from any road or bridge structure and away from any major tributaries, discharges or return flows.
    - Choose reaches with habitats that are representative of the entire stream (i.e. riffles that looks like all the other riffles in the area).
    - You may have to walk around the entire are of your 200 foot segment to find habitats to sample, they do not need to be right next to each other.

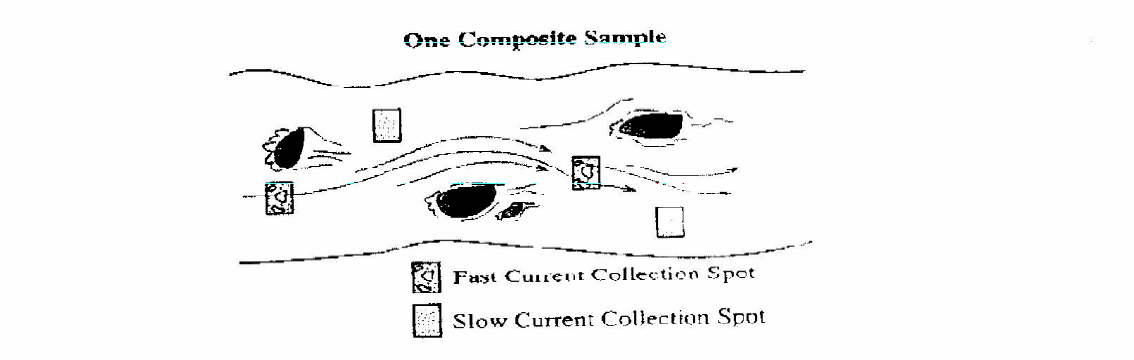
3. On part 1 of the data sheet, draw a map of the 200 foot section, scanning 100 feet above and below the area sampled, including the riparian zone. Draw boulders, snags, riffles, pools, dams, pipes, ditches, tributaries, bridges, wetlands, riprap and any landmarks that help identify your spot. ROCKY substrate folks: draw a square for each kick sample and the number of the kick (1-4) inside the square of where you will be sampling. SANDY substrate folks include riparian and instream vegetation, sandbars, etc.

4. On part 2 of the data sheets, there is a location for recording the longitudinal profile of the stream reach. Record this information (see data sheet section below).

Rocky Substrate Collection

A team approach here can be very effective, if all teams understand their role prior to arriving in the field. Assign one team to collect water quality samples, one to draw the map, one to collect bugs, one to time and record bug collectors information, one to conduct physical habitat assessment, and one to conduct longitudinal profile.

1. Determine four specific sites (two fast riffles and two slow riffles) where you will collect a kick net sample. Your sample is a composite of four separate kick sub-samples collected into the net. You must identify four locations where you will collect these sub-samples. With minimum of disturbance to the stream substrate, find two riffle areas where the water is flowing fast (1.5-2.5 feet per second) and two riffle areas that are flowing slower (0.5 to 1.5 feet per second), but still flowing. Use the floating device, timing and tape measure to estimate flows if you need too. See diagram below:



2. Approach the most downstream riffle spot for the first kick. Visualize an area on the stream bottom that is equivalent to about a 5.5 x3 feet (or 1 x 1 .7 meter) square area. Another way to measure this kick area is to lay the net down and make a mental map of the area that roughly covers from the length of the handle to the width of the net. This will be the kick area.

3. Place the net in the riffle making sure the net is on the stream bottom and if possible water does not flow over the top of the net. It is best if you can see water flowing through the net. Eddies, dead flow areas or areas water is flowing back upstream behind large rocks will not work as flowing water is needed to carry the bugs into your net as you disturb the substrate. Once your kick area defined, the net is set and the water is flowing through the net, you are ready to conduct your first kick.

4. Conduct kick #1:

    a. One person will hold the net open downstream.
    b. Second person will kick and disturb from upstream to down.
    c. A third person will time for 60 seconds.
    d. A fourth person the recorder.
    e. Begin at the downstream end of your rectangle with the net close enough to your feet so that dislodged organisms will go into the net and not around it (not more than one foot away).
    f. The timer starts timing 60 seconds.

g. The kicker uses their toe and heels to disturb, dislodge, uproot the upper layer of substrate and dig into the river bottom sediment. Do not kick the larger substrate out of the way, larger rocks or debris (logs, vegetation, and trash) should be picked up and brushed while immediately upstream of the net, so bugs will flow into the net. The goal is to get all bugs no matter where they are in that rectangle to flow into the net. Smaller debris like twigs and leaves should be kicked into the net and examined later for clinging bugs.
h. Have the data recorder label this kick #1 and identify it as a fast or slow riffle collection.

5. Complete Part 3 of the substrate data sheet Substrate Composition for kick #1 for Rocky Substrate, columns 1 and 2. All of these questions focus on the kick area being sampled only. You will need to circle rocky or sandy substrate and record the total time spent sampling each individual kick area (if followed instructions it would be 60 seconds/ kick). There are 3 columns of information and two major rows. The first row addresses slow/fast riffle and average depth of water in the kick area, answer these.

6. Columns 1 and 2 address inorganic substrate composition and organic composition of the habitat sampled. Kickers and recorders will work together on this step. For column 1, inorganic component, use a ruler to measure various substrate sizes and the size guides on part 3 of the data sheet. KICKER estimates the percent of each substrate size and the RECORDER records the estimate in the appropriate shaded box. Check that total percent of inorganic material adds up to 100%. An example of this would be a kick area with 25% cobble, 50%pebble and 25%silt. For column 2 organic substrate components, estimate the percent of the total rectangle that is covered by various types of organic matter. This may not add up to 100%; it is dependant upon how much organic material is covering the substrate (i.e. the kick area described above containing 10% leaf litter).

7. Repeat steps 1 through 5 for kicks #2, #3 and #4. Raise the net out of the water between each kick so that no organisms are lost. Carry the net to each riffle location; do not remove the bugs in-between kicks. After all sites are sampled, process the sample as described below.

8. The same team of another team then completes Part 4 of the data sheet. Habitat terms and descriptions are provided in the data sheet instructions.

Sandy Substrate Collection

1. Determine specific areas you will kick or dip your net. Sandy substrate tends to shift and doesn't have the large interstitial space many bugs prefer. In this habitat, the bugs will be in the water just above the substrate, or in aquatic vegetation in the stream amongst debris or along the banks. Identify **ALL** potential habitat types (vegetated banks, submerged vegetation, snags/debris, water column or sandy substrate) in the segment and plan to sample each habitat minimizing deep dipping into the sand. You are really dip netting more than kick netting. Below is a description of each habitat type and discussion on how best to sample each type. Identify the variety of habitats along the 200 foot section and divide 4 minutes (i.e. 5 habitats/4 min = 48 seconds each) into those habitats. Remember you are compositing all collections into one sample. Move in a downstream to upstream direction with minimal or no wading if possible.

    - Snags and other woody debris: Fallen branches, washed out or inundated shrubs/trees and small logs, which have been submerged in the water for a long time (not just fallen), provide excellent colonization habitat. Accumulated woody material in pools (deeper slower water) is considered snag habitat. To sample this habitat you would jab into the snag (with the net) and kicking around the snag with a net held downstream.

    - Overhanging and Vegetated banks: Occur when lower banks are submerged and have roots and emergent plants associated with them. Submerged areas of undercut banks are good habitats. They are sampled in a fashion similar to snags by jabbing and disturbing the area upstream of the net. Bank habitat can be kicked first (with larger net) to dislodge organisms with net placed downstream to retrieve any bugs.

    - Aquatic submerged macrophytes (large plants): Seasonal in their occurrence and may not be a common feature of many streams, particularly those that are of high gradient. These plants live submerged in the water and bank and can be seen with the unaided eye. Collect sample from aquatic plants that are rooted on the bottom of the stream or in the bank, and are submerged in the water by drawing the net through the vegetation from the bottom to the surface of the water. In shallow water, sample by bumping or jabbing the net along the bottom in the rooted area, avoiding sediments when can.

    - Sand and other fine sediment: Usually the least productive macroinvertebrate habitat in streams; this habitat may be the most prevalent in some streams. Collect sample from banks with no vegetation or soft soil by bumping the net along the surface of the bottom rather than dragging the net through the soft substrates, this reduces the amount of debris in the sample.

2. Approach the most downstream location. Visualize an area in the habitat you have selected that is equivalent to about a 1 x 1 .7 meter area. Another way to measure this area is to lay the net down and make a mental map of the area that it covers from the length of the handle to the width of the net. This will be the area that you will probe, dip and collect your sample in. This is the area you have 60 seconds to kick, sweep and disturb. Note mental markers as to the beginning and end of this rectangle.

3. Conduct kick #1.

    a) One person operates the net
    b) Second person assists with larger debris and substrate
    c) A third person can time
    d) A fourth person fourth is needed to record
    e) Sample the 1X1.7.m area using the techniques described above. Record the amount of time spent sampling the habitat in part 3, column 3 of the data sheet.
    f) Move to the next location; be careful to not lose organisms in switching habitats or dipping into the water. Repeat for all habitats selected.

4. Complete Part 3 of substrate data sheet Substrate Composition kick/habitat #1 for Sandy Substrate, column 3 (See data sheet section).

5. Repeat steps 1 through 4 for all remaining kicks. Raise the net all together out of the water between each kick so that no organisms are lost from the net. Carry the net to the next riffle location. Be careful not to lose any bugs when placing net in the stream. After all sites are sampled, process the sample as described below.

6. The same team or another team can complete Part 1, which is a diagram illustrating where the samples were collected. If the space is too small include your own sheet.

7. The same team or another team can complete Part 2, which is a longitudinal profile of a representative segment transect wet water width. Over time this will illustrate channel movement.

8. The same team of another team then completes Part 4 of the data sheet. Habitat terms and descriptions are provided in the data sheet instructions.

Sample Processing

The goal for processing is to get into the sample jar all the macroinvertebrates within the kick net, but with as little water and large debris as possible. This allows the preservative to work on the organisms effectively. If the sample is too watery, the preservative is diluted and organisms will become mushy and difficult to identify. If you process (wash and scrub) large material correctly, the lab can focus on identification bugs versus processing material. Small bunches of organic material such as algal mats need to be left in the sample.

1. Once the sample is collected and composite from all sample habitats or locations, carry the net to the shore. Fill the bucket a 1/2 to 2/3 full of stream water. Gather the sample material into one corner of the net. Grab the corner of the net from the bottom outside, holding the clump in your hand(s) and turn the net inside out into the clean sample bucket. Knock or wash any obvious macroinvertebrates, debris, algae clumps or masses into the bucket. Rinse the net from the OUTSIDE into the bucket if necessary. Examine the net closely for organisms that may want to stay behind. Pluck these organisms off with forceps and place directly into the sample jar with half the alcohol.
2. Look in bucket for large rocks or debris you can handle, bare twigs or leaves (not algae masses). Pick them up one at a time. Hold them over the sieve and look for organisms. Rinse the object with squirt bottle over the sieve; pluck the organisms off with forceps and place in the sample jar. Do not rinse over the alcohol filled sample jar.

3. Separate the organisms from the debris by "swirling" the sample in the bucket. Add more water if you need to and really swirl! The lighter organisms and debris will rise to the top of the water and the heavier sediment will not. Look for bugs to float to the top. Pour off the top water and floating material into the sieve, leaving the sand and gravel in the bucket.

4. Repeat the swirling until lighter material and bugs no longer rise to the top. Swirling needs to be aggressive enough to dislodge clinging organisms. This will take a MINIMUM of 15 swirls; maybe 20 to make sure all organisms are dislodged. Use more water if you need to. Limit scraping or any movement that would smash the bugs. Pick up a handful of gravel or substrate and look closely to see if it moves or you can see any bugs. If so, swirl again.

5. If you have algal masses, place them on the sieve and let as much water as possible drain out of the mass. Do not smash the mass as you will smash the bugs. Water from these masses will dilute the alcohol. The bugs in these masses are hard to see with the naked eye and require further sorting. When drained, place mass into sample jar.

6. After the last swirl and all floating bugs have been GENTLY placed into the preserved sample jar with forceps, pour the contents of the bucket onto the sieve in ***manageable*** batches. Spread the material and look for bugs. Then place sieve material onto a white tray looking for organisms one last time. Pluck bugs from sieve or tray and place in preservative. If you are collecting a QA/QC sample, place any remaining algae mats or clumps of debris in a second sample jar, draining as much water as possible. If you are not collecting a QA/QC sample, dump thoroughly processed debris from the pan. Repeat in batches until all debris has been processed.

7. If you have been selected to collect a QA/QC sample, you will have received 4 jars partially filled with alcohol. Process the sample as described below placing all your picked bugs in one jar. All processed debris throughout the process (leaves, sticks, rocks, sand and twigs) is placed in the other jar rather than being disposed of. This debris will be processed for bugs you might miss thus serving as a QA sample. Check the QA sample line on the macroinvertebrate label whereas your "normal" bug sample will have a similar label without the QA line checked.

8. Once all debris from the net is processed, rinse the net, sieve and pan thoroughly in the river, until no debris is visible. It is best to let net dry as soon as possible to avoid mold growth.

9. Label your sample and any QA sample you may have accordingly with a magic marker on the outside of the jar. Include sample #, station #, date and time. Place the other label inside the macroinvertebrate samples. The inside label is smaller and requests collector's name. These labels are located on page 22 of this sampling protocol.

10. **The evening of sampling, carefully decant off the first half the alcohol and then pour in rest of alcohol. If you have collected a QA/QC sample, decant that one also and replace with fresh alcohol.** Decanting will lessen the amount of water in the bottle that would hasten the degeneration of the bugs. Place sample label(s) from Step 2 of Laboratory Preparation in jar(s) and cap snugly.

11. Ship or deliver the sample within **two** weeks of collection. Include all data sheets and chain of custody (there is a spot to label "bugs", also downloadable on our website). Complete a Field Data sheet to go with the macroinvertebrate and physical habitat data sheets. Include water quality data if it was collected (preferred). Keep a copy of data sheets. A macroinvertebrate sample, with or without a chemical sample is a sampling event.

Macroinvertebrate Collection Data Sheet Instructions

Top Section of the Data Sheet:

- Organization/school name, river and station name
- Date and time
- Check box for Rocky or Sandy Substrate
- Sample Method, circle Modified D-net or describe other if another River Watch approved method was use.
- Circle the number of "kicks" performed. This should total four for rocky substrate.

Part 1 of the Data Sheet

Part 1a

Draw a picture of the reach in which you sampled. You want to diagram from a birds eye view of the 200 foot segment and 5 to 10 feet of bank on each side for the segment you have chosen. To orient the diagram, pretend you are a bird looking down at your site. Look upstream and identify left and right bank, circle the appropriate bank on the drawing, on the top of the box circle left or right bank and do the same at the bottom of the box. If this space is too small to draw in, provide your own drawing and write in this space: "See enclosed drawing".

1b Circle the direction the flow is going in the diagram.

1c Sketch the stream banks and major objects such as boulders, debris, pools, dams, tributaries, ditches, pipes, riprap, etc. Label items you feel need labeling to understand.

1d Draw a square resembling each kick or sample area and put a number in the box represent which kick it is.

1e Describe where the station is relative to your water quality station IF this is not that same station.

Part 2: Average Depth Profile of Representative Sample Transect

What this seeks to identify is a cross sectional measurement from wet water width (waters edge to water's edge) that consists of a series of depth measurements. Using a marked rod (PVC pipe, broom handle) that is marked off in feet and inches, record the depth of the stream at increments of every one foot. If you need more than 35 spaces, use the back of the data sheet or an additional piece of paper. This measurement should be taken in an area representative of where you will be collecting your macro invertebrate sample. As recording this data may disturb the very habitat you are sampling, this measurement should be taken after the macro invertebrate sampling has been completed. Over time this data will illustrate channel movement within that reach.

Note: You will need to measure bank full width as well. Both bank full and wet water measurements can be done as part of this step and the bank full measurement can be recorded in **Part 4, Section F**. Measure bank full width by noting the area from the end of the high water mark on one bank, to the edge of the high water mark on the opposite bank.

Part 3: Habitat Description for Rocky and Sandy Substrate

This is a microphysical habitat description of each rectangular kick area that was sampled. RECORDER and KICKER need to work together on this. You will be recording organic and inorganic substrate type and composition, riffle speed (for rocky habitats) and average depth of the kick area. Columns 1 address inorganic substrate composition of the **rocky** habitat sampled, and column 2 addresses organic composition of the habitat sampled **be it rocky or sandy**. Column 3 address inorganic substrate composition of the habitat sampled for **sandy** habitats.

Column 1 Inorganic Habitat Composition for Rocky Substrate: This column address inorganic substrate composition of the habitat sampled. You will first need to circle whether the area sampled was a fast or slow riffle. You need to quantitatively describe what the stream substrate is comprised of. To do this, use a ruler to measure various substrate sizes. There is a range of size that correlates to how the substrate is classified (be it boulder, gravel, sand etc.) in column 1. Once this is determined, estimate the amount of each specific type of substrate that is represented in your kick area. Check that total percent of inorganic material adds up to 100%. An example of this would be a kick area with 25% cobble, 50%pebble and 25%silt.

Column 2-Organic Habitat Composition for Rocky/Sandy Substrate: This column addresses the organic substrate components of the kick area. Different organic components are described below. This value may not add up to 100% as it is a **% of the amount of organic in the entire kick area**. The value may be anywhere from 0% to 100%, it is dependant upon how much organic material is covering the substrate (i.e. the kick area described above containing 10% detritus/leaf litter).

Detritus is any sticks, leaves, floating plant material or algae. Basically anything organic you could pick up with your hands is coarse organic material (CPOM). Look for fine slippery algae on large rocks, this is Periphyton. This is food for the bugs.
Muck-mud is very fine, yucky, black, slimy material and will sometimes have and odors of sulfur like in a wetland soil, this is fine organic material (FPOM). This is another form of food for bugs.
Marl is gray, finely broken shell like fragments. It is unlikely you will find this in most Colorado streams.

Record the average depth of the water sampled, row 1 column 2.

Part 3 Inorganic Habitat Composition for Sandy Substrate: This column address inorganic substrate composition of the habitat sampled. In Column 3 you will need to describe each habitat that was sampled. Recorder can identify the habitat type as vegetated bank, submerged vegetation, snags/debris, water column or sand/substrate. Record the amount of time that each habitat was sampled and note any worthy characteristics of each site like size, community structure (one or multiple species of plant) etc. Also record the % composition of each habitat type for each area sampled. An example: If you sampled 4 separate areas and you sampled in this 1st area that had 90% vegetated banks and 10% substrate, record as such.

Part 4: Entire Segment Physical Habitat Description

This section evaluates Habitat Features, Watershed Features, Localized Erosion, Riparian Vegetation, Aquatic Vegetation and Instream Features **for the entire 200 foot segment you have mapped in Part 1**. There are 6 sections in this description and definitions to all terms found in these sections are provided below (as discussed in the USEPA Rapid Bioassessment+6 Protocols). Observe the entire reach and be as objective and consistent as possible. Don't for get recorder's signature and the date at the bottom.

Section A Habitat Feature Descriptions

This item describes all the different habitat types that could be sampled for macroinvertebrates. In rocky substrate streams we are only sampling one habitat type, the riffle, which in theory is cobble. In sandy substrate streams we are sampling several habitats—snags, debris, vegetated banks and sand. It is helpful to know how much of the other habitat types are present for future sampling, especially if the riffles are not that numerous or large. Estimate the percentage within the 200' reach the percent cobble, snags, vegetated banks and sand present.

- Snags and other woody debris are fallen branches, washed out or inundated shrubs/trees and small logs, which have been submerged in the water for a long time (not just fallen), provide excellent colonization habitat. Accumulated woody material in pools (deeper slower water) is considered snag habitat.

- Overhanging and Vegetated banks occur when lower banks are submerged and have roots and emergent plants associated with them. Submerged areas of undercut banks are good habitats.

- Aquatic submerged macrophytes (large plants) are seasonal in their occurrence and may not be a common feature of many streams, particularly those that are high gradient. These plants live submerged in the water and bank and can be seen with the unaided eye.

- Sand and other fine sediment are usually the least productive macroinvertebrate habitat in streams; this habitat may be the most prevalent in some streams.

Section B Watershed Features of Overall Area

This is a description of the land adjacent to *both* left and right stream banks. These are always determined by looking upstream from kick site. For each bank check the predominant (top 1 or 2 most prevalent types) land uses 300 ft adjacent to the reach.

- Forests: trees, pine or deciduous in a fairly undisturbed tract

- Field/Pastures: fields of grass, left undisturbed or used for grazing even if irrigated, not cropland, etc.

- Irrigated: irrigated land for any crop

- RR/hwy: a railroad or highway or road

- Dense housing: like a suburban or urban area
- Sparse housing:10 acres or more per house/unit
- Commercial: commingled buildings or business as on a main street in town
- Industrial: refinery, brewing company, power plants etc.
- Other: anything that doesn't fit above (please describe)

Section C Localized Erosion of Overall Area

This section evaluates local erosion and potential sources of sediment in the stream reach. A river carries a certain amount of sediment either in the water column (suspended) or moving along the bottom (bed load). How much sediment and what size particles in the sediment load are a function of the stream volume (discharge) and velocity (flow). A river is designed to carry sediment from it's headwaters to the mouth. Sediment in unnatural amounts, from sources outside the flood plain or delivered at an unnatural rate becomes a pollutant, smothering habitat and causing other effects. Natural and accelerated erosion of land causes sediment to end up in the river. Many sources of sediment to a river come from a diffuse non point source like an unchecked construction site versus a direct source like a pipe. You are evaluating evidence of diffuse or non-point sources of sediment and the amount of visual erosion. Some things to look while assessing localized erosion include looking for extensive reaches of non-vegetated banks, traveled foot/tire paths next to, down to or even crossing the stream, culverts/bridges, any bare dirt proximate to the bank, etc. Asses what percent of the stream reach you are surveying has any of this evidence using the guide below. There are three parameters to note.

1. % Bare Bank Soil: make an estimate of the area of bare soil (80%, 10%, etc.) in the riparian zone that is not bound by plants and their root structures or covered in concrete or rocks. These bare areas can be caused by wildlife, stock or people access, roads and crossings, clearing or undercut banks.

2. Erosion Amount: estimates the amount of erosion that is present on the banks within the reach. Choose the category that best describers your estimate.

3. Bank Movement and Stability: due to lack of vegetation, roots or other mechanisms to keep the soil and bank from entering the water, the banks may have become unstable and show signs of degradation. Choose the category that best describers your estimate of bank stability or degradation.

Section D Riparian Vegetation of Overall Area

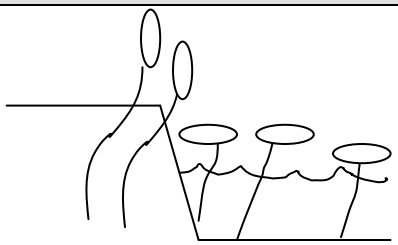
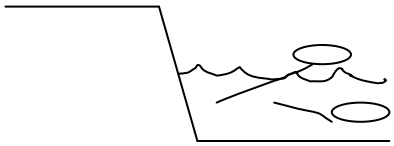
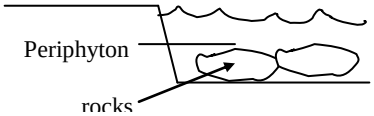
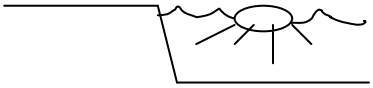
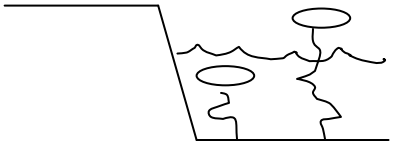
This section describes the vegetation type on each bank. Riparian vegetation along the bank and transitioning into the upland ecosystem provide food and habitat for a variety of animals in some aspect of their life cycles. It can also provide a migration corridor, soil stability, water quality filter and buffer for the water body.

In this section, please describe the predominate vegetation type for each bank. You don't need to know the species, just the type (tree, grass, shrub/bush or forbs) but if you do you know species, please document. If there is no vegetation along the riparian zone, note *other* and please describe in the space provided (i.e. pavement, dirt, etc.) Evaluate for both left and right banks. Estimate the width of each bank riparian zone; the width from the water to another vegetation type is the riparian zone width, record on data sheet.

Section E Instream Aquatic Vegetation of Overall Area

This section focuses on vegetation *in the stream* only, not on the banks. These species need water, need to be submerged or associated water for some part of their life cycle. Typical examples range from cattails, liverworts, blue, green and brown algae and periphyton. These also vary in that they are macrophytes, vascular and non-vascular and angiosperms. Aquatic vegetation is an important component of water bodies because they can provide food, oxygen and habitats for aquatic animals, supply food and habitat for birds, stabilize banks and beds, and take from the water some of the potential pollutants in runoff. Factors that affect the type and distribution of water plants include climate, flow, velocity, light, temperature and water quality. Look in the water and estimate the dominant vegetation type in the entire 200 foot stream reach using the guide below. Then estimate the percentage within the reach that is populated with vegetation.

- Rooted emergent or submerging is an aquatic plant rooted in wetland, lake or river substrate. Usually grow at the water's edge or in shallow water. Most of the plant is above water. These include common plants such as rushes and some grasses. Some are broad-leafed and some are have narrow leaves.
- Rooted floating is a rooted aquatic plant that "came" loose and is floating in the stream.
- Submerged / Floating leaf varieties These have root systems attached to the bottom of the water body and in some cases have leaves that float on the surface and / or flowers parts that emerge from the water. These include plants such as water lilies, milfoils, watercress and ribbon weed.
- Attached Algae is like periphyton, the most common in Rocky Mountains Stream.
- Free floating is a plant that prefers to grow as it floats. They are not attached at any time and occur in relatively still water. The whole plant is floating with roots suspended in the water. These include common plants like azolla and exotic plants like water hyacinth.

	Visual		
Rooted emergent or submerging		Rooted Floating	
Attached Algae		Free Floating	
Submerged / Floating Leafed			

Section F Instream Features of Overall Area

This section evaluates instream features that provide quality habitat for macroinvertebrates. This is to be evaluated for the entire 200' stream reach.

Canopy Cover: Trees and large shrubs provide shade and minimize temperature changes in the stream. It also provides food and habitat for emerged macroinvertebrates and food for fish as the insects fall into the river. Look up and down the stream, if the tree canopy covers the entire width of the open water, the canopy cover would be 100%. If any coverage occurs, estimate how much of the cover is generated from both the right and left banks and record

Stream Reach Description: Pools (slow deep water), riffles (fast shallow water), or runs (long, deep, slow, gliding pool) are the descriptors used to identify how the stream is moving through space. Identify the percentage of each type in your reach and note.

Wet Water Width: The width of the water in the stream, from one wet edge to the opposite wet edge. If you have a measuring tape and can wade in the stream, measure this.

Bank Full Water Width: The highest level that water could reach without flowing out of the banks onto adjacent land. Usually you can tell this by old wet watermarks or vegetation changes. If you have a measuring tape and can wade the stream measure this.

Average Stream Depth: The estimate or measurement of stream depth in several places along a transect the in the stream. A PVC pipe or stick (bug net) with measured tick marks works for this. You can get this from averaging results of Part 2.

Channelized: When you look within the stream reach up and downstream, or can you see the stream meander (bend) at all? Answer this with a yes or no answer. If you cannot see any meanders, then stream may be channelized due to a road, railroad or other reasons.

Macroinvertebrate Collection Data Sheets

(Sample Site Information and Depth Profile - Page 1 of 5)

Station Name_____ Date of sample ___/___/___ Time ___:___

River_____ Station number:_____

Group (School)_____ RW Net / Other_____

# of kicks: 1 2 3 4

Substrate type **Rocky** ☐ **Sandy** ☐ **QA Sample Collected** ☐

Part 1

a. Draw a picture of the sample site (from bank to bank, 200 feet above/below sample area):

Left bank or **right** bank-looking upstream (circle one)

Left bank or **right** bank looking upstream (circle one)

b. Flow direction on diagram → OR ← (circle one)

c. Draw in stream attributes such as riffle, dams, fallen trees, pools, roads, tributaries, bridges, wetlands, riprap, pipes, and other landmarks to identify reach, Label appropriately, include larger sheet if desire.

d. Draw a square representing bug sample location and a number in each square representing each 1 of 4 kicks.

e. If not at water quality station, describe (distance from water quality station, etc.):

_____

_____

Part 2

Average Depth Profile of representative sample transect

Select a spot typical of the sample area. Measure depths at 1-step intervals from bank to bank across the river and record below: (UNIT=_____) Place transect on diagram above.

1 _____	6_____	11_____	16_____	21_____	26_____	31_____
2_____	7_____	12_____	17_____	22_____	27_____	32_____
3_____	8_____	13_____	18_____	23_____	28_____	33_____
4_____	9_____	14_____	19_____	24_____	29_____	34_____
5_____	10_____	15_____	20_____	25_____	30_____	35_____

Substrate Composition for Each Kick (page 2 of 5)

Part 3 Substrate Composition kick #1:

Circle one: **Rocky (Columns 1 and 2)** **Sandy (Column 3 and maybe 2)**

Total Time Sampled **(circle one)** Rocky or Sandy Substrate habitat _____ seconds

1			2			3			
Fast riffle / Slow Riffle (circle one) 1.5-2.5 ft/sec 0.5-1.5 ft/sec			Average Depth of rectangle = _____ in _____(unit) If sampling in water						
Inorganic Substrate Components (Rocky=should add to 100%)			**Organic Substrate Components (Rocky and MAYBE Sandy Substrates)**			**Habitat sampled (Sandy =should add to 100%)**			
Substrate Type	Diameter	% Composition in sample	Substrate Type	Describe Characteristics	% Composition in sample	Habitat Type	Time sampled ___seconds	Describe Characteristics	% Composition of sample
Bedrock			Detritus	Sticks, wood, coarse plant material, CPOM		Vegetated Banks			
Boulder	>256mm, 10inches					Submerged Vegetation			
Cobble	64-256mm, 2.5-10”		Muck-Mud	Black, very fine organic material, FPOM		Snags/Debris			
Gravel	2-64 mm, 0.1-2.5“					Water Column			
Sand	0.06-2 mm, Gritty					Sand/Subs			
Silt	0.004-0.06mm		Marl	Grey, shell fragments					
Clay	<0.004, slick/slimy								
	TOTAL %					**TOTAL TIME**		**TOTAL %**	
Rocky Only			**Rocky and MAYBE Sandy (if substrate part of sample habitat)**			**Sandy Only**			

Substrate Composition for Each Kick (page 3 of 5)

Part 3 Substrate Composition kick #2:

Circle one: **Rocky (Columns 1 and 2)** **Sandy (Column 3 and maybe 2)**

Total Time Sampled **(circle one)** Rocky or Sandy Substrate habitat _____ seconds

1			2			3			
Fast riffle / Slow Riffle (circle one) 1.5-2.5 ft/sec 0.5-1.5 ft/sec			Average Depth of rectangle = _____ in _____(unit) If sampling in water						
Inorganic Substrate Components (Rocky=should add to 100%)			**Organic Substrate Components (Rocky and MAYBE Sandy Substrates)**			**Habitat sampled (Sandy =should add to 100%)**			
Substrate Type	Diameter	% Composition in sample	Substrate Type	Describe Characteristics	% Composition in sample	Habitat Type	Time sampled ___seconds	Describe Characteristics	% Composition of sample
Bedrock			Detritus	Sticks, wood, coarse plant material, CPOM		Vegetated Banks			
Boulder	>256mm, 10inches					Submerged Vegetation			
Cobble	64-256mm, 2.5-10”		Muck-Mud	Black, very fine organic material, FPOM		Snags/Debris			
Gravel	2-64 mm, 0.1-2.5“					Water Column			
Sand	0.06-2 mm, Gritty					Sand/Subs			
Silt	0.004-0.06mm		Marl	Grey, shell fragments					
Clay	<0.004, slick/slimy								
	TOTAL %					**TOTAL TIME**		**TOTAL %**	

Rocky Only **Rocky and MAYBE Sandy (if substrate part of sample habitat)** **Sandy Only**

Substrate Composition for Each Kick (page 4 of 5)

Part 3 Substrate Composition kick #3:

Circle one: **Rocky (Columns 1 and 2)** **Sandy (Column 3 and maybe 2)**

Total Time Sampled **(circle one)** Rocky or Sandy Substrate habitat _____ seconds

1			2			3			
Fast riffle / Slow Riffle (circle one) 1.5-2.5 ft/sec 0.5-1.5 ft/sec			Average Depth of rectangle = _____ in _____(unit) If sampling in water						
Inorganic Substrate Components (Rocky=should add to 100%)			**Organic Substrate Components (Rocky and MAYBE Sandy Substrates)**			**Habitat sampled (Sandy =should add to 100%)**			
Substrate Type	Diameter	% Composition in sample	Substrate Type	Describe Characteristics	% Composition in sample	Habitat Type	Time sampled ___seconds	Describe Characteristics	% Composition of sample
Bedrock			Detritus	Sticks, wood, coarse plant material, CPOM		Vegetated Banks			
Boulder	>256mm, 10inches					Submerged Vegetation			
Cobble	64-256mm, 2.5-10”		Muck-Mud	Black, very fine organic material, FPOM		Snags/Debris			
Gravel	2-64 mm, 0.1-2.5“					Water Column			
Sand	0.06-2 mm, Gritty					Sand/Subs			
Silt	0.004-0.06mm		Marl	Grey, shell fragments					
Clay	<0.004, slick/slimy								
	TOTAL %					**TOTAL TIME**		**TOTAL %**	

Rocky Only **Rocky and MAYBE Sandy (if substrate part of sample habitat)** **Sandy Only**

Substrate Composition for Each Kick (page 5 of 5)

Part 3 Substrate Composition kick #4:

Circle one: **Rocky (Columns 1 and 2)** **Sandy (Column 3 and maybe 2)**

Total Time Sampled **(circle one)** Rocky or Sandy Substrate habitat _____ seconds

1			2			3			
Fast riffle / Slow Riffle (circle one) 1.5-2.5 ft/sec 0.5-1.5 ft/sec			Average Depth of rectangle = _____ in _____(unit) If sampling in water						
Inorganic Substrate Components (Rocky=should add to 100%)			**Organic Substrate Components (Rocky and MAYBE Sandy Substrates)**			**Habitat sampled (Sandy =should add to 100%)**			
Substrate Type	Diameter	% Composition in sample	Substrate Type	Describe Characteristics	% Composition in sample	Habitat Type	Time sampled ___seconds	Describe Characteristics	% Composition of sample
Bedrock			Detritus	Sticks, wood, coarse plant material, CPOM		Vegetated Banks			
Boulder	>256mm, 10inches					Submerged Vegetation			
Cobble	64-256mm, 2.5-10”		Muck-Mud	Black, very fine organic material, FPOM		Snags/Debris			
Gravel	2-64 mm, 0.1-2.5“					Water Column			
Sand	0.06-2 mm, Gritty					Sand/Subs			
Silt	0.004-0.06mm		Marl	Grey, shell fragments					
Clay	<0.004, slick/slimy								
	TOTAL %					**TOTAL TIME**		**TOTAL %**	
Rocky Only			**Rocky and MAYBE Sandy (if substrate part of sample habitat)**			**Sandy Only**			

Stream Reach Physical Habitat

Part 4 Overall area physical habitat

Habitat Features	**A**	Indicate % of each habitat type in reach (50ft above/below sample): ☐ Cobble____% ☐ Snags_____% ☐ Vegetated Banks_____% ☐ Sand_____%			
Watershed Features	**B**	Predominant Surrounding Land Use **R**ight **B**ank: ☐ Forest ☐ Dense housing ☐ Field/pasture ☐ Sparse housing ☐ Irrigated ☐ Commercial ☐ RR/hwy ☐ Industrial ☐ Other_____ **L**eft **B**ank: ☐ Forest ☐ Dense housing ☐ Field/pasture ☐ Sparse housing ☐ Irrigated ☐ Commercial ☐ RR/hwy ☐ Industrial ☐ Other_____			
Localized Erosion	**C**	% Bare Bank Soil ☐ 80-100% ☐ 10-39% ☐ 40-79% ☐ 0-9%	Erosion Amount ☐ extensive ☐ localized/occ ☐ some evidence ☐ no evidence	Bank Movement ☐ bank failures ☐ slight ☐ mod collapses ☐ none	
Riparian Vegetation	**D**	Indicate the dominant riparian zone vegetation type and record dominant species: **R**ight **B**ank: ☐ Trees ☐ shrubs ☐ grasses ☐ herbaceous ☐ other_____ ☐ dominant species_____ **L**eft **B**ank: ☐ Trees ☐ shrubs ☐ grasses ☐ herbaceous ☐ other_____ ☐ dominant species_____			Riparian Zone Right Bank ____ ft Wide Left Bank ____ ft Wide
Aquatic Vegetation	**E**	Indicate the dominant vegetation type instream (not on banks): ☐ Rooted emergent ☐ Submerging floating leaf ☐ Rooted floating ☐ Free Floating ☐ Attached Algae		Portion of reach with aquatic Vegetation: _____%	
Instream Features	**F**	Canopy Cover: _____% of stream bank covered with Canopy/other	% of Reach Stream: ☐ Riffle_____% ☐ Pool_____% ☐☐ Run_____%	Estimated Wet Water Width _____Ft Estimated Bank Full Width _____Ft Estimated average stream depth _____Ft Channelized ☐ YES ☐ NO	

Macroinvertebrate Sample Labels For Inside Sample

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

River Name ______________________________
Station Name ______________________________
Station Number ___________________________
Date ______________ Time ______________
Sample Collector ________________________

Preserved with 95% ethanol / method 1

Optional Macroinvertebrate Collection

Method for equipment limitation (no required RW equipment) or data objectives other than RW

If your data objectives differ from the RW required macroinvertebrate protocol, for example simple education is the only objective or if you do not have a RW set of bug equipment, you can conduct the following macroinvertebrate studies.

Collection Designs

Ask your monitoring objective, what questions are you trying to answer with this bug collection, what are you trying to learn? For example:

If you intend to demonstrate the influence of an impoundment on benthic diversity, then collect benthic macroinvertebrates above and below the impounded reach.

If you are interested in testing the River Continuum Concept, then collect benthic macroinvertebrates along the headwaters, midreach, and lower reach of a river system.

