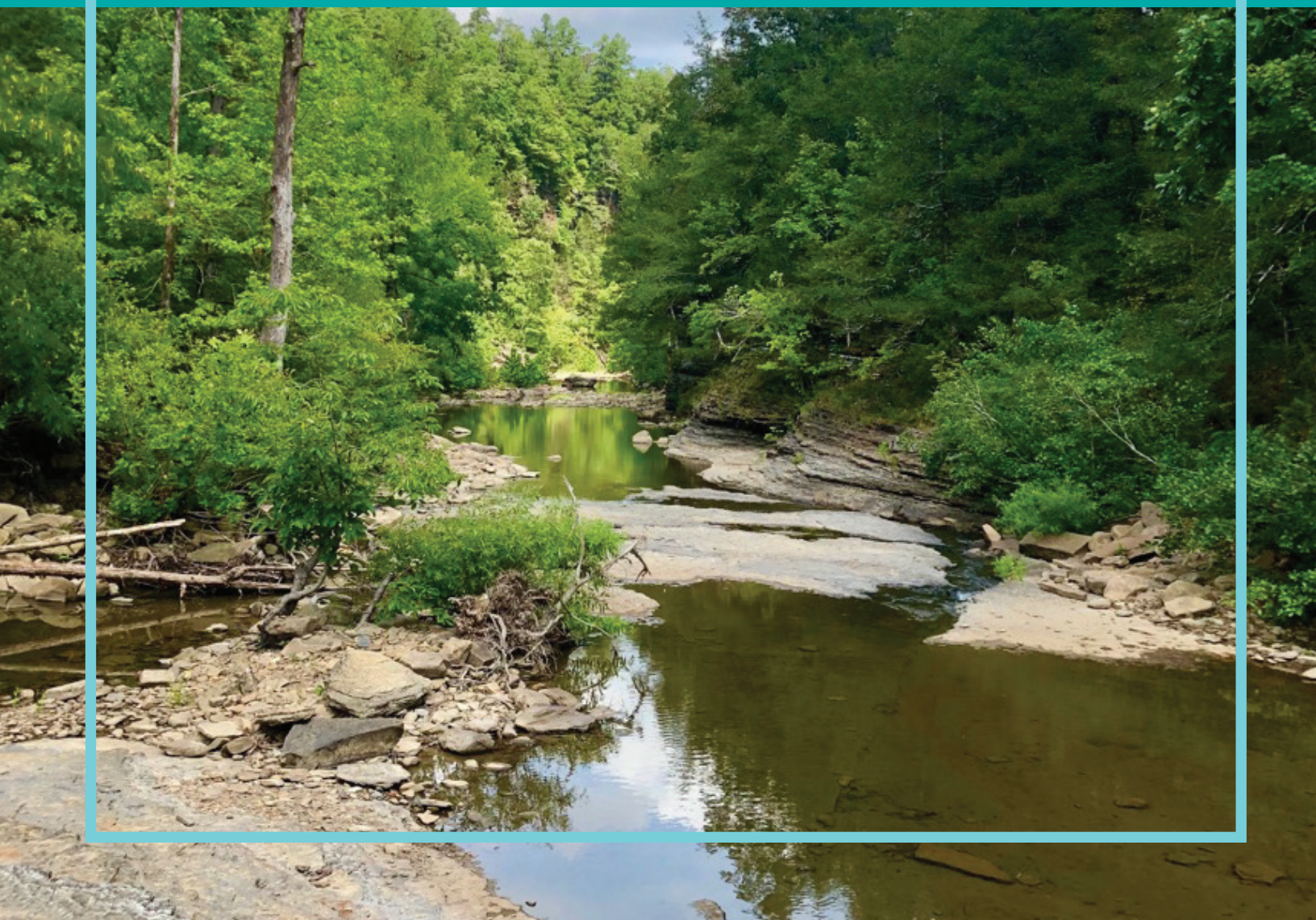
ARKANSAS DEPARTMENT OF ENERGY AND ENVIRONMENT

ENVIRONMENTAL QUALITY

WADEABLE STREAM SAMPLING PROTOCOLS



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Wadeable Stream Sampling Protocols

Arkansas Department of Energy & Environment
Division of Environmental Quality

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Compiled by the Water Quality Planning Branch

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1. Introduction/Scope and Application

The Arkansas Department of Energy & Environment, Division of Environmental Quality (DEQ), Office of Water Quality (OWQ) planning establishes these standard operating procedures (SOPs) for wadeable stream sampling and water quality monitoring. Certain procedures described in this document may vary depending on the lab used to analyze the samples. Protocols used and data types collected are dependent on study goals. All sampling personnel should be familiar with the policies and procedures of the lab used to conduct the chemical analysis of the surface water samples.

Quality Assurance/Quality Control (QA/QC) procedures applicable to all SOPs are described in the main body of Arkansas's Water Quality and Compliance Monitoring Quality Assurance Project Plan (QAPP). These procedures should be followed at all times.

2. Timing of Field Data Collection

Table 1. Field collection method timing

Discrete in Situ	DEQ collects in situ meter measurements along with water samples on a monthly or twice per quarter basis
Deployed in Situ	DEQ conducts two separate 48-hour in situ meter deployments during the critical season of the same year when stream temperatures are above 22 °C, usually July-September
Water Aliquoting	DEQ collects water samples along with in situ meter measurements on a monthly or twice per quarter basis
Flow	DEQ collects summer flow measurements in conjunction with sampling fish and fall flow measurements in conjunction with sampling macroinvertebrates
Habitat	DEQ collects habitat data in the summer in conjunction with sampling fish or in the fall in conjunction with sampling macroinvertebrates, depending on study goals
Fish	DEQ collects fish during the critical season when water temperatures are above 22 °C, usually July-September
Macroinvertebrates	DEQ collects macroinvertebrates during the fall when water temperatures have dropped below 22 °C, usually October-December

3. Safety Precautions

BASIC PRECAUTIONS

- Observe surrounding hazards including, but not limited to biting/stinging insects and venomous snakes.
- Bring a satellite-based communication device for contacting help in event of emergencies.
- Have a comprehensive first aid kit accessible to respond to injuries until trained medical professionals can arrive.

WATER COLLECTION

- Do not attempt to access streams or rivers during unsafe conditions, e.g., high winds, icy, flooded.
- Do not attempt to collect samples during unsafe weather conditions such as severe storms.
- When sampling from roadsides and bridges, wear a high-visibility vest at all times when collecting samples.
- Wear gloves at all times when handling any sample containers that contain acid preservative.
- Do not overfill sample containers containing acid preservative with sample water to prevent spillage of acid.

