STANDARD OPERATING PROCEDURE FOR AQUATIC BENTHIC MACROINVERTEBRATE COLLECTION IN RIVERS AND STREAMS

State of Utah
Department of Environmental Quality
Division of Water Quality

Revision 0
Effective May 1, 2014

Utah Division of Water Quality (DWQ) Standard Operating Procedures (SOPs) are adapted from published methods, or developed by in-house technical experts. This document is intended primarily for internal DWQ use. This SOP should not replace any official published methods.

Any reference within this document to specific equipment, manufacturers, or supplies is only for descriptive purposes and does not constitute an endorsement of a particular product or service by the author or by DWQ. Additionally, any distribution of this SOP does not constitute an endorsement of a particular procedure or method.

Although DWQ will follow this SOP in most instances, there may be instances in which DWQ will use an alternative methodology, procedure, or process.[1]

[1] *Disclaimer language above adapted from Washington State Department of Ecology SOPs.*

REVISION PAGE

Date	Revision #	Summary of Changes	Sections	Other Comments
5/1/14	0	Not applicable	Not applicable	New SOP adapted from USEPA EMAP protocol. Began document control/revision tracking.

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1.0 SCOPE AND APPLICABILITY

This document presents the Utah Division of Water Quality's (DWQ) Standard Operating Procedure (SOP) for the collection of aquatic benthic macroinvertebrates (BMI) within running waters (rivers and streams). Benthic macroinvertebrates are also commonly referred to as benthos, inverts, macroinverts, macroinvertebrates, or simply "bugs". Collection of BMI is routinely performed during DWQ's probabilistic state-wide surveys; for those procedures, please refer to the specific instructions found in the Utah Comprehensive Assessment of Stream Ecosystems (UCASE) Field Manual (http://www.waterquality.utah.gov/Monitoring/WQMonitorQAQC.html). There are, however, numerous additional opportunities to collect BMI data for streams outside of the probabilistic survey design. BMI data is desirable because:

1) BMI are relatively quick and inexpensive indicators for identifying a wide variety of pollutants

2) BMI typically exhibit a predictable community composition under natural conditions

3) BMI are a temporally-integrated water quality indicator versus water chemistry samples (which are essentially a snapshot of current conditions)

4) Some BMI are especially useful for targeted sampling due to their high sensitivity to environmental changes (e.g., impacts of remediation or pollutant discharges)

DWQ's collection methods are derived from USEPA's Environmental Monitoring Assessment Program-Western Pilot program (EMAP-West), which provides continuity and consistency for DWQ and other agencies conducting water quality assessments using BMI.

This SOP is applicable to rivers and streams. For collection of BMI in wetlands, refer to DWQ's SOP for Collection of Macroinvertebrates in Wetlands.

IMPORTANT: If BMI samples are intended for regulatory purposes by outside (non-DWQ) entities, samples must be analyzed by an accredited laboratory with documented QA/QC and analytical procedures approved by DWQ. Please first contact DWQ for questions about specific details.

2.0 SUMMARY OF METHOD

See **Section 9.0** for the procedure details.

Because biological measures require sampling an extended length of waterway for a representative picture of the ecological community, a reach length of 40 times the channel wetted width (at base flow) is established to characterize the habitat and several biotic assemblages associated within the sampling reach. Riffle habitat is targeted when sampling running waters because of greater BMI diversity, ease of collection, and consistency. However, if a site is devoid of riffles, then edge habitat is targeted.

The collection technique consists of a semi-quantitative benthic macroinvertebrate composite sample using a D-frame net. A composite sample is performed by collecting 8 subsamples made at different locations within the reach. The sampler carries a sieve bucket as they move through the reach and composites the benthic material collected in the D-net at each subsample location into the sieve bucket. The BMI collection technique itself is designed to be rapid so that one subsample requires no more than 3 minutes to perform. At each of the 8 subsample locations, the sampler attempts to collect all available BMIs located in a one square-foot area upstream of the D-net opening. BMI are collected from the largest substrates down to the smaller substrate to a depth of approximately 3 inches. The sampler rinses the material to the bottom of the net and then empties the contents of the net into the sieve bucket. This process is repeated at the remaining seven subsample locations. The result is a composite BMI sample in the sieve bucket.

Sample processing is required for the composite sample because most of the heavy inorganic benthic material collected is not of interest and the BMI in the sample must be concentrated into small jars for transfer to the analytical laboratory. Processing involves using a regular 2.5 gallon bucket and water to separate out heavy inorganic material from lighter organic material (where the BMI are most likely located). This separation process results in a much smaller volume of material which is then placed into 1 L plastic jars and preserved with 95% ethanol. Multiple jars may be required for one sample. Jars are then sealed, labeled, and stored until delivery to the laboratory.