You could also compare what the Sequential Comparison Index (SCI), a qualitative index, indicates about water quality with the Water Quality Index.
Choose the time of year, station location and sample frequency that will best answer your questions.

Optional RW Macroinvertebrate Method Supplies

1. A three-foot-long net made of screen-door-mesh should be used. This provides a consistent mesh size since there is only one size mesh for screen door. You may use the metal or nylon version. Nylon is more "user-friendly." If you wish to capture a "different" size macroinvertebrate, make a second net with a mesh size less than 0.5 mm.

2. Place the net in the water riffle; not pool habitat. Depending on flow, start about 4-5 feet upstream from the net, kick and disturb the substrate (bottom of the river) moving downstream to the net. If debris is floating past net, start closer. The kicking should last

one minute for consistency. DO NOT sample if the river is deeper than 24 inches for safety reasons.

3. Pull the net out of the water in such a manner the bugs on the net are not swept downstream from the current. Put the net on the bank and pick all sizes and colors of bugs from the net for 30 person minutes—two people picking for 15 minutes equals 30 person minutes. This is also for consistency.

4. Complete the sequential index. If you are making a reference collection, store the bugs in 70 percent ethyl alcohol (ethanol) or everclear alcohol diluted with river water. Try not to use isopropyl alcohol, the bugs become rubbery over time.

5. As a rule of thumb, you should complete at least three "kicks" or "nets" per station (transect) in riffle habitat to collect a representative sample. The more you do the more representative your sample will be. If the stream is wide enough you can do three kicks across the river. If the river is not wide enough, do your kicks in an upstream fashion: kick one, move upstream; kick two, move upstream; and kick the third.

6. Macroinvertebrates vary with season. They emerge at different times of the year filling unique niches. Because of seasonal variation you should try to sample three times a year. Refer to the biological calendar for those times of year. The calendar considers the school schedule.

7. If you want to identify the macroinvertebrates further than the sequential index use the data sheets provided. These data sheets can be transferred to the computer.

8. Identification can be taken a step further by completing the trophic level (functional feeding analysis). If the bugs are identified past the family level the tropic level (functional feeding group) can be determined. Functional feeding group or trophic level refers to how a bug "captures" its food, for example shredding detritus, filtering the water, gathering detritus, or preying on other bugs. This information can be related to the River Continuum Concept, physical habitat, and the riparian zone.

There is a percent functional feeding group summary table included for you.

9. The percent composition of each family can also be computed and provide valuable information. Does one family dominate? Are the three sensitive families (mayfly, stonefly, and caddisfly) well represented? If not, why? Etc.

It is suggested that you use *An Introduction to the Aquatic Insects of North America* by Merritt and Cummins [1] for identification. It teaches the students how to use a key and has interesting information about bugs (life cycles, habitats, unique features, etc.) in addition to the identification key.

[1] Merritt R.W. and K.W. Cummins, 1984. *An Introduction To The Aquatic Insects of North America,* Second Edition. Kendall/Hunt Pub. Comp. Dubuque, Iowa. (Available in soft or hard back).

Sequential Comparison Index

"The Sequential Comparison Index is a simple method for non-biologists to estimate relative differences in biological diversity (Cairns, et al. 1968)." This Index, like other diversity measures, assumes that reduced diversity is an indication of pollution. Reduced diversity may also be related to land uses such as impoundments and urbanization, or to stream order. When estimating relative differences in benthic diversity it is important to match approximately such physical variables as flow, bottom substrate, and amount of shading.

a. Empty the bugs from the net, or picked from a net into a white pan.

b. Ice cube trays help a lot here, but are not necessary, a white trash bag demarcated will work. Randomly pick an organism, place it in the first ice cube tray or delineated square. Pick the next organisam and compare each organism with the one preceding it. If the second organism is like the first organism, place it with it, if it is different place it in another cube or square. Pick the third organism and compare with the previous two, if it is like either of them, place it there or place it in a different cube or square, repeat until all organisms are placed.

c. Calculate the diversity index (DI):

DI = number of runs (number of ice cube squares = 8 = 0.57
number of organisms (total individuals) 14

Ind Org	x	x	x	y	z	z	a	b	f	f	h	h	r	r
Run			1	2		3	4	5		6		7		8

d. The greater this diversity index value, the greater the diversity and the better the water quality. The SCI runs from 0 to 1.0, with a value of 1 representing the greatest diversity. (General Water Quality Rating: 0-0.30= Poor; 0.31-0.60= Fair; 0.61-1.0 = Good.)

e. Each group in class can calculate a diversity index and these may be averaged for a particular station. The higher the ability the more sophisticated the lumping should be. For example HS students can lump species of mayflies, stoneflies, caddisflies whereas elementary students might just lump major families.

f. !5 organisms is a good number to work with, groups may continue until they have "processed" 50 organisms if they have 50 organisms in their sample and if there is enough time or complete the entire pan. For comparison purposes the same collection method, time of kick and number of organisms needs to be consistent between samples.

Suggested Equipment:

a. White enameled pans, white trash bags or steel vegetable dishes.
b. Quart Mason® type jars for collecting live material to be placed in aquariums.
c. Turkey basters (used to pick up small aquatic organisms).
d. Forceps
e. Meter stick (used for depth measurements)
f. Wire cloth or hardware cloth can be fashioned into a hand screen and various items or even window screen stretched between two pieces of wood.
g. Vials for collecting aquatic organisms.
h. Dissecting scopes.
I. Vegetable brush or soft toothbrush.
j. Small paint brush.
k. Buckets.
l. Hip waders (not essential, but a good idea for some).
m. Nets, dip or 3 x 3 foot screen (can be homemade).

Sugaring for Burrowing Aquatic Organisms

If you do a method that employs a different net that you need to empty into a bucket,, sugaring can help find the organisms. So can swirling a large bucket 20 times or so as the RW macroinvertebrate method employs. To determine the concentration and distribution of aquatic organisms in stream sediments, a technique that could be used is called, "sugaring".

Samples of stream sediments are placed in separate pans and water is added to cover the material by several inches. Saturating the water with sugar changes the density of the water and the lighter aquatic organisms float to the surface. The organisms can then be identified into general categories and returned to the stream.

If specific identification is needed, a preservation technique is to add formalin to the water covering the sediments in the pan (10 percent formalin solution) to replace the body fluids of the aquatic organisms with formalin. After several hours pour off all liquids in the pan and replace with water. The lighter preserved organisms will float to the surface of the water as the sediments are stirred and can be removed and identified.

Sequential Comparison Index for Macroinvertebrates

Station Name_____ Date of survey ___/___/___

River_____ School_____

Station Description _____

Total number of samples_____ Page _____ of _____

1. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____	2. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____
3. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____	4. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____
5. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____	6. Sample____ of____ A. Number of runs____ B. Number of organisms____ Diversity Index (DI) = A/B DI=____/____=_____

Station Average DI = Sum of DIs divided by number of DIs = _____
(This average is for pages _____ of _____, Samples _____ through _____)'

Data recorded by_____ Date recorded _____

Optional Macroinvertebrate Data Sheets

Introduction

Macroinvertebrate data collected from the required RW method (modified D kit net) can be further analyzed using these data sheets. The optional RW macroinvertebrate collection method (3 x 3 foot screen door net) can also use the following data sheets. Comparison from following data sheets require the same collection and laboratory procedures. These data sheets provide additional means of displaying, viewing and analyzing your bug data. The following is an overview of what is offered.

Macroinvertebrate Summary Data Sheet

Once a species lists is completed, the functional feeding group (trophic level) of each species or taxa (depending on how far you identified your collection) can be recorded on the Functional Feeding Group Analysis data sheet. You can determine each species functional feeding group from Merritt and Cummins[2](1984) identification book. This book is recommended but others may work as well. If you do not have this book and identify your collection further than order, notify Barb Horn at the DOW to send you the functional feeding descriptions from Merritt and Cummins. This book is available from Barb Horn to "check out."

The Functional Feeding Group Data Sheet provides a visual summary of the functional feeding group analysis. It is a table you complete based on your identification and research. Do the functional feeding groups you found, fit the River Continuum Concept for the stream reach they were collected? Do you have a dominance of predators? Are you in stream order 1-3 and have no shredders?

Species Composition Summary Data Sheet. This data sheet is a table that records percent composition for the major taxonomic groups. The data sheet table enclosed is an example, you can make your own list and percent composition just of the species you collected. Calculate your EPT index, the percent of may, stone and caddis flies.

[2]Merritt R.W. and K.W. Cummins, 1984. *An Introduction To The Aquatic Insects of North America,* Second Edition. Kendall/Hunt Pub. Comp. Dubuque, Iowa. (Available in soft or hard back).

Macroinvertebrate Functional Feeding Group Analysis

Station Name _____ Station Number _____

River _____ Date of survey ___/___/___

School _____

Station Description _____

Functional Feeding Group	Sample				
	1	2	3	4	5
Shredder					
Collector Grer					
Collector Filterer					
Scraper					
Predator					

Comments:_____

_____

_____

Data recorded by_____ Date recorded _____

Percent Composition Macroinvertebrates by Major Taxonomic Groups

Station Name_____ Station Number _____

River_____ Date of survey ___/___/___

Station Description_____

Taxa	Sample				
	1	2	3	4	5
Ephemeroptera (May fly)					
Plecoptera (Stone fly)					
Trichoptera (Caddis fly)					
Coleoptera (Beetles)					
Diptera (Crane flies)					
Chironomidae (Midges)					
Odonata (Dragonflies)					
Hemiptera (True bugs)					
Arachnida (Water mites)					
Turbellaria (Flatworm)					
Oligochaeta (Earthworm)					
Hirudinea (Leeches)					
Gastropoda (Snails)					

Comments_____

_____

Data recorded by_____ Date recorded _____

RIPARIAN AREA MANAGEMENT

Process for Assessing
Proper Functioning Condition

by

U.S. Department of the Interior
Bureau of Land Management
Proper Functioning Condition Work Group

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Process for Assessing Proper Functioning Condition

I. Introduction

The Bureau of Land Management (BLM) has responsibility for 269 million acres of public lands (USDI, 1992) that sustain a variety and abundance of resources. These resources are prized for their recreation, fish and wildlife, cultural, and historic values, as well as their economic values, and for such uses as livestock production, timber harvest, and mineral extraction. Riparian-wetland areas, though they comprise less than 9 percent of the total land base, are the most productive and highly prized resources found on BLM lands.

Federal policy defines wetlands as ***areas that are inundated or saturated by surface or ground water at a frequency and duration sufficient to support, and which, under normal circumstances do support, a prevalence of vegetation typically adapted for life in saturated soil conditions***. BLM Manual 1737, ***Riparian-Wetland Area Management,*** includes ***marshes, shallow swamps, lakeshores, bogs, muskegs, wet meadows, estuaries, and riparian areas as wetlands***.

BLM's manual further defines riparian areas as ***a form of wetland transition between permanently saturated wetlands and upland areas. These areas exhibit vegetation or physical characteristics reflective of permanent surface or subsurface water influence. Lands along, adjacent to, or contiguous with perennially and intermittently flowing rivers and streams, glacial potholes, and the shores of lakes and reservoirs with stable water levels are typical riparian areas. Excluded are such sites as ephemeral streams or washes that do not exhibit the presence of vegetation dependent upon free water in the soil***.

Riparian-wetland areas are grouped into two major categories: 1) lentic, which is standing water habitat such as lakes, ponds, seeps, bogs, and meadows, and 2) lotic, which is running water habitat such as rivers, streams, and springs.

A. Purpose

The Federal Land Policy and Management Act (FLPMA) of 1976 directs BLM to manage public lands in a manner that will provide for multiple use and at the same time protect natural resources for generations to come. In addition to FLPMA, numerous laws, regulations, policies, Executive orders, and Memorandums of Understanding (MOUs) direct BLM to manage its riparian-wetland areas for the benefit of the nation and its economy.

Under BLM's mandate of multiple-use management, a variety of activities such as livestock grazing, timber harvest, mineral extraction, recreation, and road and transportation corridor construction takes place on public lands. If not managed correctly, these activities can impact the quality of riparian-wetland areas.

In 1991, the BLM Director approved the *Riparian-Wetland Initiative for the 1990's*, which establishes national goals and objectives for managing riparian-wetland resources on public lands. One of the chief goals of this initiative is to restore and maintain riparian-wetland areas so that 75 percent or more are in proper functioning condition (PFC) by 1997. **The overall objective of this goal is to achieve an advanced ecological status, except where resource management objectives, including PFC, would require an earlier successional stage, thus providing the widest variety of vegetation and habitat diversity for wildlife, fish, and watershed protection**. This objective is important to remember because riparian-wetland areas will function properly long before they achieve an advanced ecological status. The *Riparian-Wetland Initiative for the 1990's* also includes a strategy to focus management on the entire watershed. Entire watershed condition is an important component in assessing whether a riparian-wetland area is functioning properly.

In the past, considerable effort has been expended to inventory, classify, restore, enhance, and protect riparian-wetland areas, but the effort has lacked consistency. The purpose of this document is to provide a thought process for assessing PFC for riparian-wetland areas on BLM-managed lands.

B. Approach

BLM depicts natural riparian-wetland areas as resources whose capability and potential is defined by the interaction of three components: 1) vegetation, 2) landform/soils, and 3) hydrology. A few resource specialists regard fish and wildlife as a fourth element because some wildlife species may alter a riparian-wetland area's capability and potential. However, most classifiers categorize fish and wildlife as a "user," but place wildlife species that can alter the capability and potential of a riparian-wetland site (i.e., beaver) as a special modifier under the hydrology component. BLM takes this approach in its inventory and classification system, Ecological Site Inventory (ESI).

Since natural riparian-wetland areas are characterized by the interactions of vegetation, soils, and hydrology, **the process of assessing whether a riparian-wetland area is functioning properly requires an interdisciplinary (ID) team**. The team should include specialists in vegetation, soils, and hydrology. A biologist also needs to be involved because of the high fish and wildlife values associated with riparian-wetland areas.

To initiate the process, in February 1992, the Director assembled an ID team of specialists to review existing Bureau definitions for PFC and to expand or develop new definitions as required. Appendix A provides the names of the specialists that were involved in this process. The ID team also developed a format for BLM to report functionality to Congress, which will include the tables in Appendix B.

C. Definitions

The terms introduced in BLM's definition of riparian-wetlands are generally understood by resource specialists. However, some confusion still exists with the term ephemeral stream. A stream is a general term for a body of flowing water. In hydrology the term is generally applied to water flowing in a natural channel as distinct from a canal. Streams in natural channels are classified as being perennial, intermittent or seasonal, or ephemeral and are defined as follows (Meinzer, 1923):

Perennial - A stream that flows continuously. Perennial streams are generally associated with a water table in the localities through which they flow.

Intermittent or seasonal - A stream that flows only at certain times of the year when it receives water from springs or from some surface source such as melting snow in mountainous areas.

Ephemeral - A stream that flows only in direct response to precipitation, and whose channel is at all times above the water table.

These terms refer to the continuity of streamflow in **time**; they were developed by the U.S. Geological Survey in the early 1920's, have a long history of use, and are the standard definitions used by BLM resource specialists. Confusion over the distinction between intermittent and ephemeral streams may be minimized by applying Meinzer's (1923) suggestion that the term "intermittent" be arbitrarily restricted to streams that flow continuously for periods of at least 30 days and the term "ephemeral" be arbitrarily restricted to streams that do not flow continuously for at least 30 days. Also, the intermittent stream is to be distinguished from an **interrupted** stream, which is a stream with discontinuities in **space**. Intermittent or seasonal streams usually have visible vegetation or physical characteristics reflective of permanent water influence; for example, the presence of cottonwood.

To understand how riparian-wetland areas operate and to implement proper management practices, thus ensuring an area is functioning properly, **the capability and potential of a riparian-wetland area must be understood**. Assessing functionality is based upon an area's capability and potential. For the purpose of this document, capability and potential are defined as follows:

Capability - The highest ecological status a riparian-wetland area can attain given political, social, or economical constraints. These constraints are often referred to as limiting factors.

Potential - The highest ecological status an area can attain given no political, social, or economical constraints; often referred to as the "potential natural community" (PNC).

In BLM's annual report to Congress, the following definitions are to be used when completing the table in Appendix B:

Proper Functioning Condition - Riparian-wetland areas are functioning properly when adequate vegetation, landform, or large woody debris is present to dissipate stream energy associated with high waterflows, thereby reducing erosion and improving water quality; filter sediment, capture bedload, and aid floodplain development; improve flood-water retention and ground-water recharge; develop root masses that stabilize streambanks against cutting action; develop diverse ponding and channel characteristics to provide the habitat and the water depth, duration, and temperature necessary for fish production, waterfowl breeding, and other uses; and support greater biodiversity. **The functioning condition of riparian-wetland areas is a result of interaction among geology, soil, water, and vegetation**.

Functional—At Risk - Riparian-wetland areas that are in functional condition but an existing soil, water, or vegetation attribute makes them susceptible to degradation.

Nonfunctional - Riparian-wetland areas that clearly are not providing adequate vegetation, landform, or large woody debris to dissipate stream energy associated with high flows and thus are not reducing erosion, improving water quality, etc., as listed above. The absence of certain physical attributes such as a floodplain where one should be are indicators of nonfunctioning conditions.

Unknown - Riparian-wetland areas that BLM lacks sufficient information on to make any form of determination.

II. Process

Most of the Bureau's riparian-wetland areas are found in Alaska and are considered functioning properly because they are in their natural state (USDI, 1991). This is not the case for BLM riparian-wetland areas in the 11 contiguous Western States, as well as small tracts in Alabama, Arkansas, Florida, Louisiana, Minnesota, Mississippi, and Oklahoma. Most of these riparian-wetland areas have been altered by human activities. However, the following process for determining whether an area is functioning properly is the same for Alaska as it is for the other states.

A. Review Existing Documents

To start the process, existing documents that provide a basis for assessing PFC should be reviewed. Technical Reference 1737-5, ***Riparian and Wetland Classification Review*** (Gebhardt et al., 1990), provides an excellent start as it reviews, in a like format, the more common procedures that are used to classify, inventory, and describe riparian-wetland areas. This document identifies ESI as being the most complete procedure because it provides a process for defining the capability of an area, its

potential, and how it functions. However, not all riparian-wetland areas will require the magnitude provided by ESI to assess functionality.

Technical Reference 1737-2, ***The Use of Aerial Photography to Inventory and Monitor Riparian Areas*** (Batson et al., 1987), Technical Reference 1737-3, ***Inventory and Monitoring of Riparian Areas*** (Myers, 1989), and Technical Reference 1737-7, ***Procedures for Ecological Site Inventory—With Special Reference to Riparian-Wetland Sites*** (Leonard et al., 1992), are three other documents that should be reviewed. These documents provide additional thought processes that will be useful in assessing functional status of riparian-wetland areas.

B. Analyze the Definition

Next, the definition of PFC must be analyzed. One way to do this is by breaking the definition down as follows:

> "Riparian-wetland areas are functioning properly when adequate vegetation, landform, or large woody debris is present to:
>
> 1) dissipate stream energy associated with high waterflows, thereby reducing erosion and improving water quality;
> 2) filter sediment, capture bedload, and aid floodplain development;
> 3) improve flood-water retention and ground-water recharge;
> 4) develop root masses that stabilize streambanks against cutting action;
> 5) develop diverse ponding and channel characteristics to provide the habitat and the water depth, duration, and temperature necessary for fish production, waterfowl breeding, and other uses;
> 6) and support greater biodiversity."

Riparian areas are functioning properly when there is adequate structure present to provide the listed benefits **applicable** to a particular area. The analysis must be based on the riparian area's capability and potential. If, for example, the system does not have the potential to support fish habitat, that criteria would not be used in the assessment.

C. Assess Functionality

1. Attributes and Processes

The third aspect of assessing PFC involves understanding the attributes and processes occurring in a riparian-wetland area. Table 1 provides a list of attributes and processes that may occur in any given riparian-wetland area. When assessing PFC, attributes and processes for the area being evaluated need to be identified.

To understand these processes, an example of an alluvial/nongraded valley-bottom type riparian area in both a functional and nonfunctional condition is provided in Figure 1 (Jensen, 1992). Using the Bureau's definitions for PFC, **State A** represents

Table 1. Attributes/Processes List *

Hydrogeomorphic
Ground-Water Discharge
Active Floodplain
Ground-Water Recharge
Floodplain Storage and Release
Flood Modification
Bankfull Width
Width/Depth Ratio
Sinuosity
Gradient
Stream Power
Hydraulic Controls
Bed Elevation
Vegetation
Community Types
Community Type Distribution
Surface Density
Canopy
Community Dynamics and Succession
Recruitment/Reproduction
Root Density
Survival
Erosion/Deposition
Bank Stability
Bed Stability (Bedload Transport Rate)
Depositional Features
Soils
Soil Type
Distribution of Aerobic/Anaerobic Soils
Capillarity
Annual Pattern of Soil Water States
Water Quality
Temperature
Salinity
Nutrients
Dissolved Oxygen
Sediment

* This list provides examples of various attributes/processes that may be present in a riparian area. By no means is it complete.

a high degree of bank stability, floodplain, and plant community development, and would be classified as PFC. The important attributes and processes present for **State A** are:

Hydrogeomorphic - Active floodplain, floodplain storage and release, flood modification, bankfull width, width/depth ratio, sinuosity, gradient, stream power, and hydraulic controls.

Vegetation - Community type (2 of 3), community type distribution (similar in the wet types), root density, canopy, community dynamics, recruitment/reproduction, and survival.

Erosion/Deposition - Bank stability.

Soil - Distribution of anaerobic soil, capillarity.

Water Quality - No change.

State B may be properly functioning or functional—at risk. It would be classified as functional if bank stabilizing vegetation is dominant along the reach and other factors such as soil disturbance are not evident. It is important to identify the species of vegetation present since they do vary in their ability to stabilize streambanks and filter sediment.

State B would be classified as at risk if bank stabilizing vegetation is not dominant (even though it may be in an improving trend from prior conditions), nondesirable species are present (e.g., Kentucky bluegrass), soil disturbance is evident (e.g., caved banks from livestock or vehicle use), or hydrologic factors such as degraded watershed conditions exist, increasing the probability of extreme flow events that would damage the reach. The following changes in attributes/processes are likely in **State B**:

Hydrogeomorphic - Bankfull width (increase), width/depth ratio (increase in width, no change in depth), active floodplain frequency (decrease).

Vegetation - Community types changed, community type distribution changed, root density, canopy, community dynamics, recruitment/reproduction, and survival.

Erosion/Deposition - Bank stability (decrease).

Soil - No change.

Water Quality - No significant change.

Figure 1. *Succession of states for alluvial/nongraded valley-bottom type.*

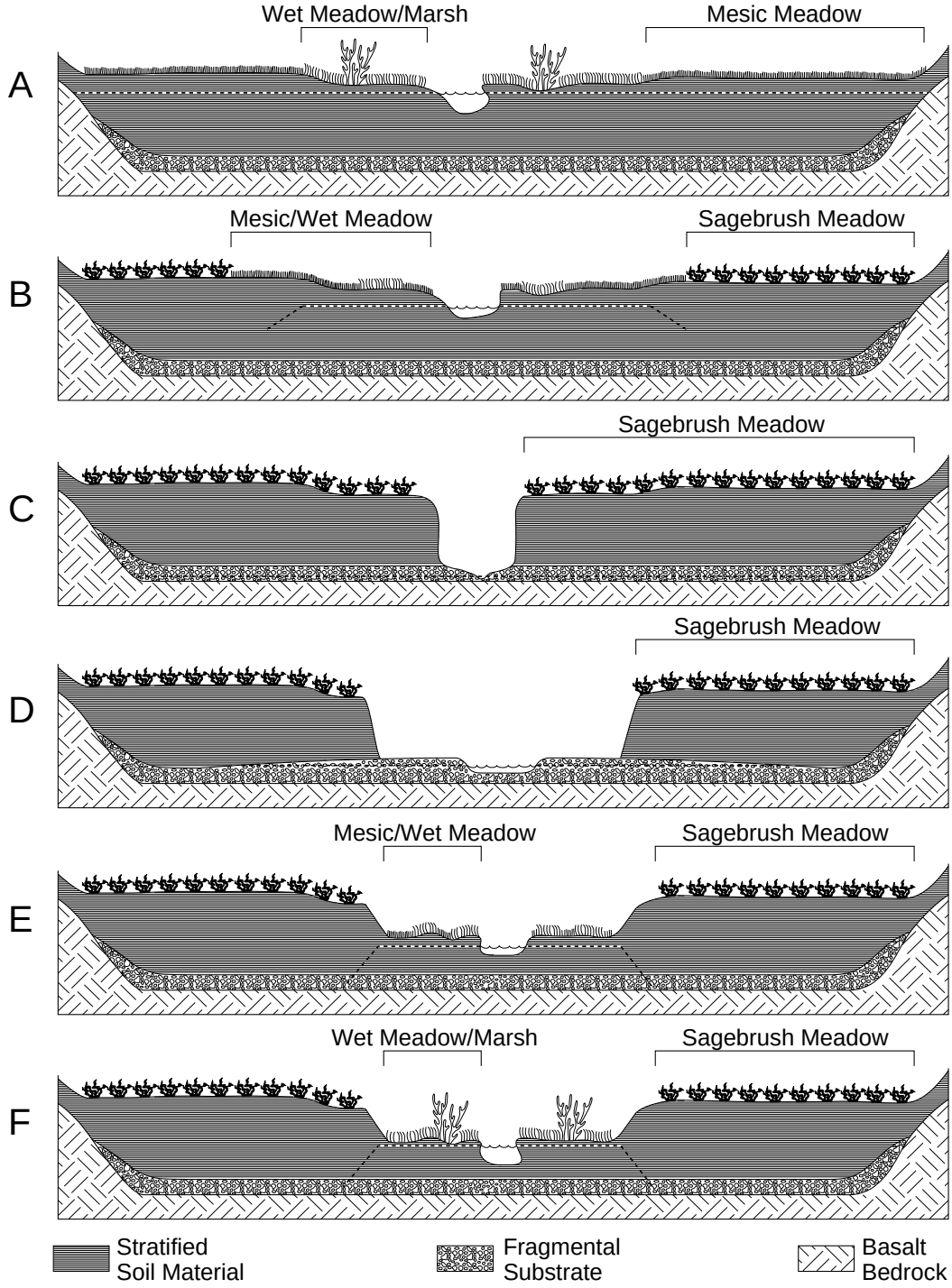

Figure 1. Succession of states for alluvial/nongraded valley-bottom type.

States C and D would be classified as nonfunctional conditions. **State C** represents incisement of the stream channel to a new base level. There is little or no bank stabilizing vegetation and no floodplain. Channel widening exhibited in **State D** must occur to restore floodplain development. Vegetation, if present, is often only temporary due to the large adjustment process occurring. The following changes in attributes/processes are likely in **States C and D**:

Hydrogeomorphic - Bankfull width (increase), width/depth (increase/increase), active floodplain frequency (decrease).

Vegetation - Riparian community types lost; community type distribution changed; root density, canopy, community dynamics, recruitment, reproduction, and survival (decrease).

Erosion/Deposition - Bank stability (decrease).

Soil - Well drained.

Water Quality - Temperature (increase), sediment (increase).

State E may again be classified as functional-at risk or functional depending on vegetation, soil, and hydrologic attributes. Establishment of the floodplain and bank stabilizing vegetation indicate reestablishment of functional conditions. However, stream segments in this state are usually at risk for the same reasons described for **State B**. Attributes and processes would revert back to those that appear in **State B**.

State F is classified as functioning properly even though the riparian area may not have achieved the greatest extent exhibited in **State A**. Banks are stabilized and exhibit channel geometry similar to **State A**. The floodplain has widened to the extent that confinement of peak flows is only occasional and aggrading processes are slowed because of the surface area available. The largest difference between **States A and F** occurs in size and extent of hydrologic influence, which regulates size and extent of the riparian area.

This alluvial/nongraded valley-bottom example is found in the Great Basin and represents only one of many types found on public lands. However, it is important to remember that there are other types and to understand that:

Riparian-wetland areas do have fundamental commonalities in how they function, but they also have their own unique attributes. Riparian-wetland areas can and do function quite differently. As a result, most areas need to be evaluated against their own capability and potential. Even for similar areas, human influence may have introduced component(s) that have changed the area's capability and potential. Assessments, to be correct, must consider these factors and the uniqueness of each system.

Appendix C contains examples of other kinds of riverine systems found on BLM managed lands (Jensen, 1992). The analogy used for Figure 1 can be applied to most of the examples found in Appendix C because differing channel types do have functional commonality. However, differing channel types may accommodate their own unique evolutionary processes. Information concerning the classification system used by Jensen can be found in BLM technical reference TR 1737-5 (Gebhardt et al., 1990).

2. Capability and Potential

Assessing functionality then involves determining a riparian-wetland area's capability and potential using an approach such as the following:

- Look for relic areas (exclosures, preserves, etc.).
- Seek out historic photos, survey notes, and/or documents that indicate historic condition.
- Search out species lists (animals & plants - historic & present).
- Determine species habitat needs (animals & plants) related to species that are/were present.
- Examine the soils and determine if they were saturated at one time and are now well drained?
- Examine the hydrology, establish cross sections if necessary to determine frequency and duration of flooding.
- Identify vegetation that currently exists. Are they the same species that occurred historically?
- Determine the entire watershed's general condition and identify its major landform(s).
- Look for limiting factors, both human-caused and natural, and determine if they can be corrected.

This approach forms the basis for initiating an inventory effort like ESI. For some areas, conducting an ESI effort will be the only way to assess an area's capability and potential.

Some riparian-wetland areas may be prevented from achieving their potential because of limiting factors such as human activities. Most of these limiting factors can be rectified through proper management. However, some limiting factors such as dams and transmountain diversions are not as easy to correct. The placement of dams and transmountain diversions can result in a riparian-wetland area's flow regime being altered, thus changing the area's capability. For example, cottonwood trees are maintained by periodic flooding, which creates point bars for seedling establishment. A dam or transmountain diversion that reduces or eliminates the potential for flooding may remove the potential for cottonwoods to remain in that area. PFC must be assessed in relationship to the area's capability.

3. Functioning Condition

When determining whether a riparian-wetland area is functioning properly, the condition of the entire watershed, including the uplands and tributary watershed system, is important. The entire watershed can influence the quality, abundance, and stability of downstream resources by controlling production of sediment and nutrients, influencing streamflow, and modifying the distribution of chemicals throughout the riparian-wetland area. Riparian-wetland health (functioning condition), an important component of watershed condition, refers to the ecological status of vegetation, geomorphic, and hydrologic development, along with the degree of structural integrity exhibited by the riparian-wetland area. A healthy riparian-wetland area is in dynamic equilibrium with the streamflow forces and channel aggradation/degradation processes producing change with vegetative, geomorphic, and structural resistance. In a healthy situation, the channel network adjusts in form and slope to handle increases in stormflow/snowmelt runoff with limited perturbation of channel and associated riparian-wetland plant communities.

Riparian-wetland areas can function properly before they achieve their Potential Plant Community (PPC) or Potential Natural Community (PNC). In fact, some would argue that riparian-wetland areas are always functioning properly, no matter what state they are in. From the perspective of fluvial geomorphology, it is true that the channel is constantly adjusting itself to the water and sediment load delivered to it from the watershed; however, BLM's definition goes beyond the processes of channel evolution and includes vegetation and biological attributes. The Bureau's definition does not mean PNC or optimal conditions for a particular species have to be achieved to be rated as functioning properly.

Figures 2 and 3 provide an example of the relationship between PFC and vegetation community succession for one area. Assuming succession continues uninterrupted (Step 1 to Step 2 in Figure 2), the channel will evolve through some predictable changes from bare ground to PNC (although not necessarily as linearly as depicted). The riparian-wetland area will progress through phases of not functioning, functioning—at risk, and properly functioning along with plant succession. In this example, PFC occurs at the mid-seral stage (Step 3). Figure 3 shows a stream cross section of each condition (A-E) displayed in Figure 2.

At various stages within this successional process, the stream can provide a variety of values for different uses (Step 4). In Figure 2, optimal conditions for grazing occur when forage is abundant and the area is stable and sustainable (mid-seral). Wildlife goals depend upon the species for which the area is being managed. If the riparian zone in Figure 2 is to provide habitat for shrub nesting birds, the optimum conditions would be from mid- to late seral. Trout habitat conditions would be optimum from mid-seral to late seral. The threshold for any goal is at least PFC because any rating below this would not be sustainable.

For some areas, PFC may occur from early seral to late seral. Desired plant community (DPC) would be determined based on management objectives through an interdisciplinary approach (Step 5). Figure 2 is an example of only one riparian-wetland area.

When rating functionality, it will be easy to categorize many riparian-wetland areas as PFC or nonfunctional. For others it will not be easy. Difficulty in rating PFC usually arises in identifying the thresholds that allow a riparian-wetland area to move from one category to another. To provide consistency in reporting PFC, BLM has established a standard checklist for field offices to initiate this process (Appendix D).

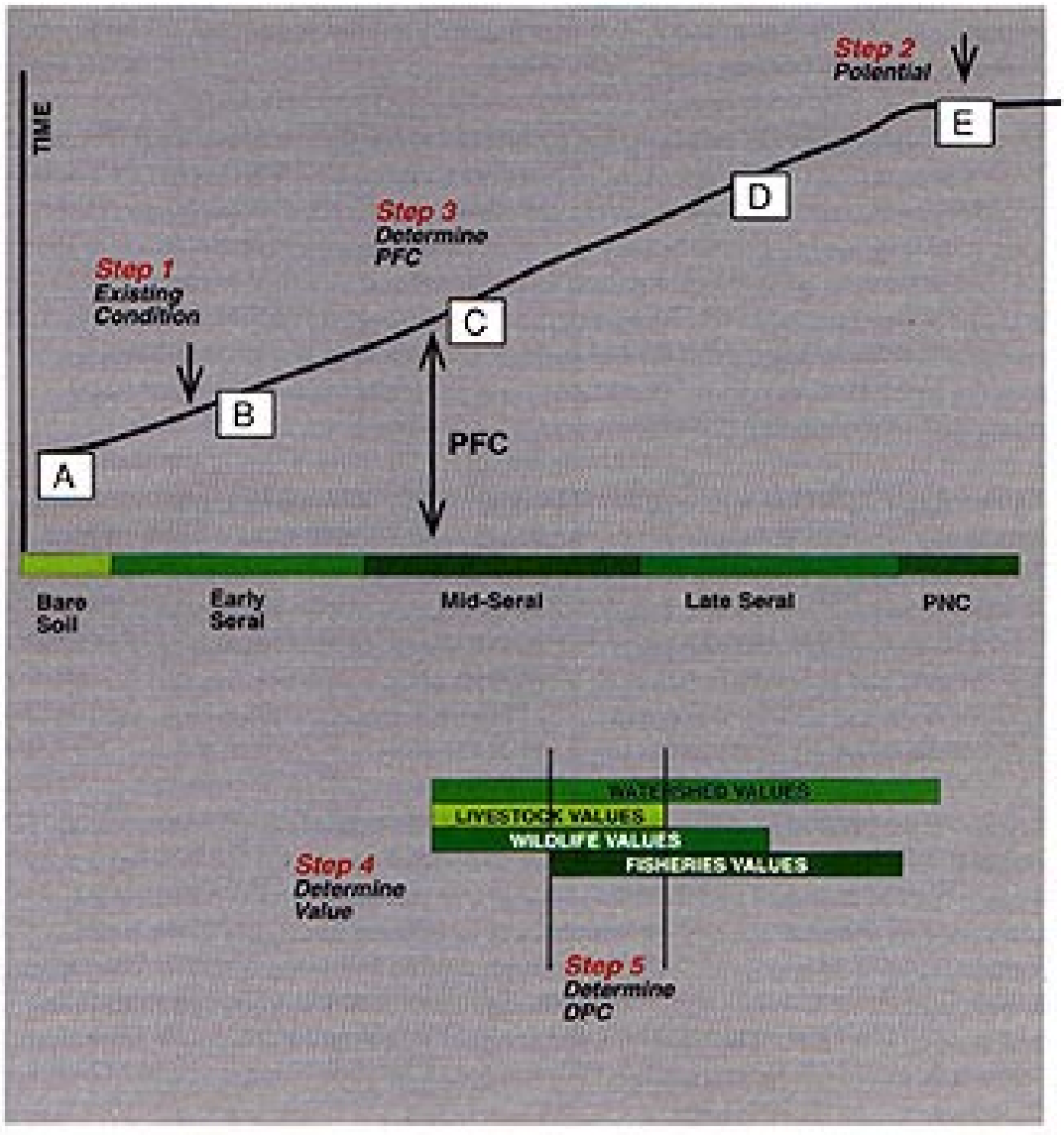

Figure 2. Succession for stream recovery.