INSTREAM SAMPLING

- Always conduct sampling with at least two people when working within a stream or river.
- When sampling in the fall, wear a high-visibility hat at all times.
- If deploying a sonde, check weather reports for entirety of upstream watershed to ensure that water will not rise to unsafe levels before the end of the deployment.
- Wear appropriate footwear for walking on slippery or uneven substrates.
- Do not enter waterbodies if current velocity and/or depth present potentially unsafe conditions.
- Use caution when working around large woody debris, boulders, mud, bedrock, and in other conditions that could present slip/trip/fall hazards.
- Wear non-breathable waders with a wading belt to stay dry and protect against physical, chemical, and/or biological hazards when electrofishing.
- At least two crew members should be First Aid and Cardiopulmonary Resuscitation (CPR) certified.
- When possible, at least two crew members should be Swiftwater Rescue certified, when conducting biological sampling.
- Wear lineman gloves to protect against electrical shock when electrofishing.
- Do not submerge bare hands or arms under the water's surface when electrofishing is occurring.
- Wear gloves and eye protection and take great care to not inhale fumes when handling formalin.
- Do no touch Unexploded Ordinances (UXO) and exit the area if found. Take photographs, latitude longitude, and report to appropriate authorities.

4. Water Sample Collection and Aliquoting

4.1 OVERVIEW

The goal of this section is to outline the procedures for collecting and aliquoting water samples for chemical analysis of rivers and streams. Certain procedures described in this SOP may vary depending on the lab used to analyze the samples. All sampling personnel should be familiar with the policies and procedures of the lab used to conduct the chemical analysis of the surface water samples.

4.2 EQUIPMENT

- Chain of custody data sheets
- Plastic, quart jug (1 per site + 1 duplicate (dup.)
- Plastic, quart jug with H_2SO_4 preservative (1 per site + 1 dup.) *Only when holding times cannot be met
- 40 mL glass vial with H_2SO_4 preservative (1 per site + 3 dup.)
- 125 mL plastic wide-mouth bottles with HNO_3 preservative (2 per site – 1 for total metals, 1 for dissolved metals + 1 dissolved metals for field blank, and + 1 dissolved metals dup.)

- 250 mL plastic wide-mouth bottle with HNO_3 preservative (1 for total metals dup. site)
- 1,000 mL brown plastic bottle (1 per site + 1 dup.) *May – September only
- 120 mL sterile IDEXX bottle (or similar)
- Disposable nitrile gloves
- Becton Dickinson (BD) Luer-Lok™ Tip 60 mL (or similar) syringe (1 per day with extras)
- Ample 0.45 µm Acrodisc® (or similar) filters
- Ample Acrodisc® (or similar) pre-filters, if needed
- Two quarts of Type I Deionized (DI) water
- Collection vessel – plastic bucket with plastic handle, plastic pitcher, or other non-metallic vessel
- High-visibility vest
- Permanent markers suitable for labeling sample containers
- Toothbrush
- Scoopula
- Squirt bottle
- 2.5" (6.0 cm) diameter rubber delimiter, with a 1.0" (2.5 cm) diameter opening
- 1 larger plastic bin
- 1 smaller plastic bin
- Small funnel

Additional apparatus and materials should always be on hand in case of meter failure, material contamination, etc.

4.3 WATER COLLECTION

At each sampling site, collect sample water for aliquoting using an appropriate collection vessel. Collection vessels should be metal free if collecting water for metal analysis. A plastic bucket with plastic handle, pitcher, bottle, or similar device is typically used as a collection vessel.

Stream samples are taken from a road crossing or other accessible location using a bucket with a plastic handle on a polyethylene rope. Lower/cast out bucket into the thalweg from the upstream side of the road crossing (or wherever access permits) and fill with sample water. In cases where water flows over the road crossing (if depth and velocity are low enough to safely sample), wade to the thalweg and collect sample water upstream of both the crossing and the sampler's legs. Lift/pull in the bucket, swirl to rinse, and dump water onto the downstream side of the road crossing (take care not to dump the water into the upstream area to be sampled). Complete this three times in total. After rinses, collect a sample by filling the bucket with water, ensuring that the sample is representative of the waterbody; avoid collecting water from a backwater pool or where conditions are shallow enough to stir up the sediment. Ensure that the rope isn't scraping along the bridge crossing or other surface, which could cause debris to fall into the sample. Document any deviations in the methodology on the datasheet.

Note the water level and flow at the time of sampling and document on the datasheet using the following index (Table 2).

Table 2. Flow severity index

FLOW CODE	FLOW DESCRIPTION
0 = Dry	Streambed is completely dry with no visible pools
1 = Intermittent	Streambed has water visible in naturally occurring isolated pools
2 = No Flow	Streambed has water from bank to bank, but flow is not detectable (e.g. a bayou)
3 = Low Flow	Flows are detectable
4 = Typical Flow	Flows greater than low flow, but stay within the stream channel.
5 = High Flow	Flows that leave the normal stream channel, but stay within the stream banks. Water may be more turbid than at "Typical flow"
6 = Flood	Flows that leave the normal confines of the stream channel and move out on to the flood plain over the stream bank (either side of the stream)

4.4 SAMPLE ALIQUOTING

Prior to aliquoting, label all sample containers with the site identification (ID). Labels must be written directly on the side of the container with permanent, waterproof ink.

If collecting metals, collect a field blank as outlined in Section 4.4.5. Field blanks are taken once per calendar day during sampling events.

One duplicate sample is taken for every 10 samples collected. The sampler can choose which site will be the duplicate. Collecting a duplicate sample typically takes about twice the amount of time that a non-duplicative site takes, so samplers generally select a site where they are safe from traffic. Refer to Section 4.4.8 for more information regarding duplicative samples.

Aliquot water sample into containers as outlined in Sections 4.4.1-4.4.7. Various containers will be required to aliquot water samples.

The number of samples and aliquots will vary depending on the project, as well as research goals and objectives. If you do not have enough volume in the sample container to aliquot all samples, repeat the water collection procedure in Section 4.3 to acquire more sample water. Keep in mind that duplicates should be aliquoted from the same container of sample water. To ensure data quality, follow all procedures described in this SOP regarding sample container preparation.