Field data and other sampling details during BMI collection is recorded on a Sample Collection Form. See **Section 10.0** for details. Numerous data are documented including GPS waypoints of the site location, a sketch of the targeted sampling stream reach including the subsample locations, and identification and description of the sampled habitat.

Lastly, personal gear and sampling equipment is decontaminated prior to leaving the site to reduce the spread of invasive species. See DWQ's SOP for Decontamination of Monitoring Equipment.

3.0 DEFINITIONS

BMI: benthic macroinvertebrates

ft^2: square foot

L: liter

mm: millimeter

MSDS: Material Safety Data Sheet

QA/QC: Qualty Control and Quality Assurance

µm: micrometer

4.0 HEALTH AND SAFETY WARNINGS

Field personnel should be aware that hazardous conditions potentially exist at every waterbody. If unfavorable conditions are present at the time of sample collection, it is recommended that the sample collection be rescheduled. If hazardous conditions arise during sampling, such as lightning, high winds, rising water, or flash flood warning, personnel should cease sampling and move to a safe location.

Field personnel should take appropriate precautions when operating equipment and working on, in, or around water, as well as possibly steep and unconsolidated banks, or edges of ponds/lagoons. All field crews should follow EPA, OSHA, and specific health and safety procedures and be equipped with safety equipment such as proper wading gear, personal flotation devices (PFDs), gloves, first aid kits, cellular phone, etc.

Be sure to wash hands or use hand sanitizer after sampling, especially when sampling sites with potential fecal contamination.

Prior to sampling be sure to review the MSDS for the preservation chemical. Pure ethanol (200-proof, 95% ethyl alcohol) is preferred for sample preservation. However, denatured alcohol may be used with caution. Wear gloves and wash off any denatured alcohol that comes in contact with skin. Denatured alcohol contains hazardous components and should not be inhaled, ingested, or come into contact with skin.

Alcohol is flammable. Keep alcohol carboy away from heat, sparks, flame, and all other sources of ignition. Do not smoke in the vicinity of alcohol or fumes.

5.0 CAUTIONS

For representative data, it is best to collect BMIs during the growing season, which can vary depending on elevation and latitude, but is generally limited to the months of May through October.

Refer to all instructions within this SOP for setting up the sampling reach and targeting the proper sample habitat in order to collect a representative sample.

6.0 INTERFERENCES

Field personnel should scout the potential sampling reach to make sure it is clear of obstacles that would prohibit sampling and data collection activities. Make every effort to avoid walking within the proposed stream reach during reconnaissance to ensure biological organisms remain unaffected.

Samples must be collected in the appropriate sample containers with the appropriate preservative; failure to preserve a sample properly can lead to inaccurate results, sample degradation, or invalidation of the sample by the laboratory.

Samples must be stored and handled appropriately; samples stored improperly may be invalidated by the laboratory.

Ensure preservative is adequately mixed within the sample. If the sample is not properly preserved, microbes will persist and ultimately destroy the sample. Additionally, ensure the samples are submitted to the laboratory no later than 6 months after collection to reduce likelihood of sample decomposition.

7.0 PERSONNEL QUALIFICATIONS/RESPONSIBILITIES

Collection of BMI is a very hands-on technique and requires in-person training from an experienced sampler. Personnel performing water sampling must be familiar with sampling techniques, safety procedures, proper handling, and record keeping. Samplers are responsible for attending refresher meetings held prior to each field season to review procedures and techniques. New staff will be trained in the field by experienced personnel.

Samplers are required to read this SOP annually and acknowledge they have done so via a signature page (see **Appendix 1**) that will be kept on-file at DWQ along with the official hard copy of this SOP.

8.0 EQUIPMENT AND SUPPLIES

The following equipment and supplies are required for benthic macroinvertebrate collection at wadeable sites:

For collecting samples	• Flagging for marking reach boundary • Kick net (D-frame with 500 µm mesh) with at least a 4-foot long handle or a modified surber with at least a 4-foot long handle. • Watch with timer or stopwatch • Plastic buckets (8-10 quart size) • Sieve bucket with 500 µm mesh openings • Plastic forceps • Wash bottle; 1 Liter capacity labeled “Stream water” • Sample jars suitable for use with ethanol such as 1-Liter HDPE Nalgene sample bottles • 95% ethanol (ETOH) in proper container • Bottle caddy • Electrical tape • Scissors or knife • This SOP or UCASE Field Manual • Waterproof neoprene gloves • Waders, boots, personal flotation device (Use waders with attached boots whenever possible, as opposed to stocking foot waders with separate boots, as organisms can easily get trapped in laces and

	inside of boots. In addition, it is preferred that personnel do not use boots with felt soles.)
For recording measurements	• Sample Labels (**Figure 2**) – labels for jar interior must be printed on waterproof paper and filled out in pencil; labels for jar exterior can be printed on regular paper and filled out in ink/sharpie • Lead pencils • Fine tipped indelible markers • Clipboard, Sample Collection Form (**Appendix 2**) and field notebook • Clear tape strips to cover sample labels

9.0 PROCEDURE

9.1 Pre-Sampling Preparation

Prior to visiting a site, inspect the D-net/modified surber and sieve bucket for holes or tears and replace or repair. It is also good practice to carry a back-up set.