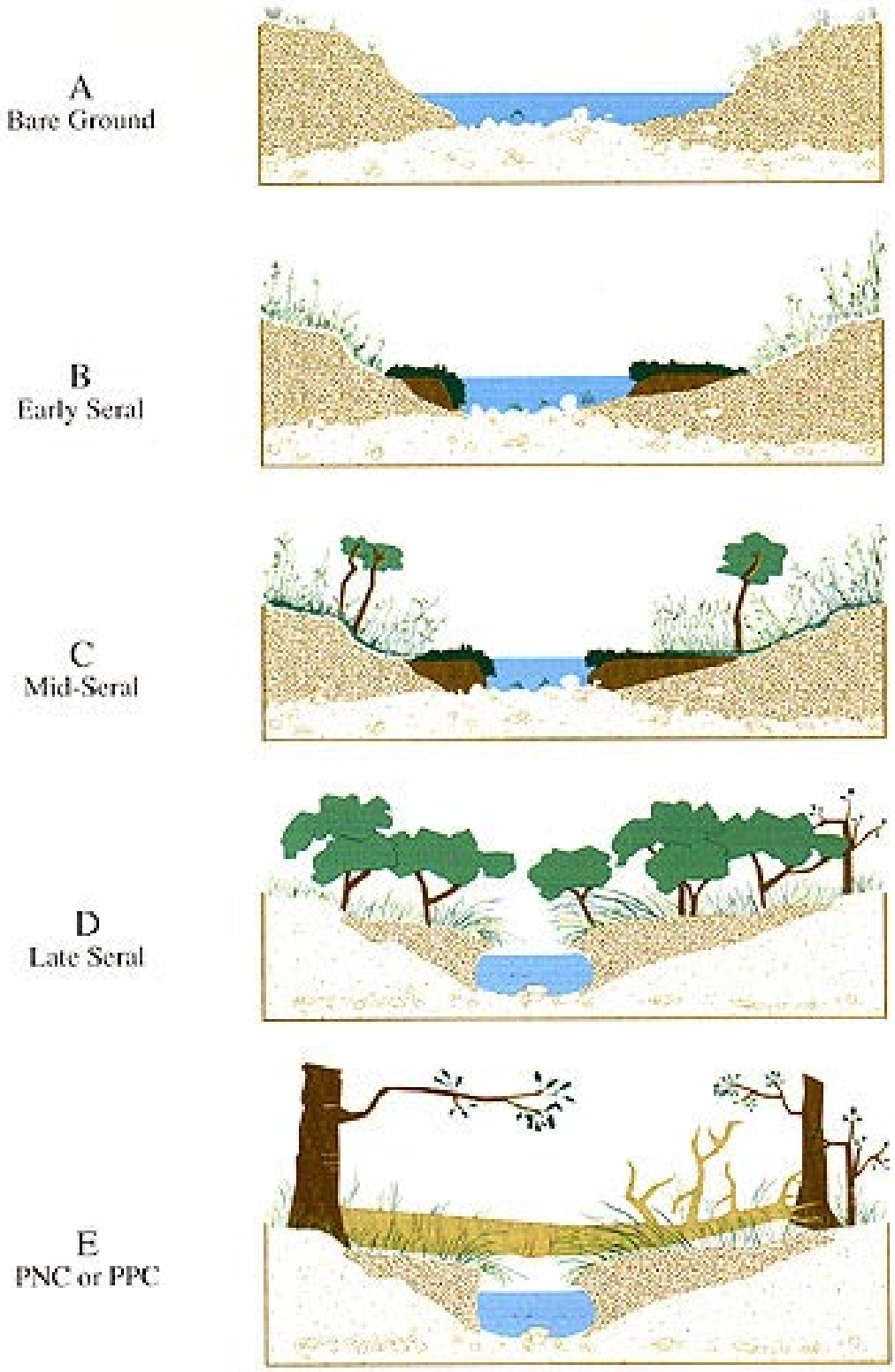

Figure 3. Stream cross sections.

BLM's checklist may not answer the question of functionality for all riparian-wetland areas. Some areas may require a more intensive inventory effort, like ESI. Elements can be added to BLM's standard checklist to address unique riparian-wetland attributes. To further assist field offices in assessing functionality, Appendix E provides examples of riparian-wetland areas and depicts the categories of PFC, functional—at risk, and nonfunctional.

The process described in this document has concentrated on lotic forms of riparian-wetland areas for two reasons: 1) they are the form of wetland BLM most frequently has to resolve conflicts on, and 2) inventory, classification, and monitoring efforts within and outside the Bureau have concentrated on this type of resource. However, the process would be the same for lentic forms of wetlands. Additional guidance will be developed for lentic wetlands as BLM gathers more information on them.

III. Instituting the Process

A. Planning

A logical manner to incorporate the information collected into a management plan is as follows (refer to Figure 2 in the Functioning Condition section):

Step 1 **Existing Condition** - Determine the existing riparian-wetland and watershed condition using BLM standard inventory methods.

Step 2 **Potential Condition** - Determine PNC by using relic areas, historic photos, etc. (ESI process).

Step 3 **PFC** - Determine the minimum conditions required for the area to function properly.

Step 4 **Resource Values** - Determine existing and potential resource values and the plant communities necessary to support these values.

Step 5 **Management Goals** - Negotiate specific objectives to reach management goals for the watershed, DPC, or Desired Future Condition.

Step 6 **Planned Actions** - Design management actions to achieve DPC.

Step 7 **Monitoring** - Design appropriate monitoring strategies to assess progress towards meeting management goals.

Step 8 **Flexibility** - Maintain management flexibility to accommodate change based upon monitoring results.

B. Management

For BLM to be successful in reaching its goal of having 75 percent of its riparian-wetland areas functioning properly by 1997, best management practices need to be set in motion. Successful management strategies address the entire watershed. Upland and riparian areas are interrelated and cannot be considered separately.

Two other documents can be helpful in assisting with this process: Technical Reference 1737-4, ***Grazing Management in Riparian Areas*** (Kinch, 1989), provides grazing management principles, concepts, and practices that have been effective in improving and maintaining desired conditions on riparian-wetland areas. For other forms of management such as recreation development, mining opportunities, timber practices, and watershed treatments, Technical Reference 1737-6, ***Management Techniques in Riparian Areas*** (Smith and Prichard, 1992), provides suggested management practices. With a change in management, most riparian-wetland areas can achieve PFC in a few years, but some will take years to achieve the identified DPC or advanced ecological status.

C. Monitoring

Management effectiveness can be assessed and progress towards meeting PFC can be documented through monitoring. Sites should be revisited periodically as part of the overall monitoring program. Areas rated at a single point in time can reflect short-term factors such as climatic conditions. Monitoring will reflect longer-term trends. Technical references such as TR 1737-3 (Myers, 1989) are tools that can be used to develop monitoring criteria.

IV. Summary

Riparian-wetland areas constitute an important resource on lands managed by BLM. BLM's goal is to have 75 percent of its riparian-wetlands functioning properly by 1997. This technical reference provides a thought process for assessing functioning condition.

The status of some riparian-wetland areas will be relatively easy to discern while the status of others will be less evident. Appendix D contains the minimum national standards that BLM field offices will use in making this assessment. For hard-to-discern areas, Ecological Site Inventory may be the only method to determine capability and potential and assess functionality. Using either method will require an interdisciplinary team to adequately address the complexities associated with riparian-wetland areas and to report their functioning condition.

Appendix B contains the forms for reporting functioning condition. Riparian areas are reported in four categories: proper functioning condition, functional—at risk, nonfunctional, and unknown. Areas with and without specific riparian management and objectives are reported separately. The Definitions section of this technical reference describes the meanings of these terms.

Trend is reported for areas that are identified as functional—at risk, and is a key consideration in interpreting the data. Areas identified as functional—at risk with a downward trend are often the highest management priority because a decline in resource values is apparent. Yet these areas often retain much of the resiliency associated with a functioning area. There is usually an opportunity to reverse this trend through changes in management. Functional—at risk areas with an upward trend are often a priority for monitoring efforts. These areas are improving but remain at risk. Monitoring these areas assures that upward trends continue.

Conversely, trend is not reported for areas that are nonfunctional. While these areas could theoretically still be in decline, most of the riparian values have already been lost. The presence of sufficient riparian-wetland attributes and processes to warrant a determination of trend usually results in a designation of functional—at risk.

It is common for an area in PFC to continue to have an upward trend. Many sites that are properly functioning must continue to improve to meet site-specific objectives. However a downward trend may put the area at risk. Once proper functioning condition is reached, trend relates to specific objectives. Therefore, it is not part of this data report.

The lack of specific information will place many riparian-wetland areas into the category of unknown. In order for BLM to make an adequate assessment of progress towards its goal, it is imperative that areas for which no data exists be evaluated and added to the data base. As information is acquired and resource values are identified, best management practices need to be set in motion. Successful management strategies have to address the entire watershed, as upland and riparian-wetland areas are interrelated and cannot be considered separately.

Examples provided in this document have concentrated on lotic riparian areas for two reasons: 1) they are the form of wetland BLM most frequently has to resolve conflicts on, and 2) inventory, classification, and monitoring efforts within and outside the Bureau have concentrated on this type of resource the most. However, the thought process for assessing functionality of lentic areas would be the same. In the future, a technical reference will be developed with more specific information for lentic wetlands.

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Glossary of Terms

Active Floodplain - The low-lying land surface adjacent to a stream and formed under the present flow regime. The active floodplain is inundated at least once or twice (on average) every 3 years.

Advanced Ecological Status - A community with a high coefficient of similarity to a defined or perceived PNC for an ecological site, usually late seral or PNC ecological status.

Annual Pattern of Soil Water States - A description of field soil water over the year as applied to horizons, layers, or standard depth zones. Water state is reported by layers.

Hydraulic Control - Features of landform (bedform and bed material), vegetation, or organic debris that control the relationship between stage (depth) and flow rate (discharge) of a stream.

Hydrogeomorphic - Features pertaining to the hydrology and/or geomorphology of the stream system.

Potential Plant Community - Represents the seral stage the botanical community would achieve if all successional sequences were completed without human interference under the present environmental conditions.

Riparian-Wetland Ecological Site - An area of land with a specific potential plant community and specific physical site characteristics, differing from other areas of land in its ability to produce vegetation and to respond to management. Ecological site is synonymous with range site.

Seral Stage - One of a series of plant communities that follows another in time on a specific site.

Stream Power - A measure of a stream's ability to erode and transport sediment. It is equal to the product of shear stress and velocity.

Vegetation Community Dynamics - Response of plant communities to changes in their environment, to their use, and to stresses to which they are subjected. Climatic cycles, fire, insects, grazing, and physical disturbances are some of the many causes of changes in plant communities. Some changes are temporary while others are long lasting.

Vegetation Community Succession - Primary succession is a sequence of plant community changes from the initial colonization of a bare soil toward a PNC. Secondary succession may involve sequences of plant community change from PNC due to perturbations, or a sequence toward PNC again following a perturbation. Vegetation community succession may be accompanied by subtle but significant changes in temporal soil characteristics such as bulk density, nutrient cycling, and microclimatic changes, but is differentiated from major physical state changes such as landform modification or long-term elevation or lowering of a water table that would change the PNC of an ecological site.

Appendix A

Interdisciplinary Team

Team Member	Discipline
Ron Clark - WO-222 (Now CO-930)	Watershed Specialist
Mike Crouse - OR-932	Management/Biologist
Wayne Elmore - OR-050	Riparian-Wetland Specialist/ Wildlife Biologist
Jim Fogg - SC-212	Hydrologist
Ron Hooper - AZ-932	Riparian-Wetland Coordinator/ Hydrologist
Steve Leonard - NV-931	Range Scientist
Don Prichard - SC-213	Riparian-Wetland Coordinator/ Fishery Biologist
Dan Tippy - TC-200 (Now OR-050)	Riparian-Wetland Training Coordinator/Soils
Don Waite - WO-222	Management/Economist
Jack Williams - WO-240	Fisheries Program Manager

Appendix B

Reporting Tables

Table 1. Functioning Condition Status

State:_____

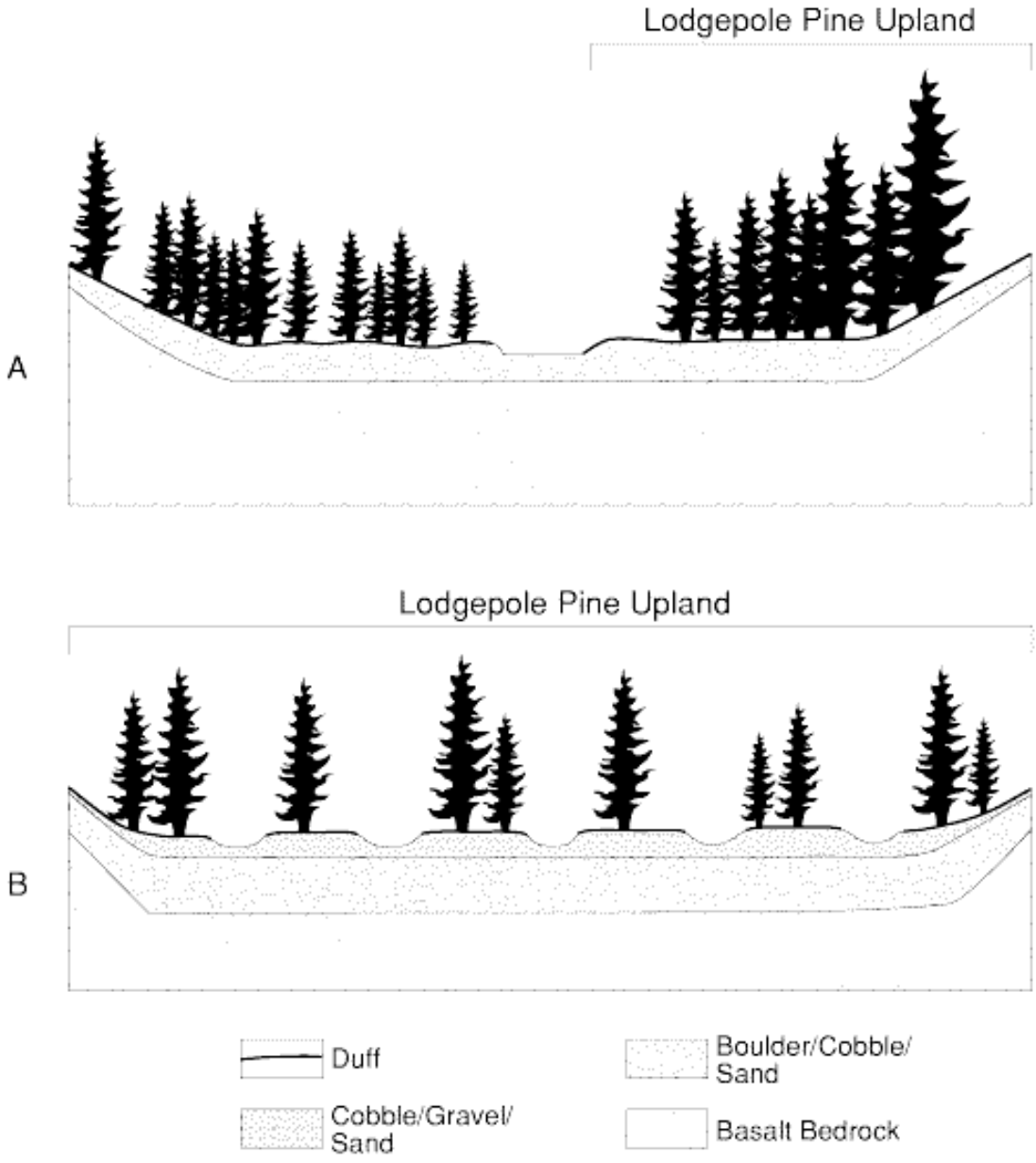
Habitat Types	Proper Functioning Condition	Functional—At Risk			Non-functional	Unknown	Total
		Trend Up	Trend Not Apparent	Trend Down			
Riverine Miles (Lotic)							
Nonriverine Acres (Lentic)*							

* Report only acres associated with lentic riparian-wetland areas. Do not include acres associated with lotic riparian-wetland areas.

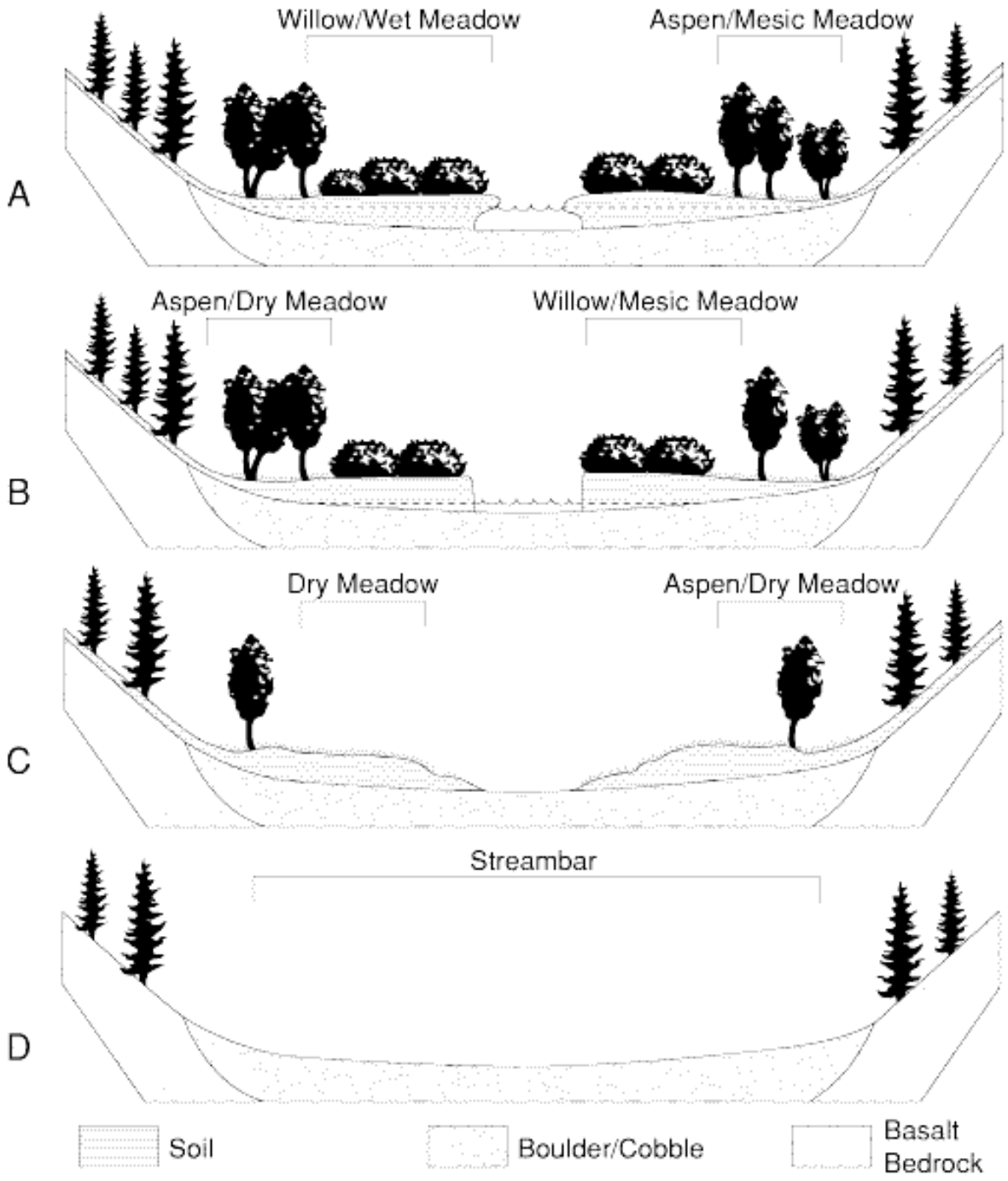
Appendix C

Channel Evolution Examples

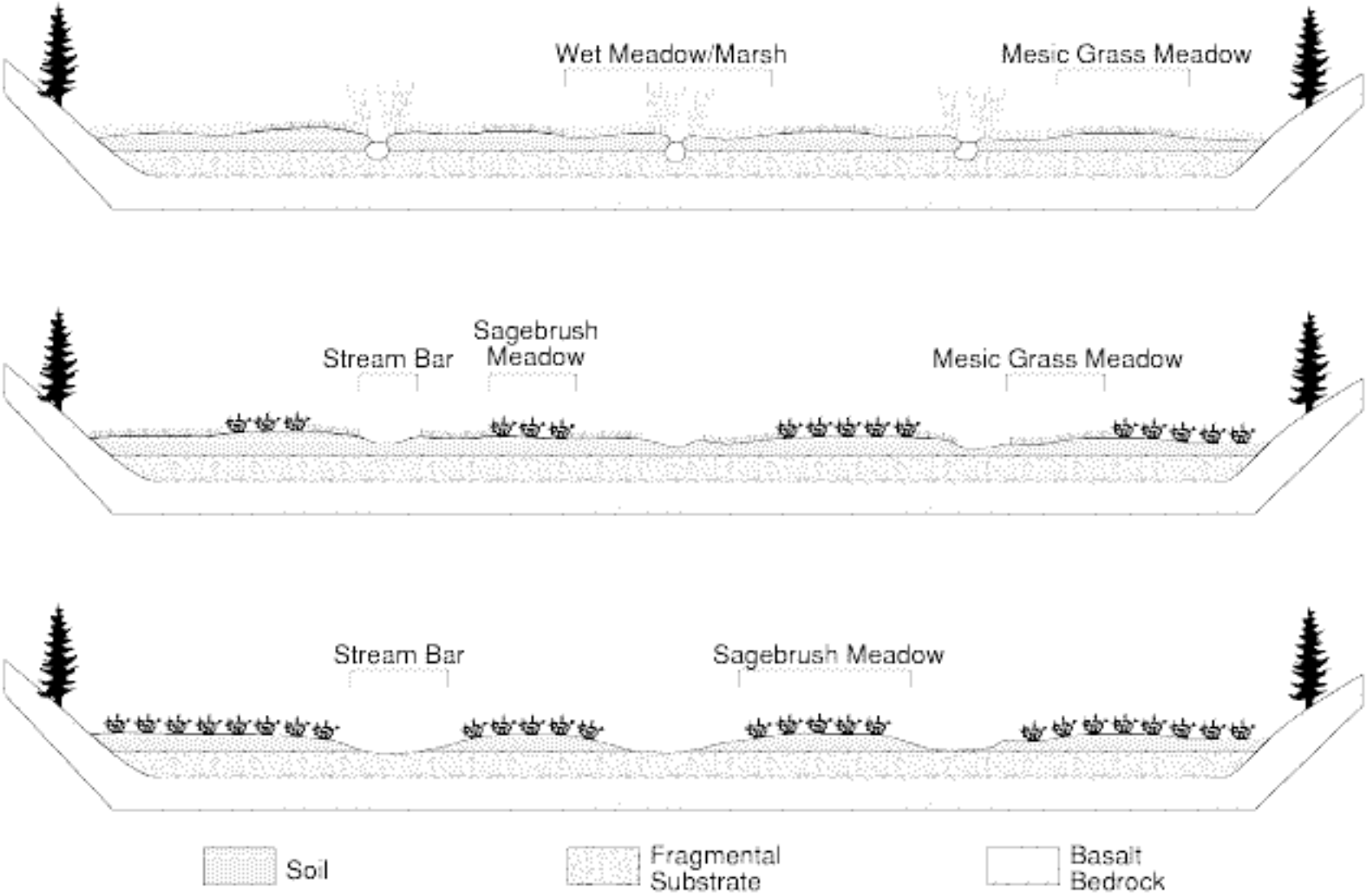
Glacial Valley-Bottom Type



Succession of States for Fluvial/V-Shaped Depositional Valley-Bottom Type



Succession of States for Alluvial/Graded Valley-Bottom Type



Appendix D

Riparian-Wetland Functional Checklist

General Instructions

1) This checklist constitutes the **Minimum National Standards** required to determine proper functioning condition of lotic riparian-wetland areas.

2) As a minimum, an **ID team** will use this checklist to determine the degree of function of a riparian-wetland area.

3) An ID team **must review existing documents**, particularly those referenced in this document, so that the team has an understanding of the concepts of the riparian-wetland area they are assessing.

4) An ID team **must determine the attributes and processes important** to the riparian-wetland area that is being assessed.

5) Mark one box for each element. Elements are numbered for the purpose of cataloging comments. The numbers do not declare importance.

6) For any item marked "**No**," the severity of the condition must be explained in the "**Remarks**" section and must be a subject for discussion with the ID team in determining riparian-wetland functionality. Using the "**Remarks**" section to also explain items marked "**Yes**" is encouraged but not required.

7) Based on the ID team's discussion, "**functional rating**" will be resolved and the checklist's summary section will be completed.

8) Establish photo points where possible to document the area being assessed.

Standard Checklist

Name of Riparian-Wetland Area: _____

Date: _____ Segment/Reach ID: _____

Miles: _____ Acres: _____

ID Team Observers: _____

Yes	No	N/A	HYDROLOGY
			1) Floodplain above bankfull is inundated in “relatively frequent” events
			2) Where beaver dams are present they are active and stable
			3) Sinuosity, width/depth ratio, and gradient are in balance with the landscape setting (i.e., landform, geology, and bioclimatic region)
			4) Riparian-wetland area is widening or has achieved potential extent
			5) Upland watershed is not contributing to riparian-wetland degradation

Yes	No	N/A	VEGETATION
			6) There is diverse age-class distribution of riparian-wetland vegetation (recruitment for maintenance/recovery)
			7) There is diverse composition of riparian-wetland vegetation (for maintenance/recovery)
			8) Species present indicate maintenance of riparian-wetland soil moisture characteristics
			9) Streambank vegetation is comprised of those plants or plant communities that have root masses capable of withstanding high streamflow events
			10) Riparian-wetland plants exhibit high vigor
			11) Adequate riparian-wetland vegetative cover is present to protect banks and dissipate energy during high flows
			12) Plant communities are an adequate source of coarse and/or large woody material (for maintenance/recovery)

Yes	No	N/A	EROSION/DEPOSITION
			13) Floodplain and channel characteristics (i.e., rocks, overflow channels, coarse and/or large woody material) are adequate to dissipate energy
			14) Point bars are revegetating with riparian-wetland vegetation
			15) Lateral stream movement is associated with natural sinuosity
			16) System is vertically stable
			17) Stream is in balance with the water and sediment being supplied by the watershed (i.e., no excessive erosion or deposition)

(Revised 1998)

Remarks

Summary Determination

Functional Rating:

Proper Functioning Condition _____
Functional—At Risk _____
Nonfunctional _____
Unknown _____

Trend for Functional—At Risk:

Upward _____
Downward _____
Not Apparent _____

Are factors contributing to unacceptable conditions outside the control of the manager?

Yes _____
No _____

If yes, what are those factors?

____ Flow regulations ____ Mining activities ____ Upstream channel conditions
____ Channelization ____ Road encroachment ____ Oil field water discharge
____ Augmented flows ____ Other (specify) _____

Appendix E

Riparian-Wetland Examples

Texas Creek—Colorado
September 1976
Nonfunctional

Texas Creek—Colorado
September 1976
Nonfunctional

Texas Creek—Colorado
June 1978
Functional—At Risk

Texas Creek—Colorado
September 1976
Nonfunctional

Texas Creek, located in south-central Colorado on public lands administered by the Canon City District Office, would have been rated ***nonfunctional*** in 1976 based on the Bureau's definitions. Texas Creek is a small coldwater perennial stream that originates in the Sangre De Cristo Mountains, flowing for approximately 24 miles before it enters the Arkansas River. Inventories conducted in 1976 classified the stream as a laterally unstable area that was moderately confined, severely impacted from continuous grazing, and providing limited fish and wildlife values.

The September 1976 photograph clearly demonstrates why Texas Creek would have been rated ***nonfunctional***. This riparian area was clearly not providing adequate vegetation, landform, or large woody debris to dissipate stream energies associated with high flows. With each storm event, the stream channel migrated, erosion accelerated, sediment was not filtered, flood-water retention and ground-water recharge were limited, and water quality was altered. Wildlife values were limited to principally a watering site, and the brown trout population, less than 13 fish per 500 feet of stream, was well below the area's capability or potential.

For the most part, placing a stream into the category of ***nonfunctional*** would be a simple task. However, there are areas (natural and altered) that will always look like this.

Texas Creek—Colorado
June 1978
Functional—At Risk

Management actions were changed in 1977 to reverse the trend of Texas Creek and to allow the area to progress towards its capability and potential. Changes included improved fencing, and rest and implementation of deferred seasonal grazing or winter grazing. Quality of habitat in Texas Creek began to improve immediately after changing management practices, and the June 1978 photo displays the results. Using the Bureau's definitions, Texas Creek would have been rated as ***functional—at risk*** in June 1978, with an upward trend.

Comparing the changes between the 1976 photo and the 1978 photo shows that Texas Creek was in an upward trend and had started to function physically. With increased vegetation, stream energies had been reduced, sediment had been filtered and captured, streambanks had developed, flood-water retention and ground-water recharge had increased, stream width had decreased, erosion was reduced, and water quality improved. With these physical changes, wildlife and fishery values had increased. The brown trout population more than doubled from 1976.

Yet, the area was still at risk because soil and vegetation attributes still made it susceptible to degradation. The area contained too much bare soil and lacked desirable species of vegetation. The dominant species present lacked root masses that stabilize streambanks against cutting action.

Texas Creek—Colorado
October 1978
Proper Functioning Condition

Texas Creek—Colorado
October 1978
Proper Functioning Condition

Texas Creek—Colorado
July 1987
Proper Functioning Condition

Texas Creek—Colorado
October 1978
Proper Functioning Condition

By the end of the 1978 growing season, Texas Creek progressed to where it had crossed its threshold as described in Figure 2 in the Functioning Condition section. Using the Bureau's definitions, in October 1978, Texas Creek would have a rating of ***PFC***. **Yet, by no means had Texas Creek achieved its capability or potential. However, it may have achieved its management objectives and obtained its desired plant community (early seral versus PNC).** The early seral vegetation community that had established itself in the October 1978 photo possessed the ability to dissipate stream energies associated with high flows for Texas Creek. The instability that was present in Texas Creek in June 1978 had dissipated and the soil and vegetation attributes that placed Texas Creek into the category of ***functional—at risk*** were no longer present. Attributes such as reduced erosion; improved water quality; floodplain development; trapment of woody debris; improved retention of flood-water and ground-water recharge; diverse ponding; channel characteristics that provide habitat and water depth, duration, and temperatures necessary for fish production; and other wildlife values had been greatly strengthened.

Adjusting the rating of an area from ***functional—at risk*** to ***PFC*** may not be easy. For Texas Creek it was easy because 12 years of data had been collected. For most areas, BLM does not have that luxury. **That's why an ID team is necessary.** For some areas, the only way to assess functionality is with an effort like ESI.

Texas Creek—Colorado
July 1987
Proper Functioning Condition

Placing areas that have achieved late seral or PNC, as Texas Creek had in this July 1987 photo, into the appropriate category is easy. Using the Bureau definitions, Texas Creek would have a rating of ***PFC***. The difference between the October 1978 photo and the July 1987 photo is that the vegetation community was early seral for 1978 and late seral for 1987. However, both communities were functioning properly. Management defines its Desired Plant Community for an area, which in turn defines BLM's management options.

For example, bighorn sheep and brown trout are present in the Texas Creek watershed. If the desired species for management is bighorn sheep, which prefer early seral vegetation around watering sites, the desired plant community for Texas Creek would be early seral (October 1978 photo). At the same time, brown trout production is possible, but not at optimal numbers. Yet, the area can **function properly**. Optimal numbers of brown trout for this area would occur by managing for mid-seral to late seral. However, this would not be to the liking of the bighorn sheep.

Riparian-wetland areas can be managed to provide greater biodiversity as well as to allow the **entire area to function properly**. Most riparian-wetland areas can function properly in all seral stages, thus giving BLM greater management flexibility.

Forested Coastal Stream—Oregon
Nonfunctional

The below photograph gives an example of a coastal stream, located in Oregon, that would be rated as ***nonfunctional*** relative to BLM's definitions for proper functioning condition. The riparian area is clearly not providing adequate vegetation, landform, or large woody debris to dissipate stream energies associated with high flows. During precipitation events, the stream channel migrates, erosion continues, sediment is not filtered, flood-water retention and ground-water recharge are limited, and water quality is altered. Wildlife values are limited, and the area is not providing diverse ponding or channel characteristics that provide habitat and water depth, duration, and temperature necessary for fish production. The area provides little biodiversity.

Forested Coastal Stream—Oregon
Functional—At Risk

Forested Coastal Stream—Oregon
Proper Functioning Condition

Forested Coastal Stream—Oregon
Functional—At Risk

Establishment of alders provides the capacity to dissipate some stream energies that occur with flow events in this area. This capability results in captured sediment and bedload, reduced erosion, and improved water quality, and aids floodplain development and improves flood-water retention and ground-water recharge. In other words, the area has started to function physically.

In spite of functioning, this area would be rated as ***functional—at risk*** because a vegetation and hydrologic attribute still make the area susceptible to degradation. While the alder plant community does provide root masses that stabilize streambanks against cutting action, it probably is insufficient for major flow events. Large woody debris (hydrologic controls) is also lacking, which inhibits capture of sufficient bedload to aid in the development of habitat that provides water depth, duration, and temperature necessary for fish production, waterfowl breeding, and other uses, thus supporting greater biodiversity.

This area will function properly before it obtains PNC. As the alder community ages, it will topple into the stream providing woody debris that aids in the capture of bedload. Also, as the alders depart, conifer climax species will dominate the site and provide the necessary bank stability. All this will occur before optimal numbers of wildlife and fish species (greater biodiversity) are achieved.

Forested Coastal Stream—Oregon
Proper Functioning Condition

The photograph to the left depicts a forested riparian-wetland area that achieved the rating of ***PFC***. The photograph clearly shows a coastal stream that contains adequate vegetation and large woody debris that is dissipating stream energy associated with high waterflows, thereby reducing erosion and improving water quality. The plant community has developed root masses that have stabilized streambanks against cutting action, filtered sediment, and captured sufficient bedload. This has aided floodplain development and has improved flood-water retention and ground-water recharge. The natural process has created diverse ponding and channel characteristics that provide the habitat and the water depth, duration, and temperature necessary for fish production, waterfowl breeding, and other uses, thus supporting greater biodiversity.

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This technical reference outlines the Bureau of Land Management's (BLM's) process for assessing the functioning condition of riparian areas on public lands. Emphasis is placed on the interaction of vegetation, landform/soils, and hydrology in defining capability and potential of an area. The importance of using an interdisciplinary team is also stressed. The document describes four categories of functioning condition—proper functioning condition, functional—at risk, nonfunctional, and unknown, and discusses management strategies for each.

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Sampling

Overview and Check List

This list is an overview of everything involved with a sampling event. You can use it as a check list to make sure all is covered. Specific instructions for each follow in this section as well as datasheets and instructions.

Sample Prep (in the lab)

Decide what you will be sampling today, water quality, macroinvertebrates, physical habitat, optional (though recommended) velocity / discharge and or photograph

Decide how many stations will be sampled today

Determine if QA/QC samples will be collected today

- Metals = blank/duplicate every 5th trip
- Nutrient duplicate (if instructed by us to do so)
- Macroinvertebrate QA sample (if instructed by us to do so)

Label your bottles

- Metals (every time)
- Nutrients (hi/low flow biannually)
- Macroinvertebrate (inside/outside labels)

Preserve all metal samples (**follow nitric acid protocols**)

If time allows and it's a metal QA/QC sample, collect blank sample in your lab

Make sure caddy is stocked, don't forget waders, waste container, safety plan, gloves, etc.

If macroinvertebrate collection, check macroinvertebrate check list

If physical habitat collection, check physical habitat check list

If velocity / discharge, photo, check for: data sheets, float object, timer, distance measuring device, depth measuring device, camera

In the Field

Order of collection function (walking composite, bucket composite or grab). If collecting everything always collect water quality first (metal and/or nutrient), macroinvertebrate collection second, physical habitat third, velocity / discharge fourth

Collect filtered and not filtered metals with syringe (blank and dup if necassary)

Collect DO, 300 ml BOD

Take and record temperature

Collect 16 ounce sample for alkalinity/hardness/pH

Collect nutrient (if requested)

Collect macroinvertebrates (if requested)

Assess physical habitat if possible

Measure velocity/discharge if desire

Take annual photo if time permits

Complete field data sheet for field

Check site for litter

Back in the Lab

Put metals and or nutrients in refrigerator until it’s time to ship

If sample at real time USGS/State Engineer Gauge, go online and get flow, record on field datasheet

Titrate DO within 8 hours (keep cold/dark place), complete data sheet

Titrate alkalinity/hardness within 24 hours, complete data sheet

Take pH within 24 hours at room temperature, record on field datasheet

Complete field data sheets

If collected macroinvertebrates, that evening decant alcohol in sample jar and refresh with remaining alcohol

Enter data on webpage

Copy original data sheets (all), file copy in your records

With data sheets and chain of custody, ship metals at least every 3 months, nutrients within 48 hours, not on a Thursday or Friday and by 15th of each month, macroinvertebrates within two weeks of collection

Before shipping, check supplies for refills, include sample bottles in the shipment for appropriate chemicals

Send velocity / discharge, take photo, conduct optional macroinvertebrate, physical habitat analysis

Clean and store all field and lab equipment properly

Sampling Instructions

Identifying a Sampling Event

Each time you go to the river and collect or assess something, it is a sampling event. For example if you only measure temperature, that is a sampling event. If you collect and assess everything, metals, nutrients, bugs and physical habitat that is an event. If you measure dissolved oxygen every hour for 24 hours, each hour is one sampling event. A sampling event is a one time occurrence. River Watch needs to be able to identify each sample event as unique occurrence. Using the following data, each sample event is uniquely identified by:

- station number
- sample date
- sample time

All three items are required. Thus, if one bottle or 14 sample bottles are collected at one time and analyzed for one or dozens of parameters, this is **one** sampling event, and gets only the above listed unique combination of identifiers. You can collect two samples in one day, but each of the sampling events would occur at a separate time, thus have two unique combinations of station number, sample date and sample time. If a nutrient sample or a macroinvertebrate sample is taken at the same time as metal sample, the sample identifiers would be the same.

In the past River Watch used sample number, a combination of station number and a consecutive frequency number, such as 93.040 for the unique sample event identifier. **River Watch no longer uses this sample number as the unique identifier, but instead will use the combination of station number, sample date and time.** There should never be two different sampling events that have the same combination of identifiers.

Prioritization of what constitutes a sampling event. For River Watch data objectives this is what is recommended:

- when collecting a metals sample always collect pH, temperature, alkalinity and hardness (field parameters)
- when collecting dissolved oxygen always collect temperature
- when collecting nutrients, collect metals and other field parameters (temperature, pH, etc.)
- when collecting macroinvertebrates, collect water quality samples metals and field parameters at a minimum, nutrients if possible (it tells us the water quality for the bugs when you sampled)
- if collecting macroinvertebrates, ALWAYS complete physical habitat analyses and take a photo if possible
- if not collecting macroinvertebrates conduct a stream reach physical habitat analyses and photo if possible
- keep an annual photo log of site (via our instructions)

Each station should use a **sample tracking sheet** to track what is been collected or assessed when at that location. The sample date and time will be recorded along with a note of what parameters are collected, metals, blank or duplicate metals, nutrients, macroinvertebrates and physical habitat. The sample tracking sheet is for your records only. Below is an example of how to use the tracking sheet.