4.4.1 DISSOLVED METALS

A 125 mL plastic bottle with 0.5 mL of 1:1 HNO_3 preservative.

1. Gently swirl sample in the collection vessel to homogenize.
2. Triple rinse the BD Luer-LokTM Tip 60 mL (or similar) syringe by drawing up sample water from the collection vessel and expelling the rinsate away from work area three times.
3. Draw up 60 mL of sample water in the triple rinsed syringe.

4. If using an Acrodisc® (or similar) pre-filter, securely attach it to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Then securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the pre-filter utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the pre-filter.
5. If a pre-filter is not needed, securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Ensure the filter is securely attached so that no sample leaks out prior to filtration.
6. Rinse the filter by pushing 5-10 mL of sample through syringe and expel it away from work area.
7. Filter 50 mL of sample water into the sample container. It may be necessary to use more than one filter. If so, repeat the steps above for each additional filter used to fill the sample container with a total of 50 mL of sample water.
8. Tightly secure the lid back on the sample container so that it does not leak.
9. Refer to Section 4.4.8 for instructions on taking a duplicate sample.
10. Place syringe back in the protective plastic sleeve.
11. Placing the sample bottle on ice is NOT required.

4.4.2 TOTAL RECOVERABLE METALS

A 125 mL plastic bottle with 1.0 mL of 1:1 HNO_3 preservative.

1. Gently swirl the sample in the collection vessel to homogenize.
2. Draw up sample water in the triple rinsed syringe. If there is enough control over the collection vessel, you can pour directly from the collection vessel to the 125 mL bottle, but over-pouring will result in loss of preservative and an inadequate sample. Use of the syringe for smaller volumes in containers that contain acid is encouraged.
3. DO NOT filter the sample.
4. Slowly depress the triple rinsed syringe to fill the labeled plastic bottle with approximately 100 mL of sample water from the collection vessel. Ensure that the bottle is not over-filled in order to prevent loss of acid preservative and sample.
5. Tightly secure the lid back on the sample container so that it does not leak.
6. Refer to Section 4.4.8 for instructions on taking a duplicate sample.
7. Dispel any extra water in the syringe away from the sample area.
8. Place the syringe back in the protective plastic sleeve.
9. Placing the sample bottle on ice is NOT required.

4.4.3 DISSOLVED ORGANIC CARBON (DOC)

A 40 mL glass vial with septa and 0.1 mL 1:1 H_2SO_4 preservative.

1. Gently swirl the sample water in the collection vessel to homogenize.

2. If the syringe has not yet been rinsed, triple rinse the BD Luer-LokTM Tip 60 mL (or similar) syringe by drawing up sample water from the collection vessel and expelling the rinsate away from work area three times.
3. Draw up the sample water in the triple rinsed syringe.
4. If using an Acrodisc® (or similar) pre-filter, securely attach it to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Then securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the pre-filter utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the pre-filter.
5. If a pre-filter is not needed, securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Ensure that filter is securely attached so that no sample leaks out prior to filtration.
6. Rinse the filter by pushing 5-10 mL of sample through syringe and expel it away from work area.
7. Filter 40 mL of sample water into the sample container. It may be necessary to use more than one filter. If so, repeat the steps above for each additional filter used to fill the sample container with a total of 40 mL of sample water. The vial should be filled to the neck with some airspace at the top to prevent spillage of H_2SO_4.
8. Tightly secure the lid back on the sample container so that it does not leak.
9. Refer to Section 4.4.8 for instructions on taking a duplicate sample.
10. Place syringe back in the protective plastic sleeve.
11. Immediately place sample container in the holding case, place on ice and store at ≤ 6.0 °C until relinquished to the lab.

4.4.4 ALL OTHER PARAMETERS

A 946 mL (quart) plastic container.*

1. Gently swirl sample in collection vessel to homogenize.
2. Fill labeled 946 mL (quart) plastic container with sample water.
3. Tightly secure the lid back on the sample container so that it does not leak.
2. Refer to Section 4.4.8 for instructions on taking a duplicate sample.
3. Immediately place sample container(s) on ice and store at ≤ 6.0 °C until relinquished to the lab.
4. *If holding times cannot be met, bring an additional labeled 946 mL plastic container with H_2SO_4 preservative for each sample (and duplicate) and follow procedures above. Take care to not spill contents.

4.4.5 DISSOLVED METALS FIELD BLANK

A 125 mL plastic bottle with 0.5 mL of 1:1 HNO_3 preservative.

Dissolved metals field blanks are collected to assess contamination that may occur from field sampling equipment. Field blanks are taken once per calendar day during sampling events and should be taken after collecting the final field aliquot of the day. Type I DI water is used as the sample and is collected in a 125 mL plastic wide-mouth bottle with 1.0 mL of 1:1 HNO_3 preservative. To collect a field blank:

1. Label the appropriate sample container with "Field Blank".
2. Triple rinse all components of the collection vessel with Type I DI water.
3. Add at least 250 mL of Type I DI water to the collection vessel. Ensure that the water passes through all components of the collection vessel.
4. Triple rinse the BD Luer-LokTM Tip 60 mL (or similar) syringe by drawing up sample water from the collection vessel and expelling the rinsate away from work area three times.
5. Draw up 60 mL of Type I DI water in the triple rinsed syringe.
6. If Acrodisc® (or similar) pre-filters were used during sampling, be sure to use one for collecting the field blank. Securely attach it to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Then securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the pre-filter utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the pre-filter.
7. If pre-filters were not used during sampling, securely attach a 0.45 µm Acrodisc® (or similar) filter to the tip of the syringe utilizing the filter packaging to handle and attach the filter. Do not touch the filter with bare or gloved hands when attaching it to the syringe. Ensure that filter is securely attached so that no sample leaks out prior to filtration.
8. Rinse the filter by pushing 5-10 mL of Type I DI through syringe and expel it away from work area.
9. Filter 50 mL of Type I DI water into the sample container.
10. Tightly secure the lid back on the sample container so that it does not leak.
11. Place syringe back in the protective plastic sleeve.
12. Placing the sample bottle on ice is NOT required.

4.4.6 *E. COLI* SAMPLING

E. coli sampling typically occurs during the primary contact recreation season (May 1 to September 30). For assessment of individual sample criteria, at least eight (8) samples, evenly spaced throughout the primary contact recreation season, are needed. For calculation and assessment of geometric mean criteria, a minimum of five (5) samples spaced evenly and within a thirty (30)-day period are needed.