At the site, find an area downstream of the reach and wash all equipment with stream water. Visually inspect the nets and buckets and make sure no particles are present on/inside of them. If they are, continue to wash the gear until it is clean. Rinse out the spray bottle and fill it with stream water.

9.2 Setting up the Sampling Reach

At the sampling location, record the channel width at various points to determine the average wetted width, and then calculate the reach length by multiplying 40 times the average channel wetted width (during base flow). Mark each end of the reach using stakes/flags.

While within the boundary of the reach (preferably mid-reach), collect and record GPS coordinates (in decimal degrees). Record these coordinates on the Sample Collection Form (**Appendix 2**).

Complete a rough sketch map of the sampled stream reach. Be sure to note any interesting features or landmarks/directions that can be used to find the reach for future visits, in addition to the GPS coordinates.

9.3 Sample Collection

NOTE: Determining where to collect subsamples within the reach is a somewhat subjective process based on the experience and best judgment of the sampler. Therefore, it is imperative that new samplers first be trained in the field by experienced samplers.

9.3.1 Gather the clipboard and Sample Collection Form, stopwatch, D-net, and sieve bucket. A scratch piece of paper can be used to record information as

well if it is easier to manage; transfer the data later onto the field form, preferably before leaving the field site.

9.3.2 Begin walking upstream along the reach. While walking along the reach, look for desirable habitat, which are riffle/runs with coarse substrates (coarse gravel or bigger). Your goal will be to collect 8 subsamples within the reach, targeting riffle habitat. In order to reduce human bias, alternating locations in the stream should be sampled (e.g. left-25% of channel width, center- 50% of channel width, right- 75% channel width). Start randomly with one of these locations, and then consistently follow the pattern of left (L), center (C), right (R) and repeat until 8 subsamples are collected (see **Figure 1** as an example). ***Multiple sub-samples can be collected in one riffle habitat if riffle habitat in the reach is limited.***

Figure 1. Sampling locations within a reach.

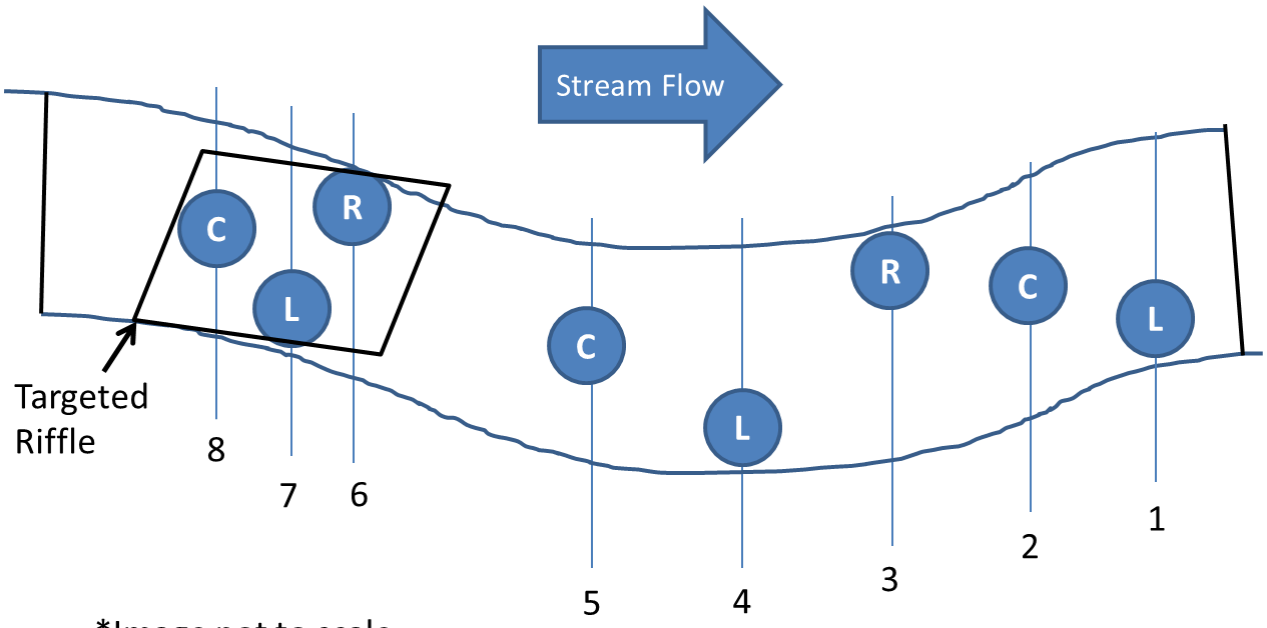


*Image not to scale.