SAMPLE TRACKING SHEET

Station Name *At Mouth*   Sheet *1* of *2*

River *Plum Creek*   Station Number *999*

Volunteer Group *River Watch*

***Remember every 5th metal sample event should include a duplicate and blank sample.**

Sample Collection Preparation

SAMPLE DATE	SAMPLE TIME	**DESCRIPTION**		
		Metals	Nutrients	Macroinvertebrates
07-06-06	0900	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
8-10-06	0930	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
9-12-06	0905	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
10-10-06	1000	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
11-11-06	0930	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☒ NF Blank ☒ F Duplicate ☒ NF Duplicate ☒	TSS / CS ☒ NP ☒ Duplicate TSS / CS ☐ NP ☐	Yes ☒ QA sample ☐
12-09-06	1000	Filtered (F) ☒ Non-Filtered(NF) ☒ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐
		Filtered (F) ☐ Non-Filtered(NF) ☐ F Blank ☐ NF Blank ☐ F Duplicate ☐ NF Duplicate ☐	TSS / CS ☐ NP ☐ Duplicate TSS / CS ☐ NP ☐	Yes ☐ QA sample ☐

In the Lab

Determine what you will be collecting or assessing this sampling event, field parameters (metals and baseline parameters), quality control samples such as metal blanks and duplicates, nutrients, macroinvertebrates, physical habitat or discharge (optional). Record on the sample tracking sheet this sampling event for each station.

Prepare datasheets for each station: Field Data Sheet, Alkalinity, Hardness, and Dissolved Oxygen. Also, prepare data sheets for Macroinvertebrate collection and physical habitat, if appropriate, and Discharge (optional). Complete the top portions including date, station name, station number, organization and river name.

Metals:
For metal samples prepare (label and preserve) 2, two-ounce bottles for filtered and not-filtered samples.

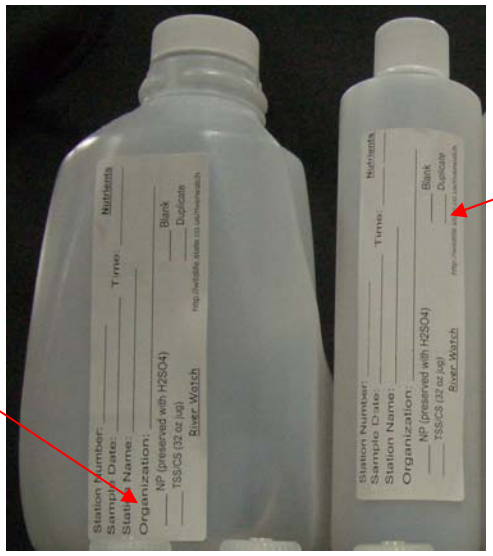
If you run out of labels and you are planning on sampling before you request more, please use a sharpie (or another non leaking marker) to label the bottles. Please make sure group name, station, date, time and what kind of sample it is (filtered, non-filtered etc.) is clearly marked on each bottle.

All metal collection samples are preserved with nitric acid. **Please use caution!** Using the proper safety gear, place 12 drops of HNO_3 (nitric acid) in each of your metal collection sample bottles.

If you spill your sample in the field, rinse the sample collection bottle twice with sample water and repeat steps for filling. Once back in the lab, put 12 drops of HNO_3 in the collected sample.

If you are on your fifth metals sample since the last blank and duplicate sample was taken, you need to prepare duplicate and blank sample bottles. Please review instructions for duplicates before sampling. Remember, all blank and duplicate samples need the 12 drops of HNO_3 too.

Nutrients:
If you are collecting a nutrient sample, gather those two bottles and prepare label. Nutrient samples are generally collected twice per year, once during high flow in the spring, and once during low flow in the fall.



If you run out of labels and you are planning on sampling before you request more, please use a sharpie (or another non leaking marker) to label the bottles. Please make sure group name, station, date, time and what kind of sample it is (filtered, non-filtered etc.) is clearly marked on each bottle.

Macroinvertebrates and physical habitat assessment:
If you are collecting a macroinvertebrate sample and conducting a physical habitat assessment, prepare the bug sample bottle. Macroinvertebrates are sampled once per year in the fall. A physical habitat assessment should always be conducted with a bug sample but can be conducted independently. A water sample should be collected with each bug collection to tell a more complete story.

Sample Labeling

It is important the datasheets and metals, nutrients and macroinvertebrate sampling bottles are labeled correctly. Also, it is important to mark/label the filter you will be using to collect the metal sample. **Filters must only be used once**; marking them helps remind you that this filter has been "used".

Be sure and fill out your sample labels completely. Be sure to check if metal is filtered, non-filtered, and if the sample is a blank or duplicate. **For example:**

Metal Label:

Station Number _____6000__
Sample Date __7/17/05_____ Time: 900__
Station Name _Sample Bridge_____
Organization _River Watch_____
__X__Non-Filtered _____Blank
_____Filtered _____Duplicate

Metal Label (for duplicate):

Station Number _____6000__
Sample Date __7/17/05_____ Time: 900__
Station Name _Sample Bridge_____
Organization _River Watch_____
__X__Non-Filtered _____ Blank
_____Filtered _X___Duplicate

Nutrient Sample:

Station Number _____
Sample Date _____ Time: _____
Station Name _____
Organization _____
____NP (250 ml H2SO4 preserved) ____Blank
____TSS/CS (500 ml jug) ____Duplicate

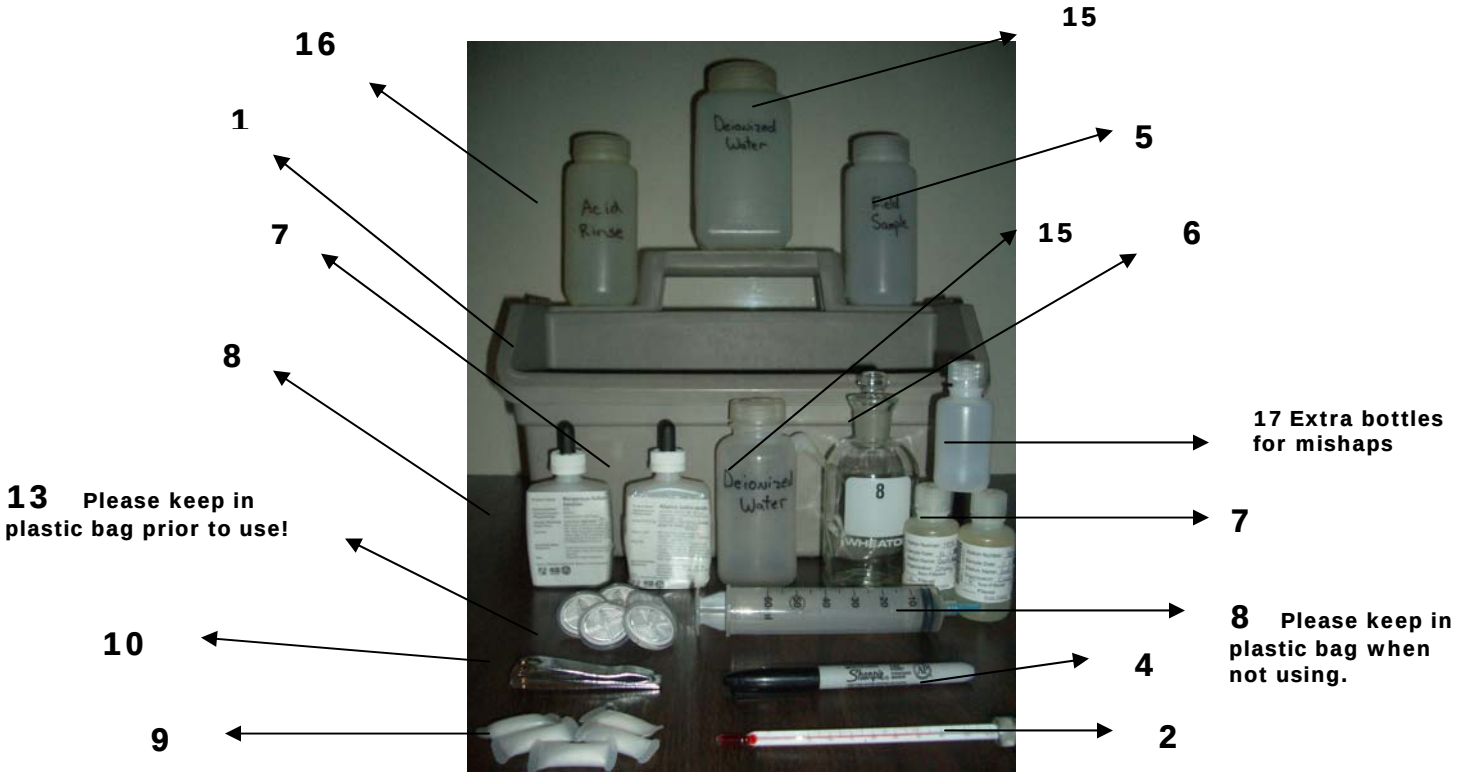
Macroinvertebrate Sample (for outside bottle):

Station Number _____
Sample Date _____ Time: _____
Station Name _____
Organization _____
____Rocky Substrate ____Sandy Substrate
____QA sample

See macroinvertebrate label for inside the bottle in macroinvertebrate section.

Gather the equipment, supplies, and sample bottles to take in the field. Here is a check list of what you will need:

1. caddy
2. Thermometer
3. Field data sheet(s)
4. Sharpie
5. 16 ounce sample bottle for sample collection
6. 300 ml BOD bottle (one per station),
7. alkaline iodide
8. manganous sulfate
9. sulfamic pillows
10. clippers
11. 2, 2 ounce pre-labeled and preserved metal bottles (N and NF - per station)
12. syringe (one per station)
13. 2 filters (two per station) – (plus extras for mishaps)
14. 16 ounce bottle of nitric acid rinse
15. Large container of Deionized water and a DI squirt bottle
16. 4, 2 ounce pre-labeled and preserved metal bottles **IF** collecting blank and duplicate (per station)
17. CS/ TSS 500 ml pre-labeled nutrient bottle (per station) **IF** collecting nutrient sample
18. NP 250 ml pre-labeled and preserved nutrient bottle (per station) **IF** collecting nutrient sample
19. Macroinvertebrate and/or physical habitat equipment, labeled sample jar and macro/phys habitat datasheet **IF** collecting
20. Waders
21. Safety plan and appropriate materials (goggles, gloves, bug spray, etc.)
22. Extra unpreserved 2 oz metal sample bottle (for mishaps)



Sample Collection

There are two ways of to collect a sample:

- A composite sample is collected by taking several small sub-samples and combining them.
- A grab sample is collected at one point in the stream.

A composite is always preferred to provide a representative sample. The priority of collection method should follow this order after access and safety issues are addressed: first a walking (or wading) composite, next an upstream bridge composite, then a downstream composite and last choice a grab sample. The grab sample is taken only if the sampler cannot access the entire stream either from a bridge or by crossing it because of high flows or other unsafe conditions.

Walking (wading) Composite Sample

If access and safety permits, for all collection containers, wade across the stream following an imagined transect line, collecting a sub-sample of water at appropriate frequencies, based upon stream width. Wade and collect in such a manner that you do not disturb the substrate and water column where you dip the collection container.

The exception to this is method is the dissolved oxygen BOD sample bottle. Find a representative flow, not too fast or too slow and stagnant to hold the BOD bottle under water for 2-3 minutes. See specific sections for detailed sample instructions, metals, field and nutrient.

Remember:
It is important that the sampler and the other field crew never walk in the stream above where the sample is collected.

Composite Bucket Sample

For sample stations located at a bridge and on a river that is too deep and/or dangerous to wade across, a bridge composite sample should be taken. An upstream sample is preferred, however, if it is safer to sample downstream that is okay.

1. Use the two buckets provided. Use these buckets for sampling **ONLY**, otherwise we risk contamination. Use one bucket to collect water and the other to hold or “composite” the sample. The composite bucket holds the water you will collect your samples from.

2. Dip one bucket into a representative flow. This water will be used for rinsing the sample buckets.

3. Flush 60 mL of DI water through the syringe twice by opening the syringe and pouring the DI into it, (120 mL total). Assemble the syringe.

4. Rinse the syringe three times with sample water; the 16 ounce sample bottle, and the BOD bottle (if you plan on collecting the DO sample from the composite bucket) and rinse the other bucket. Empty the rinse water bucket. Keep the rinsed sample syringe and containers in a clean place.

5. With the sample bucket, go to the upstream left bank and fill the bucket with a representative sample of water. Gently pour the sample into the composite bucket, filling it 1/3 full. Remember to always toss the extra water downstream behind you.

6. Go to the middle of the bridge. Lower the sample bucket down to the river and fill the bucket with a representative sample of river water. Gently pour the sample into the composite bucket, filling it 2/3 full.

7. Go to the upstream right bank and repeat this procedure. Gently pour sample into the composite bucket. The composite bucket should be close to full with river water from three areas.

Remember, **do not** let your bucket touch the bottom or the sides of the bridge, you want to test the water column, not the stream sediment.

The composite bucket now holds the water you will collect for analysis and perform the chemical tests on. The order of the collection is important due to possible contamination.

*Because you do not want to contaminate your samples, if you are going to take dissolved oxygen using the composite bucket, take this sample first. Very carefully and slowly, pour sample water into the BOD bottle. You MUST pour this slowly as you do not want to introduce any more oxygen into the sample. Make sure to overflow the bottle a little and have no air bubbles in the bottle. Once the bottle is full, continue to “fix” the sample.

* If you would rather not pour the sample from the bucket into the BOD bottle, you can collect your other samples first and then collect another composite sample. Once you have collected another composite sample, place the BOD bottle slowly in the bucket, making sure to tilt the bottle up to release all the air. Once the bottle is full, remove from the bucket and continue to “fix” the sample.

Next, if you are collecting a nutrient sample, pour the sample from the bucket into the 32 ounce jug and from the jug, fill the 8 ounce preserved nutrient container. Be careful not to overflow the bottle as it contains a preset amount of sulfuric acid to preserve the sample. Refill the 32 ounce jug from the composite bucket.

Next, pour water from the bucket and fill the 16 ounce sample bottle. This will be used for field parameter tests.

Next collect your metals samples, both filtered and non-filtered. You have taken everything from the composite bucket, so you can go ahead and put the syringe in the bucket for these samples.

Finally, take the temperature from the bucket.

Grab Bottle Sample

Use a grab sample when flows are too high to complete a walking composite or if a bridge is not available. When collecting a grab sample:

1. Select a site you can safely reach into the water where the water is flowing and not stagnant.

2. Flush 60 mL of DI water through the syringe twice by opening the syringe and pouring the DI into it, (120 mL total). Assemble the syringe.

3. Rinse the syringe three times with sample water then fill metals bottles (see instructions).

4. Rinse 16 ounce sample bottle twice with sample water, disposing the water downstream, then fill sample bottle.

5. Fill BOD bottle for dissolved oxygen test and take temperature.

Remember:
It is important that the sampler and the other field crew never walk in the stream above where the sample is collected.

Frozen Sample Site

1. **Proceed only if your teacher or team leader has ascertained the ice will support your weight and movements.**

   If possible, obtain an ice auger and auger through the ice at your station. Use the auger with care and periodically assess the stability of the ice around you. Note that you augured a hole in the Field Data Sheet “Comments” section.

OR:

2. Walk up or down stream to the first open water and collect a sample there. On the Data Entry Form “Comments” section, note where you collected the sample. In either case make sure the ice and snow has thoroughly melted in the stream before sampling.

3 If neither above option is possible, **DO NOT** collect a sample, and note the reason in the Field Data Sheet and/or Data Entry Form “Comments” section and file. Discard collection bottles. **SAFETY FIRST!**

Metal Sample Collection

Metals are generally collected monthly with basic River Watch. When collecting a metal sample a filtered (dissolved fraction) and non-filtered (total fraction) collection is required. In addition, River Watch requests that when a metal sample is collected, data for hardness, alkalinity, temperature and pH measurement and discharge if possible are also collected. This data provides information for interpretation about mitigating factors and factors that influence the toxicity of metals to aquatic life.

Filtered and Non-filtered sample collection

****Remember, before leaving the lab, all METAL collection bottles should have been preserved with 12 drops of nitric acid. Please follow instructions on HNO_3^- use!!! Be careful not to spill bottles!**

Filling the sample bottles, remember the chant "rinse-rinse-collect-rinse-rinse":

Non- Filtered:

1. Completely flush (fill and squirt) syringe twice with deionized water.
2. Rinse and shake syringe twice with 10 ml sample water.
3. Fill syringe with sample water, from walking composite, composite bucket or grab location.
4. Open cap to non-filtered bottle and set aside.
5. Gently squirt into non-filtered sample bottle without touching, fill to the neck of the bottle. Do not overfill (if you do, you must dump, rinse, fill again, preserve sample back at lab and note on field datasheet). Syringe holds 60 ml; sample bottle is 60 ml, fill to neck/shoulder of bottle; refill syringe if necessary.
6. Recap non-filtered bottle and set aside.

Filtered:

1. Remove cap from filtered bottle and set aside.
2. Fill syringe with sample water.
3. Mark the filter with sharpie and place marked filter on syringe tip (screws in).
4. Seed (empty) up to 10 mL of sample through filter, **NOT** over the open sample bottle.
5. Gently squirt sample through the filter into the filtered bottle. If filter becomes clogged, use second filter; remember to seed this filter too. Syringe holds 60 ml. Sample bottle is 60 ml, fill to neck/shoulder of bottle. Refill syringe if necessary.
6. Recap the bottle and set aside.
7. Remove filter from syringe and dispose in a trash receptacle. Remember, filters can only be used once.
8. Check the boxes on the field data sheet to match your collection, non-filtered and filtered.

<u>Now it's time to clean. Back in the lab or in the field (within high water mark):</u>

1. Do the dishes: “wash” the syringe with acid rinse. Pour 10 ml or so of acid rinse into the syringe and swirl. Push the acid rinse down the sink or within the high water mark.
2. Rinse syringe with deionized water. Shake excess water from syringe and store syringe in a clean Ziploc bag.

Metal Blank Sample

A blank sample is a quality control sample where the sample water is deionized water not river water. From preparation to collection to analysis, a blank metal sample is treated just like a normal sample; the difference is the actual water in the bottle and the fact that the “sample” is poured into the syringe. A blank sample serves as a quality control sample by testing for contamination in the method that is used to “collect” normal river water for metals analyses. That is why it is easy to remember how to collect a blank, same way you collect a normal river filtered and a non filtered metal sample, only your sample water is deionized water. In theory, there are no metals in de”ion”ized water. Thus, if you are not introducing metals to the sample via the collection procedure, when we analyze the blank metal sample for metals we should get zero. What does it mean if we get a result? It is an indication that metals have been introduced to the blank sample and thus possible the river sample as well. We cannot validate your metals samples without blank checks. Please collect a blank metal filtered and non-filtered metal sample every fifth trip to a station.

You can collect a blank before you leave for the sample event, at the site or when you return.

1. Label two (2) additional metals bottles, one filtered and one non-filtered, as you normally would for one of your stations. Remember to check the “Blank” line on the label for both samples.
2. Flush the syringe twice with deionized water. **DO NOT STICK THE SYRINGE IN THE CONTAINER.** Flush by pouring deionized water into the syringe; never stick your syringe into anything but the river. This is the first step of collecting a “normal” sample too. If you think your finger is contaminating the sample let about 10 ml drain.
3. Rinse the syringe twice with sample (deionized) water, again by pouring 10 ml or so in the syringe, shaking and squirting. This is the second step in a normal sample collection.
4. Now you are ready to collect the blank sample. Fill the syringe with deionized water from your deionized water container.
5. Find the bottle labeled with “non-filtered” and “blank” and fill it to the neck/shoulder, do not over fill-or start over.
6. Refill the syringe with deionized water.
7. Grab a filter, mark it as used. Place it on the syringe and seed 10 ml through it.
8. Find the bottled labeled with “filtered” and ”blank” and fill it to the neck/shoulder, do not over fill.
9. Check the boxes on the field data sheet that match your collection for blank collection.

Back in the lab or in the field (within high water mark)

1. Do the dishes; “wash” the syringe with acid rinse. Pour 10mls or so of acid rinse into the syringe and swirl. Push the acid rinse down the sink or within the high water mark.
2. Rinse syringe with deionized water. Shake excess water from syringe and store syringe in a clean Ziploc bag.

Metal Duplicate Sample

A duplicate metal sample is a quality control sample that is a “second” sample containing the same “slug” of water as the normal metal sample. A duplicate metal sample serves as a quality control sample by checking for the reproducibility of the sample crew collection method. If collection methods are adequate and followed, and the ICP is working properly, metal analyses results should be very similar between the metal duplicate and normal sample.

You will take both the normal sample and the duplicate sample at the same time.

1. Label two (2) additional metals bottles as you normally would for one of your stations. Check the “Duplicate” line on the label. Be sure to have your normal bottles on hand too.
2. Flush the syringe twice with deionized water. **DO NOT STICK THE SYRINGE IN THE CONTAINER.** Do this by pouring deionized water into the syringe; never stick your syringe into anything but the river. Assemble the syringe.
3. Rinse the syringe twice with sample water. Fill the syringe with sample water.
4. Open both normal and duplicate “non-filtered” bottles. Gently squirt some into each bottle, alternating bottles until both are full to the neck/shoulder. You will need to refill your syringe at least once to fill both bottles.
5. Collect another syringe full of sample water.
6. Grab a filter, mark it as used. Place it on the syringe and seed 10 ml through it.
7. Open both normal and duplicate “filtered” bottles. Gently squirt some sample water into each bottle, alternating bottles until both are full to the neck/shoulder. You will need to refill your syringe. Remove filter before refilling syringe, minimize handling and place in a clean dry place. Replace filter before continuing to fill the bottles. If need to replace actual filter, seed second filter with 10 ml of sample water first.
8. Check the boxes on the field data sheet that match your collection for duplicate collections.

Back in the lab or in the field (within high water mark)

9. Do the dishes; “wash” the syringe with acid rinse. Pour 10 – 20mls or so of acid rinse into the syringe and swirl. Push the acid rinse down the sink or within the high water mark.
10. Rinse syringe with deionized water by disassembling the syringe and pouring DI water into it. Shake excess water from syringe and store syringe in a clean Ziploc bag.

Instructions for Making Acid Rinse

Cleaning our syringe and equipment is a quality assurance activity. Metals tend to accumulate onto plastic. Acid rinse is a “soap” that prevents build up of unwanted material from each sampling event. This helps reduce sources of contamination.

1. Use proper nitric acid handling procedures and personnel protective equipment.
2. Fill the acid rinse bottle with deionized water.
3. Place 36 drops or approximately 1 mL of nitric acid into the acid rinse bottle.
4. Shake well.
5. When you run out of acid rinse, make more. This is a weak solution and should not be irritating to most individuals upon touch.

Nutrient Sample Collection

Nutrient samples will be collected twice a year, once in the fall during a low flow period and once in the spring during a high flow period. The annual River Watch Calendar provides the nutrient sampling schedule for participants. For special projects, additional samples may be collected.

You will receive two nutrient sample bottles for each sample event per each site. One container is a 32 ounce juice jug; the second container is an 8 ounce cylindrical and already contains 0.75 mL of **concentrated sulfuric acid** preservative. Be careful to not spill any of the sulfuric acid. Please store these bottles in a clean, dry, cool place in an upright manner.

Again, River Watch requests that you try and coordinate your nutrient sampling event with a regular (metal and field parameter collection) scheduled sampling event. Having all the data from the same sample event tells a deeper broader story about your river.

Label each bottle prior to sampling as per instructions. Place containers in carrying caddy to take to the field.

1. Take the juice jug and either:

   A. Collect a walking composite from a cross section.
   B. Pour sample into jug from the composite bucket.
   C. Collect sample from the bank if a composite is not possible.

2. Pour a portion of jug contents into the 250 ml cylindrical container. **Do not put the cylindrical jug in the stream. BE CAREFUL to not splash, spill or overfill the bottle with the sulfuric acid preservative.**
3. Once your cylinder is full of sample, recap the bottle and set aside.
4. Refill the jug in the same manner and set aside.
5. Check the boxes on the Field Data Sheet that refer to collecting a nutrient sample. If you are only collecting a nutrient sample and no other sample, still complete all relevant information on the Field Data Sheet.

Back in the lab, refrigerate samples as soon as possible.

Complete chain of custody, and ship within 48 hours. The cooler should contain enough blue ice to keep the sample chilled for two days.

Please do not collect a sample on Friday if you cannot ship until Monday, it will take two more days to arrive. Please do not ship you nutrient samples on a Friday as they will sit over the weekend or longer in a warehouse prior to reaching River Watch.

NUTRIENT QUALITY CONTROL SAMPLE

Approximately 10% of the volunteer groups will receive an extra set of nutrient bottles labeled "**DUPLICATE**." These serve as a field quality control and assurance sample. You collect these identically to the regular sample, alternating pouring from the jug into the cylinder till both cylinder bottles are full. Fill both jugs from the composite sample bucket.

1. You collect these identically to the regular sample, alternating pouring from the juice jug into the 250 mL preserved cylinder till both cylinder bottles are full. Check the label on the bottle "NP" and "duplicate". Mark the Field data sheet box to indicate a nutrient duplicate.

2. Fill both jugs from a walking composite, the composite bridge sample from the bucket or at the grab/bank location. You want the same slug of water in both bottles. Check the label on the bottle "CS/TSS" and "duplicate". Mark the Field data sheet box to indicate a nutrient duplicate.

3. Follow the same storage and shipping instructions as a normal nutrient sample.

Temperature

How

1. Take the temperature directly in the river in representative location or in the composite bucket if access to the river is not possible or safe.

2. Allow a couple of minutes or more for the thermometer to equilibrate.

3. Measure and record to the nearest whole degree Celsius.

4. Record the temperature on the Filed Data Sheet.

Hint: you might want to tie a string to the thermometer for easier retrieval and to reduce opportunities for losing thermometer.

Back in the lab

1. Store all samples in the refrigerator or where appropriate until you're ready to perform the analyses.

2. Clean all equipment. Store all your equipment clean.

3. If possible conduct all titrations and pH tests when you return. If not, do so within the following holding times:

    - **pH** within 24 hours at room temperature. If the temperature of the water is below 20^{o}C, you should let the sample warm-up to room temperature for a better reading (20-25 ^{o}C is optimal). This should only take a half hour or so. **DO NOT** artificially heat the sample. Tuck sample bottle in pocket or under arm pit for warming.
    - **Alkalinity and Hardness** within 24 hours if sample bottle kept in cold place.
    - **Dissolved Oxygen** within 8 hours, once fixed (first three chemicals), if capped and stored in cold dark place.

4. Complete all datasheets completely and check or completeness and accuracy. (See following pages.)

5. Enter data via website.

6. Copy data sheets and file.

7. Prepare chain of custody and shipping when ready.

NOTES

Field Data Sheet Instructions

A Field Data Sheet should be completed and submitted along with your other data sheets and samples *for each* sampling event. The Field Data sheet serves as the official "record" of what happened on that particular sampling event. The Field Data Sheet also serves as the pH worksheet.

1. Complete station name, station number, volunteer group (school or organization), river name, **date and time**. (Use 24-hour time method. For instance if you collected a sample at 3:05 in the afternoon, the time would be 15:05). Time and date are very important – **they are how we track your sample**.

2. Note any comments regarding weather anything that could affect the results of your collection. For example, note in the comments if you sampled after a big storm or if there is construction/drilling near your sampling location.

3. In the boxed area titled: "Samples Collection Method" these should be filled out in the field during collection. Make sure to fill out the box detailing what kind of samples you have collected. **It's important that your collections marked here match your chain of custody**. We check this box against your samples when processing, so completing this section will help to processing the sample faster. Make sure to note is this was a grab or composite sample.

4. Record results from the field and your laboratory on the appropriate line. If an analysis was not conducted, then fill in the result area with a -9 (we use -9 to indicate that the information is not available). Every line should have some piece of information in it for a complete Field Data Sheet.
5. Use this sheet for recording the results of the pH analysis and temperature measurement.
6. The Field Data sheet is used to consolidate the field analytical results from the other field data sheets:

    - Alkalinity
    - Hardness
    - Dissolved Oxygen
    - Flow

7. Make sure to sign and date ALL data sheets and keep a copy for your records and send River Watch the originals along with your samples.

8. In order to better capture our volunteer's efforts, we have added the volunteer timesheet to the field data sheet.

Field Data Sheet

Station Name _____ Station Number _____

River/Stream _____ Date of sample ___/___/___

Volunteer Group _____ Time of sample ___ : ___

Air T°/Weather/Comments:_____

_____

Sample Collection Method: **Grab** **Composite**

Samples collected for River Watch analysis: Check all that apply:

Metals	**Metals QA/QC**		**Nutrients**		**Biological**
Filtered (F)	F Blank	NF Blank	TSS/CS	NP	Macroinvertebrate
Not Filtered (NF)	F Duplicate	NF Duplicate	Duplicate		Macro QA sample
No metals			TSS/CS	NP	

PARAMETERS — RESULTS

Flow
Gauge Estimate _____ ft^3/second

River Temperature: _____ Celsius

pH buffer calibration (S.U. = standard unit)

pH 7 \ temperature: _____ S.U.\ _____ ^oC

pH 10 \ temperature: _____ S.U.\ _____ ^oC

pH sample \ ATC Temp Reading: (pH \ATC temp) _____S.U.\ _____ ^oC

Phenolphthalein Alkalinity: _____ mg/L $CaCO_3$

Total Alkalinity: _____ mg/L $CaCO_3$

Hardness: _____ mg/L $CaCO_3$

Dissolved Oxygen : _____/ mg/L_____ % Saturation

Other : _____ (unit)

Data recorded by _____ **Date recorded** _____

Volunteer Time Capture						
Name (use other side if necessary)	**Hours**	**Mileage**	**Gas**	**Equipment**	**Mailing**	**Other**

- ✓ Enter this data into the River Watch Database at http://wildlife.state.co.us/riverwatch
- ✓ Attach all original data sheets to this form and submit to River Watch, keep a copy for your files
- ✓ Record “-9” for all analysis not performed

pH Measurement Instructions

pH Supplies

The pH carrying case kit should have:

2 buffers solutions (pH 7.0, and 10.0) in small 60 ml bottles (pH 4.0 is available if needed)
1 pH meter
1 bottle with KCl solution
1 ATC (Automatic Temperature Compensation) probe - the skinny probe
1 empty bottle (for sample)
1 pH probe
Instruction manual
Teflon tape (you provide) or cap for pH probe

When testing, place the liquid waste bottle, paper towels and a squirt bottle of deionized water nearby.

pH Measurement Steps

1. Rinse the square sample bottle 2 times with sample, add sample, cap and allow sample to come to room temperature (at least 20 ^{o}C).
2. Remove Teflon tape or cap from probe.
3. Soak pH probe (not ATC probe) in KCl solution for at least three minutes prior to first use of the day.
4. Rinse the pH and ATC probes with deionized water and then dry with paper towel.
5. Rinse probes with pH7 buffer solution from the pH7 buffer squeeze bottle.
6. Place both probes into the square pH7 buffer sample bottle and lightly stir.
7. Turn the meter on [I/O] and press [C].
8. Press the [pH] key.
9. Press the [STD] key. You should see a" ▸ " displayed by STD 1.
10. When the [eye] stops blinking, note the pH reading of the buffer and record on pH 7 buffer line of Field Data Sheet.

    If it is between 6.85 and 7.15, you can proceed to the next step. If not, repeat the procedure. If after repeating, it still is not in range try replacing the buffer, re-rinse the probes and try steps 5-10 again. If it still does not work, see pH troubleshooting section.

11. Rinse probes with pH10 buffer solution from the pH10 buffer squeeze bottle.
12. Put the probes in the square pH10 buffer bottle and stir.
13. Press only the [STD] key. You should see " ▸ " displayed by STD 2.
14. When the [eye] stops blinking, read the meter and record on pH 10 buffer line of Field Data Sheet.

    If the pH reading is between 9.85 and 10.15, you can proceed to the next step. If not, try replacing the 10 buffer and repeat steps 5-14 again.

15. Rinse probes excessively with sample water.
16. Place probes in pH sample bottle and lightly stir. Press the [pH] key. When the [eye] stops blinking, read the sample pH and temperature. Continue pressing [pH] key until 2 successive readings are the same. Before recording the reading, check and see if you have four triangles, one pointing to each ATC probe, pH probe, standard 1 and standard 2. If this is true, record this pH reading in the "pH sample" line of Field Data sheet. If you do not have four triangles pointing to those four specific items, calibration did not hold or something is not well with the meter. Go to the trouble shooter guide and investigate. The reading, in theory is not defensible.
17. If taking the pH of another sample, change sample water in pH vessel. Repeat steps 16 - 17.
18. If you are done for the day, turn the pH meter off [I/O]. Rinse both probes with deionized water and dry.
19. Use the Teflon tape to cover the white line on the pH probe or cap probe. NOTE: When original tape wears out, replace it with white Teflon pipe tape.

Possible Problems with pH Meter

These instructions are in the pH manual under "Measuring pH".

If the triangle points to the ATC label, that indicates the ATC probe is plugged in and actively reading a temperature.

If the triangle does not point to the ATC label then the ATC probe is NOT plugged in and the temperature compensation default is 25 degrees Celsius. The probe will read the pH but compensate at a temperature of 25 degrees Celsius.

If [Err] displays where a temperature should, check ATC connection and report if problem persists.

[lighting bolt] Indicates the battery is low and need to be changed. See the pH manual. Request new battery when ordering supplies.

[▼] Suggests the pH probe may be malfunctioning or your standards (or calibration) is wrong. If this happens try two things:

- Soak the pH probe in KCl or pH 4.0 buffer for 10 or more minutes.
- Try a new batch of pH buffers. Empty the little bottles of old buffers. Rinse these three bottles with deionized water several times. Pour new buffers in the small bottles. Buffers can get contaminated. Now, recalibrate the meter and read your pH again.
- Make sure the pH probe is clean. Check the tip of the probe for white crystals. Make sure the hole near the top was open when you tried to read the buffers or pH.

Remember your pH meter is stupid—it will always give a reading regardless of if you perform the analysis correctly or not. You are the scientist— look at meter results and reality check them. If there appears to be a problem respond accordingly.

Check your pH reading and see if it makes sense, especially if you have seasonal data to compare. A good scientist ALWAYS checks his/her answer for plausibility.

Check the range of buffers during calibration. If the buffers do not calibrate within the acceptable ranges, change buffers and calibrate again.

River Watch will replace batteries and KCl solution. Please call or email if you are having difficulties with the pH meter.

Alkalinity Titration Instructions

When testing, place the liquid waste bottle, paper towels and a squirt bottle of deionized water nearby.

1. Complete top portion of the Alkalinity Datasheet.
2. Rinse the graduated cylinder and the "A" labeled Erlenmeyer flask once with deionized water and twice with sample water.

Part I – Phenolphthalein Alkalinity

3. Fill graduated cylinder with 50 mls of sample. Pour into the "A" Erlenmeyer flask. Record amount of sample used on line 1.
4. If known, record your pH value on line 2. Answer the question: Is pH greater than 8.3? Based on the pH value, what color do you predict your sample will be?
5. Add 15 drops of phenolphthalein indicator to Erlenmeyer flask. Answer question on line 3: Did the solution turn a faint pink? If answer is **YES**, go on to step 6 below.

   If your answer is **NO** and the sample did not turn pink, but instead turned a cloudy white or remained clear, record phenolphthalein alkalinity as 0.0 mg/L on line 5 and note this in the field data sheet comment section. It may mean the pH sample was too cold when pH was read, thus the pH reading is off slightly. Go on to Part II.

6. Self zero the pipet with H_2SO_4, (sulfuric acid). Be sure **NO** air bubbles are in the stem of the flask by releasing a few drops and self zero the pipet again. Also make sure the tip of the pipet is not crusted with H_2SO_4.
7. Place a white piece of paper under the flask. Place the flask under the pipet and add H_2SO_4 **drop by drop**. Swirl the flask after each drop. Do this until the next drop turns the solution colorless. This is your endpoint for phenolphthalein alkalinity.

   Read the pipet carefully. Record the reading on the data sheet on line 4. Starting point should have been "0".
8. Subtract starting point from endpoint. Multiply that difference by 40 (see line 5). This is the phenolphthalein alkalinity in mg/L of C_aCO_3. Record phenolphthalein alkalinity value on line 5.

   For example: endpoint = 0.7 ml, start = 0.0 ml, 0.7 ml x 40 = **14.0 mg/L** phenolphthalein alkalinity as $CaCO_3$.

You are **NOT** through; continue to Part II for BGMR alkalinity.

Part II – Total Alkalinity

9. Place 6 drops of BGMR indicator into the same "A" Erlenmeyer flask used above and swirl (color should be a turquoise). Answer the question on line 6.

a. If your phenolphthalein alkalinity was **less than or equal to** zero (< 0), automatically zero your buret with the bulb.

b. If your phenolthalein alkalinity was **greater than** zero (> 0), **DO NOT** zero the buret.

10. Place the flask under the pipet and add H_2SO_4 **drop by drop**. Swirl the flask after each drop. This reaction is relatively fast. The solution may turn pink, but return to blue. The color change proceeds from turquoise to blue-gray to a clear gray, then a pink-gray and finally a pink-peachy-pink. The color changes from blue-gray to pink-peachy-pink are usually a drop a part. **Your endpoint is the pink-gray color not the pink-peachy-pink**. Stop when you are at your endpoint (change should be gradual if you go drop by drop).

    Past the pink-gray endpoint, the solution will stay a pink-peachy-pink, regardless of any additional H_2SO_4 you add. Learn your river's color transition. A viable technique is to titrate through the endpoint color **if** you read the buret after every drop. Thus, you have a reading for every color change and can choose the best endpoint.

11. Read the pipet carefully. Record the reading on the data sheet on line 7. Starting point should have been "0".

12. Subtract starting point from endpoint. Multiply that difference by 20. This is the Total Alkalinity in mg/L of $CaCO_3$. Record total alkalinity value on line 8.

13. For example: endpoint = 2.5 ml, start = 0.0 ml, 2.5 ml x 20 = **50 mg/L** Total Alkalinity as $CaCO_3$.

14. Dispose the solution in the flask into a waste bucket or sink. Rinse out Erlenmeyer flask and graduated cylinder with deionized water and store UPSIDE DOWN.