A sterile 120 mL polyethylene IDEXX bottle

1. Gently swirl sample in collection vessel to homogenize.
2. Fill labeled 120 mL plastic container, fill to at least the 100 mL fill line with sample water.
3. Tightly secure the lid back on the sample container so that it does not leak.

4. Refer to Section 4.4.8 for instructions on taking a duplicate sample.
5. Immediately place sample container(s) on ice and store at ≤ 6.0 °C until relinquished to the lab.

4.4.7 CHLOROPHYLL *A* SAMPLING

4.4.7.1 Sestonic Chlorophyll *a* Sampling (Phytoplankton)

A 1,000 mL brown plastic bottle.

Chlorophyll *a* is collected to assess productivity of a waterbody between May and September. Depending on the physical and chemical state of the waterbody, chlorophyll *a* can be bound up in rooted, filamentous, or sestonic (within the water column) plants. The following methodology described is for sestonic chlorophyll *a* only.

1. Swirl sample in collection vessel to homogenize. Do not aerate the sample.
2. Fill labeled container with sample water, leaving a little head space.
3. Close the sample container, ensuring that the lid is properly threaded and tightened so that the sample does not leak or get contaminated.
4. Immediately place sample container(s) on ice and store at ≤ 6.0 °C.
5. If not returning samples to the lab for processing within 24 hours, refer to Section 4.4.7.3 on how to filter samples in the field.

4.4.7.2 Benthic Chlorophyll *a* Sampling (Periphyton)

A 1,000 mL brown plastic bottle.

Periphyton is collected in high gradient streams during periods of stable flow within the critical season (July-September). In the case of extreme events, such as scouring floods or droughts, allow three weeks for periphyton recolonization. Collect periphyton samples before any other data collection occurs to avoid dislodging sample material by walking through the riffles. For streams with distinct riffle-pool patterns, collect 6 periphyton samples from each of the first two riffles within the stream reach for a total of 12 samples. Preferably, selected riffles should be greater than 3 meters long to allow for adequate sampling space. If only one riffle occurs within the reach, collect all 12 samples within the single riffle. If riffles are dry, collect samples within a shallow run. If there is no available riffle or run habitat within the reach, collect samples from the shallow tail area of pools.

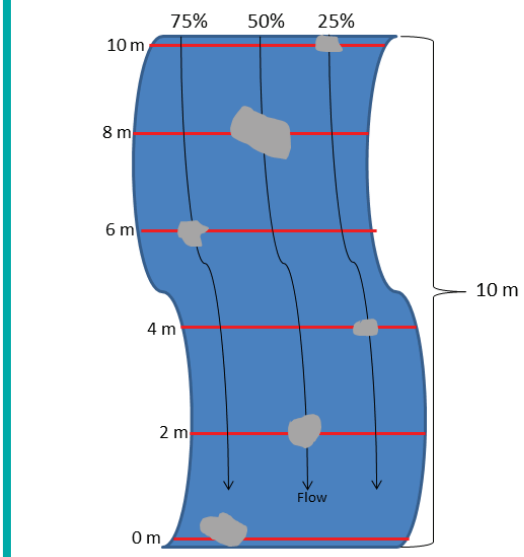


Figure 1. Diagram of periphyton sampling locations within a delineated riffle

1. Determine the number of riffles within the reach. Refer to Section 7.3 for reach delineation guidelines. If at least two riffles occur within the reach, collect 6 rocks in one riffle and 6 rocks in a second riffle.

*If only one riffle occurs within the reach, collect all 12 samples within the single riffle. If riffles are dry, collect samples within a shallow run. If there is no available riffle or run habitat within the reach, collect samples from the shallow tail area of pools.

2. Starting at the bottom of the reach, walk upstream until you approach the first riffle > 3 meters long that occurs in the reach.
3. Periphyton is sampled along 6 evenly spaced transects perpendicular to flow. Measure the length of the riffle from toe to head and divide the total riffle length by 5 (divide by 11 if only sampling a single riffle) to determine transect spacing. The first transect is at toe of riffle (0 m), and last at head (Figure 1).
4. Starting at the left wetted edge, walk perpendicular to flow along transect 1 until you reach 25% of the total stream width.
5. Without looking, reach down and gently pick up the cobble (150-500 mm rock) nearest the toe of your right foot, taking care to not disturb the periphyton on the "top" surface of the rock. If the cobble is too large to lift or is heavily colonized by macrophytes, select a different cobble from as close to the same area as possible.
6. Gently place the rock in a large plastic pan with the "top" surface where the periphyton is located facing up, and carry to the stream bank for processing. Make every effort to avoid disturbing established periphyton during transport.
7. The remaining samples will be taken in the locations detailed in Table 3.

Table 3. Riffle transect locations

RIFFLE TRANSECT	LOCATION
1	25% of wetted width
2	50% of wetted width
3	75% of wetted width
4	25% of wetted width
5	50% of wetted width
6	75% of wetted width

8. Once all six rocks are collected, transfer one rock to a smaller white pan for processing.
9. Prior to removing any periphyton, examine and remove any conspicuous attached invertebrates or invertebrate cases.
10. Place a 2.5" (6.35 cm) diameter rubber delimiter, with a 1.0" (2.54 cm) diameter opening, in the center of each cobble on "top" surface where the periphyton is located. Total area sampled per target riffle is 30.4 cm^2.
11. Using a clean toothbrush and scoopula, remove all periphyton within the delimiter. Avoid disturbing periphyton outside the delimiter.
12. Rinse dislodged algae from cobble into white pan using stream water in a squirt bottle (using no more than 400 mL of stream water per composite riffle).

13. Carefully examine all equipment for dislodged periphyton. Rinse dislodged periphyton into the pan if it originated from the sample area; otherwise discard.
14. Repeat for all 6 collected rocks.
15. Transfer the composited sample for each riffle into an amber HDPE sample bottle using a small funnel. Scrub and rinse the inside of the pan to collect any residual periphyton.
16. Walk upstream to the second riffle within the reach and repeat the riffle sampling process for a total of 12 rocks. Total area sampled per site is 60.8 cm^2.
17. Label the amber HDPE sample bottle with the site name, date, time, and any other information necessary.
18. Store sample on ice to transport and relinquish to DEQ Office of Water laboratory (or other approved laboratory) for processing with holding times not to exceed 24 hours.
19. If not returning samples to the lab for processing within 24 hours, refer to Section 4.4.7.3 on how to filter samples in the field.