9.3.3 At each of the 8 collection points in the reach, determine habitat type (pool, glide, riffle, or rapid), and the dominant substrate (fines/sand, gravel, coarse, or other). On the Sample Collection Form use the box titled “Other” to include sample site information such as occurrence of wood, leaves, edge habitat, overhanging vegetation, bedrock, hardpan, etc. At each subsample location, target coarse substrates such as large gravel (pea-size and larger) to small boulders (basketball-size and smaller) rather than substrates at either

spectrum. If coarse substrates are lacking, woody debris, macrophytes, or leaf packs could be targeted; please, identify and document these situations.

9.3.4 Follow the sample collection instructions specific to habitat type as listed in **Sections 9.3.5 (Riffle/Run Habitat)** and **9.3.6 (Edge and Pool/Glide Habitat)**.

NOTE: Sampling edge habitat along the stream banks provides the best alternative to riffle samples (due to BMI diversity, ease of collection and consistency). Often, overhanging vegetation, sticks, and other material will offer protection and stability for BMI to colonize. In many cases, Utah streams will lack or be absent of riffles and coarse sediments (e.g. desert streams in Southern Utah are predominantly characterized by glides and fine/sandy sediments). It is important to target edge habitat in these cases to get a representative BMI sample.

9.3.5 Procedures for Riffle/Run Habitats:

1) With the net opening facing upstream, quickly and firmly position the net securely in the stream bottom to eliminate gaps under the frame. This will ensure flow space is limited (i.e. flow is directed into the net, not under). Avoid large rocks that prevent the net from sitting properly on the stream bottom.

   NOTE: This is easier said than done in most cases, especially in high gradient cobble-dominated streams. Do the best you can and make an attempt to get this net as flush with the substrates as possible. If there are issues or concerns, record them on the Sample Collection Form.

2) Holding the net in position on the substrate, visually define an area that is one net-width wide and one net-width long upstream of the net opening. The area within this quadrat is $1ft^2$. Your goal is to collect all available BMIs located in this one square-foot area upstream of the net opening. It is helpful to hold the net in place with your knee while using your hands to disturb the substrate.

3) Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the sieve bucket. Pick up loose rocks or larger substrate particles in the quadrat. Use your hands to dislodge organisms from their surfaces and wash them into the net. When lifting and washing, ensure that the substrate remains in front of the net opening and flows are directed into the net. Scrub all rocks that are golf ball-sized or larger and which are halfway into the quadrat. Discard scrubbed rocks/substrate back into the stream outside of your targeted quadrat.

4) When all large (>golf-ball size) substrates are removed from the area in front of the net, focus on the smaller substrate to a depth of approximately 3 inches. Hold the net securely. Start at the upstream end of the quadrat, and vigorously perturb the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch if desired) with your

hands. If the substrate is too difficult to dislodge or the water depth is greater than your elbow, use your boots to disturb the area. Conduct this perturbation activity for no more than 30 seconds. Immerse the net in the stream several times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any water or material enter the mouth of the net during this operation (dip the net material only, not the mouth). Empty the contents of the net into the sieve bucket and repeat seven more times as you move upstream. Alternatively, you do not need to empty the net if it has only very little material at this point.

NOTE: For samples located within dense beds of long, filamentous aquatic vegetation (e.g. algae or moss), kicking within the quadrat may not be sufficient to dislodge organisms in the vegetation. Usually these types of vegetation are lying flat against the substrate due to current. Use a knife or scissors to remove only the vegetation that lies within the quadrat (i.e. not entire strands that are rooted within the quadrat) and place it into the net.

NOTE: If flow is too little or slow to sweep organisms into the kick net, stir up the substrate with your hands and sweep the water through the fixed net.

5) Go to the next area with sampleable habitat.

9.3.6 Procedures for Edge-Pool/Glide Habitats:

NOTE: Sample edge habitat if you are at a site where beaver ponds are common or the site lacks desireable habitat (e.g. riffles, coarse substrates).

NOTE: This technique takes practice and beginners should be trained in the field by experienced personnel.

1) Visually define a quadrat that is one net-width wide and one net-width long at the sampling point (1 ft^2).

2) Sweep the area in a figure eight motion with the net for 30 seconds. Or, stir up any overhanging vegetation, sticks, or other material with your hands or feet and then sweeping them in a figure eight motion for 30 seconds. Be sure not to constantly drag the frame of the net against the bed of the stream as you will scoop up lots of fines and muck. Bounce the end of the sample frame on the bottom of the substrate as you sweep the net about 1 inch above the substrate.

3) After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.