15. Does this result make sense? You are the first point of validation, is it similar to last time, what you know, etc.? Provide any comments that help us understand what your experience was.

16. Sign and date for a complete datasheet. Copy the result to the Field Data Sheet.

Common problems

- Misreading the buret—check twice, or get a second opinion.
- Passing the endpoint because you:
  did not allow enough time between drops for reaction to occur.
  did not add one drop at a time.
- Titrating only for phenolphthalein alkalinity and forgetting to titrate BGMR alkalinity.
- Final multiplication is wrong.

Alkalinity Data Sheet

Station Name _____ Station Number _____

River _____ Date of sample ___/___/___

Volunteer Group _____ Time of sample: ____:____

PART I - Phenolphthalein Alkalinity

1. Amount of sample used (should be 50ml): _____**mL**

2. pH_____ Is pH greater than 8.3? **Yes** **No**

3. Add phenolphthalein indicator. Did solution turn pink? **Yes** **No**
   If **YES** → continue with step 4.
   If **NO** → record phenolphthalein alkalinity as 0.0 mg/L, and then go to part II.

4. Titrate from a pink to a clear, record mL of H_2SO_4 you added on the line provided.
   end point ___ mL **-** start point ___mL = ___ mL H_2SO_4 used
   0.7 mL - 0 mL = 0.7 mL H_2SO_4 used

   _____ **mL H_2SO_4**

5. Multiply mL of H_2SO_4 used by 40. Record this as the phenolphthalein alkalinity below.

   Example: 0.7 ml H_2SO_4 titrant used x **40** = 14.0 mg/L $CaCo_3$

Phenolphthalein Alkalinity (Carbonate) Result: _____**mg/L $CaCo_3$**

(Note: If you have phenolphthalein alkalinity, DO NOT rezero the buret before continuing.)

PART II - Total Alkalinity

6. Add BGMR indicator. Did solution turn blue? **Yes** **No**

9. Titrate from turquoise to pink-gray. Record ml of H_2SO_4 added.
   end point ___ mL **-** start point ___mL = ___ mL H_2SO_4 used
   2.5 mL - 0 mL = 2.5 mL H_2SO_4 used

   _____ **mL H_2SO_4**

8. Multiply ml of H_2SO_4 used by 20. This is the total alkalinity.
   Example: 2.5 mL H_2SO_4 titrant used x **20** = 50.0 mg/L $CaCo_3$

Total Alkalinity (Carbonate and Bicarbonate) Result: _____**mg/L $CaCo_3$**

Comments: _____

_____

_____

Data Recorded by: _____ Date: _____

Hardness Titration Instructions

When testing, place the liquid waste bottle, paper towels and a squirt bottle of deionized water nearby.

1. Complete the top portion of the "Hardness Data Sheet".
2. Rinse the graduated cylinder and "H" Erlenmeyer flask (flask) once with deionized water and twice with sample water.
3. Fill the graduated cylinder with 50ml of sample, and then pour into the flask, do not spill.
4. Add 15 drops of ammonia buffer to flask and swirl.
5. Place a small amount of the EBT indicator into the flask and swirl.* Place sheet of white paper under flask.

   *Use the metal scoop and to add about 1/8 inch of EBT. Remember, more can be added if needed, but cannot be taken out of the sample. The sample should be purple (magenta), you should be able to just see through the solution. The key here is to produce a consistent purple.

Answer questions 1 and 2 on the hardness datasheet.

6. Self-zero the EDTA buret. Record the stating point on line 4. Be sure air bubbles are not in the nozzle of the buret by releasing a few drops and self-zeroing the buret again. Also remove any crust that may be on the tip of the buret.
7. Place flask under EDTA buret and add EDTA drop-by-drop. Swirl the flask after each drop. Be sure to give yourself plenty of time between drops to swirl the flask sufficiently. Keep adding a drop at a time until the next drop turns the solution from purple to a blue.

   This is a slower reaction than alkalinity, thus needs more time in between drops to react. This solution should stay blue, and if not add another drop of EDTA. The shade of "blue" will correlate to the purple. If your purple was dark, the blue will be dark blue. Likewise, if the purple was light, the blue will be light. The first blue you see is your endpoint.

8. Read the buret carefully and record the end point on line 4. Subtract the starting point from the endpoint (The starting point should have been "0"), and record the milliliters of EDTA used line 4.
7. Multiply the milliliters of EDTA used by 20. This is the total hardness in mg/L of C_aCO_3. Record the hardness result value in line 5.

   Example: if the endpoint = 7.4 mL and the start = 0.0 mL, so the difference is 7.4 mL. Now multiply 7.4 mL x 20 to get 148 mg/L hardness as C_aCO_3.

8. Dispose the solution in the flask into a waste bucket or the sink. Rinse out Erlenmeyer flask and graduated cylinder with deionized water and store UPSIDE DOWN.
9. Does this result make sense? You are the first point of validation, is it similar to last time, what you know, etc.? Provide any comments that help us understand what your experience was.
10. Sign/date for a complete datasheet. Copy the result to the Field Data Sheet.

Hardness Data Sheet

Station Name _____ Station Number _____

River _____ Date of sample ___/___/___

Volunteer Group _____ Time of sample: ____:____

---

1. Amount of sample used (should be 50ml): _____**mL**

2. Add ammonia buffer and EBT indicator.
   Did solution turn purple? **Yes** **No**

3. Titrate from purple to first drop changes solution to blue.

4. Record the mL of EDTA you added.
   end point ___ mL **-** start point ___mL = ___ mL EDTA used
   7.4 mL - 0 mL = 7.4 mL EDTA used _____ **mL EDTA**

5. <u>Multiply mL of EDTA used by 20</u> to get the Total Hardness result, and record below.

   Example: (7.4 mL EDTA titrant used) x 20 = 148.0 (mg/L) total hardness as $CaCO_3$

Total hardness _____ **(mg/L) $CaCO_3$**

Comments: _____

_____

_____

Data recorded by _____ Date recorded _____

Common mistakes

- Misreading the buret—check twice, get a second opinion.
- Purple is too deep or dark making endpoint hard to see and reaction not accurate.
- Participant does not allow enough time between drops for reaction to occur.
- Participant forgets to use ammonia buffer, color changes will never occur.
- Final multiplication is wrong.

Dissolved Oxygen Winkler Titration Instructions

Safety
The Winkler titration test uses a number of potentially hazardous chemicals, please take care the chemicals **do not come into contact with eyes, skin,** or clothes - wear safety glasses and rubber gloves.

When testing, place the liquid waste bottle, paper towels and a squirt bottle of deionized water nearby.

- Alkaline **potassium iodide azide is a strong base and can cause severe burns.**
- The **azide is very poisonous**.
- **Sulfamic acid can cause eye burns** and can cause skin and respiratory tract irritation
- Manganous sulfate can irritate eyes and skin.

Standard Winkler Titration Method Dissolved Oxygen

In the Field

1. Record the temperature of the river on line 1 of datasheet.

2. Rinse 300 mL, BOD in sample water.

3. Collect a water sample the BOD bottle. Submerge the bottle and hold at a 30-45 degree angle. Overflow the bottle for one to two minutes to remove any trapped air bubbles.

4. Add 1 ml Manganese Sulfate Solution and 1 ml Alkaline Iodide-Azide Reagent (wearing gloves and goggles).

5. Immediately insert the stopper so that no air is trapped in the bottle. Invert several times to mix.

6. A flocculent precipitate will form. It will be orange-brown if oxygen is present or white/pale yellow if oxygen is absent.

7. Wait until the floc in the solution has settled at least half way down the bottle, invert again and let the floc settle again before moving on to next step.

8. Remove the stopper and add the contents of one Sulfamic Acid Powder Pillow.

9. Replace the stopper without trapping air in the bottle and invert several times to mix prepared sample.

   The floc will dissolve and leave a golden/yellow color if oxygen is present. The sample is now ’fixed” and needs to be titrated within 8 hours (kept in a cold dark place).

In the Lab

1. Rinse the 500 ml Erlenmeyer flask and graduated cylinder with deionized water.
2. Measure 200 ml of the prepared sample using a graduated cylinder, then pour into the 500 mL Erlenmeyer flask.
3. Rinse and fill the 25 mL buret with 0.025 N Sodium Thiosulfate ($Na_2S_2O_3$), by filling the buret to the 5 mL mark. Let 3 mL out to the 8 mL mark. If you go past 8 mL, fill back to 8 mL with more $Na_2S_2O_3$.
4. Record starting point on line 2 of Winkler Dissolved Oxygen datasheet.
5. Titrate with $Na_2S_2O_3$ to the prepared sample drop-by-drop, swirling the flask until the sample turns a pale, straw yellow color.
6. Compare color to the remaining sample in the BOD bottle. If solution in Erlenmeyer flask is more gold than yellow, add more Sodium Thiosulfate.
7. Add 5 - 20 drops of Starch Indicator Solution, enough drops to make a dark blue or green or brown, point is that it is dark enough to see the solution turn colorless. If 5 drops work, stop. If 20 drops do not work, change the starch.
8. Continue to titrate with Sodium Thiosulfate from the dark blue to colorless or clear endpoint. Watch out for floating particles that may stay colored when solution is clear.
9. Record end point on line 3 of Winkler Method Dissolved Oxygen datasheet.
10. Calculate by subtracting starting point from end point, and record mL dissolved oxygen on line 4 of Winkler Dissolved Oxygen datasheet.
11. 1 mL titrant used equals 1 mg/L dissolved oxygen.
12. Calculate the percent saturation of dissolved oxygen, using the chart on the datasheet.
13. Find your water temperature on the top scale and dissolved oxygen value on the bottom scale.
14. Draw a straight line between the water temperature and dissolved oxygen measurement (oxygen mg/liter).
15. Read the saturation percentage at the intercept on the sloping scale.
16. Record the percent saturation on the percent saturation line.
17. Drain buret, Erlenmeyer flask, graduated cylinder. Then rinse with deionized water and store upside down or store buret upright with remaining $Na_2S_2O_3$ and place foil or some other closable plastic to cover opening.
18. Provide any comments that help us understand what your experience was.
19. Sign and date for a complete datasheet. Copy the result to the Field Data Sheet.

Dissolved Oxygen Winkler Data Sheet

Station Name _____ Station Number _____

River _____ Date of sample __/___/____

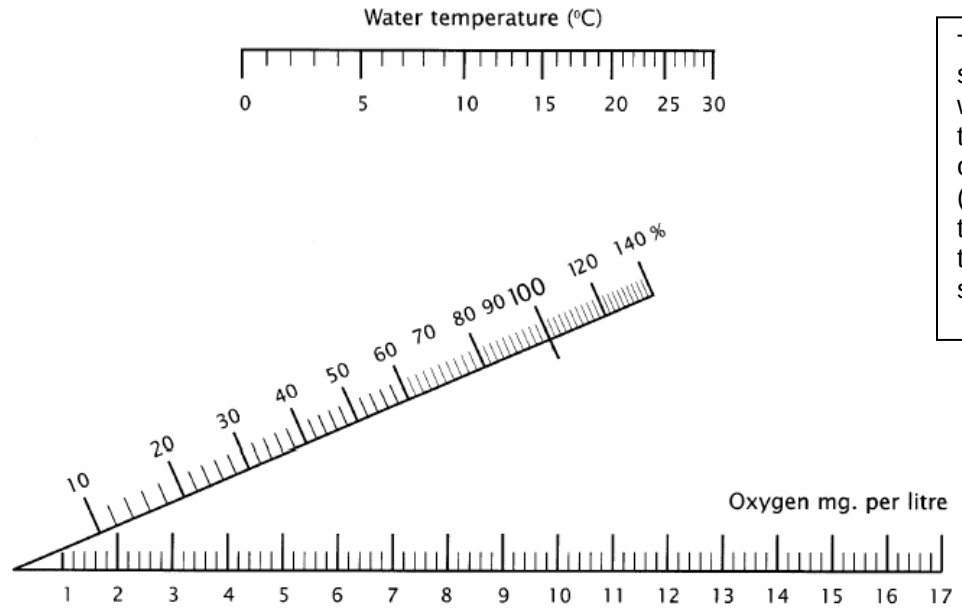
Volunteer Group _____ Time of sample ___ : ___

---

1. River temperature _____ **° Celsius**
2. Titrate start point (yellow) _____ **ml**
3. Titrate end point (clear) _____ **ml**
4. Subtract starting point from end point: # of mL at start point - # of mL at end point = milliliters of titrant used

   milliliters of titrant used = mg/L dissolved oxygen _____ **mg/L D.O.**

5. Determine the percent saturation of dissolved oxygen using the chart below.



To use the chart, draw a straight line between the water temperature at the test site and the dissolved oxygen measurement (Oxygen mg/L), and read the saturation percentage at the intercept on the sloping scale.

Percent Saturation_____**%**

Comments: _____

_____

Data recorded by_____ Date recorded _____

Stream Velocity and Discharge

How much water in the stream that is diluting a particular pollutant concentration is an important piece of information to add to our story. Whenever possible we want to locate a station by a USGS or State Engineer Stream Gauge. Instructions to access this flow data are included below. For educational purposes the following activity is provided to help understand how velocity and flow is measured and how it might influence chemical, physical and biological components of a stream ecosystem.

Stream discharge can be measured directly using the methods listed below. At some stations there are stream gage stations. Using the methods described in the Retrieving Stream Flow Gage Data from the Internet for River Watch Sampling section, the data can be downloaded from the internet.

To measure discharge you need to know the velocity of the cross sectional area of the stream. The velocity is the speed the stream is moving. The cross sectional area over a given distance is the volume of the stream area. By multiplying the stream velocity by the stream volume you get the amount of water that is moving past a given point at a given time or discharge. Discharge is normally measures in cubic feet of water moving past a point in one second (cubic feet per second or CFS).

Stream Velocity

Ideally pick a 50-100 yard section of stream that is a free flowing riffle without any “hang-up” areas. Measure the exact length in feet and record.

Place (gently release) at the surface of the water an orange or other floatable object. Another participant will be downstream at the end of the station to catch the orange or floatable object.

Another participant records time between release and capture of the orange or floatable object. Measure the time travel over the known distance at lease three times, and record time. Average the time of the three and record.

Divide the distance the object floated (in feet) by the average time (in seconds) to get stream velocity (feet/second).

Discharge

Pick a representative stream reach preferably include the stretch used for velocity. At the beginning, middle, and end of the segment, measure and record the wet stream width.

At those same three locations, measure and record the depth, 1/4, 1/2, and 3/4 across the channel. You should have nine depth measurements. Average the nine depths and record.

Choose a bottom stream type.

Take the discharge formula and plug in the appropriate values. You will need a velocity value and average travel time from your velocity test.

Complete the math and you have discharge in cubic feet per seconds.

Stream Velocity Data Sheet

Station Name _____ Date of survey ___/___/___

River/Stream _____

Volunteer Group _____

---

1. Starting point description: _____
2. Ending point description: _____
3. Distance in between _____ feet
4. Seconds for orange to travel: First time _____ seconds

   Second time _____ seconds

   Third time _____ seconds

   Average _____ seconds
5. Distance in between stations

   Average number of seconds equals _____ feet/second

Continue to the Stream Discharge Data Sheet

Comments:

_____

_____

Data recorded by_____ Date recorded _____

Stream Velocity & Discharge Data Sheet

Station Name_____ Station Number _____ Date of survey ___/___/___

River_____ Volunteer Group_____

1. Travel distance of floatable object _____ feet
2. Travel time of floatable object: First time _____ seconds
   Second time _____ seconds
   Third time _____ seconds
   Average _____ seconds
3. Velocity (divide travel distance by travel time) **(v)** _____ **feet/sec**
4. Channel width: at beginning of segment _____feet
   at middle of segment _____feet
   at end of segment _____feet
   Average (w) _____**feet**
5. Channel depths:

	beginning of segment	middle of segment	End of segment
1/4 across	feet	feet	feet
½ across	feet	feet	feet
3/4 across	feet	feet	feet

Average of average depths (d) _____**feet**

6. Stream bottom type (choose one) **(a)**
   a. (0.8) rough, loose rocks, coarse gravel
   b. (0.9) smooth, mud, sand, hardpan rock
7. Discharge calculation:

$$r = v*w*d*a$$

r = discharge in cubic feet per second.
v = velocity of stream in feet per second**.**
w = average width of channel section tested (average of three measurements above).
d = average depth in feet (use average from nine measurements)
a = constant whose value depends on the nature of the stream bottom:

8. Stream Discharge _____$feet^3$/second

Data recorded by_____ Date recorded _____

Retrieving Stream Flow Gage Data from the Internet

The Colorado Water Resource Division and the U.S. Geologic Survey Water Resource Division maintain a couple of hundred stream gauge stations throughout Colorado. To access all the active stream flow gage data go to http://www.dwr.state.co.us.

The first step is to determine if there is a stream flow gage in the vicinity of your River Watch monitoring station. For most of the stream flow gage stations, there is only a name given to indicate which stations are gauged. You can view the list on the list on the Water Resource website. Contact the River Watch office if you are unable to determine whether a stream flow gage station is near your monitoring site (303-291-7412).

There are many River Watch monitoring stations that do not have stream flow gage monitoring stations near them. But if one is nearby, the flow data can be used to calculate load rates in pounds per day (concentration X flow rate = load). Load is used extensively in managing pollutant discharges to streams.

Steps to downloading flow data

Go to the website http://www.dwr.state.co.us and locate the appropriate stream flow station from the list. You can narrow your search by the selecting only the stream flow stations in the county or water division your monitoring station is located in. Bookmark your stream flow station site.

If the stream flow station is operated by the Colorado Water Resource Division, follow the steps below. If they station is operated by U.S. Geologic Survey follow the second set of directions.

Colorado Water Resource Division

1. Select your station from the list of stations, and click on the retrieve button. The screen will show an instantaneous flow and a graph of flows over the last 10 days.

2. Select the “Retrieve self-timed tabular data” hotlink.

3. Data is available on 15 minute time increments for the last three days. Find the stream flow under the “DISCHRG” column closest in time to when you sample was collected. This is the stream discharge in cubic feet per second (cfs).

4. Record result on Field Data Sheet, and check the “gage” box.

5. Data older then three days is not available online.

U.S. Geologic Survey, Water Resource Division

1. Select your station from the list of stations, and click on the retrieve button. The USGS symbol will appear in the upper left corner along with data about the station and a graph of stream flow from the last seven days.

2. In the “Output format” dropdown menu, select “Table”. You can get data up to 31 days old, by typing “31” the “days” window, then select the “get data” button.

3. Data is available on 15 minute time step for the last three days. Find the “stream flow”, this is closest in time to when you sample was collected. This is the stream discharge in cubic feet per second (cfs).

4. Record result on field data sheet and check the “gage” box.

5. Data older then three days is available online two different ways.

6. If the stream flow data you need is between 31 and 370 old then go to the “Available data for this site” box and select “Recent Daily” from the pull down menu.

7. In the “Output format” dropdown menu, select “Table”. You can get data up to 730 days old, by typing “730” the “days” window. Select the “get data” button.

8. Data is available by the “Mean Daily Value”. Find the day, month, and year you sampled and record the result on the Field Data Sheet and select the “gage” box. This is the stream discharge in cubic feet per second (cfs).

9. For data older then 730 days go the “Available data for this site” box and select “Surface-water: Daily stream flow” from the pull down menu.

10. In the “Retrieve Daily stream flow data for Selected Sites” section fill in the date(s) for your sample collection date, select the button for “Tab-separated data”, then select “display in browser” from the dropdown window, and push the “submit” button.

Look for the flow data for the sample collection date under the “dv_cd” column. Record the result on the Field Data Sheet and check the “gage” box. This is the stream discharge in cubic feet per second (cfs).

Chain of Custody Form

A chain of custody is a quality control measure. It is a form that tracks the custody of a sample from its birth (collection) to its death (analysis) and helps ensure that no tampering or contamination occurred along that pathway. A chain of custody is required for samples shipped or delivered to Denver.

For each unique sampling event (station number plus date plus time), record the station name, station number, date, time and check the appropriate boxes that describe what samples are present (filtered, not filtered, filtered blank, etc.) for metals and nutrients (TSS, NP) for that specific sampling event.

An effective way to complete the chain of custody is to lay out all the samples you are shipping on a table in order of date. Complete the chain from the actual sample containers versus the data sheets. While you are completing the chain for each sample, check the associated datasheets for accuracy and completeness. This method almost guarantees what is on the chain actually exists and is in the cooler.

Place these forms in a plastic bag along with your data sheets in the cooler. Samples are not valid without a chain of custody form and will not be processed without completed data sheets. Refer to shipping instructions to complete shipping tasks.

Chain of Custody

Volunteer Group ______________________________ Date shipped _______ /_____/______

Shipped by ______________________________ Signature _________________________

Field Data Sheets included - Yes No Field Data entered via web - Yes No

F = filtered NF= non-filtered FB = filtered blank NFB = non- filtered blank FD = filtered duplicate NFD = non-filtered duplicate

Station Name	Station Number	Date	Time	Metal Samples	Nutrients	Macro Sample
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___
				Filtered (F) _____ Non-Filtered(NF)_____ F Blank _____ NF Blank _____ F Duplicate _____ NF Duplicate _____	TSS / CS___ NP___ Duplicate TSS / CS___ NP___	Yes ___ QA sample___

Nutrients unpreserved (32 ounce jug): **TSS** = total suspended solids and **CS** = Chloride, Sulfate
Nutrients preserved with H_2SO_4 (8 ounce cylinder): **NP** = Nitrate + Nitrite, Ammonia, Total Phosphorous

Total number of metals bottles in cooler _____
Total number of nutrient bottles ____
Total number of macroinvertebrate samples ____

Ship samples to River Watch, 6060 Broadway, Denver Colorado 80216.

River Watch Staff Section

Total number of metals bottles received _____ Date Samples Received _______
Total number of nutrient bottles received _____
Total number of macroinvertebrate samples received _____ Received by ______________________________

Shipping Metals, Nutrient or Macroinvertebrate Samples

- Copy all data sheets for your records. Remember all originals must be shipped with your samples.
- After you have copied your data sheets, enter data online if possible, then ship.
- Place completed chain of custody form along with data sheets in a Ziploc bag.
- Ensure all bottles being shipped are tightly closed.
- Check the samples are labeled correctly.
- Place samples in a sealable bag.
- Place enough absorbent material in the sealable bag (newspaper, etc) to absorb the contents of the largest container.
- Seal the bag and place in the cooler.
- For nutrients put frozen blue ice or enough ice to last 48 hours in the cooler.
- For metals and macroinvertebrates blue ice is not needed.
- Surround the samples and fill the empty space with packing paper or newspaper so the samples do not roll around.
- Check supplies, if need any that require shipping a sample bottle, now is a good time to include it, include a bottle and a request for equipment slip.
- Secure the cooler closed with packing tape. This will inhibit tampering during shipment.
- Use the mailing labels provided.
- If a problem arises with shipping, please notify River Watch. Whenever asked by anyone “What are you shipping?” reply “Water samples.”
- Do not ship samples on a Thursday or Friday. They will get stuck in a warehouse over the weekend.

When to Ship

Each volunteer group is asked to ship their metals samples at least once every quarter. The holding time for metal samples is 6 months. Shipping metal samples on a quarterly basis allows maximum time for River Watch to process and analyze the data.

Nutrients should be shipped within 48 hours of collection, with enough ice to last for 48 hours. Do not ship on Fridays or on a day when the sample would arrive on a Saturday or Sunday. The holding time for the nutrients is 28 days.

Macroinvertebrate samples should be shipped within two weeks of collection. Enclose any alcohol that was not used.

Mailing Labels

FROM: **Name:**_____
Organization:_____ **Kit#**_____
Address:_____
City, State, Zip:_____

To: Colorado Division of Wildlife
River Watch
6060 Broadway
Denver, CO 80216

FROM: **Name:**_____
Organization:_____ **Kit#**_____
Address:_____
City, State, Zip:_____

To: Colorado Division of Wildlife
River Watch
6060 Broadway
Denver, CO 80216

FROM: **Name:**_____
Organization:_____ **Kit#**_____
Address:_____
City, State, Zip:_____

To: Colorado Division of Wildlife
River Watch
6060 Broadway
Denver, CO 80216

Web Enabled Data Entry

As part of your performance criteria you need to enter and submit your data at least once per quarter in a contract year. More is appreciated. Data entry by you allows our staff to focus scarce resources on more sample analyses, reporting, site visits and the like.

Along with electronic entry of your data, you are required to keep a copy of the hard data and send us the original copies. We validate your data entry from the original copies (a quality control measure) as well as the actual results.

1. Once you have completed filling out your Field Data Sheet, or prior to shipping samples go the River Watch website and enter your data into the electronic data forms (http://wildlife.state.co.us/riverwatch).

2. Go to the "Data" page and enter in your kit number and password as a login procedure. If you do not know your kit number and password, please contact a River Watch staff person phone or by email.

3. Once you have logged in, select your station for data to enter and complete the datasheet online from your paper field datasheet. We encourage you to do this as part of every sampling event equal to the titrations.

4. If you did not collect information for a particular parameter (i.e. flow) than please write '-9' in allotted space and leave that space blank when entering online. When finished, hit the "submit" button and your data is sent to our database.

5. Copy all data sheets.

6. Mail all original data sheets with your samples.

7. If you do not have any samples to ship, (for example all you have is field data), no metals, nutrients or macroinvertebrates, you still need to enter the data electronically, copy the datasheets and send originals.

NOTES

Photographic Record of Your Stations (Optional)

Completing a photographic record of your River Watch station(s) could prove valuable in the future. The object is to document change over time—geologic time. The beach erosion that has occurred on the Colorado River in the Grand Canyon is an excellent example that illustrates the invaluable importance of such documentation. The large beaches present in the canyon have been eroding away since the Glen Canyon Dam/Lake Powell was built. This was due to the extreme flow fluctuation due to dynamic releases from the reservoir. These extreme fluctuations are no longer allowed because old photographs documented the way the beaches used to be. A new water release plan, the result of extensive research, is in place now with the intent to restore old beaches and reduce erosion of existing beaches.

This photographic record will become part of history, as will your water quality data. When photographing your station, pick a transect that can be repeated. Sometimes it helps to mark the transect with stakes or flags. Pick a consistent direction/angle to take the picture and include large permanent structures if possible. The following information should be included with every photograph. Use the form on the following page.

- Station name and location
- Direction of photograph (upstream, downstream, etc.)
- Date of photograph
- Time of photograph
- Type of film/exposure or digital
- Time of year
- Relevant comments

Digital photographs are preferable, and pictures can be emailed to River Watch. If your volunteer group does not have a digital camera, River Watch can send you a disposable camera, that can be returned for processing after the pictures are taken.

Photographic Record Data Sheet

Station Name_____ Date of picture ___/___/___

River_____ Station number:_____

Volunteer Group_____

Upper Terminus:
1. Location T_____ R_____ S_____

2. Elevation _____ feet

Lower Terminus:
Location T_____ R_____ S_____

Elevation _____ feet

Approximate flow: _____ Approximate width:_____

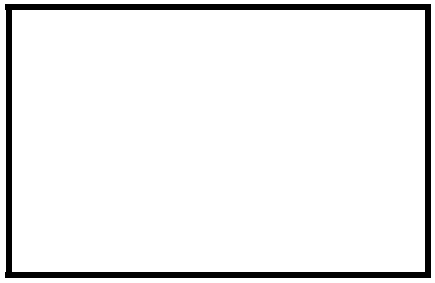
Station description
_____

_____

Photo: Film: _____ Exposure _____

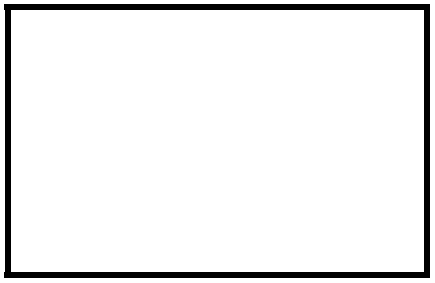
Time picture taken: _____ Time of year (season) _____

Name of photographer: _____
Diagram of Angle



Circle one:

Flow Direction:
Place a ☆ to show where you stood when you took the picture.



Attach photo here.

Comments: (comparison with other photos, weather conditions, etc.)
_____

Fish Information

The following worksheets are provided as an exercise to illustrate how CDOW uses fish data. In this lesson you may want to contact our local fisheries biologist to either demonstrate how fish are collected or to give a presentation on the electro fishing methodology, why the data is useful and how it is used. If a biologist cannot do the above, ask for current data available near your station. Finally, if no information is available use the numbers provided in the example or make up realistic numbers. The enclosed example is real data.

Once raw data is obtained, perform the following analyses just as CDOW biologists. The analysis includes calculating number of fish per acre, pounds of fish per acre and a length frequency distribution.

The number and pounds of fish per acre provide an estimate for total species or individual species over an area. Biologists use this information to make fishery management decisions. Some examples of those type of decisions include what species to stock if any, what size to stock, where to stock, should a species be introduced or eradicated, has a local habitat improvement project or water quality clean-up project impacted (positively or negatively) a particular population?

A length frequency distribution graphically displays the number of fish collected in each length category. This distribution assists in determining the various ages present of a certain species or for all species. It may also indicate whether reproduction has occurred. This distribution can infer relative growth if compared with other distributions from the same site earlier in time. Take the raw data in this example or data you have obtained and in Quattro Pro on your computer, compute a length frequency distribution, and then graph that distribution.

In the example of brown trout, there are probably three age classes of fish present and no young-of-year (1-year old fish). Young-of-year are generally 20-50 mm. or 1-2 inches. One-two year old fish might be represented here as the 100-180 mm. fish or 4-7 inches. Larger fish could be 2-5 plus years old. The only sure age assessment would be analyzing fish scales. As fish get older, the length frequency distributions overlap.

Fish Population and Biomass Data

Station Name_____ Station Number _____

River_____ Date of survey ___/___/___

School_____

Station Description_____

SUMMARY DATA FROM FIELD COLLECTION

1. Length of electrofished stream reach: _____feet

2. Widths of electrofished stream reach were:

a. _____ feet b. _____ feet c. _____ feet d. _____ feet

3. Average stream width
Add widths in step 2 (a, b, c and d) and divide by 4 _____feet

4. Total weight of captured fish _____grams

5. Total number of fish captured _____fish

6. Average weight of fish captured
Divide the sum in number 4 by the sum in number 5 (above) _____grams

7. Total number of fish captured in the first pass _____fish

8. Total number of fish captured in the second pass _____fish

SEBER LE CREN TWO PASS POPULATION ESTIMATE

9. Use the following equation to calculate fish populations:

$$N = \frac{(C_1)^2}{(C_1 - C_2)}$$

N = Population estimate (number of fish present—this is what we are looking for).
C_1 = Total catch on the first electrofishing pass (step 7 above)
C_2 = Total catch on the second electrofishing pass (step 8 above)

N = fish _____fish

This population estimate, N, needs to be related to the area that was electrofished and then related to a standard unit of area in order to compare results between stations.

Total area electrofished equals the length (step 1 above) multiplied by the average width (step 3 above) or step 3 x step 1 = $feet^2$

_____ x _____ = _____feet^2

Use the following formula to estimate the number of fish present per standard unit of area; here, we used an acre:

43,560 feet^2
Acre = 43,560 per acre = _____ Number of sampling units

Area electrofished feet^2 (Step 10)

12. Use the following formula to estimate the fish population:

N (Step 9) x sampling units (Step 11) = number of fish = _____ fish per acre
Acre per acre

BIOMASS ESTIMATE

13. Average weight of fish (step 6) times the number of fish, per acre, (step 12).

_____ x _____ = _____total grams per acre

14. Use the following formula to calculate total pounds:

Total grams per acre (Step 14) _____total pounds per acre
454 grams per pound

Comments_____

_____

_____

Data recorded by_____ Date recorded _____

EXAMPLE DATA SHEET

FISH POPULATION AND BIOMASS DATA

Station Name Above Alpine Lake Station Number _____

River Chalk Creek Date of survey __/__/__

School Blank HS

Station Description This Analysis is for brown trout only.
(Could be completed for brook trout only or total trout)

SUMMARY DATA FROM FIELD COLLECTION

1. Length of electrofished stream reach: 724 feet
2. Widths of electofished stream reach were:
   a. 35 feet b. 30 feet c. 37 feet d. 28 feet
3. Average stream width
   Add widths in step 2 (a, b, c and d) and divide by 4 32.7 feet
4. Total weight of captured fish 3.608 grams (brown only)
5. Total number of fish captured 45 fish (brown only)
6. Average weight of fish captured
   Divide the sum in number 4 by the sum in number 5 (above) 80.18 grams
7. Total number of fish captured in the first pass 61 fish
8. Total number of fish captured in the second pass 14 fish

SEBER LE CREN TWO PASS POPULATION ESTIMATE

9. Use the following equation to calculate fish populations:

$$\hat{N} = \frac{(C_1)^2}{(C_1 - C_2)}$$

$\hat{N}$ = Population estimate (number of fish present—this is what we are looking for).

C_1 = Total catch on the first electrofishing pass (step 7 above) $c_1 = 36$

C_2 = Total catch on the seconf electrofishing pass (step 8 above) $c_2 = 9$

$\hat{N}$ = fish 45 fish

This population estimate, $\hat{N}$, needs to be related to the area that was electrofished and then related to a standard unit of area in order to compare results between stations.

10. Total area electrofished equals the length (step 1 above) multiplied by the average width (step 3 above) or step 3 x step 1 = feet^2
    32.7 x 724 = 23674.8 feet^2

11. Use the following formula to estimate the number of fish present per standard unit of area; here, we used an acre:

$$\frac{\frac{43{,}560 \text{ feet}^2}{\text{acre}}}{\text{Area electrofished feet}^2 \text{ (step 10)}} = \frac{43{,}560}{\text{(step 10)}} = \underline{1.839}$$ Number of sampling units per acre

↳ 236 74.8

12. Use the following formula to estimate the fish population:

$$\hat{N} \text{ (step 9)} \times \frac{\text{sampling units}}{\text{acre}} \text{ (step 11)} = \frac{\text{number of fish}}{\text{per acre}} = \underline{82.75}$$ fish per acre

45 × 1.839 =

BIOMASS ESTIMATE

13. Average weight of fish (step 6) times the number of fish, per acre, (step 12).

80.18 x 82.75 = 6,635 total grams per acre

14. Use the following formula to calculate total pounds:

$$\frac{\text{Total grams per acre (step 14)}}{\text{454 grams per pound}}$$ 14.6 total pounds per acre

Comments Suggestions: 1) complete this exercise with brook trout only, then with all trout together and compare numbers. (sample data is enclosed).
2) what do these numbers mean?
3) collect similar data from your stream and do this analysis

Data recorded by _____ Date recorded _____

RAW DATA

Stream Widths 35' 30' 37' 28'

COLORADO DIVISION OF WILDLIFE
FISH SAMPLING SURVEY

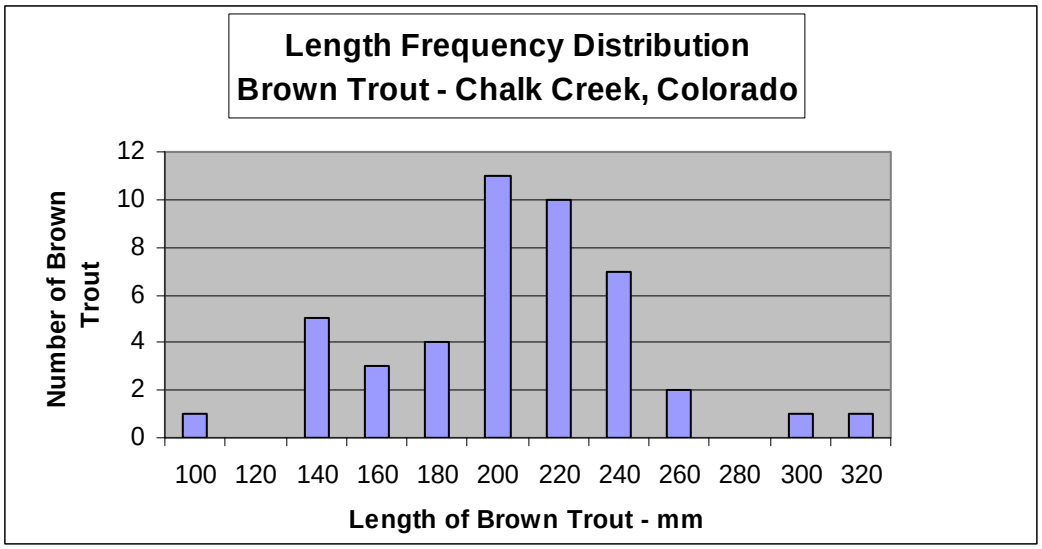
WATER Chalk Creek
SAMPLE METHOD Electrofish
DATE 10/29/93 CREW DOW
DISTANCE 724' PASS (1) 2
STATION NUMBER 3
STATION LOCATION Above Alpine Lake

SPECIES	LENGTH (mm)	WEIGHT (g)	SPECIES	LENGTH (mm)	WEIGHT (g)	SPECIES	LENGTH (mm)	WEIGHT (g)
Brown	258	134	Brown	225	111	Brook	182	61
L	199	68	L	306	292	B	137	27
	189	69		270	108		233	116
	191	61		233	105		168	39
	160	41		230	109		98	9
	199	73		205	89		121	18
	122	17		186	70		178	18
	161	38		203	72		109	11
	251	152		203	74		107	9
	195	78		235	102		151	30
	167	44		293	130		203	72
	222	105		212	101		172	42
	132	63		205	95		117	13
	215	84		214	93		159	39
	182	58					96	9
	190	61					108	10
	183	64					108	11
	185	57					109	9
	135	20					101	8
	228	127					105	8
	132	22					65	3
	150	32					163	36

Brook ↓

Length	Weight
203	6
212	88
174	49

Total 1+2 = 61+19 = 75 Total Fish 1st Pass = 61
Total L 1+2 = 36+9 = 45 Total Brown 1st Pass = 36
Total B 1+2 = 25+5 = 30 Total Brook 1st Pass = 25



Quick Fish Fact Sheet

by Ashley Rollings and Julie Wilson, CWN

Brown Trout

Characteristics: The average length of the brown trout is 16 inches and generally they weigh only a few pounds. In streams the coloring is a light brown with silvery sides and pronounced black spots on the back whereas in large lakes or in the sea the overall coloration is silvery.