Figure 2. Periphyton being removed from cobble

Figure 3. Processed cobble

4.4.7.3 Filtering Chlorophyll *a* Samples in the Field

Filtering Equipment

This list may need to be modified depending on availability of equipment, sampler preference, or laboratory policy.

- Vacuum pump (manual or electric)
- Filter cup manifold
- Manifold funnels with screen
- Filter cups
- 47-mm diameter Whatman GF/F filters
- Filtrate flask and tubing
- Forceps
- Millipore 47 mm petri dishes
- Aluminum foil
- Ice chest and ice (or other refrigeration capable of maintaining samples at ≤ 6 °C)
- Graduated cylinders (variety of sizes)
- Type I DI (16 oz. wash bottle)
- Ziploc bags
- Permanent marker

Filter samples in the field that cannot be delivered to a state accredited laboratory for processing within 24 hours. Field filtration and processing typically occurs in an environment such as a hotel room where lighting and other environmental conditions can be reasonably controlled. Filtration and processing should never occur in direct sunlight or in other conditions where environmental factors cannot be reasonably controlled.

1. Rinse filter towers and graduated cylinders.
2. Set up the filtering manifold by connecting it to a vacuum pump and use forceps to place a glass fiber filter on each of the filter towers needed for use. Place filters with the textured side up.
3. Place a filtering cup onto each of the filtering towers.
4. Remove sample from ice/refrigeration.
5. Gently invert the sample multiple times to homogenize.
6. The volume of water needed to filter will vary depending on the algal or suspended sediment content of the sample. Enough water will need to be filtered to give a green or brown appearance to the filter.
7. Pour a known amount of water into the filtering cup and turn on the vacuum pump – you may need to open the individual valves on the filter towers. Record the amount of water filtered.
8. Label a disposable 37 mm petri dish with the site name, volume filtered, time collected, time filtered, and date.
9. Once all the water has passed through the filter, use forceps to remove the filtering cup and carefully fold the filter in half so that the side with the filtered material is not exposed. If all of the water did not pass through the filter, re-start the filtering process.
10. Place the folded filter in the labeled petri dish.
11. Rinse filter towers and graduated cylinders between samples.
12. Repeat this process for all samples that will not meet 24 hour holding time.
13. Once complete, stack all dishes together and wrap in aluminum foil to block out light.
14. Store the wrapped petri dishes in freezer bag and maintain at ≤ 6 °C for transport to a State accredited laboratory.
15. Do not let water seep into the bags the filters are held in.

4.4.8 DUPLICATE SAMPLE COLLECTION

Duplicate collection procedures may differ depending on laboratory requirements. At DEQ, duplicate samples are taken for all parameters at a rate of 10% (1 duplicate per 10 samples). Take all duplicates at a single selected site. Duplicate sites are selected at the discretion of the sampler and are usually selected based on ease of collection and distance of sample processing location from the road.

For the dissolved metals duplicate, filter 100 mL of sample water into the container. For the total metals duplicate site, pour 100 mL of sample water into a 125 mL container and pour 200 mL of sample water into a 250 mL container. For a duplicate dissolved organic carbon sample, fill three additional vials with filtered sample water. To collect a duplicate sample for all other parameters, fill one additional container with sample water. Label all applicable containers with their sample ID and "dup". If holding times cannot be met, collect a duplicate 946 mL plastic container with H_2SO_4 preservative.

If the study requires sestonic chlorophyll *a* collection (May – September), collect a chlorophyll *a* duplicate at the same location as the other duplicates. No duplicate sample is required for periphyton sampling. If the study requires *E. coli* collection, collect an *E. coli* duplicate at the same location as the other duplicates.

4.4.9 RELINQUISHING SAMPLES TO THE LABORATORY

Procedures for relinquishing samples to an accredited laboratory may vary between labs. All sampling personnel must coordinate with a laboratory representative to ensure that the lab's relinquishing procedures are understood. All relinquishing procedures must be followed in order to retain the sample's viability and to maintain an accurate chain of custody (COC).

5. Discrete In Situ Meter Measurements and Unattended Deployments

5.1 OVERVIEW

The goal of this section is to describe basic protocols for storing, calibrating, deploying, retrieving, and QA/QC data for multiparameter sondes for discrete and short-term continuous deployment in lakes and streams. Always consult the sonde's manual for specific instructions.

5.2 CALIBRATION AND SAMPLING EQUIPMENT

- National Institute for Science and Technology (NIST) - certified thermometer (monthly check)
- Bucket of room-temperature water (monthly check, see temperature calibration in Section 5.3)
- Handheld unit for in situ meter
- Field cord to connect handheld unit to in situ meter (if applicable)
- Multi-parameter in situ meter
- Appropriate standards and Type I DI water
- Clean sonde guard/calibration cup
- Deployment bulkhead caps (model dependent)
- Protective PVC (or similar) tubes with holes to allow flow, painted with black, grey, green, brown, or similar colors for camouflage (unattended deployments)
- Steel security cables, locks, and keys (unattended deployments)
- Sonde deployment datasheets
- Computer equipped with appropriate cord and software (for data retrieval)
- Extra batteries
- Cinder blocks or other anchoring devices (if needed for unattended deployments)

5.3 CALIBRATION

Record calibration data on a calibration datasheet or lab notebook. Each meter should have its own calibration logbook. Scan and save all calibration datasheets when sampling is completed.

Before each calibration, clean probes using the central wiper (if available) and a lint-free cloth such as lab-tissue. Use care while cleaning sensors.

Prior to sonde use, calibrate according to methods outlined in the user manual/manufacturer's guidelines. Perform proper maintenance if any sensor is not reading or calibrating correctly.

Calibrate sonde probes in the following order (as applicable). Rinse probes with Type I DI water between standards. Record both the pre-calibration and post-calibration values:

Temperature

- Check temperature monthly during use.
- Place a NIST thermometer and the sonde in the same bucket of water (can be Type 1 DI, tap, or sample water).
- For sondes used at only certain times of year (unattended deployments), check temperature monthly during use and before deployment.
- Record in the calibration book.