9.3.7 General Procedures for Any Habitat Type

1) If the net is not full after collection of a subsample, move onto the next sampling location and make your next kick, leaving any material from the previous kick in the net. If the net

is full of detritus and/or substrates, invert the net to transfer the sample into the sieve bucket. To prevent bugs from being damaged during transport in the bucket, any large substrates need to be removed. To do this, carefully inspect coarse substrates and wash off any organisms still clinging into the bucket (using stream water) before discarding the substrate.

2) Determine the predominant substrate size/type you sampled within the sampling quadrat. Fill in the approprate circle for the dominant substrate type for the transect on the Sample Collection Form. If carrying a clipboard and the data sheets is too cumbersome, you can keep record data in a small notebook or scratch piece of paper and transfer it to the actual data sheet when you are finished, preferably before leaving the field site.

NOTE: If there are co-dominant substrate types, you may fill in more than one circle; note the co-dominants in the comments section of the form.

- **F**ine/sand: not gritty (silt/clay/muck <0.06 mm diam.) to gritty, up to lady bug-sized (2 mm)
- **G**ravel: fine to coarse gravel (ladybug to tennis ball-sized; 2 mm to 64 mm)
- **C**oarse: cobble to boulder (tennis ball to car-sized; 64 mm to 4000 mm)
- **O**ther: bedrock (larger than car-sized; >4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc. Note type of "other" substrate in comments on field form.

3) Identify the habitat type where the sampling quadrat was located. Fill in the appropriate circle for channel habitat type for the transect on the Sample Collection Form.

- **P**ool; still water; low velocity; smooth, glassy surface; usually deep compared to other parts of the channel.
- **GL**ide: water moving slowly, with smooth, unbroken surface; low turbulence.
- **RI**ffle: water moving, with small ripples, waves, and eddies; waves not breaking and surface tension is not broken; "babbling" or "gurgling" sound.
- **RA**pid: water movement is rapid and turbulent; surface with intermittent "white water" with breaking waves; continuous rushing sound.

9.4 Sample Processing and Preservation

1) After sampling is complete, it is beneficial to separate the organic material from the heavier, inorganic material in the sieve bucket. Gently, dump the composited material from the net/sieve bucket into a plastic bucket (non-sieve); do not worry about removing *all* the material from the sieve bucket. Inspect the net for any remaining bugs that may still be clinging to it. Using a wash bottle full of stream water and/or forceps, flush/pick them off the net and into the bucket.

2) Next, fill the plastic bucket with stream water a few inches above the material line. Slowly swirl the contents in the bucket for about 7 seconds so that lighter (organic) material (sticks, leaves, organisms) in the bucket come to the surface and heavier material (inorganic substrates) stay at the bottom. While the material in the bucket is suspended and swirling, slowly pour the water into the sieve bucket making sure not to dump any of the heavier material at the bottom of the bucket with it. Repeat this step several times until no more bugs are seen crawling around in the plastic bucket. This process is known as sieving.

NOTE: If there is an abundance of pebbles or cobbles in your sample, you will need to rinse (scrub if necessary) these off in the bucket with ample amount of site water and then discard them.

NOTE: Do not attempt to sieve samples with an abundance of filamentous algae. If this is the case, simply include all of the algae into the sample jar since it is difficult to effectively process these kinds of samples in the field.

3) Ultimately, you will end up with a 2.5 gallon plastic bucket containing coarse gravel and sand, and a sieve bucket containing organisms and detritus (it is okay if some fine sediment is present in the sieve bucket). Be sure to inspect the bucket for caddisfly cases as sometimes the cases are composed of predominantly gravel-sized material. Once the sieving process is complete, you can dump the heavy material left in the 2.5 gallon bucket into the stream or on the ground.

4) Place the material in the sieve bucket into a 1-L jar making sure not to fill it more than 40% full with sample material; use multiple jars if necessary. Be sure not to grab such a large handful where material will become dislodged on the mouth of the bottle when you are filling it. Keep in mind some material will stick to your hands during each transfer. It is a good idea to rinse your hands in the sieve bucket each time you put material in a jar.

5) As the volume of material becomes less abundant at the bottom of the sieve bucket you will need to wash the remaining contents to one side of the bucket in order to get the rest of the sample by gently agitating the bottom outside portion of it.

6) Once you think you have removed everything from of the sieve bucket, carefully examine it for any remaining organisms. If there are still visible organisms use a pair of forceps to pick the bugs out. Or, alternatively, you can tip the sieve bucket upside down and spray the bottom side of it with rinse water into a funnel place in the mouth of the sample jar, washing the BMI in to the jar.

NOTE: If you choose to spray the sieve bucket as a final precaution, but end up filling the sample jar with too much water (>1/3 full) pour it off into the sieve bucket and re-spray with a smaller volume of water.