Habitat: abundant from high mountain streams to broad rivers flowing onto the plains. These are a cold water fish that require cobble substrate in order to spawn and feed.

Cutthroat (Native) Trout

Characteristics: Cutthroat trout can be distinguished from rainbows by heavier spotting toward the tail and the presence of a red slash on their “throat.”

Habitat: These fish are found in high mountain streams and lakes. These are a cold water fish that require cobble substrate in order to spawn and feed.

Smallmouth Bass

Characteristics: The best way to distinguish the smallmouth from its cousin, the largemouth bass, is by the “smallies” jaws that does not extend beyond the eye. They maintain broken vertical lines on their sides and many have a reddish eye. Smallies are frequently caught along rip-rap shorelines with small jigs or crayfish imitations, and can be a great fish for impatient kids who may need a lot of action.

Habitat: Introduced to Colorado in 1951, small-mouth have been stocked in warm- and cool-water reservoirs and lakes in many parts of the state.

Green Sunfish

Characteristics: This fish is similar in appearance to the bluegill, but has a larger mouth and is olive in color with short, rounded pectoral fins and yellow trim on the fins.

Habitat: This stocky fish is found in both streams and impoundments and spawns in shallow areas from June to mid-August. Like most sunfish, this sporty panfish can be taken with crickets, worms, and other bait rigged under a bobber, or with small lures, jigs, and flies.

Colorado pikeminnow *(Ptychocheilus lucius)*

Characteristics: The Colorado pikeminnow (formerly Colorado squawfish) is a torpedo-shaped fish with an olive-green and gold back, silver sides and white belly. These fish spawn between late June and early September and when they are 5-6 years old and at least 16 inches long.

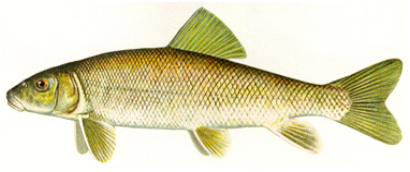
Habitat: The Colorado pikeminnow thrives in swift flowing muddy rivers with quiet, warm backwaters.

Brassy Minnow

Characteristics: body scales have about 20 lines that fan out from the focus of the scale; color dark dorsally, shading to yellowish-brassy sides, creamy belly.

Habitat: These fish are found in the Eastern Plains of Colorado. The brassy minnow prefers habitat that has slow moving water and lots of stream cover such as tumbleweeds. These fish eat small macroinvertebrates, algae and periphyton from the stream bed.

White Sucker



Identifying characteristics: Single dorsal fin, sucking mouth with no barbels, long cylindrical body. The white sucker has coarser scales, the longnose has fine scales, but visually, with only one in hand, they are difficult to distinguish.

Habitat: Suckers are mainly found in areas with deep slow moving water. These fish mainly feed on algae and macrophytes on the substrate.

Long Nose Dace

Identifying characteristics: olive-green to brown on back and upper sides shading to white on the belly, mottled appearance due to presence of darkened scale pockets. Adults get to be about 4-5 inches in length.

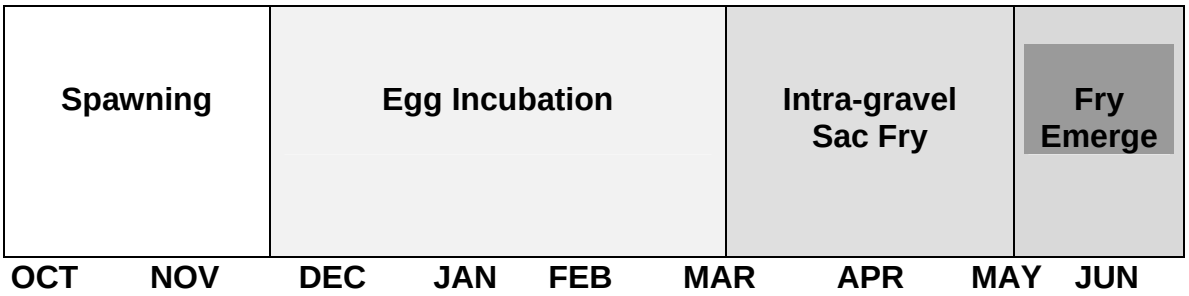
Habitat: These fish are mainly found in areas with swift moving shallow waters with cobble bottoms. These fish eat insect larvae and small macroinvertebrates.

Fathead Minnow

Identifying characteristics: mouth small and terminal, rounded blunt snout, first dorsal ray of the dorsal fin is shorter, thick and split away from remainder of the fin.

Habitat: Fathead minnows are tolerant of extremes in environmental conditions, able to withstand high temperatures, high nutrient concentrations, low dissolved oxygen levels, high turbidity and fairly stagnant pools.

The Affect of Flow on Fish



Spawning — High flows during this period cause trout to spawn on gravel that could be dewatered at normal water levels.

Egg Incubation — A drastic stream flow reduction during this period dewater can completely dewater the spawning redds.

Sac Fry in Gravel — A stream flow reduction that dewaters the spawning redds after the eggs have hatched, but before the fry have emerged, is lethal.

Fry emerge — High stream flows during this period can cause high fry mortalities since it reduces the amount of low velocity areas in the stream**.**

Table of approximate time and duration of spawning, and critical early development life stages for brown and rainbow trout in 11 physical habitat simulation study streams.

River	Species	Adult Spawning	Egg Incubation	Egg Hatching	Fry Emergence
Arkansas	brown	10/15 – 11/15	10/15 – 4/1	3/1 – 5/1	4/1 – 6/1
Blue	brown	10/15 – 11/15	10/15 – 6/1	4/1 – 6/1	5/15 – 7/1
Cache La Poudre	brown	10/15 – 11/15	10/15 – 6/1	4/1 – 6/1	5/15 – 7/1
Cache La Poudre	**rainbow**	4/15 – 5/30	4/15 – 7/15	6/15 – 7/15	7/1 – 8/1
Colorado	brown	10/15 – 11/15	10/15 – 4/1	4/1 – 6/1	5/15 – 6/15
Colorado	**rainbow**	4/15 – 4/30	4/15 – 6/15	6/1 – 7/1	6/15 – 7/15
Frying Pan	brown	10/15 – 11/15	10/15 – 5/1	4/1 – 6/1	5/15 – 6/15
Frying Pan	**rainbow**	4/1 – 5/1	4/1 – 6/15	6/1 – 7/1	6/15 – 7/15
Gunnison	brown	10/15 – 11/15	10/15 – 4/1	3/15 – 5/15	5/1 – 6/15
Gunnison	**rainbow**	4/1 – 5/1	4/1 – 6/15	6/1 – 7/1	6/15 – 7/15
Rio Grande	brown	10/15 – 11/15	10/15 – 5/1	4/1 – 6/1	5/15 – 6/15
S Fk Rio Grande	brown	10/15 – 11/15	10/15 – 6/1	5/1 – 7/1	6/1 – 7/15
S Platte	brown	10/15 – 11/15	10/15 – 5/1	4/1 – 6/1	5/1 – 6/15
S Platte	**rainbow**	4/1 – 5/15	4/1 – 6/1	6/1 – 7/1	6/15 – 7/15
St Vrain	brown	10/15 – 11/15	10/15 – 5/1	4/1 – 6/1	5/15 – 7/1
Taylor	brown	10/15 – 11/15	10/15 – 5/1	4/1 – 6/1	5/15 – 7/1

Colorado Division of Wildlife

Whirling Disease Facts

A report on whirling disease in Colorado — UPDATE — JANUARY 1997

COLORADO DIVISION OF WILDLIFE
6060 Broadway, Denver, CO 80216
(303) 297-1192

EFFORTS AGAINST WHIRLING DISEASE CONTINUE

The whirling disease puzzle

Whirling disease continues to be a topic of discussion and a source of question to Colorado's fish managers as they look ahead to trout management in 1997. The past year saw continued efforts to control the spread of the parasite that has caused so much concern in the past several years. Significant efforts have been made along several fronts, including new regulations and a Policy concerning the use of stocked fish, fish production clean-up efforts, increased field research and fish sampling to assess the full extent of the impacts of whirling disease, and completion of a special report that gives specific management recommendations.

Whirling disease (WD) is an infectious disease of trout and salmon, caused by a microscopic parasite so small that thousands of them could fit on the head of a pin. The name of the organism is *Myxobolus cerebralis*; loosely translated, it means small round myxospore of the brain. The parasite has a complicated, two-stage life cycle illustrated on the next page.

The name "whirling disease" comes from the way small fish act when heavily infested with the parasite. Spores from the parasite can enter a fish's body, especially a young fish, and infect its skeletal tissue while it is still soft. The spores favor the head and spinal column of fish. When young fish have enough parasites in their body, they begin to exhibit signs of the disease distinctive whirling or swimming in circles and skeletal deformities like sunken heads, bent backs and a possible black color where there should be none. If young trout absorb enough spores at the right time, the disease can be fatal. Trout larger than 4 inches can carry spores, but their hardened skeleton keeps them from being affected by the parasite.

Whirling disease is not spread directly from fish to fish but through an intermediate host, the tubifex worm. The disease may also be spread by birds and mammals that consume infected fish and pass undigested spores through defecation into other waters.

Introduced into Colorado in the late 1980s, the whirling disease parasite now is found in parts of streams in 14 of Colorado's 15 major river drainages, including the Colorado, South Platte, Gunnison, Arkansas and Rio Grande. Most streams and rivers in Colorado continue to remain free of the parasite, but several key fisheries suffer from the parasite's impact on newly hatched fry of rainbow and brown trout. The efforts to control the stocking of fish exposed to the par-

WHIRLING DISEASE FACTS

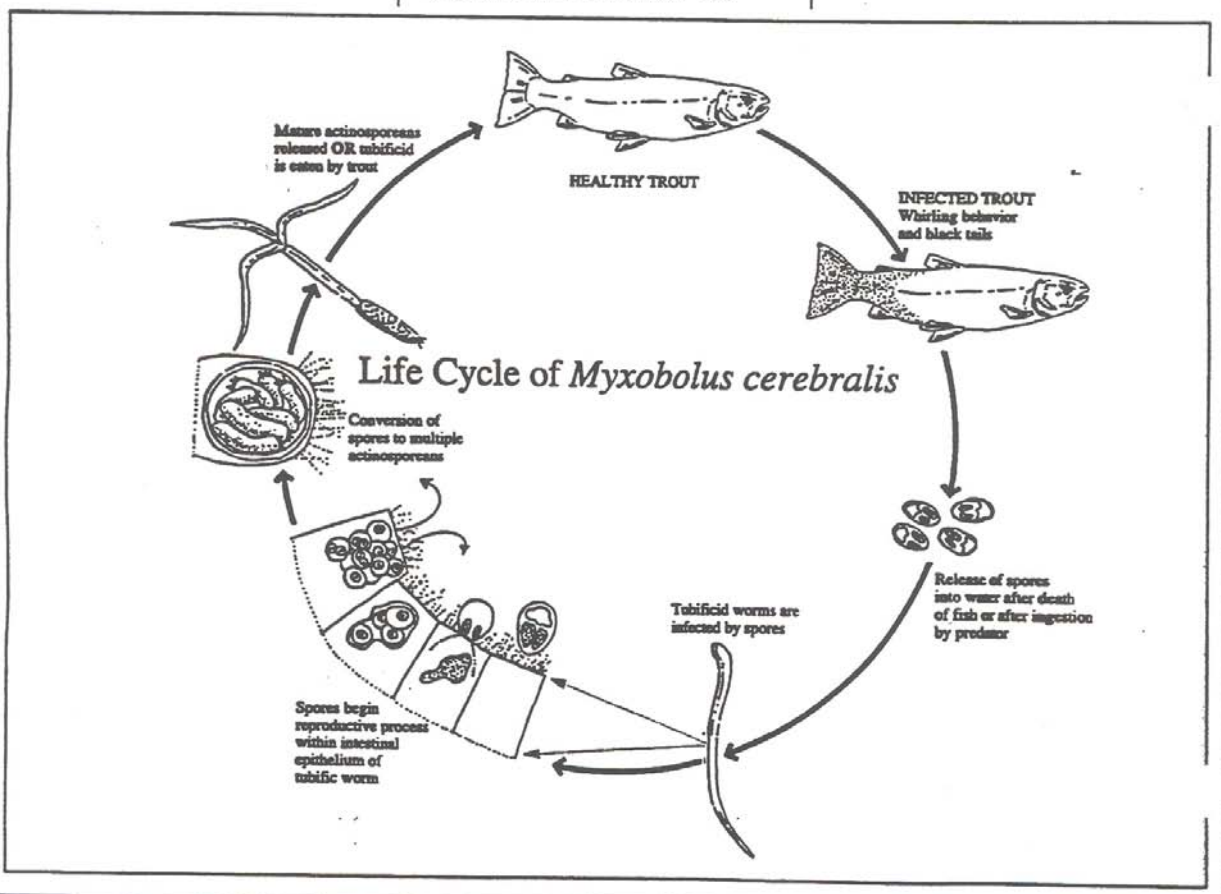
asite and hatchery clean-up efforts have helped to virtually stop the spread of the disease. The majority of Colorado's waters remain free of the parasite.

Unfortunately, the whirling disease spore is resistant to freezing and other harsh environmental factors. Once it is established in a stream, the parasite may persist indefinitely.

Fish production

WHIRLING DISEASE PREVENTION AT HATCHERIES

Division of Wildlife hatcheries have taken a very proactive stance toward the reduction and elimination of whirling disease and have begun to implement the recommendations of the blue ribbon committee report. Fish rearing procedures have changed on positive units to produce fish with minimal WD exposure. The use of constant rearing facility cleaning, rearing fry to larger sizes before putting them out in raceways and keeping fish over concrete appear to be effective ways to reduce infection levels. The Division is applying $750,000 of capital development funds to increase production of negative trout on clean units, applying $1,000,000 to clean-up the Rifle, Mt. Shavano and Roaring Judy units, and requesting additional funds for water filtration systems for the fish production units from the Legislature. Special efforts to protect the Durango Unit and clean-up the Bellvue research Unit are being made in 1997. We believe that completion of the projects will give the state the required number of whirling disease negative trout for stocking by 1999. These projects will also greatly enhance fish health and eliminate other potentially harmful diseases.



WHIRLING DISEASE FACTS

Fish research/Fish Health Lab

With the field research program, the Division is attempting to better understand the impacts of WD on wild fish populations. Research efforts in 1996 focused on assessing continued WD impacts on wild trout recruitment in the South Platte, Cache laPoudre, Colorado, Gunnison, and Rio Grande rivers. Populations in these streams are impacted and are being managed by re-stocking with fingerlings. Research seems to indicate that small fish lightly exposed to the triactinomyxon stage of WD may have an immune response that protects them from infection. Other research has been directed at developing techniques to identify and quantify the triactinomyxon spores in flowing water; and determining the distribution and abundance of tubifex worm habitat in Colorado River cutthroat trout streams, and the possible occurence of *Myxobolus cerebralis* (Mc) DNA. Development of special laboratory processes to identify Mc DNA was also completed in 1996.

The fish health lab has been fully staffed as of Jan. 6, 1997, and has continued the complex process of analyzing the many WD samples taken by field and research biologists. In 1996, over 2,100 fish heads and field samples were analyzed. The lab also has coordinated the analysis of samples at various labs throughout the country. Colorado leads the country in efforts to fully understand the distribution and spread of whirling disease. Special research projects with CSU are studying the links between observed clinical signs of WD and presence of the parasite; and the lab is studying the susceptibility of various trout species to WD.

Fish stocking policy

In 1996, the Colorado Wildlife Commission took definitive action on approving specific regulations to implement the previously approved Policy on the use of fish exposed to WD. The regulations very clearly indicated the Commission's intent to protect those waters of the state that are free of the WD parasite while continuing to recognize the need for stocked fish in Colorado's fishery management programs.

The new regulations give the Director of the Division of Wildlife the authority to designate all cold-water streams, with few exceptions, as "protected habitat." This means that only WD-free fish may be stocked into those waters. Protected habitats also include waters in wilderness and wilderness study areas, native trout restoration waters, waters important to spawning areas, and waters adjacent to clean fish production facilities.

Fish stocking

In light of the current research and knowledge about whirling disease, the new Commission regulations also allow the continued stocking of fish lightly exposed to the parasite. As a result of careful analysis, only a limited number of streams and lakes known to be positive for whirling disease will continue to be stocked with fish that have been lightly exposed on positive fish production units. These waters are considered to be "restricted habitats" and meet a set of criteria that takes into account how many years they have been stocked, the risk of further spread of the parasite and the importance of the recreational sport fishery. Fish exposed to whirling disease are also targeted to reservoirs in low-elevation, dead-end Foothill or eastern plains locations where no salmonid reproduction occurs. In addition, more than 600,000 4-inch rainbow trout are targeted to be stocked stocked into the Colorado, Gunnison, Rio Grande and other rivers to supplement existing fish populations and make-up for lost year classes and diminished natural reproduction .

Whirling disease and fishing

A continuing and important question is how whirling disease continues to affect fishing in Colorado. Certainly, the disease has impacted fishing in a few specific locations throughout the state; however, it is incorrect to assume there will no longer be quality angling in Colorado.

First, whirling disease only affects young salmonids, and research is beginning to show that judicious stocking of larger fingerlings can successfully replenish stream trout populations and allow continuation of good fishing. Over time then, key rivers such as the Gunnison, Colorado, and South Platte will continue to offer quality angling.

Because of efforts to prevent further spread of the disease, and a shortage of clean fish to stock, many traditionally stocked reservoirs and streams in western Colorado will not receive as many fish as in the past, or they may not receive any fish at all. However, Colorado's western waters offer fine fishing for native cutthroat trout, brown trout and brook trout. High-

WHIRLING DISEASE FACTS

mountain lakes and small streams are often under-used and offer a lot of opportunity for the adventurous angler. The Division is also purchasing approximately 100,000 commercially raised trout for stocking in select waters and has an agreement with the U.S. Fish and Wildlife Service on a stocking trade that will allow the stocking of 800,000 6-8-inch fish in waters that are free from the WD parasite.

Large streams, such as the Colorado River, that have seen losses of young fish, still have excellent populations of adult rainbows and browns. We recommend that you treat those fish carefully, though, if you do fish. Catch-and-release angling seems to be in order, to maximize protection for those fish. Anglers seeking warm- and cool-water species, such as bass, catfish and walleyes, will see no change in their favorite fisheries. Furthermore in many waters, particularly along the Front Range and the eastern plains, increased stocking of catchable trout will continue this year as many of these waters are approved for stocking of WD-exposed fish. Remember, whirling disease does not affect adult fish in terms of their edibility or health and poses no threat to humans.

Whirling disease outlook

The Division is committed to a long-term program that continues to protect our aquatic resources from the spread of the whirling disease parasite. There is still much to learn about this parasite and how it operates in fish populations. The Division also is committed to those anglers and citizens who utilize fisheries for recreation and economic interests. The Division will present information on our current research, policy and management activities periodically to keep citizens appraised of changes in the status of the parasite.

What can you do?

As an angler, you can help prevent the spread of the disease by making sure you wash your boots and equipment if you have been wading where it is muddy. The tubificid worm stage of whirling disease likes mud, so if you fish the Colorado, Gunnison, Poudre or Rio Grande rivers, clean up before you leave and scrub your waders when you get home. Using chlorine to wash boots and equipment can also help prevent the transfer of spores. Do not transport any live fish from one place to another and do not dispose of fish entrails, skeletal parts or other by-products in any water.

Where do we go from here?

If you have issues or concerns about management of whirling disease in Colorado, please provide your comments to:

Colorado Division of Wildlife
Aquatic Wildlife Section
6060 Broadway
Denver, CO 80216
ATTN: WD comments

Terms and definitions

HOST - An organism that provides nutrients and also living substrate for a parasite.

INTERMEDIATE HOST - An organism that provides temporary nutrition and substrate for a parasite, usually during an early-middle stage of the parasite's life, and is necessary for the parasite to complete its life cycle.

PARASITE - An organism that obtains nutrition from another organism, without causing immediate death.

SALMONIDS - Members of the family Salmonidae. In Colorado, common members of this family include brown, brook, cutthroat, rainbow and lake trout, kokanee and whitefish.

SPORE - The whirling disease parasite's asexual reproductive cell, a resting stage adapted to resist unfavorable environmental conditions.

WD-NEGATIVE - Fish that have no evidence (signs) of whirling disease (spores), or fish from hatcheries or wild environments that tested negatively for whirling disease.

WD-SUSPECT - Fish reared in a hatchery that tested positively for whirling disease.

U. S. EPA ENVIRONMENTAL RESPONSE TEAM

STANDARD OPERATING PROCEDURES

SOIL SAMPLING

CONTENTS

SUPERCEDES: SOP #2012; Revision 0.0; 11/16/94; U.S. EPA Contract 68-C4-0022.

1.0 SCOPE AND APPLICATION

The purpose of this standard operating procedure (SOP) is to describe the procedures for the collection of representative soil samples. Sampling depths are assumed to be those that can be reached without the use of a drill rig, direct-push, or other mechanized equipment (except for a back-hoe). Analysis of soil samples may determine whether concentrations of specific pollutants exceed established action levels, or if the concentrations of pollutants present a risk to public health, welfare, or the environment.

These are standard (i.e., typically applicable) operating procedures which may be varied or changed as required, dependent upon site conditions, equipment limitations or limitations imposed by the procedure. In all instances, the actual procedures used should be documented and described in an appropriate site report.

Mention of trade names or commercial products does not constitute U.S. Environmental Protection Agency (EPA) endorsement or recommendation for use.

2.0 METHOD SUMMARY

Soil samples may be collected using a variety of methods and equipment depending on the depth of the desired sample, the type of sample required (disturbed vs. undisturbed), and the soil type. Near-surface soils may be easily sampled using a spade, trowel, and scoop. Sampling at greater depths may be performed using a hand auger, continuous flight auger, a trier, a split-spoon, or, if required, a backhoe.

3.0 SAMPLE PRESERVATION, CONTAINERS, HANDLING, AND STORAGE

Chemical preservation of solids is not generally recommended. Samples should, however, be cooled and protected from sunlight to minimize any potential reaction. The amount of sample to be collected and proper sample container type are discussed in ERT/REAC SOP #2003 Rev. 0.0 08/11/94, *Sample Storage, Preservation and Handling*.

4.0 INTERFERENCES AND POTENTIAL PROBLEMS

There are two primary potential problems associated with soil sampling - cross contamination of samples and improper sample collection. Cross contamination problems can be eliminated or minimized through the use of dedicated sampling equipment. If this is not possible or practical, then decontamination of sampling equipment is necessary. Improper sample collection can involve using contaminated equipment, disturbance of the matrix resulting in compaction of the sample, or inadequate homogenization of the samples where required, resulting in variable, non-representative results.

5.0 EQUIPMENT

Soil sampling equipment includes the following:

- Maps/plot plan
- Safety equipment, as specified in the site-specific Health and Safety Plan
- Survey equipment or global positioning system (GPS) to locate sampling points
- Tape measure
- Survey stakes or flags
- Camera and film
- Stainless steel, plastic, or other appropriate homogenization bucket, bowl or pan
- Appropriate size sample containers
- Ziplock plastic bags
- Logbook
- Labels
- Chain of Custody records and custody seals
- Field data sheets and sample labels
- Cooler(s)
- Ice
- Vermiculite
- Decontamination supplies/equipment
- Canvas or plastic sheet
- Spade or shovel
- Spatula
- Scoop
- Plastic or stainless steel spoons
- Trowel(s)
- Continuous flight (screw) auger
- Bucket auger
- Post hole auger
- Extension rods
- T-handle
- Sampling trier
- Thin wall tube sampler
- Split spoons
- Vehimeyer soil sampler outfit
  - Tubes
  - Points
  - Drive head
  - Drop hammer
  - Puller jack and grip
- Backhoe

6.0 REAGENTS

Reagents are not used for the preservation of soil samples. Decontamination solutions are specified in ERT/REAC SOP #2006 Rev. 0.0 08/11/94, *Sampling Equipment Decontamination*, and the site specific work plan.

7.0 PROCEDURES

7.1 Preparation

1. Determine the extent of the sampling effort, the sampling methods to be employed, and the types and amounts of equipment and supplies required.

2. Obtain necessary sampling and monitoring equipment.

3. Decontaminate or pre-clean equipment, and ensure that it is in working order.

4. Prepare schedules and coordinate with staff, client, and regulatory agencies, if appropriate.

5. Perform a general site survey prior to site entry in accordance with the site specific Health and Safety Plan.

6. Use stakes, flagging, or buoys to identify and mark all sampling locations. Specific site factors, including extent and nature of contaminant, should be considered when selecting sample location. If required, the proposed locations may be adjusted based on site access, property boundaries, and surface obstructions. All staked locations should be utility-cleared by the property owner or the On-Scene-Coordinator (OSC) prior to soil sampling; and utility clearance should always be confirmed before beginning work.

7.2 Sample Collection

7.2.1 Surface Soil Samples

Collection of samples from near-surface soil can be accomplished with tools such as spades, shovels, trowels, and scoops. Surface material is removed to the required depth and a stainless steel or plastic scoop is then used to collect the sample.

This method can be used in most soil types but is limited to sampling at or near the ground surface. Accurate, representative samples can be collected with this procedure depending on the care and precision demonstrated by the sample team member. A flat, pointed mason trowel to cut a block of the desired soil is helpful when undisturbed profiles are required. Tools plated with chrome or other materials should not be used. Plating is particularly common with garden implements such as potting trowels.

The following procedure is used to collect surface soil samples:

1. Carefully remove the top layer of soil or debris to the desired sample depth with a pre-cleaned spade.

2. Using a pre-cleaned, stainless steel scoop, plastic spoon, or trowel, remove and discard a thin layer of soil from the area which came in contact with the spade.

3. If volatile organic analysis is to be performed, transfer the sample directly into an appropriate, labeled sample container with a stainless steel lab spoon, or equivalent and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container, and mix thoroughly to obtain a homogenous sample representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; or, if composite samples are to be collected, place a sample from another sampling interval or location into the homogenization container and mix thoroughly. When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

7.2.2 Sampling at Depth with Augers and Thin Wall Tube Samplers

This system consists of an auger, or a thin-wall tube sampler, a series of extensions, and a "T" handle (Figure 1, Appendix A). The auger is used to bore a hole to a desired sampling depth, and is then withdrawn. The sample may be collected directly from the auger. If a core sample is to be collected, the auger tip is then replaced with a thin wall tube sampler. The system is then lowered down the borehole, and driven into the soil to the completion depth. The system is withdrawn and the core is collected from the thin wall tube sampler.

Several types of augers are available; these include: bucket type, continuous flight (screw), and post-hole augers. Bucket type augers are better for direct sample recovery because they provide a large volume of sample in a short time. When continuous flight augers are used, the sample can be collected directly from the flights. The continuous flight augers are satisfactory when a composite of the complete soil column is desired. Post-hole augers have limited utility for sample collection as they are designed to cut through fibrous, rooted, swampy soil and cannot be used below a depth of approximately three feet.

The following procedure is used for collecting soil samples with the auger:

1. Attach the auger bit to a drill rod extension, and attach the "T" handle to the drill rod.

2. Clear the area to be sampled of any surface debris (e.g., twigs, rocks, litter). It may be advisable to remove the first three to six inches of surface soil for an area approximately six inches in radius around the drilling location.

3. Begin augering, periodically removing and depositing accumulated soils onto a plastic sheet spread near the hole. This prevents accidental brushing of loose material back down the borehole when removing the auger or adding drill rods. It also facilitates refilling the hole, and avoids possible contamination of the surrounding area.

4. After reaching the desired depth, slowly and carefully remove the auger from the hole. When sampling directly from the auger, collect the sample after the auger is removed from the hole and proceed to Step 10.

5. Remove auger tip from the extension rods and replace with a pre-cleaned thin wall tube sampler. Install the proper cutting tip.

6. Carefully lower the tube sampler down the borehole. Gradually force the tube sampler into the soil. Do not scrape the borehole sides. Avoid hammering the rods as the vibrations may cause the boring walls to collapse.

7. Remove the tube sampler, and unscrew the drill rods.

8. Remove the cutting tip and the core from the device.

9. Discard the top of the core (approximately 1 inch), as this possibly represents material collected before penetration of the layer of concern. Place the remaining core into the appropriate labeled sample container. Sample homogenization is not required.

10. If volatile organic analysis is to be performed, transfer the sample into an appropriate, labeled sample container with a stainless steel lab spoon, or equivalent and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container, and mix thoroughly to obtain a homogenous sample representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; or, if composite samples are to be collected, place a sample from another sampling interval into the homogenization container and mix thoroughly.

    When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

11. If another sample is to be collected in the same hole, but at a greater depth, reattach the auger bit to the drill and assembly, and follow steps 3 through 11, making sure to decontaminate the auger and tube sampler between samples.

12. Abandon the hole according to applicable state regulations. Generally, shallow holes can simply be backfilled with the removed soil material.

7.2.3 Sampling with a Trier

The system consists of a trier, and a "T" handle. The auger is driven into the soil to be sampled and used to extract a core sample from the appropriate depth.

The following procedure is used to collect soil samples with a sampling trier:

1. Insert the trier (Figure 2, Appendix A) into the material to be sampled at a 0^{o} to 45^{o} angle from horizontal. This orientation minimizes the spillage of sample.

2. Rotate the trier once or twice to cut a core of material.

3. Slowly withdraw the trier, making sure that the slot is facing upward.

4. If volatile organic analyses are required, transfer the sample into an appropriate, labeled sample container with a stainless steel lab spoon, or equivalent and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container, and mix thoroughly to obtain a homogenous sample representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; or, if composite samples are to be collected, place a sample from another sampling interval into the homogenization container and mix thoroughly. When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

7.2.4 Sampling at Depth with a Split Spoon (Barrel) Sampler

Split spoon sampling is generally used to collect undisturbed soil cores of 18 or 24 inches in length. A series of consecutive cores may be extracted with a split spoon sampler to give a complete soil column profile, or an auger may be used to drill down to the desired depth for sampling. The split spoon is then driven to its sampling depth through the bottom of the augured hole and the core extracted.

When split spoon sampling is performed to gain geologic information, all work should

be performed in accordance with ASTM D1586-98, "Standard Test Method for Penetration Test and Split-Barrel Sampling of Soils".

The following procedures are used for collecting soil samples with a split spoon:

1. Assemble the sampler by aligning both sides of barrel and then screwing the drive shoe on the bottom and the head piece on top.

2. Place the sampler in a perpendicular position on the sample material.

3. Using a well ring, drive the tube. Do not drive past the bottom of the head piece or compression of the sample will result.

4. Record in the site logbook or on field data sheets the length of the tube used to penetrate the material being sampled, and the number of blows required to obtain this depth.

5. Withdraw the sampler, and open by unscrewing the bit and head and splitting the barrel. The amount of recovery and soil type should be recorded on the boring log. If a split sample is desired, a cleaned, stainless steel knife should be used to divide the tube contents in half, longitudinally. This sampler is typically available in 2 and 3 1/2 inch diameters. A larger barrel may be necessary to obtain the required sample volume.

6. Without disturbing the core, transfer it to appropriate labeled sample container(s) and seal tightly.

7.2.5 Test Pit/Trench Excavation

A backhoe can be used to remove sections of soil, when detailed examination of soil characteristics are required. This is probably the most expensive sampling method because of the relatively high cost of backhoe operation.

The following procedures are used for collecting soil samples from test pits or trenches:

1. Prior to any excavation with a backhoe, it is important to ensure that all sampling locations are clear of overhead and buried utilities.

2. Review the site specific Health & Safety plan and ensure that all safety precautions including appropriate monitoring equipment are installed as required.

3. Using the backhoe, excavate a trench approximately three feet wide and approximately one foot deep below the cleared sampling location. Place excavated soils on plastic sheets. Trenches greater than five feet deep must be sloped or protected by a shoring system, as required by OSHA regulations.

4. A shovel is used to remove a one to two inch layer of soil from the vertical face of the pit where sampling is to be done.

5. Samples are taken using a trowel, scoop, or coring device at the desired intervals. Be sure to scrape the vertical face at the point of sampling to remove any soil that may have fallen from above, and to expose fresh soil for sampling. In many instances, samples can be collected directly from the backhoe bucket.

6. If volatile organic analyses are required, transfer the sample into an appropriate, labeled sample container with a stainless steel lab spoon, or equivalent and secure the cap tightly. Place the remainder of the sample into a stainless steel, plastic, or other appropriate homogenization container, and mix thoroughly to obtain a homogenous sample representative of the entire sampling interval. Then, either place the sample into appropriate, labeled containers and secure the caps tightly; or, if composite samples are to be collected, place a sample from another sampling interval into the homogenization container and mix thoroughly. When compositing is complete, place the sample into appropriate, labeled containers and secure the caps tightly.

7. Abandon the pit or excavation according to applicable state regulations. Generally, shallow excavations can simply be backfilled with the removed soil material.

8.0 CALCULATIONS

This section is not applicable to this SOP.

9.0 QUALITY ASSURANCE/QUALITY CONTROL

There are no specific quality assurance (QA) activities which apply to the implementation of these procedures. However, the following QA procedures apply:

1. All data must be documented on field data sheets or within site logbooks.

2. All instrumentation must be operated in accordance with operating instructions as supplied by the manufacturer, unless otherwise specified in the work plan. Equipment checkout and calibration

activities must occur prior to sampling/operation, and they must be documented.

10.0 DATA VALIDATION

This section is not applicable to this SOP.

11.0 HEALTH AND SAFETY

When working with potentially hazardous materials, follow U.S. EPA, OHSA and corporate health and safety procedures, in addition to the procedures specified in the site specific Health & Safety Plan..

12.0 REFERENCES

Mason, B.J. 1983. Preparation of Soil Sampling Protocol: Technique and Strategies. EPA-600/4-83-020.

Barth, D.S. and B.J. Mason. 1984. Soil Sampling Quality Assurance User's Guide. EPA-600/4-84-043.

U.S. Environmental Protection Agency. 1984 Characterization of Hazardous Waste Sites - A Methods Manual: Volume II. Available Sampling Methods, Second Edition. EPA-600/4-84-076.

de Vera, E.R., B.P. Simmons, R.D. Stephen, and D.L. Storm. 1980. Samplers and Sampling Procedures for Hazardous Waste Streams. EPA-600/2-80-018.

ASTM D 1586-98, ASTM Committee on Standards, Philadelphia, PA.