Conductivity

- If collecting in situ conductivity, calibrate conductivity before each sampling event.
- Calibrate using specific conductance with one calibration standard greater than 1000 µs/cm. Ensure that there are no air bubbles in the sensor and that the sonde is reading in the correct units.
- Record pre and post-calibration values in the calibration book.
- Check calibration standard against a second source, monthly.
- Record monthly checks in the calibration book.

pH

- Calibrate pH before each sampling event.
- Conduct a 2-point calibration using pH 7 and pH 4 or 10 (will depend on the expected pH of the sample water DEQ typically uses pH 4).
- Record pre and post-calibration values in the calibration book.
- During calibration for each standard, write down the millivolts (mV) (mVs will not have a pre- and post-calibration value).

  mVs can provide information about the effectiveness of the pH sensor.

  - While calibrating at ~25°C, the mV range between standards should fall between 165 and 180. If the range falls outside of these values, the pH sensor needs replacement.
  - mVs for pH 7 should be 0 ± 50

- mVs for pH 4 should be 177 ± 50
- mVs for pH 10 should be -177 ± 50.

EXAMPLES		
mV for pH 4 = 173	mV for pH 7 = -30 173 – (-30) = 203	mV range = 203; pH sensor needs **replacement**
mV for pH 4 = 147	mV for pH 7 = -20 147 – (-20) = 167	mV range = 167; pH sensor is **OK**

Dissolved Oxygen (DO)

- Calibrate DO before each sampling event.
- Ensure there are no water droplets on the DO or temperature probe.
- Record the barometric pressure using an internal barometer, a local barometer or an online resource. If not done automatically in the instrument's programming, adjust the reading for elevation. Refer to a solubility table to check appropriate calibration values.
- Calibrate DO using percent saturation. Put a small amount (~1/8 inch) of water into the calibration cup. Loosely thread the calibration cup (do not seal) to the sonde. Wait for the % DO and temperature to equilibrate.
- Record pre and post-calibration values in the calibration book.

To ensure accuracy of measurements, calibrate specific conductance, DO, and pH within 12 hours of each deployment. If temperature is not accurate, send the instrument in for servicing.

5.4 STORAGE

- Rotate meters that are used with regularity in and out of service to maintain usable condition throughout the year.
- Store sondes, Type I DI, and calibration standards at room temperature prior to calibration.
- Store sondes not in use for over 4 weeks in pH 4 buffer.
- Store sondes used more regularly (at least once per 4 weeks) in a small amount (~0.5 inches) of tap water.
- Condition sondes coming out of long term storage in tap water for at least one hour before calibration and use.

5.5 DISCRETE SAMPLING PROCEDURES

Unless otherwise specified, all water quality samples collected will have paired in situ data readings with a calibrated multiparameter sonde. See Section 3.3 for correct water collection methodology.

For water quality meters without calibration cups:

Collect water quality meter readings **after** all aliquots have been completed. Pour sample water over the probe three times to rinse. Submerge probe into the collection vessel ensuring all probes are fully submerged. Wait for readings to stabilize. Record all relevant information.

For water quality meters with calibration cups:

Pour water from the bucket into the calibration cup, place the probe in the cup, swirl to rinse, and discard rinse water. Complete this three times in total. After three rinses, fill the calibration cup with sample water and place the probe into the cup ensuring all probes are fully submerged. Wait for readings to stabilize. Record all relevant information.

5.6 POST SAMPLING QUALITY ASSURANCE CHECK

When sampling is complete for the day, perform a post-field QA check in the lab or other controlled environment by checking readings against calibrations standards. Place standards in the calibration cup in the same order as calibration (conductivity, pH, DO) and record readings in the calibration book. See Table 4 for maximum allowable deviations from calibration standards for post-field checks. If a value falls outside of the maximum allowable limit ranges (Table 4), flag the data recorded from whichever probe malfunctioned as unusable.

Table 4. Acceptable value ranges for post-field QA check (TCEQ, 2012)

MEASURED FIELD PARAMETER	MAXIMUM ALLOWABLE LIMITS FOR WATER-QUALITY SENSOR VALUES
Temperature	±0.2 °C (pursue factory maintenance) ±0.5 °C (flag data)
Specific conductance	±5%
Dissolved Oxygen	±6% saturation, ±0.5 mg/L
pH	±0.5 pH units

5.7 UNATTENDED SONDE DEPLOYMENT AND RETRIEVAL PROCEDURES

Duration of unattended sonde deployment depends on study objective, but is typically deployed for a minimum of 48 hours.

5.7.1 PROGRAMMING SONDE

Refer to sonde user's manual for exact procedures on how to program the model being used.

1. Name the file with a unique name in a known location.
2. Set all desired parameters to record.

3. Set desired measuring increment (typically once every 15 minutes, but no longer than once an hour).
4. Program sonde to record data for the duration of deployment.

5.7.2 SHORT-TERM DEPLOYMENT

1. Ensure sonde batteries are fully charged or charged long enough to last throughout deployment. Write the battery percentage on the datasheet.
2. Check local weather to avoid deploying sondes during heavy rain events where readings could be difficult to interpret or equipment could be lost or damaged.
3. Choose a suitable location to deploy the calibrated sonde. Deploy in a run (if available). Avoid deploying sonde in areas of stagnant water. Locate suitable vegetation or boulder(s) to anchor sonde. Examine vegetation or boulder(s) to ensure they are stable and unlikely to be washed away in a large flushing event. In the event of a scoured bank or lack of natural anchoring points, use a cinder block.
4. Remove the calibration cup from the sonde and replace with a sonde guard (if applicable) then place the sonde in a protective sleeve (i.e. PVC tube).
5. Ensure that the sonde won't slip out of the protective cover anytime during deployment.
6. Orient the sonde so sensors are facing downstream to prevent fouling or sediment build-up on sensors.
7. Thread cables or chains through the sonde's bail (handle) and through the holes in the top of protective sleeve. Wrap cables around the chosen anchor and use a padlock to secure. Ensure that sensors are far enough below the water's surface so if water levels drop the sonde will remain submerged.
8. Take care to ensure the sonde/sonde guard is not sitting directly on the streambed.
9. Hide the sonde and cables as much as possible to prevent tampering.
10. Record the timing of deployment, coordinates, description of the deployment location, and field conditions for easier retrieval.
11. Take a photograph of each deployment location to add context for data interpretation.

5.7.3 SONDE RETRIEVAL

1. Locate the sonde, remove from protective sleeve, and rinse in stream water to remove any accumulated sediment or algae.
2. End the sonde deployment according to sonde user manual.
3. Check that data were recorded and saved in a uniquely named file.
4. Record relevant information on the datasheet.
5. Check your sonde readings with a post-field QA check in the lab or other controlled environment by checking readings against calibration standards.
6. Record the post-field QA check values in the calibration book.