7) Place a properly and completely filled out sample label (**Figure 2**) inside each jar (each label for the site should be filled out exactly the same except for the Jar # of total Jars, e.g., Jar 1 of 3). *Labels to be placed **inside** the jar must be printed on waterproof paper and filled out by hand using a pencil. Ink will fade eventually due to the ethanol.*

Figure 2. Example of a properly filled-out sample label. Template location is U:\WQ\PERMITS\MONITORS\Labels\UCASE Labels

BENTHOS COMPOSITE SAMPLE (95% ETOH)

Site Name: CALF CK BL LOWER FALLS

STORET: 5994070 # Bottles: 1 of 1

Personnel: EO, HH Date: 2/29/2012

Equipment/Method: D-net; 8 subsamples

8) Completely fill the jar with 95% ethanol (no headspace). It is very important that sufficient ethanol be used or the organisms will not be properly preserved. Existing water in the jar should not dilute the concentration of ethanol below 70%.

NOTE: Samples can be transported back to the vehicle before adding ethanol if necessary. However, if site is a fair distance from vehicle (e.g. long hike into site) a liter of ethanol should be taken to the site with you. Fill the bottles to at least the detritus line, then completely fill the rest of the bottle once back at the vehicle.

9) Replace the cap on each jar. Slowly tip the jar to a horizontal position and then gently rotate the jar to mix the preservative. Do not shake the jar. After mixing, seal each jar lid with electrical tape.

10) Place a sample label (**Figure 2**) on the outside of each jar making sure it coincides with the interior label. Cover it with clear tape to maintain label integrity. *Labels to be placed on the **outside** of the jar may be printed on regular paper and partially filled out with ink/sharpie prior to sampling.*

11) Store filled jars in an empty cooler or jar tote during transportation until they can be stored in the appropriate location before shipment to a lab. Samples do not need to be refrigerated or stored on ice.

9.5 Reducing the Spread of Invasive Species

Before leaving the site, sampling equipment and personal gear (boots, waders, etc.) need to be thoroughly decontaminated in order to prevent the spread of aquatic invasive species. Refer the DWQ's SOP for Decontamination of Monitoring Equipment for full instructions.

Briefly:

1) Before leaving the site, use a -bristled brush to remove mud, plant material, and debris from boots, nets, and any other monitoring gear that has come in contact with the stream.

2) Remove wading gear immediately after exiting the stream and make sure gear does not come in contact with other equipment. If you use separate wading boots, remove the insoles from the boots (these procedures also apply if wet-wading in shoes or sandals).

3) It is recommended that you wear latex gloves and eye protection when using Sparquat 256. This product is an industrial cleaner and standard safety precautions should be followed.

4) Place waders, boots and insoles, sandals, etc. and any other sampling equipment that has come in contact with the stream into the Sparquat 256 solution for a minimum of 10-15 minutes (the solution may be reused several times).

5) Remove the gear from solution and inspect it to make sure all organisms have been removed.

6) Rinse by immersing and agitating the gear in the bucket of clean rinse water (tap water). Do not use stream water to rinse gear as this may reintroduce organisms.

7) Do not discard the Sparquat 256 solution or the rinse water in the field; dispose of the liquid down a drain that is routed to a wastewater treatment plant.

10.0 DATA AND RECORDS MANAGEMENT

Be sure that sample labels both inside and outside the jar are filled out exactly the same. If your sample is placed in multiple jars, be sure to note the number of jars on the labels (e.g., 1 of 2, 2 of 2) as well as on the Sample Collection Form.

Be sure that site information [Monitoring Location ID (i.e. site code) if applicable, site description, and GPS coordinates in decimal degrees] are recorded accurately on the Sample Collection Form.

Fill out the Sample Collection Form completely. Include subsample dominant substrate type and size, channel characteristics, and other details discussed throughout **Section 9.0** of this SOP.

Include with the Sample Collection Form a rough sketch of the sampled reach, ideally including the locations of the 8 subsamples. See **Figure 3** for an example of a filled out form and sketch (note subsample locations were not marked in this example).

Figure 3. Example of a completed Sample Collection Form and sketch of sampled reach (on next 2 pages).

STREAM VERIFICATION FORM-WADEABLE STREAMS (cont.)

Reviewed by (initial): RB

SITE NAME: Straight Canyon Ck DATE: 10/05/2011 TEAM:

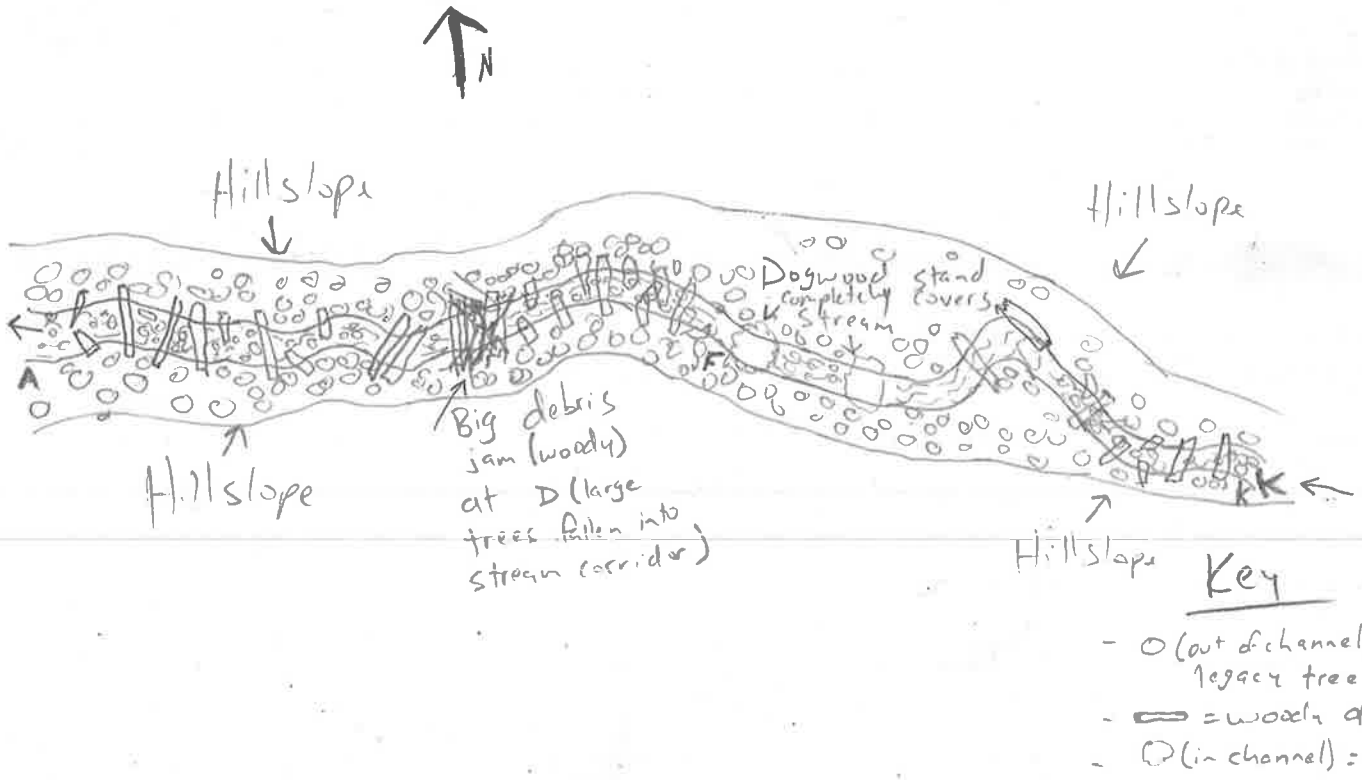
SITE/STORET ID: 494 1020 UCASE ID (if applicable): UT 09 ST- 741 VISIT: 0 (1) 2 3

LAND OWNERSHIP INFO/GENERAL COMMENTS ABOUT SITE LOCATION: Site is on Fishlake NF. Hike required to get to site. Hike is ~1.2 miles long but an established USFS Trail exists the whole. Overall, hiking conditions are moderate in difficulty

STREAM REACH DETERMINATION

Channel Width Used to Define Reach (m)	Total Reach Length Intended (m)	COMMENTS
< 1	150	

SKETCH MAP - Arrow indicates North



PERSONNEL

NAME	DUTIES: Bio/Chem Sampling	DUTIES: Habitat
B. Holcomb	○	●
S. Tuhir	○	●
R. Brown	●	○
	○	○
	○	○

SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial): ABB

SITE/STORET ID: 4941020 DATE: 10/05/2011

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place on ice. Lab: State of Utah Health Lab within two weeks of collection. Shop storage: Fridge	Comments/Flags
Total Chemistry	(Y) / N	Filtered Nutrient	(Y) / N		
Non-Filtered Nutrient	(Y) / N	Filtered Metals	(Y) / N		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
01	Y / (N) __ __	●D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		A		B		E		J	
Dom. Substrate	Channel	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	○F	○P	○F	○P	○F	○P	○F	○P
Gravel	Glide	●G	○GL	●G	○GL	●G	○GL	○G	○GL
Coarse	Riffle	○C	●RI	○C	●RI	○C	●RI	●C	●RI
Other: Note in Comments	Rapid	○O	○RA	○O	○RA	○O	○RA	○O	○RA
SUBSTRATE SIZE CLASSES		J		J		K		K	
		Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
F/S – ladybug or smaller (<2 mm)		○F	○P	○F	○P	○F	○P	○F	○P
G – ladybug to tennis ball (2 to 64 mm)		●G	○GL	○G	○GL	●G	○GL	✓G	○GL
C – tennis ball to car sized (64 to 4000 mm)		○C	●RI	●C	●RI	○C	●RI	●C	●RI
O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.		○O	○RA	○O	○RA	○O	○RA	○O	○RA

Note:

- Always perform 8 kicks at every site.
- Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments.
- If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach.
- If kicks are made in beaver influenced areas, please explain in the comments section.