APPENDIX A
Figures
SOP #2012
February 2000

FIGURE 1. Sampling Augers

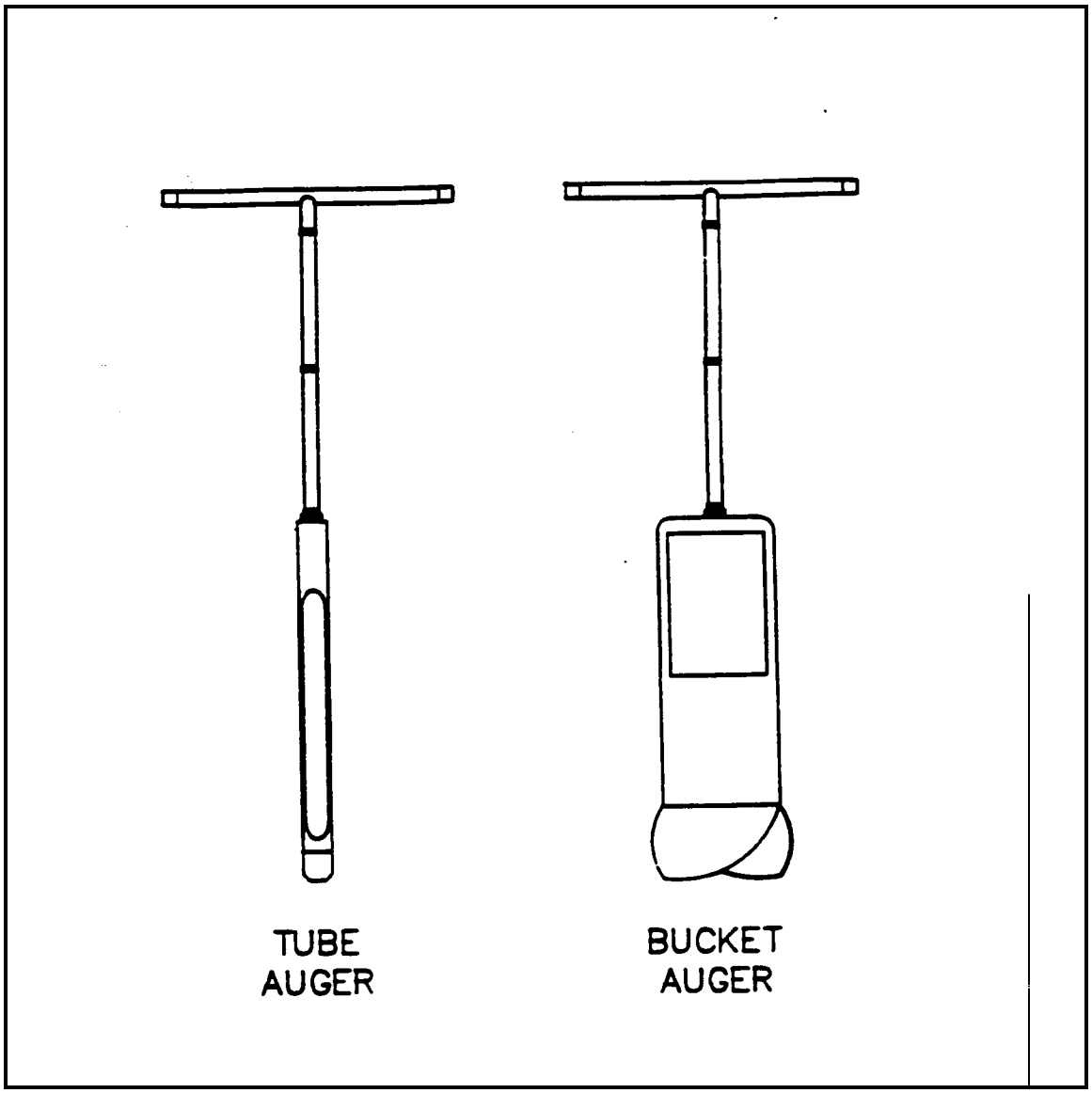
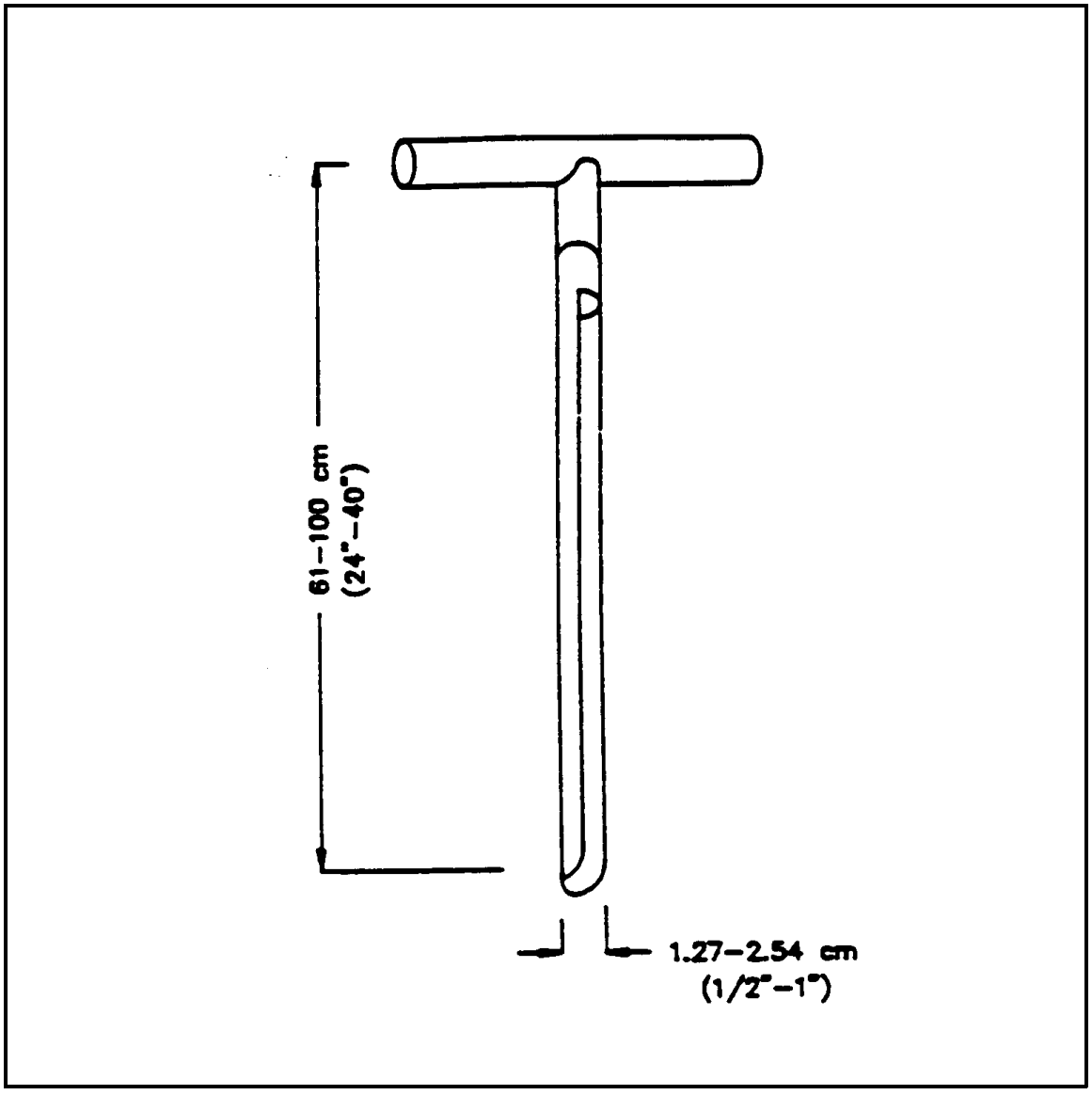


FIGURE 2. Sampling Trier



Implementation Guidance for Determining Sediment Deposition Impacts to Aquatic Life in Streams and Rivers

(As intended for higher gradient, cobble-bed, course-grained streams)

As Revised, May 2005

Colorado Department of Public Health and Environment
Water Quality Control Commission
Water Quality Control Division
4300 Cherry Creek Drive South
Denver, Colorado 80246-1530

Developed in conjunction with the
Colorado Sediment Task Force

Commission Policy 98-1
Expires May 31, 2013

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INTRODUCTION

This guidance provides an interpretation of the Colorado Water Quality Control Commission's (Commission) "narrative standards" as they apply to sediments which may form deposits detrimental to the attainment of aquatic life uses. The Basic Standards and Methodologies for Surface Water, Regulation 31 (5 CR 1002-31), are the basis for establishing this guidance. In particular, section 31.11 of this regulation provides the following language:

> *All surface waters of the State are subject to the following basic standards; however, discharge of substances regulated by permits which are within those permit limitations shall not be a basis for enforcement proceedings under these basic standards:*
>
> *(1) Except where authorized by permits, BMP's, 401 Certifications, or plans of operation approved by the Division or other applicable agencies, state surface waters shall be free from substances attributable to human-caused point source or nonpoint source discharge in amounts, concentrations or combinations which:*
>
> *(a) for all surface waters except wetlands;*
>
> *(I) can settle to form bottom deposits detrimental to the beneficial uses. Depositions are stream bottom buildup of materials which include but are not limited to anaerobic sludges, mine slurry or tailings, silt, or mud;...*

Although the deposition of sediment on the bottom of surface waters could have an impact to any of the beneficial uses for which Colorado surface waters are classified, this guidance is intended to apply only to the assessment of impacts to aquatic life uses in streams and river environments. Assessment of impacts to other uses or to reservoir and lake systems is not covered in the guidance and would require a site-specific assessment. Guidance to address these other impacts is being developed through the Colorado Sediment Task Force under the direction of the Division.

Streams Types Covered By This Guidance

This guidance is intended to apply to the assessment of impacts to aquatic life uses in higher gradient, cobble-bed, course-grained, mountainous stream and wadeable river environments. (For example, Rosgen stream types A1-A4, B1-B4, C1-C4.) The guidance can also apply to transition-zone streams that fit the above description. It is not intended to cover sandy-bottom, lower-gradient plains streams, large unwadeable rivers, and lakes and reservoirs. The Division with the assistance of the Sediment Task Force is currently working on guidance to assess these other waterbodies, as well as other beneficial uses.

Introduction to Sediment Impairment

The scope of this guidance is limited to the assessment of bottom deposition of sediment. It is not intended to address sediment suspended in the water column or turbidity. Turbidity and suspended sediment are aspects of sediment transport, which is a complex relationship of streamflow, the type and size of sediment in rivers. However, it is important to understand that an increase in suspended sediment concentrations will reduce light penetration, and a sustained high concentration of suspended sediment can reduce primary production. Increased suspended sediment can cause problems with water treatment, clog irrigation canals, and reduce reservoir storage capacity.

Sediment can be dichotomously classified in overlapping ways – clean or contaminated, and organic or inorganic. This guidance addresses only clean sediment, not sediment that is contaminated by toxic substances such as heavy metals. Organic matter can become abundant enough to cause water quality problems, typically below outfalls where decay can depress dissolved oxygen levels. The distinction between inorganic and organic fractions is not always made in the monitoring or study of sediment, nor is it the intent of this guidance to do so. Inorganic sediment, the product of physical weathering of geologic materials and sediment caused by human induced erosion, is the main focus of this guidance.

This guidance applies to sediment causing stress to aquatic life through the deposition of materials. The guidance is not intended to provide a complete analysis of aquatic life use attainment; it is necessary to perform other analysis (e.g. chemical and toxicity analysis) to determine a full range of possible stressors which may be impacting aquatic life. Only human-caused discharges in amounts, concentrations, or combinations are considered in this guidance. Therefore, natural erosive processes over a variety of geologic conditions must be considered in the implementation of this guidance, in order to determine natural or background conditions.

Excessive deposition of sediment on the bottom substrate of streams and rivers is an important cause of impacts to aquatic life. These impacts usually result from the loss of critical habitat for many fish, aquatic invertebrates, and algae. These kinds of impacts have been addressed in a detailed review in by Waters (1995) and in other literature reviews. Impacts to fish can include the smothering of fish spawning gravels and cobble surfaces with fine sediment resulting in decreased intergravel oxygen and a reduction in survival and growth rates; loss of fish food sources; and loss of pool and other habitat types through changes in stream channel morphology. Impacts to aquatic invertebrates can include the smothering and infilling of the interstitial spaces normally found in clean such as gravel and cobble. This loss of habitat space can result in changes to the normal aquatic invertebrate community including changes in abundance, community structure, distribution, and in the loss of sensitive species.

One of the fundamental questions regarding sediment in streams and its effect on biota is particle size. Stream channels and floodplains are constantly adjusting to the amount of water and sediment supplied by the watershed. Four physical characteristics of a stream are in a dynamic state of equilibrium called Lane's Balance. These characteristics are streamflow, channel slope, sediment load, and particle size. If one of these characteristics changes in a stream, one or more of the other three must also change to accommodate and achieve equilibrium again. A change in sediment load is the first thing to change in response to a disturbance to restore equilibrium and it is the most sensitive measures of change. Chapman and Mcleod (1987) found that bed material size is related to habitat suitability for fish and Macroinvertebrates and that excess sediment decreased both density and diversity of aquatic insects. Specific aspects of sediment-invertebrate relationships may be described as follows: 1) invertebrate abundance is correlated with substrate particle size; 2) fine sediment reduces the abundance of original populations by reducing interstitial habitat normally available in large-particle substrate (gravel, cobbles); and 3) species type, species richness, and diversity all change as substrate particle size changes from large (gravel, cobble) to small (sand, silt, clay) (Waters, 1995).

This guidance is designed to provide a consistent approach for the Division, for other agencies, and stakeholders, to gather data to document the effects of bottom deposits on aquatic life uses. The guidance also provides a means for the Division and the Commission to consider the impacts of bottom deposits on the attainment of the aquatic life uses. In Colorado, surface waters may be assigned any of the following four aquatic life classifcations: class 1 coldwater, class 1 warmwater, class 2 coldwater and class 2 warmwater. The guidance presents a procedure for determining whether a particular stream segment is attaining the narrative standard based on the concept of comparing the actual sediment conditions of a study stream with the **expected conditions** for the same stream. A wide variety of factors including, aquatic life use classification, geology, elevation, climate, hydrology, and land use will influence the selection of appropriate expected conditions.

For the purposes of determining the status of water quality as required in §305(b) of the federal Clean Water Act, and establishing a listing of waterbodies requiring TMDL's under §303(d) of the Act, the standards attainment categories found in Section 4 shall be used by the Division. Classified stream segments or portions of classified segments which are determined to be not attaining the narrative sediment standard after such an analysis may be proposed by the Division for 303(d) listing. Streams which are attaining the standard should not be listed for 303(d) purposes. This guidance is intended for identifying impairment due to sediment but is not intended to address the development of TMDL's for sediment, and therefore does not address how to solve sediment problems or how to identify sediment sources or allocate loads.

1. APPROACH TO ASSESSING SEDIMENT IMPACTS TO AQUATIC LIFE

The assessment approach described in this guidance is based on the combined concepts of the use of thresholds and comparing the **actual conditions** of a specific study stream reach or segment with the **expected conditions** for the same stream to determine attainment of the narrative standard. This guidance uses the term **expected condition** rather than the EPA terminology of reference condition. Expected condition is used in this guidance in an attempt to avoid the concern that sometimes arises when reference condition is narrowly interpreted to mean pristine or minimally impacted streams. Expected condition is intended to include a wide range of aquatic conditions that can reflect more than only minimal impact, including those impacts associated with historical and dominant land and water use activities. Nevertheless, it can still serve as a reasonable and readily attainable target or goal for improvement to the aquatic life use in a sediment impacted water-body.

This approach is directly patterned after the reference condition approach found in U.S. Environmental Protection Agency (EPA) guidance for a number of programs including water quality standards, assessment and reporting, biocriteria development, rapid bioassessment protocols (RBP), use attainability analysis, and §319 monitoring. The expected condition approach, and its many modifications, is widely used across the country. By adopting this guidance, Colorado can assess and report sediment conditions in a manner consistent with other states and can take advantage of the experience gained by other states in their assessments.

Section 2 of this guidance provides detail on selecting an expected condition and those factors that need to be considered in such a selection. It provides a tiered approach that starts with site-specific expected condition sites and progresses through regional conditions. Finally it employs the use of expert opinion to determine what uses are attainable in areas where water and land resources are heavily managed, resulting in multiple and essentially irreversible impacts.

It should be noted, that to fully utilize the EPA approach requires the development of regional or statewide biocriteria. These biocriteria are then used for the direct assessment of use impairment or condition. In Colorado, regional or statewide biocriteria are currently under development and have not yet been developed. Although we still lack the ability to compare the aquatic life in impacted conditions to regional biocriteria, we can still provide a sound sediment assessment framework in Colorado by using a case-by-case or site-specific expected conditions approach to assess impacted stream segments until regional or statewide narrative or numeric biocriteria become available.

Assessment Study Design

Before any assessment work is undertaken, a study design and plan must be formulated through a stakeholders process with involvement of the Division staff. A number of issues have to be considered at this stage and detailed guidance on this can be found in the references section. There are several important aspects to consider and these are listed below.

Whenever practical, assessment studies should be conducted through a cooperative arrangement among the various stakeholders, state and federal agencies and others. Ideally, study groups should consist of multi-disciplinary teams built of personnel with the appropriate skill levels required to complete an assessment. These teams would select study methods, assessment endpoints and indicators, and complete an overall design including frequency and locations of sampling. Quality control and data quality objectives need to be formulated in quality assurance plans that are implemented as part of each study.

It is recommended that stakeholders interested in performing sediment deposition assessment work consult with the Division before initiating the assessment to insure that the design of the work is appropriate for the specific study stream, and to meet the needs of the Division and Commission for decision-making.

Proper site selection and determination of sample size are very important pieces of the assessment. Pebble counts should be conducted in the same sample reach as the collection of macroinvertebrates. Pebble counts must also be conducted using the same procedure in both the expected condition site and study site. Sampling reach location should be selected with care. A sampling reach should capture the "big picture" of the situation in the stream and be representative of the majority of the conditions in the stream. For example, it is not acceptable to "skip" certain areas of the stream because of the existence of beaver dams and for ease of sampling. Beaver dams are natural conditions in the stream and need to be captured in the assessment. Neither is it acceptable to choose a "good corner" of the stream to sample and not cover an area representative of the entire stream reach. The sampling site should include at least two riffle-run-pool sequences where possible, or at least 20x the bankfull width. The assessor should document what procedure was followed to select the sampling reach.

The number of counts in a pebble count necessary to characterize the reach is also a very important piece of the assessment. A <u>minimally</u> statistically acceptable number is 100 counts. The CDPHE pebble count SOP requires 400 counts. Bevenger and King (1995) have provided a table of sample sizes necessary to detect different levels of change. Four hundred counts are more than is required to detect a 10% change. To detect a change of 0.10 (20% fines in the expected condition site) requires about 200 – 300 counts in the study and expected condition sites. Performing more counts (300 – 400) to characterize the expected condition reaches would be beneficial to better characterize natural variability and reduce error, as these reaches will become a data set which can be used for multiple projects. The Division highly encourages the assessor to conduct 300 – 400 counts during their assessment.

2. EXPECTED CONDITION

A key element in implementing the narrative sediment standard is determining the expected condition for each candidate stream with suspected sediment deposits detrimental to the aquatic life use. An expected condition should be based on an individual expected condition site, a combination of expected condition sites, or an estimated condition, depending on the availability of acceptable sites. The expected condition establishes the basis for making comparisons and for detecting aquatic life use impairments. Initially, expected conditions will likely be established on a site-specific basis for each candidate stream. Whether expected conditions are applicable to a larger population of similar streams depends on several factors, including the spacial scale of interest, extent of impairment of the expected condition, and the need for site-specific information. This guidance presents a tiered approach to establishing the expected condition, and the specific characteristics of acceptable expected condition sites.

Approaches to Establishing Expected conditions

A tiered approach to establishing the expected condition (Figure 1) is based on the quality of expected condition sites, and is consistent with EPA technical guidance (EPA 1996). The first step to identifying an expected condition is to conduct a preliminary assessment to determine the feasibility of using expected condition sites. Expected condition sites refer to locations in the same or similar stream and habitat type at which data can be collected for comparison with candidate streams of interest. Typical expected condition sites include sites that are upstream from point and/or nonpoint sources; sites that occur at the recovery end of a gradient of impact; sites in nearby comparable watersheds; and regional expected condition sites that may be applied to a group of candidate streams of the same stream type.

Tier 1 -Expected condition sites are acceptable and are minimally disturbed. Expected condition sites would be characterized as “natural”. EPA describes these sites as the “biological integrity expectation”. An example of a stream type for which tier 1 expected condition sites may be available would be some mountain headwater streams.

Tier 2 -Expected condition sites are acceptable but are more than minimally disturbed. No “natural” sites exist; therefore the best available sites are selected and sampled for determination of expected conditions. EPA describes these sites as the “interim expectation”. An example of a stream type for which tier 2 expected condition sites may be available would be some segments of large rivers on the plains. This interim expectation could be revisited after restoration efforts have been initiated and evaluated, and may become the final expectation.

Tier 3 -Expected condition sites are not acceptable or no expected condition sites exist. Expected conditions would be based on models, historical data, data from neighboring sites, ecological information, and/or expert opinion as appropriate. EPA

describes this type of expected condition as the “hypothetical expectation”. The expected condition may be regarded as temporary until more realistic attainment goals can be developed. Some examples of stream types for which tier 3 expected conditions may be appropriate would be stream types that are significantly impaired statewide but have some recovery potential (i.e., expected condition sites are unacceptable) or very unique stream types (i.e., no expected condition sites exist).

Determining the expected condition primarily from expected condition sites is based on the premise that streams minimally affected by human activity will exhibit biological conditions representative of what is most natural and attainable for streams in the region. Anthropogenic effects include human influences, for example, watershed disturbances, habitat alteration, non-point source runoff, point source discharges, and atmospheric deposition. Sites that are undisturbed by human activities may be ideal expected condition sites. However, land and water use practices and atmospheric pollution have so altered water resources that truly undisturbed sites are rarely available. In practice, most expected condition sites will reflect some of these impacts. The selection of expected condition sites may be made from those sites with the least anthropogenic influences. Expected condition sites should represent the best attainable conditions that can be achieved by similar streams within a particular ecological region (EPA 1996). They reflect the actual potential of the candidate stream, that is, stressors that can be controlled are controlled, although other stressors may be irreversible. The use of actual expected condition sites to establish expected conditions is always important, as such sites represent achievable goals and can be regularly monitored (EPA, 1996).

If expected condition sites are not acceptable or there are no expected condition sites, then the alternative is to derive expected conditions using models, historical data, data from neighboring sites, ecological information, ecoregion and/or expert opinion. Guidance on the use of these methods to derive expected conditions can be found in *Biological Criteria: Technical Guidance for Streams and Small Rivers* (EPA, 1996). This approach may be the only means of examining some significantly altered systems. The expected condition may be regarded as temporary until more realistic attainment goals can be developed.

Although this guidance presents three tiers or individual approaches for establishing expected conditions, expected conditions may be established using multiple approaches. For example, expected conditions may be determined for a specific study stream using a combination of data from expected condition sites, and historical data, along with expert opinion and best professional judgment.

In addition, the inherent variability between streams can be accounted for if a suite of expected condition reaches is used as opposed to one expected condition site. Additional expected condition sites of the same stream type or similar morphology may be necessary to survey if the expected condition site chosen is questionable by the trained data collectors. The use of multiple expected condition reaches is a good approach to assessing impairment of aquatic life due to sediment.

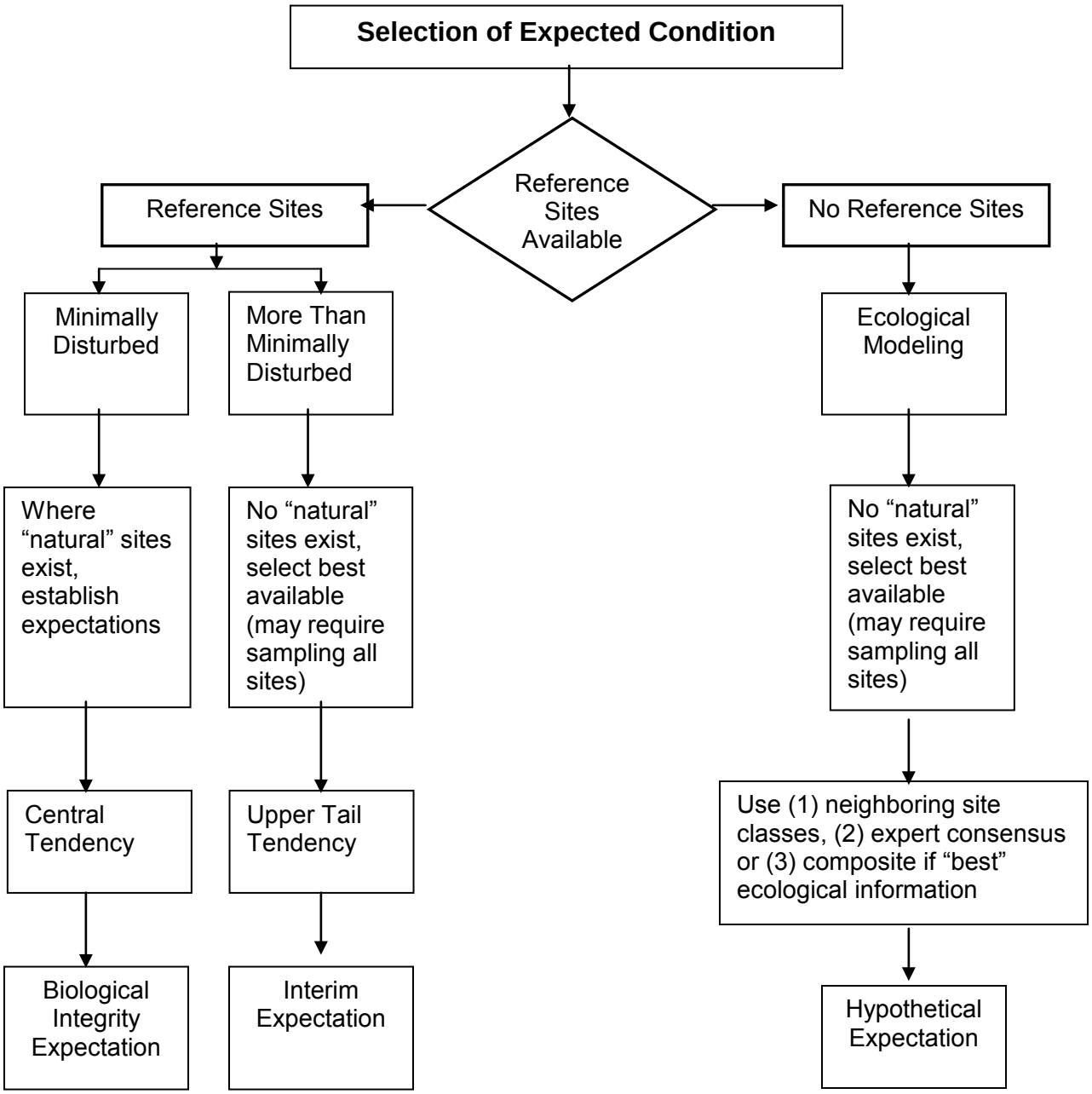


Figure 1. A tiered approach to establishing the expected condition. (After: *Biological Criteria – Technical Guidance for Streams and Small Rivers*; USEPA 1996, p.30)

Characteristics of Acceptable Expected condition Sites (Tier 1 and Tier 2)
Ideally, the expected condition and study sites should share similar or common characteristics such as elevation, geology, hydrology, hydraulics, watershed size, in-stream habitat (pools, substrate, etc), and riparian habitat. Characteristics that cannot change over time should be used as primary attributes of similarity between expected condition and study sites. Examples of parameters to study between expected condition and study site are included in table 1.

The overall goal in the establishment of the expected condition from expected condition sites is to describe the expected biota and habitat at sites of interest. Expected condition sites must be carefully selected because they will be used as benchmarks against which specific study streams will be compared. The conditions at expected condition sites should represent the best attainable conditions that can be achieved by similar streams within a particular geographic region. Two primary considerations guide the selection of expected condition sites within each class: representativeness and minimal impairment.

Representativeness - Expected condition sites must be representative of the stream and habitat types of interest. In general, the following characteristics are typical of minimally disturbed (tier 1) expected condition sites:

* Physical characteristics typical of the region (e.g., ecoregion (Hughes et al 1986) climate, topography, surficial geology, soil).
* Natural stream morphology typical of the region (e.g., Rosgen (1996) channel type, pools, riffles, runs, backwaters, and glides).
* Representative diversity of substrate materials (fines, gravel, cobbles, boulders, woody debris) appropriate to the region.
* Banks representative of undisturbed streams in the region (generally covered by riparian vegetation with little evidence of bank erosion, or undercut banks stabilized by root wads.) Banks should provide cover for aquatic biota.
* Natural color and odor - in some area, clear, cold water is typical of the waterbody types in the region; in others, the water is turbid or stained.
* Extensive, natural riparian vegetation representative of the region.
* Presence of animals, such as birds, mammals, amphibians, and reptiles, that are representative of the region and derive some support from aquatic ecosystems.

For expected condition sites that are identified as more than minimally disturbed (tier 2), decisions will need to be made and documented regarding whether such sites are representative of the candidate stream type and reflect the best attainable conditions that can be achieved by the candidate stream.

Minimal Impairment - Sites that are undisturbed by human activities are ideal expected condition sites. However, truly undisturbed sites are rarely available. Therefore, minimally impaired sites must be used to determine the selection of expected condition sites. This would include acceptable expected condition sites described as “minimally disturbed” (tier 1) as well as “more than minimally disturbed” (tier 2). For locations

where even such minimally impaired expected condition sites are significantly degraded, the search for suitable expected condition sites could be extended over a wider area, to include sites outside the watershed or Colorado. This may be particularly true for unique streams or very large rivers.

The purpose of selecting minimally impaired sites to represent expected conditions is primarily goal-setting. Sites with notable degraded conditions that can be controlled should not be accepted as expected condition sites.

A critical element in establishing expected conditions, particularly for situations where undisturbed sites are not available, is to determine if a site is “minimally impaired”. How much degradation can be allowed? What constitutes an acceptable expected condition site will differ among geographic regions because stream morphology, physiography, soil conditions, vegetation, and dominant land uses differ among regions. After considering all watersheds within an ecoregion of interest, the following factors should be considered in selecting “minimally impaired” expected condition sites. In general, these characteristics are typical of ideal minimally disturbed (tier 1) expected condition sites.

* No upstream impoundments or significant diversions.
* No known point source discharges or contaminants in place.
* No known spills, pollution incidents, or hazardous waste sites.
* Low human population density.
* Low agricultural activity.
* Low road and highway density.
* Minimal nonpoint source problems (e.g., agriculture, urban, logging, mining, feedlots, acidic deposition).
* No known intensive fish stocking or other management activities that would substantially shift the community composition.

For expected condition sites that are identified as more than minimally disturbed (tier 2), decisions will need to be made and documented regarding whether such sites are the best available sites and reflect the best attainable conditions that can be achieved by the candidate stream (i.e., acceptable expected condition sites).

Table 1. Expected condition Site Selection Characteristics

Water	Land	Vegetation
Area	Geology % Area	Cover Type % Area
Perimeter	Biotite	Trees
Basin Length	Glacial Moraine	Shrubs
Basin Aspect	Alluvium	Grass
Compactness Coefficient	Basalt	Non-Vegetated
Drainage Density	Shale/Sandstone Interbedded	Bank Vegetation
Stream Order at Mouth	Granite	
Total Stream Length	Shale	
Bifurcation Ratio	Elevation	
Watershed Size	Accessibility	
Channel Morphology	Bank Structure	
Stream Type	Gneiss	
Stream Velocity	Schist	
Water Depth	Magmatite	
Substrate Type		
Stream Gradient		
Watershed Yield		

3. MEASURING PHYSICAL HABITAT CONDITION AND BIOLOGICAL CONDITION

Introduction

In order to assess the stream bottom for excess sediment that may impair aquatic life and significantly alter the physical properties of the bottom, physical measurements of the stream bottom substrate must be made alongside measurements being made of the biological component if the sediment threshold is exceeded. Physical measurements or indicators of the stream bottom need to take into account those attributes or characteristics that potentially promote the best physical habitat or environment for aquatic life independent of water quality. This concept can be seen in Figure 2, which shows the conceptual relationship between habitat and biological quality. In this figure, the dashed red line indicates the expected stream habitat to biological condition curve. Figure 2 can best be summarized by the following four points relating to specific areas of the graph.

1. The upper right-hand corner of the curve is the ideal situation where optimal habitat quality and biological condition occur.
2. The decrease in biological condition is proportional to a decrease in habitat quality.
3. The lower right-hand corner is where degraded biological condition can be attributed to something other than habitat quality.
4. The upper left-hand corner is where optimal biological condition is not possible in a severely degraded habitat.

Section 3 of the guidance presents methods to be used in evaluating in-stream physical habitat, through the measurement of **stream bottom substrate** indicators. It also identifies methods for evaluating the **biological condition** of macroinvertebrates or fish. Methods for assessing biological impairment due to causes other than sediment deposition are not considered in this guidance. To determine the overall attainment of the sediment standard the combination of results from substrate evaluation and biological condition are plotted in the Sediment Standard Attainment Matrix in Section 4. Assessment categories and the percent comparability to the expected condition in the matrix are based on those in Figure 2.

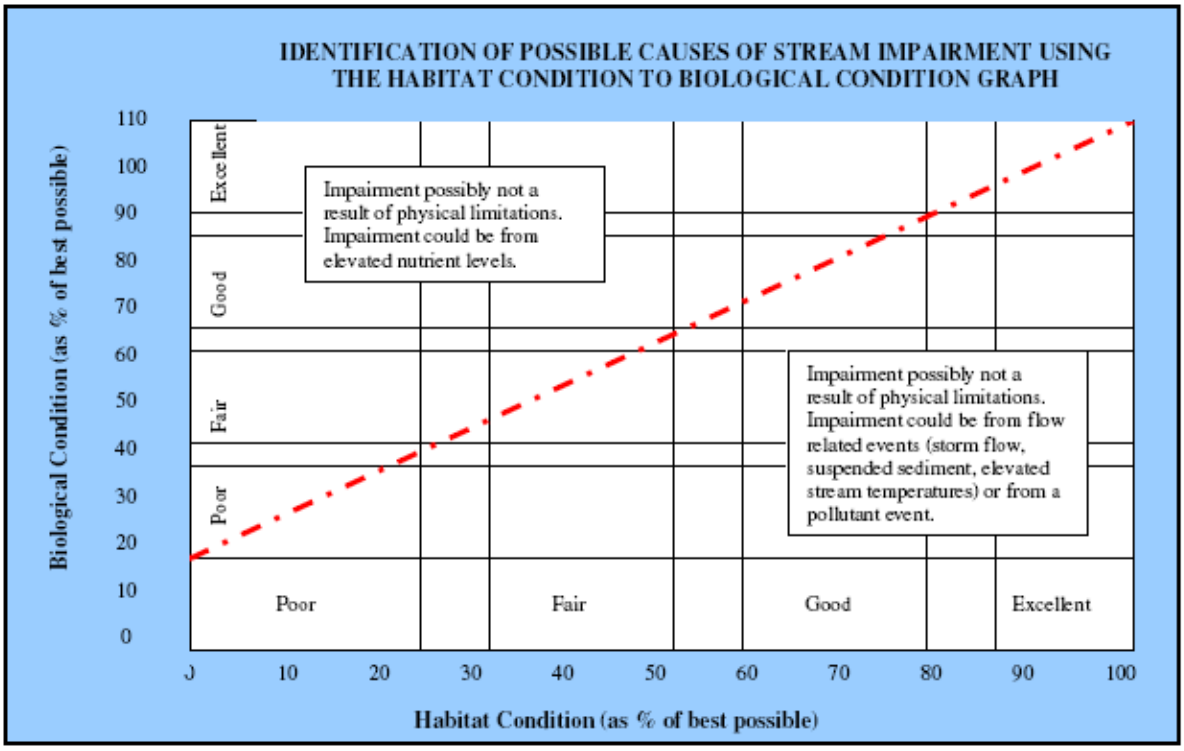


Figure 2. Conceptual Relationship between Habitat and Biological Condition
(Dashed line represents expected stream habitat to biological condition curve.)

Footnote: The above figure shows the general relationship between habitat and biological condition. However, it should be noted that sustainable, healthy biological communities can exist that are adapted to poor habitat conditions. Expected condition stream habitat quality may be poor, but it can have a robust, sustainable biologic community, with unique and important adaptation of species assemblages.

Natural Sources of Sediment

All stages of a sediment impairment assessment should consider natural sources of sediment. If a determination is made that the sediment responsible for the observed impairment is being contributed from natural sources then the sediment deposition analysis should be terminated. The evidence used to determine that natural sources are responsible should be well documented. This guidance does not discuss methods for determining if the sediment observed in a channel is the result of natural geologic sources and processes. However, if a study group determines that natural geologic sources and processes contribute all or most of the sediment to a candidate reach, then further assessment of the attainment of the narrative standard would not be warranted.

Temporal Scale Considerations

EPA guidance for sediment TMDL development (EPA 1999) discusses several important temporal factors that should be considered during each phase of a sediment impairment analysis such as the seasonal variability of sediment discharges and associated beneficial use impacts. Like most nonpoint source pollutants, sediment discharges are not continuous in magnitude and effect, and are more likely to increase as runoff increases.

The EPA guidance points out that sediment discharges vary substantially in their timing, depending primarily on the sources, watershed geology and landform, and precipitation/runoff patterns. Some sources are always vulnerable to erosion (e.g. bank erosion and continuously cultivated land), while other sources are vulnerable only during and shortly after land disturbing activities. In addition, some areas do not function as significant sediment sources except in response to extreme events. Analysts should assess whether sampling schedules and field methods are capable of adequately accounting for, or detecting temporal variability. The sampling schedule and field methods used during the assessment should be well documented in the project SAPP (Sampling and Analysis Project Plan) to address these concerns.

Stream Bottom Substrate Evaluation

Chapman and McLeod (1987) suggest that geometric particle size and percent of the bed surface covered by fines should be used to define habitat quality. These criteria can be determined by performing a pebble count. Pebble counts provide not only particle size distributions (D50, D84, etc…) and percent class sizes (% sand, % cobble, etc…), but offer a relatively fast and statistically reliable method for obtaining this information.

Sufficient and varied sizes of stream bottom substrate are necessary for biological colonization, protection, and reproduction. However, the full biological potential may not be realized if the substrate surfaces are surrounds by fine sediment. In streams containing excess amounts of sediment, the coarser particles become surrounded or partially buried by fine sediment. Insect populations decline substantially as interstitial

spaces become smaller and filled. Embeddedness quantitatively measures the extent to which larger particles are surrounded or buried by fine sediment (Mc Donald et al., 1991).
By performing a pebble count and/or measuring embeddedness, the amount of aquatic habitat can be characterized compared to an expected condition, and then cautiously evaluated for impairment due to stream bottom deposits. If it is determined excess stream bottom deposits exist beyond the expected condition, then confirmation of impairment takes place when a stream site is biologically assessed.

Pebble Count

The pebble count (Wolman, 1954) may be performed separately or as part of a larger stream inventory and assessment study (Rosgen, 1996). The Division has a pebble count protocol and recommends that assessing parties make use of the protocol when performing pebble counts. Other appropriate pebble count methods include Wolman, Bevenger and King, Bunte and Abt. Pebble counts may be recorded, tallied, and represented either by using forms in the SOP or on a computer laptop at streamside using the Expected condition Reach (channel materials) software package (Mecklenberg, 1998) which can be downloaded from the State of Ohio Department of Natural Resources website (http: www.dnr.state.oh.us/soilandwater/streammorphology.htm). Another software that can be used is the Size-Class Pebble Count Analyzer VI 2001.xls (651KB) by John Potyondi and Kristin Bunte from the US Forest Service's Stream System Technology Center (aka "Stream Team") website (www.stream.fs.fed.us) under their Download PDF Documents and Software Tools menu. Specific information concerning the program's use, application, sample size, data input, statistical analysis, and case studies are included in various document sections of the software and should be read prior to setting up a study and collecting data.

In a study of 1134 streams located in four northwestern states, Relyea et al. (2000) suggested that changes to invertebrate communities as a result of fine sediment (2mm or less) occur between 20 – 30% fines. A strong correlation between the health of macroinvertebrate communities and percent surface fines for particles <2mm has been shown in her work. The most sensitive species were affected at 20% surface fines. For streams with aquatic life of fish concerns, measurement of particles <6.35mm are commonly used to describe spawning gravel quality and includes the size range typically generated by land management activities (Weaver and Fraley, 1991). Weaver and Fraley (1991) observed a significant inverse relationship between the percentage of material <6.35mm and the emergence success of trout species.

The Division has considered various particle sizes between ≤2mm and ≤8mm for a defined particle size for this guidance. Various state and federal agencies in Colorado have conducted studies using the range of particle sizes, but the prevalent size used to define percent fines is 6.35mm. 6.35mm is a particle size well grounded in fisheries literature as the 0.25 inch threshold considered detrimental to coldwater fish species. (Chapman 1988). This protocol does not preclude the use of studies using other

particle sizes if the data is available. Site-specific studies may utilize a differently defined particle size, such as 2mm for percent fines. According to the above-mentioned work done by Relyea in Idaho, 2mm is protective of macroinvertebrates, although in some trout streams, 2mm may not be a large enough particle size to protect the fisheries aquatic life.