5.8 DATA VALIDATION

Once data have been properly collected from both discrete and continuous deployment, post-field QA checks should be performed on each sensor (Section 4.6). A full calibration is not necessary, but a known standard should be placed in the calibration cup in the same order as calibration (conductivity, pH, DO) and readings recorded on the calibration log.

6. Flow

6.1 OVERVIEW

The goal of this section is to describe basic protocols for equipment calibration, transect selection, and collecting velocity measurements using select flow meters. Flow is generally paired with biological and/or water quality sample collection depending on study goals. When using flow meters, always consult the flow meter's manual for specific instructions.

6.2 EQUIPMENT

- Flow datasheet
- FlowTracker2 or Marsh McBirney flowmeter (or similar)
- USGS top-setting wading rod
- Measuring tape with tenths/foot increments at least 100 feet long
- Batteries, extra battery pack (filled) for FlowTracker2
- Screwdriver
- Bucket and rebar (for calibration of Marsh McBirney flowmeter)

6.3 TRANSECT SELECTION

The first step for an accurate flow measurement is to select a cross-section with suitable characteristics. The location for flow measurements should be as representative as possible of your stream reach. Try to avoid areas that are over-widened or manipulated by manmade structures (like bridges and culverts), if possible. Flow may be collected any time of the year as long as site selection requirements are met.

When selecting a site for flow measurement consider the following (Turnipseed and Sauer 2010):

- Stream channel is wadeable and reasonably straight, streambed is stable and free of large rocks, weeds, and physical obstructions that would create eddies, slack water, or turbulence that could influence velocity measurements.
- Water surface is moving and smooth with steady uniform flow conditions typically found in riffles, runs, or glides.
- Depth and velocity should be relatively uniform.
- Stream width is wide enough for at least 10 cross-sections.

- Site is ideally located close to site of interest (sonde deployment location, within delineation reach, or near monitoring station).
- Ideally the velocities are, for the most part, greater than 0.5 ft/s, and depths that are greater than ~0.5 ft. These conditions are not always possible to find in the field.

It is usually not possible to satisfy all of these conditions. Select a reach that exhibits as many of these qualities as possible.

6.4 FIELD PROCEDURES FOR MEASURING FLOW

Streamflow is expressed in cubic feet per second (ft3/s). A streamflow measurement is made by subdividing a stream cross-section into segments and measuring the depth, width, and average velocity within each segment. The total streamflow is the summation of the flow in each of the segments of the stream cross-section. Streamflow measurements can be made in a variety of ways. DEQ uses the mid-point method to measure velocity (Figure 4). This SOP describes methods for an acoustic doppler velocimeter (SonTek FlowTracker2) or an electromagnetic flowmeter (Marsh McBirney). The following procedure for measuring flow applies to both types of meters.

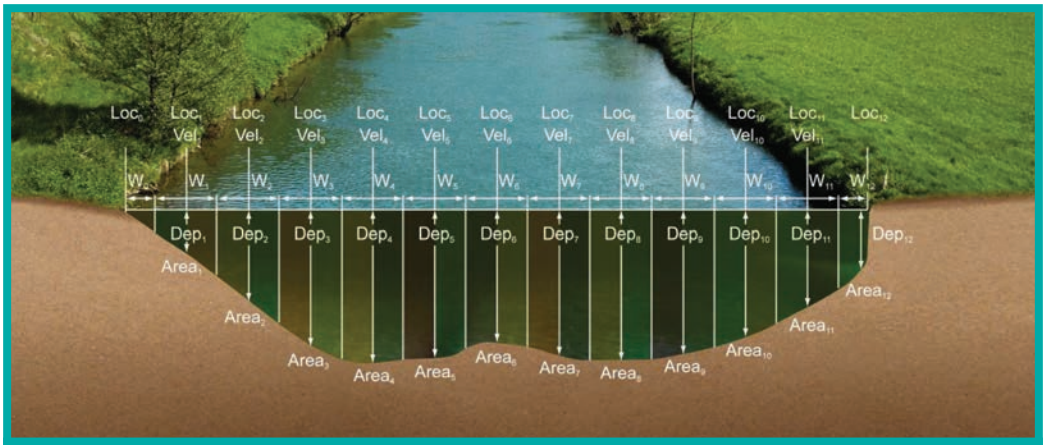


Figure 4. Mid-point method for measuring velocity across a transect. (FlowTracker2 User's Manual, 2019).

The procedure for collecting depths and velocity is as follows:

1. Select cross-section according to Section 6.3 in this document. You can remove rocks and debris within the cross-section and from the reach of stream immediately upstream to improve the cross-section flow, but you cannot change the cross-section once you commence collecting velocity and depth.
2. Ensure meter has been calibrated before collecting data. Refer to calibration instructions in Sections 6.8-6.9 for specific details on calibration for each type of meter. Check to ensure the meter is functioning properly and the correct units (ft/sec) are displayed.
3. Measure wetted width by stretching a tape measure across the stream perpendicular to its flow. Tightly suspend the measuring tape across the stream, approximately one-foot above water level and secure at both ends.
4. Record the total wetted distance indicated by the tape from water's edge to water's edge.