Process: Fill a clean wide-mouth bottle with ~30-40% of composite sample (use multiple bottles if necessary).
Preservation: Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position.
Lab: Utah State University Bug Lab; submit in large batch at end of field season.
Shop storage: Store on shelf with other bug samples.

BENTHOS COMMENTS: Not many bugs seen in field. Hydropsychid & Stone-flies seen in bug net. Overall population was low due to lack of flow (very small stream), but this is natural (not from diversions).

11.0 QUALITY ASSURANCE AND QUALITY CONTROL

If intended for regulatory purposes, BMI samples must be analyzed by an accredited laboratory with documented QA/QC and analytical procedures approved by DWQ.

Frequency and type of quality control samples such as field replicates, laboratory duplicates, and measures of laboratory inter-analyst variability will be prescribed in project-specific Sampling and Analysis Plans (SAPs).

Experienced DWQ personnel (or other DWQ-authorized personnel) will conduct field audits for non-DWQ cooperators collecting BMI data for DWQ programmatic efforts or for compliance purposes at a frequency prescribed in the project-specific SAP.

12.0 REFERENCES

Peck, D.V., J.M. Lazorchak, and D.J. Klemm (editors). Unpublished draft. Environmental Monitoring and Assessment Program -Surface Waters: Western Pilot Study Field Operations Manual for Wadeable Streams. EPA/XXX/X-XX/XXXX. U.S. Environmental Protection Agency, Washington, D.C.

Related DWQ Documents:

Standard Operating Procedure for Decontamination of Monitoring Equipment

Utah Division of Water Quality: Quality Assurance Program Plan for Environmental Data Operations

Utah Comprehensive Assessment of Stream Ecosystems (UCASE) Field Operations Manual

13.0 APPENDIX

Appendix 1 - SOP Acknowledgement and Training Form

This SOP must be read and this form signed annually. This form must be kept with the current version of the SOP.

Document Title:	
Document Revision Number:	
Document Revision Date:	

Please sign below in accordance with the following statement: "I have read and understood the above referenced document. I agree to perform the procedures described in this SOP in accordance with the document until such time that it is superseded by a more recent approved revision."

Printed Name	Signature	Date

SOP Acknowledgement and Training Form (continued)

Trainee: Sign below to acknowledge that training on this SOP was received, understood, and all questions/concerns were addressed by the trainer.

Trainer: Sign below to acknowledge that training on this SOP was completed for the individual listed and that trainee is competent to perform the procedures described within.

Date of Training	Trainee Printed Name	Trainee Signature	Trainer Printed Name	Trainer Signature

Appendix 2 – Sample Collection Form

UCASE WATER CHEMISTRY AND MACROINVERTEBRATE SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place water chem on ice and chlorophyll-*a* on dry ice Lab: State of Utah Health Lab within one week of collection. Shop storage: Fridge and freezer	Comments/Flags
Total Chemistry	Y / N	Filtered Metals	Y / N		
Non-Filtered Nutrient	Y / N	Chlorophyll-*a* (water column)	Y / N		
Filtered Nutrient	Y / N		Volume filtered: _____mL		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
___ ___	Y / N ___ ___	○D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		____		____		____		____	
Dom. Substrate	Channel	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	○F	○P	○F	○P	○F	○P	○F	○P
Gravel	Glide	○G	○GL	○G	○GL	○G	○GL	○G	○GL
Coarse	Riffle	○C	○RI	○C	○RI	○C	○RI	○C	○RI
Other: Note in Comments	Rapid	○O	○RA	○O	○RA	○O	○RA	○O	○RA

SUBSTRATE SIZE CLASSES
F/S – ladybug or smaller (<2 mm)
G – ladybug to tennis ball (2 to 64 mm)
C – tennis ball to car sized (64 to 4000 mm)
O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.

		____		____		____		____	
		Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
		○F	○P	○F	○P	○F	○P	○F	○P
		○G	○GL	○G	○GL	○G	○GL	○G	○GL
		○C	○RI	○C	○RI	○C	○RI	○C	○RI
		○O	○RA	○O	○RA	○O	○RA	○O	○RA

Note:
- Always perform 8 kicks at every site.

- Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments.

- If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach.

- If kicks are made in beaver influenced areas, please explain in the comments section.

Process: Fill a clean wide-mouth bottle with ~40% of composite sample (use multiple bottles if necessary).
Preservation: Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position.
Lab: Utah State University Bug Lab; submit in large batch at end of field season.
Shop storage: Store on shelf with other bug samples.

BENTHOS COMMENTS:

Updated: 01/2013