When conducting a pebble count, the assessor uses the pebble count software streamside at the study site, to calculate the percent fines for ≤6.35mm. If the percent fines are ≤ 20%, the study site should be evaluated as fully supporting (FS) for substrate. Percent fines of ≤ 20% is the percent fines stated in literature and recent studies as a threshold for damage to habitat conditions and macroinvertebrates. On the other side of the coin, the threshold value for damage that is not supporting of aquatic life use is percent fines ≥ 40%. If the percent fines for a study site is ≥ 40%, the stream should be evaluated as not supporting (NS) for substrate. The assessment will then move along to the biological assessment and there is no need to compare substrate analysis with the expected condition in an expected condition reach.

If the results for percent fines of the study stream are not one of the threshold values listed above (≤20% and ≥40%), the expected condition should be identified and assessed for substrate analysis as well. The study stream would then be assessed as a percentage of the expected condition and the percentage would then be applied in the final assessment matrix.

Embeddedness

A preferable technique for ascertaining embeddedness is the Burton and Harvey method (1990). This method should only be used on cobble-bottom or cobble-dominated streams, where the greatest percent fraction of any group is cobble. This method is labor intensive and its use is recommended when data from the pebble count and biological sampling does not provide a satisfactory answer as to the degree of impairment. Embeddedness measurements should be performed on the same stream reach where the pebble count was performed, only upstream of the actual pebble count transects, so as not to measure the areas disturbed by the earlier measurements.

Studies by Bjorn et al. (1974, 1977) concluded that approximately one-third embeddedness (33%) or less is probably the normal condition in proper functioning streams. Above this condition, however, insect populations decline substantially as habitat spaces become smaller and filled. After completing embeddedness measurements at the study site, calculate the percent embeddedness. If the percent embeddedness is ≤ 33%, the study site should be evaluated as FS for substrate. If the percent embeddedness is ≥ 60%, the study site should be evaluated as NS for substrate. The assessment will then move along to the biological assessment and there is no need to compare substrate analysis with an expected condition reach. If the results for embeddedness are not within the threshold values above (≤33% and ≥60%), an expected condition should be identified and assessed in a expected condition reach for embeddedness as well. The study stream would then be assessed as a percentage

of the expected condition and the percentage would then be applied in the final assessment matrix.

Although percent fines and percent embeddedness are the preferred methods for ascertaining substrate support status, there are other methodologies available. Table 2 contains a list of methods for commonly measured indicators with expected conditions that can be used to compare the substrate of the study reach with the expected condition. It is important that whatever method is chosen, the data collection sampling, amount and intensity must be the same for expected condition and impacted sites and under similar climate/flow conditions.

The list in table 2 is not exhaustive, and some assessments may use other established or documented methods. These additional methods do not have established threshold values or ranges. If the assessor wishes to utilize these other methods to determine substrate impairment, an expected condition will have to be selected and the results expressed as a percentage of the expected condition. There are basically only two requirements in selecting an indicator(s). First the indicator(s) must be quantitative. Second, the result of measuring the indicator at the candidate reach must be expressed as a percentage of the result at the expected condition reach. Detailed documentation of the selected indicator and how it was measured in the field should be included in every sediment impact assessment.

Degree of Aquatic Life Use Support for Substrate

The information collected during the stream bottom substrate evaluation is applied to the use support matrix in Table 3. Percent fines and percent embeddedness **not falling within the threshold values** are compared to the expected condition values for percent fines and percent embeddedness and expressed as percent of the expected condition. The use support categories for substrate are as follows: 90 – 100% of expected is FS, 73 – 89% of expected is Supporting, Impacts Observed, and $\leq$72% of expected is NS.

Additional statistical analysis is not necessary to compare the measured condition with the expected condition to compare to the support categories. There is error associated with conducting pebble counts and field analyses, but these are addressed with the methodology utilized and with the streamside software used to calculate % fines. The percentages associated with use support categories are comparable to percentages used by other states and agencies for substrate analysis. Designating a number signifying acceptable or unacceptable aquatic life health is difficult without a single best answer. The above percentages designated for use support for substrates are similar to the concept of the ratios used in RBP protocols and T-Walk (USFS) protocols to compare measured with expected conditions.

Table 2. Selected stream bottom substrate indicators and references.

INDICATOR	QUANTITY MEASURED	REFERENCES
Intergravel living space using embeddedness	Salmonid living space available in coarse particle substrate	Burton and Harvey, 1990
CDPHE-WQCD Riffle/Run Habitat Analysis Parameter 4	Percent of stream bed composed of fines $\leq$2mm, $\leq$6.35mm	Colorado Department of Public Health and Environment, Water Quality Control Division, (not dated)
CDPHE-WQCD Glide/Pool Habitat Analysis Parameter 6	Percent of pool bottom affected by sediment deposition	Colorado Department of Public Health and Environment, Water Quality Control Division, (not dated)
V^* for pools	Volume of pool occupied by fine sediment	Lisle and Hilton, 1992
In-situ flow through samplers	Accumulation of fine particles in interstitial spaces of coarse particle substrate	Carling and McCahon, 1987; Frostick et al., 1984
Freeze core sampling	Subsurface particle size distribution	Petts, 1988; Lisle, 1989
In-situ sampling of known volume	Subsurface particle size distribution	Lambert and Walling, 1998; MacDonald et al., 1991, p.119; Platts et al., 1983, p.17
Embeddedness	Extent to which large particles are embedded or buried by fine sediment	MacDonald et al., 1991, p. 121
Pebble Counts	Surface particle size distribution	Wolman, 1954, Bevenger and King, 1995

Table 3: Degree of aquatic life use support affected by stream bottom deposits (sediment) evaluated by increase in either fines or embeddedness, relative to an expected condition.

Pebble Count Fines **≤ 2mm, ≤6.35mm** (% Of Expected)	**% Embeddedness** (% Of Expected)	**Degree of Aquatic Life Use Support** **For Substrate (Presumptive)**
90 – 100%	**90 – 100%**	**Fully Supporting[1]**
73 - 89%	**73 - 89%**	**Supporting, Impacts Observed**
≤ 72%	**≤ 72%**	**Not Supporting[2]**

[1]Raw percent values of ≤ 20% fines, ≤ 33% embeddedness calculated at a study site should be evaluated as supporting for substrate regardless of the percent attained at the expected condition site.
[2]Raw percent values of ≥ 40% fines, ≥ 60% embeddedness calculated at a study site should be evaluated as not supporting for substrate regardless of the percent attained at the expected condition site.

Bioassessment

The bioassessment step is accomplished by assessing the condition of the benthic macroinvertebrate community and/or the fish community at the same location that the stream bottom substrate assessment is conducted. Benthic macroinvertebrates will be assessed in most studies because they are generally better indicators of impairment due to sediment deposition than are fish. However, there can be situations where fish assessments should be conducted because they will provide a more sensitive or definitive assessment of the impacts to aquatic life. The results of the bioassessment are combined with the stream substrate evaluation results in the final assessment matrix to determine whether standards are attained (Section 4).

The recommended field and laboratory protocols for the benthic macroinvertebrate assessments are the *Standard Operating Procedures for the Collection and Processing of Benthic Macroinvertebrates* (Basic Protocol) and the *Standard Operating Procedures for the Collection and Processing of Benthic Macroinvertebrates by the Enhanced Protocol,* which are found in Water Quality Monitoring in Colorado (Colorado Water Quality Forum, 1995, draft). Similar protocols such as the EPA's Rapid Bioassessment RBP-III for benthic invertebrates (Plafkin et al., 1989) are also recommended. Sampling of fish populations should be conducted according to Colorado Division of Wildlife methods for inventory and population estimates.

> The choice of the Basic (or RBP-III) or Enhanced Protocol depends on the resources available and the desired degree of analytical rigor. Benthic macroinvertebrate data generated by these protocols is typically used to calculate various indices of community structure such as those found in RBP III. Sometimes these measures of community structure are not sensitive enough to detect sediment impacts. In order to provide more sensitive measures of sediment impacts it is recommended that biomass, abundance, and the presence of sediment tolerant and intolerant taxa be measured in addition to the common measures of community structure found in the RBP.

Application of the biological assessment or degree of impairment is a percentage comparison of the sum of selected metric scores at the study site compared to a selected expected condition (site). The value will be expressed as a percentage of expected condition. Apply the value calculated to the use support matrix Table 4. The use support categories for biological assessment are as follows: 80 – 100% of expected condition is FS, 51 – 79% of expected condition is Supporting, Impacts Observed, $\leq$50% of expected condition is NS.

Table 4: Biological Integrity Attainment Matrix.

% Comparison to Expected	Biological Condition Category	Attributes[1]
80 – 100%	**Supporting**	**Comparable to best situation to be expected within ecoregion. Balanced trophic structure. Optimum community structure (composition and dominance) for stream size and habitat quality.**
51 – 79%	**Supporting, Impacts Observed**	**Community structure less than expected. Composition (species richness lower than expected due to loss of some intolerant forms. % Composition of tolerant forms increases.**
≤ 50%	**Not Supporting**	**Fewer species due to loss of most intolerant forms. Reduction in EPT index. Densities of organisms dominated by one or two taxa.**

[1]Biological attributes from EPA's Rapid Bioassessment Protocols for Use in Stream and Rivers, (Plafkin et al., 1989).

In order to assure the appropriate metrics are being analyzed to show impairment due to excess sediment, biological metrics are listed in table 5 that have shown to be sensitive to sedimentation. Determining which metrics to use in an assessment will require best professional judgment.

Table 5.a **Macroinvertebrate Metrics Sensitive to Sedimentation Effects**
* See footnote below

Metric Categories	**Metric**	**Definition**	**Predicted response to increasing perturbation**
Richness	Total Taxa	Number of distinct taxa in the macroinvertebrate assemblage	Decrease
	Ephemeroptera Taxa	Number of Mayfly taxa	Decrease
	Plecoptera Taxa	Number of Stonefly taxa	Decrease
	Tricoptera Taxa	Number of Caddisfly taxa	Decrease
Composition	Percent Plecoptera	Percent of sample that is stonefly nymphs	Decrease
Pollution Tolerance	Hilsenhoff Biotic Index	Abundance-weighted average tolerance of organisms to pollution (Hilsenhoff 1987)	Increase
Diversity	Percent Five Dominant Taxa	Percent of sample in the most abundant five taxa	Increase
Feeding Group	Scraper Taxa	Number of taxa that scrape periphyton from substrates	Decrease
Habit	Clinger Taxa	Number of taxa that have fixed retreats or adaptations for attachment to surface in flowing water	Decrease

(Jessup and Gerritson 2000)

Footnote: Recent EPA studies in mountainous areas have shown that the number of clinger taxa provides the strongest indication of sediment impairment. The percentage of clinger taxa is also a supplemental indicator.

Table 5.b Macroinvertebrate Metrics and Changes Following Disturbances

Metric	Definition	Change
Number of Taxa	Number of distinct taxa	Decrease
Number of EPT Taxa	Number of distinct taxa in EPT	Decrease
Simpson's Dominance Index	An index measuring the dominance of the community by one or a few taxa	Increase
Percent Dominant Taxon	Relative abundance of the most common taxa	Increase
Hilsenhoff's Biotic Index	Calculated using tolerance values for invertebrates	Increase
Percent Elmidae	Relative abundance of the riffle beetles (Coleoptera: Elmidae)	Decrease
Percent Hydropsychidae	Relative abundance of the net-spinning caddisflies (Tricoptera: Hydropsychidae)	Increase
Percent Hirudinea	Relative abundance of leeches	Increase
Percent Chironomidae	Relative abundance of midges (Diptera: Chironomidae)	Increase
Percent Oligochaeta	Relative abundance of aquatic worms	Increase
Percent Gatherers	Relative abundance of this functional group	Variable
Percent Scrapers	Relative abundance of this functional group	Decrease
Percent Shredders	Relative abundance of this functional group	Decrease
Percent Filterers	Relative abundance of this functional group	Increase
Percent Miners	Relative abundance of this functional group	Increase

Table 5.c Fish Metrics and Response to Increasing Perturbation

Metric Categories	Metric	Definition	Predicted Response to Increasing Perturbation
Richness and Composition	Number of cold water native species	Number of native fish species typically found in cold water streams. Excludes introduced or tolerant native fish species.	Decrease
	% Cold water individuals	Percent of individuals found in cold water streams. Includes introduced trout species.	Decrease
	% Sensitive native individuals	Percent of native individuals sensitive to perturbations	Decrease
Reproductive Function	Number of age classes	Number of age classes (use measured size classes to infer) reflects the availability of unembedded cobble	Decrease
Abundance	Catch per unit effort	Number of cold water individuals per minute of single-pass electrofishing	Decrease

(Jessup and Gerritson 2000)

Secondary Channel Characteristics

Macroinvertebrate analyses are time consuming and often expensive for agencies and individuals with too few resources. Channel characteristics can be used as secondary measures to confirm the results of substrate analyses. If the stream bottom substrate analysis provides assessed numbers between 20% and 40% for percent fines or 33 and 60 percent embeddedness and fines or embeddedness are 89 – 73% of the expected condition, secondary channel characteristics are used to verify the presence of sediment deposits that may impair the aquatic life use. If these channel measures are similar to expected conditions (>72% of expected), the substrate is evaluated as fully supporting and no additional assessment is needed. If the channel measures are significantly different from expected conditions, the assessor would then move on to the biological assessment.

Stream channel assessments should be done at the reach scale and should analyze stream channel condition and geomorphology. A comparison between expected and suspected impaired conditions is necessary. The assessor should be aware of riparian

condition, since riparian vegetation is extremely important in maintaining channel stability, natural filter, groundwater/surface water interactions, etc. This will be the case for perennial and some intermittent streams. For ephemeral streams, vegetation may not be critical, and an assessment of channel morphology characteristics will be sufficient. The morphologic variables and riparian components to collect are suggested blow.

Channel Characteristics

The following are some common channel metrics or parameters that indicate good habitat and channel stability. These channel metrics are compared to expected conditions according to the percentages for "Habitat Quality" from Figure 1:
90 - 100% of Expected Condition = Supporting
89 – 73% of Expected Condition = Partially Supporting
≤ 72% of Expected Condition = Nonsupporting

Use of past and recent aerial photographs to determine changes in sinuosity and stream length. Has sinuosity and concomitant stream length decreased over time?
Percent of raw banks for the reach compared to expected conditions
RSI – Riffle Stability Index (very applicable to cold water biota) not applicable to plains
Riffle-riffle spacing
Pool-pool spacing
Bank Stability % eroding banks
Bank erosion potential (Rosgen 1996)
Channel stability based upon bankfull indicators (e.g. entrenchment, width/depth ratio, channel materials (D16, D50, and D84), sub pavement particle size distribution, and slope
V*
D50 – median particle size
Pool Frequency
Intergravel DO (dissolved oxygen)
Suspended sediment/dissolved solids
Riparian vegetation assessment using BLM/USFS guidance – *A user guide to assessing proper functioning condition*, or similar methodology

The assessor should select 3 metrics to measure from these channel measurements. If 2 out of 3 are "Supporting", the stream can be evaluated as FS for sediment. If 2 out of 3 are "Partially Supporting", the stream would be evaluated as "Supporting, Impacts Observed" for sediment and the assessment would then proceed to the biological analysis. If 2 out of 3 are "Nonsupporting", the stream would be evaluated as "Not Supporting" for sediment and the assessment would then proceed to the biological analysis. It is important for the assessor to document the methodologies utilized and the comparison between expected condition reach and study reach.

Steps of Sediment Analysis

This guidance is intended to represent a common approach to assessing streams for the impacts of sediment deposition. The guidance may be utilized by agencies, watershed groups, or other stakeholders; however, it is recommended that those proposing to utilize this guidance consult with the Division before designing a stream study. Such consultation may help improve the focus of such a study, and also help insure that such a study is performed properly, such that it can be utilized by the Division and Commission. Figures 4 and 5 show the flowchart for determining aquatic life use impairment due to excess sediment.

Step 1. Identify candidate sediment impacted segments

This step is a screening level identification of stream reaches or segments where sediment impacts are known to occur or suspected to occur. Existing information can be compiled from information in the §303(d) list, and the §305(b) report, watershed protection program reports, and in reports from other governmental agencies. In addition, data can be gathered by screening level reconnaissance surveys. Other means to identify these segments can include land use information, agency resource assessments, anecdotal reports, and public comment, where such are found to meet a threshold of reliability.

Step 2. Perform sediment substrate analysis

This step is explained in full in section 3. The assessor will perform a pebble count or measure percent embeddedness. Percent fines and/or percent embeddedness will be used in table 3 to determine if the values are within the threshold values. If % fines is $\leq$ 20% and/or % embeddedness is $\leq$ 33%, the assessment is assumed FS for substrate regardless of expected condition and the assessment is complete. If the values are not within the threshold values, an expected condition must be defined and substrate values expressed as a percent of expected condition. If the % fines are $\geq$40% and/or % embeddedness is $\geq$60%, the assessment for substrate is considered NS and the assessment continues on to biological assessment.

Step 3. Establish expected condition criteria

Step 3 is the process of characterizing and classifying the study stream by identifying the watershed, ecoregion, flow regime, channel morphology or type, geological, physical, and other relevant chemical, and biological attributes that are crucial for the selection of a matching expected condition. This information is then used to match the candidate stream to the expected condition to the maximum extent practicable. Data is collected through field assessments and by mapping and GIS techniques.

Step 4. Identify expected condition

Step 3 and Step 4 are closely related and when completed provide the expected condition that provides the basis of comparison to the specific study stream site or stream reach. In step 4, the actual expected condition is identified through a tiered approach that can range from site-specific sites to the use of conceptual or modeled

expected conditions developed by expert consensus. This process is described in more detail in Section 2. Field surveys and mapping techniques similar to those used in Step 3 can be used to identify actual expected condition streams or sites, with the expected condition classified according to Step 3 criteria.

Step 5. Comparison of study segment with expected condition

Step 5 provides the comparison of the stream bottom substrate habitat (as it relates to sediment deposition) and accompanying aquatic life with that of the expected condition. This requires the use of the methods identified in Section 4 for collecting the data at the expected condition and study sites. The field data collection for this step can be performed concurrently with Steps 3 and 4 or can be conducted later in the process or at multiple times during the assessment. This process provides the information necessary to determine the percentage of expected condition for the habitat and biological metrics or assessment endpoints

Step 6. Secondary Channel Measurements

Step 6 is an option for those streams whose values are fall between the threshold values for substrate for raw data, i.e. 20 – 40% fines and 33 – 60% embeddedness. Assessors may choose to use secondary channel measurements discussed in section 3. If the values indicate FS, the assessment is complete. If the values do not show FS, the assessment moves on to biological assessment.

Step 7. Assess condition or degree of sediment impacts

This is the final step in the process of determining the status of the aquatic life uses as impacted by sediment deposition. At this step, categories of narrative standard attainment are assigned, based on the combination of percentage of expected condition for physical habitat and percentage of expected condition for biology. Section 4 shows suggested matrices of narrative sediment standard attainment.

FIGURE 4

Figure 4. Flow chart for Determining Sediment Impairment Assessment

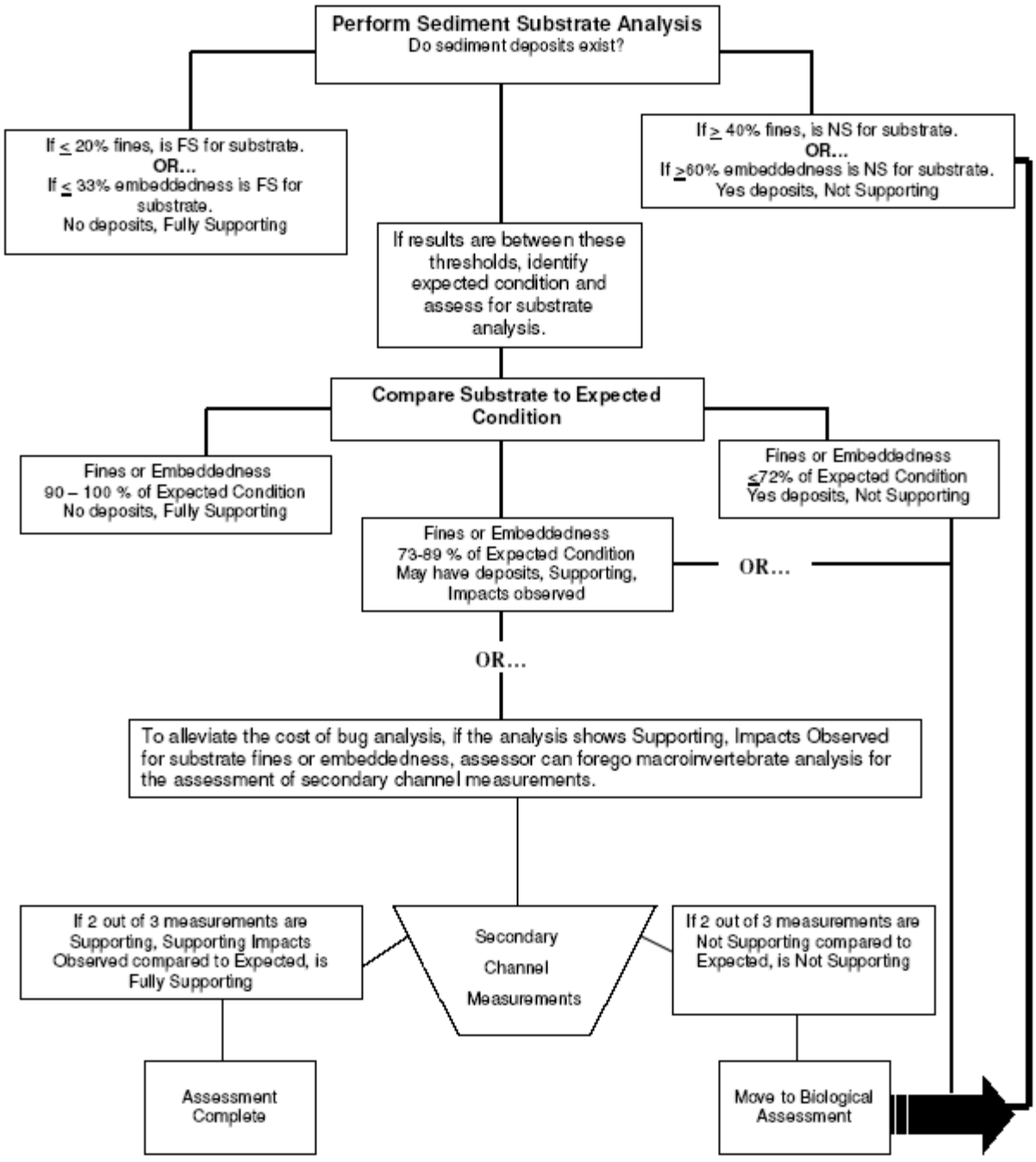
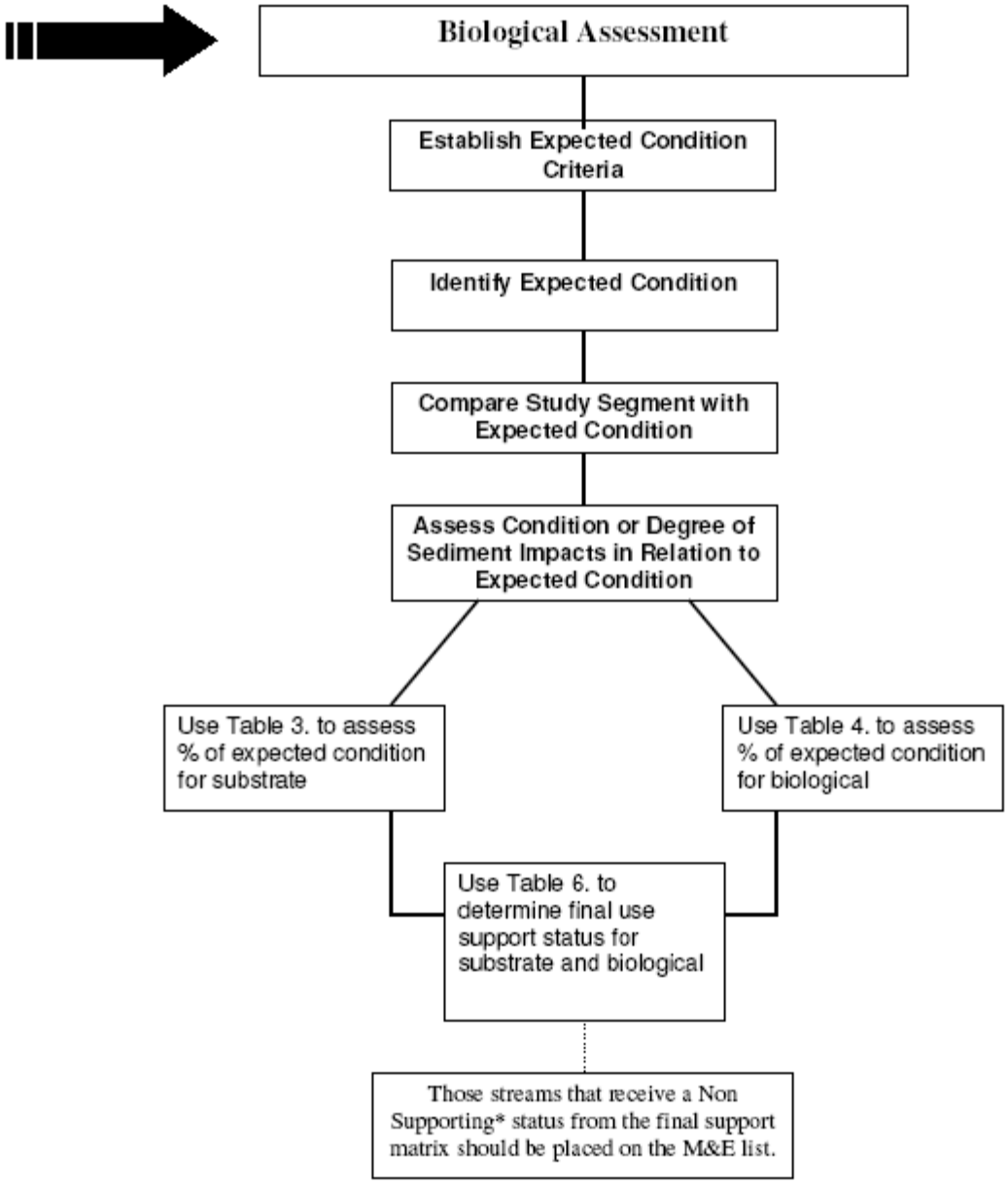


FIGURE 5

Figure 5. Flowchart for Determining Biological Impairment for Sediment Assessment



4. ATTAINMENT OF THE NARRATIVE STANDARD

The narrative standard states that the waters of the state will be free from substances which “can settle to form bottom deposits detrimental to the beneficial uses ”. The process to determine whether the narrative standard is attained is described below and involves comparing the stream substrate condition to the biological condition present at the same location. This process requires the use of a reasonable expected condition, which allows for the determination of percent of expected condition.

The standards attainment criteria in Table 6, the final attainment matrix, have been extrapolated from Figure 2, which illustrates the general relationship between habitat quality and biological conditions. Figure 2, and EPA's Rapid Bioassessment Protocols, indicate that the aquatic biological community varies with habitat quality and that as habitat quality declines discernible biological impairment results, assuming the absence of other confounding instream effects (water chemistry or toxic substances).

This guidance is designed to determine impacts to the aquatic life uses that result from the physical deposition of sediment. In order for there to be a non-attainment of the narrative standard there must be a concurrent demonstration of biological impact and sediment deposition to the stream substrate. For those assessments where either substrate alone or biology alone shows an impact as a percent of expected condition then the sediment standard is attained.

In the case of moderate to severe biological impacts found in streams attaining the narrative standard, the impairment is due to chemical toxicity or physical factors (flow, temperature, flooding) that can cause discernible biological impairment and must be considered. In these cases a finding of nonsupport of the aquatic life uses may be made, but some cause other than deposition of sediment must be observed, and listed as the cause of such nonsupport. Streams showing a determination of impairment biologically, but not physically, should be assessed for further determination of the source of impairment. It is then important that a complete habitat assessment and chemical studies and other sampling and monitoring protocols be utilized at locations in order to insure a full understanding of stream health.

Table 6: Final assessment matrix for determining aquatic life use support categories by combining physical (% fines and embeddedness) and biological assessments as sediment indicators.

Biological → **% of expected** **Physical ↓** **% of expected**	**NS** **≤ 50%**	**Supporting, Impacts Observed** **51 – 79%**	**Supporting** **80 – 100%**
NS **≤72%**	Not Supporting	Supporting, Impacts Observed	Supporting, Impacts Observed
Supporting, Impacts Observed **89 – 73%**	Not Supporting, Other Pollutant Likely*	Supporting, Impacts Observed	Supporting
Supporting **90 – 100%**	Not Supporting, Other Pollutant Likely*	Supporting	Supporting

*** Impairment in this support level for aquatic life is probably not due to sediment. It is likely the result of other impairment, alone or in combination with sediment. These streams should be evaluated for impairment source determination.**

Raw percent values of < 20% fines, < 33% embeddedness calculated at a study site should be evaluated as supporting for substrate regardless of the percent attained at the expected condition site.

Raw percent values of > 40% fines, > 60% embeddedness calculated at a study site should be evaluated as not supporting for substrate regardless of the percent attained at the expected condition site.

5. APPLICATION OF THE GUIDANCE

The Commission is hopeful that this guidance will prove to be a useful step toward providing a consistent approach to implementation of the statewide narrative basic standard that addresses sediment deposition for those streams which this guidance is intended to address, i.e. high gradient, montaine streams and not sandy – bottom plains streams. In approving this guidance the Commission recognized that there might be a number of technical issues that will need further refinement and that as the guidance is used and data is gathered, the guidance will periodically need to reviewed and updated. The Commission determined that, where possible, the Division should focus on segments with stakeholders, broadly defined, in its implementation of this guidance, for both conducting and participating in sediment impact evaluations; that an advisory group should be reconvened to help evaluate implementation of the guidance, and the Division should maintain a data base listing sediment assessment projects. Should the experience gained from implementation indicate that the guidance needs to be modified, or supplemented, appropriate action can be taken at that time. There are also several new developments on clean sediment guidance that the Federal Government has been working on that could prove to be helpful additions/revisions for this guidance. For example, the EPA will soon release clean sediment criteria guidance. Once published, the Division will evaluate how this guidance can be updated to reflect EPA's recommendations. The EPA is also developing a Fine Sediment Index (FSI) applicable to mountain streams. An FSI would be an ideal goal for the Division to reach in the future. For information about this guidance please contact the Water Quality Control Division at (303) 692-3500 and ask for the Monitoring Unit.

Appendix A – References

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Appendix B– Examples of Calculation

Examples assume that an appropriate stream reach has been selected for the stream in question and that the study reach adequately captures the evident stream features. The examples also assume that the assessor has selected the appropriate number of counts to be conducted in the pebble count.

Example 1.

The first step is to determine if sediment deposits are present. Using the CDPHE SOP for pebble counts, 400 counts have been recorded. For the stream in question, the ≤6.35mm particle size will be assessed for impairment of aquatic life.

Soapy Creek: After the pebble count was performed, the assessor uses the Potyondi and Bunte Size-Class Pebble Count Analyzer VI 2001.xls to calculate percent fines. The calculation is performed streamside.

Percent fines ≤6.35mm = 10.7%

Following the assessment flowchart, because the stream meets the threshold of ≤20% fines, Soapy Creek is automatically determined to be Fully Supporting for substrate and no further assessment is necessary.

Example 2.

The first step is to determine if sediment deposits are present. Using the CDPHE SOP for pebble counts, 400 counts have been recorded. For the stream in question, the ≤6.35mm particle size will be assessed for impairment of aquatic life.

Barrel Creek: After the pebble count was performed, the assessor uses the Potyondi and Bunte Size-Class Pebble Count Analyzer VI 2001.xls to calculate percent fines. The calculation is performed streamside.

Percent fines ≤6.35mm = 42.3%

Following the assessment flowchart, because the stream meets the threshold of ≥40% fines, Barrel Creek is automatically determined to be Non Supporting for substrate and the assessment continues on to biological assessment.

For the biological assessment of Barrel Creek, a reference site (Wagon Creek) has been selected and macroinvertebrates have been collected for both Barrel Creek and Wagon Creek using the same protocols.

The macroinvertebrate metric used for this example is total number of EPT taxa.

Barrel Creek = 3 of 9 taxa present are EPT taxa
Wagon Creek = 8 of 15 taxa present are EPT taxa

3 / 8 = 37.5% of expected condition

Using table 4, the biological integrity attainment matrix, ≤50% of expected condition is Non Supporting.

Looking at table 6, the final attainment matrix, a NS for substrate and a NS for biological is determined to be Non Supporting. This stream would therefore be eligible for 303(d) listing.

Example 3.

The first step is to determine if sediment deposits are present. Using the CDPHE SOP for pebble counts, 400 counts have been recorded. For the stream in question, the ≤2mm particle size will be assessed for impairment of aquatic life.

Alcohol Creek: After the pebble count was performed, the assessor uses the Potyondi and Bunte Size-Class Pebble Count Analyzer VI 2001.xls to calculate percent fines. The calculation is performed streamside.

Percent fines ≤2mm = 39.7%

Following the assessment flowchart, the study creek may have deposits detrimental to aquatic life. Because the percent fines falls between the two thresholds, a comparison to expected condition for substrate is required.

A expected condition creek, Straight Creek, is selected and the same pebble count protocols are applied.
Straight Creek is determined to have 9% fines ≤2mm.

To calculate percent of expected condition the following calculations are made:

Alcohol Creek has 39.7% fines, which means that 60.3% of Alcohol Creek is >2mm.
Straight Creek has 9% fines, which means that 91% of Straight Creek is >2mm.

.603 / .91 = .6626 * 100 = 66.3% of expected condition.

Looking at table 3, the substrate attainment matrix, 66.3% of expected condition is Non Supporting. The assessment would then move on to biological assessment.

The macroinvertebrate metric used for this example is percent taxa EPT.

Alcohol Creek = 5 of 11 taxa were EPT
Straight Creek = 8 of 15 taxa were EPT

5 / 11 = 0.45
8 / 15 = 0.53

0.45 / 0.53 = .849 * 100 = 85% of expected condition for biological.

Using table 4, the biological integrity attainment matrix, 85% of expected condition is Supporting, Impacts Observed.

Looking at table 6, the final attainment matrix, NS for substrate and Supporting, Impacts Observed for biological, the final determination is Supporting, Impacts Observed.

Example 4.

The first step is to determine if sediment deposits are present. Using the CDPHE SOP for pebble counts, 400 counts have been recorded. For the stream in question, the ≤6.35mm particle size will be assessed for impairment of aquatic life.

Migraine Creek: After the pebble count was performed, the assessor uses the Expected condition Reach Channel Materials software package to calculate percent fines. The calculation is performed streamside.

Percent fines ≤6.35mm = 25%

Following the assessment flowchart, the study creek may have deposits detrimental to aquatic life. Because the percent fines falls between the two thresholds, a comparison to expected condition for substrate is required.

A expected condition creek, Tylenol Creek, is selected and the same pebble count protocols are applied.
Tylenol Creek is determined to have 24% fines ≤6.35mm.

To calculate percent of expected condition the following calculations are made:

Migraine Creek has 25% fines, which means that 75% of Migraine Creek is >6.35mm.
Tylenol Creek has 24% fines, which means that 76% of Tylenol Creek is >6.35mm.

.75 / .76 = .986 * 100 = 98.6% of expected condition.

Looking at table 3, the substrate attainment matrix, 98.6% of expected condition is Fully Supporting for substrate and no further assessment is necessary.

Example 5.

The first step is to determine if sediment deposits are present. Using the CDPHE SOP for pebble counts, 400 counts have been recorded. For the stream in question, the ≤8mm particle size will be assessed for impairment of aquatic life. .

Dead Cow Creek: After the pebble count was performed, the assessor uses the Potyondi and Bunte Size-Class Pebble Count Analyzer VI 2001.xls to calculate percent fines. The calculation is performed streamside.

Percent fines ≤8mm = 30%

Following the assessment flowchart, the study creek may have deposits detrimental to aquatic life. Because the percent fines falls between the two thresholds, a comparison to expected condition for substrate is required.

A expected condition creek, Happy Cow Creek, is selected and the same pebble count protocols are applied. Happy Cow Creek is determined to have 20% fines ≤8mm.

To calculate percent of expected condition the following calculations are made:

Dead Cow Creek has 30% fines, which means that 70% of Dead Cow Creek is >8mm.
Happy Cow Creek has 20% fines, which means that 80% of Happy Cow Creek is >8mm.

.70 / .80 = .875 * 100 = 87.5% of expected condition

Looking at table 3, the substrate attainment matrix, 87.5% of expected condition is Supporting, Impacts Observed.

Following the assessment flowchart, the assessor has two options at this point. The assessor may either go to biological assessment, or has the option of assessing secondary channel characteristics. For the sake of the example, the assessor chooses the channel characteristics.

The assessor chooses 3 metrics to measure for both Dead Cow and Happy Cow Creeks. They are:

Bank Stability % eroding banks
Riffle Stability Index (RSI)
Pool Frequency

These are compared between both the study stream and expected condition stream for % of expected condition.

Bank Stability:
Dead Cow Creek = 15% eroding banks
Happy Cow Creek = 16% eroding banks
Here the study stream has a better percentage than the expected condition. This metric would be Fully Supporting according to table 3.

Riffle Stability Index (RSI) (Greater than 70 RSI is a good value range.):
Dead Cow Creek = 72 RSI
Happy Cow Creek = 80 RSI
72 / 80 = .9 * 100 = 90% of expected condition. This metric is Fully Supporting according to table 3.

Pool Frequency:
Dead Cow Creek = 10%
Happy Cow Creek = 60%
.10 / .60 = .1666 * 100 = 16.6% of expected condition. This metric is Not Supporting according to table 3.

For secondary channel characteristics, 2 metrics are fully supporting, and 1 metric is Not Supporting. Dead Cow Creek would therefore be determined as Fully Supporting for substrate and the analysis is considered final.
(If 1 metric was fully supporting, 1 was partially supporting, and 1 was not supporting, the determination would be fully supporting. If 2 metrics were not supporting, and 1 was fully supporting, the determination would be not supporting. At this point, the assessment would move on to biological assessment.)