5. Divide the total wetted stream width into equally sized intervals never less than 1/2 foot increments.
    - For streams less than or equal to 10 feet wide, the minimum number of intervals is 10 (Starting bank, 9 wetted interval measurements, and ending bank).
    - For streams greater than 10 feet wide, the preferred number of intervals is 20 (starting bank, 19 wetted interval measurements, ending bank).
6. Attach the velocity meter probe to the top-setting wading rod.
7. Stand downstream of the tape and to the side of the midpoint of the first interval (closest to the bank). Note which bank you start at.
8. At the midpoint of the interval, place the top-setting wading rod in the stream so the base plate rests on the stream bed.
9. Read the water depth off of the graduated main rod. Depths on the top-setting wading rod are a vernier scale marked with lines. Each single mark represents 0.10 ft; each double mark, 0.50 ft; and each triple mark, 1.00 ft.
10. Adjust the position of the sensor to the correct depth at each midpoint to begin velocity reading. Press down the handle of the top setting wading rod to move the smaller diameter sliding suspension rod to the correct position. Feet are marked on the sliding suspension rod and tenths of feet are labeled on the static portion of the rod near the handle (e.g. if the water depth was 1.6 ft, you would align the 1 ft mark on the sliding rod with the 6 mark on the tenths of feet portion near the handle). The top setting wading rod is designed so the user can easily set the sensor at 20, 60, and 80 percent of the total depth. See below to determine depth of sensor (Refer to Appendix B for top-settling rod instructions).
    - If the water depth is less than or equal to 2.5 ft., adjust the position of the probe on the top-setting wading rod so it is at 60% of the measured depth below the surface of the water. When the scale on the sliding suspension rod is aligned with the vernier scale on the handle to the water depth observed on the hexagonal rod, the attached instrument probe is automatically set to 60% depth from water surface.
    - If the water depth is greater than 2.5 ft., take a two point measurement at 20% and 80% of the depth from the water surface. The average of these two readings is considered the water velocity for the respective measurement point. To set the probe at the 20% depth, first multiply the water depth by two, and then use the calculated number to line up the foot scale as with the 60% depth. The same method is used for the 80% depth, except the calculated value is the water depth divided by two. (See Appendix B for examples)
11. Face the probe upstream at a right angle to the cross-section (this should be parallel to the flow). The probe should be held in a level position. Do not adjust the angle of the probe, even if local flow eddies hit at oblique angles to the cross-section.
12. Wait 40 seconds to allow the meter to equilibrate then measure the velocity. Negative recordings are not preferred (usually occurs because of backwater) but are sufficient and should be recorded assuming you have a properly calibrated meter. For details on velocity readings specific to the meter you are using refer to Sections 6.5-6.6.
13. Move to the midpoint of the next interval and repeat Steps 7 through 11. Continue until depth and velocity measurements have been recorded for all intervals.

14. Before leaving the site, make sure the flow datasheet is filled out. When using the Marsh McBirney, fill out the entire datasheet. The FlowTracker 2 records transect data automatically, therefore you do not need to record transect data on the flow datasheet. However, you will fill out the top portion of the flow datasheet and record the final discharge reading displayed by the FlowTracker2.

6.5 FLOW MEASUREMENTS USING SONTEK FLOWTRACKER2

The FlowTracker2 velocimeter measures velocities across the transect and is used in conjunction with the top-setting wading rod, which measures stream depth, to determine site-specific discharge measurements. The same field procedures outlined for use with electronic or mechanical flow meters apply to the FlowTracker2. The main difference is that the FlowTracker2 records all of the data and calculates the flow. The FlowTracker2 also has an internal Geographic Positioning System (GPS) with a manual or automatic function.

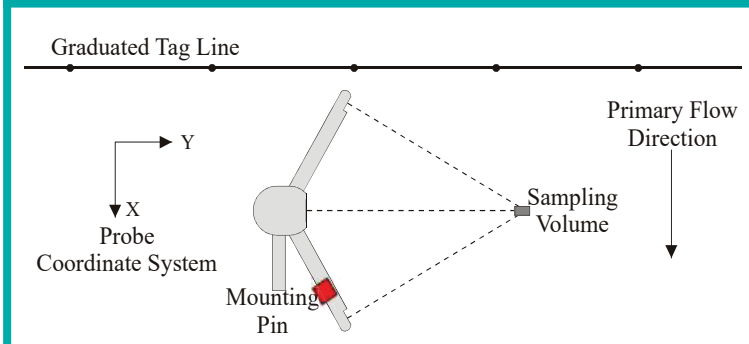
The following procedure is an overview. Refer to the manual for exact procedures and guidance for this instrument.

The FlowTracker2 must be perpendicular to the graduated tagline or measuring tape that was setup across the measurement section.

The procedure for measuring velocity using FlowTracker2 is as follows:

1. Turn the instrument on. From the main home screen, select the Measurement function.
2. The software will navigate to "New File Type" screen. For DEQ uses, we will always select Discharge on this screen.
3. The software will then navigate to "New File Template" screen. The options are to create a new template based on SonTek default settings or select an existing template created by the user. For our purposes, we will select New File Template. Scroll through the options using the up and down arrow keys and press enter.
4. The software will navigate to "New Data File" screen. Enter the Site Number, Site Name, Operator, and Comment. Select Ok at the bottom right of the screen.

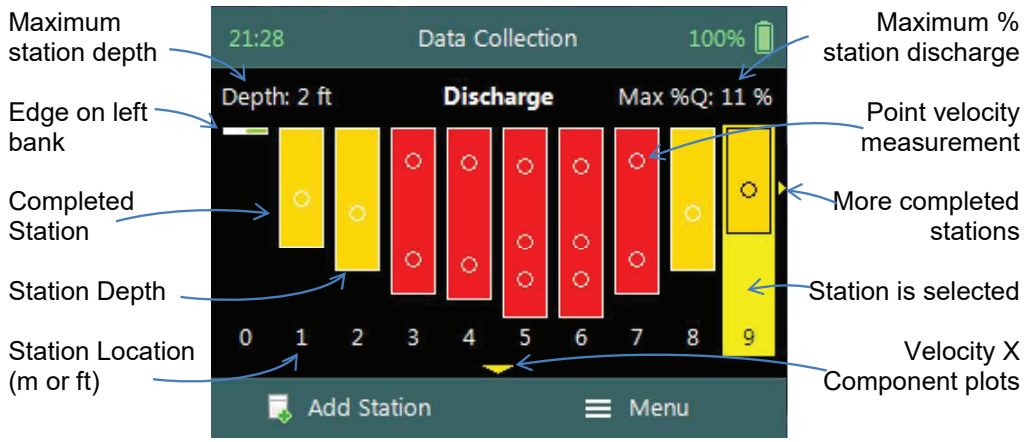
Figure 5. FlowTracker2 probe orientation relative to stream flow



5. Perform Automated Beam Check in the stream before data collection starts. See Section 6.8.
6. Proceed to the "Data Collection" function. The Data Collection screen is the main screen of the data collection process during a discharge measurement. All the software functions required to perform a discharge measurement is available from the data collection screen. From the "Data Collection" screen select Add Station* to begin collecting flow data for each station in the transect. The Add Station screen consists of the following common parameters that are required for all station types: a) Location, b) Depth, c) Station Type, d) Correction Factor, e) Comment. *FlowTracker2 calls each interval across a transect a "station".

7. For the first station on the transect, on the "Add Station" screen, enter 0 for location and depth which is usually 0. Enter your starting bank (either Left Bank or Right Bank) under Station Type. This defaults to a 0 velocity reading.

Figure 6. The key features of the data collection window (FlowTracker2 User's Manual, 2019).



8. For the next stations on the transect, select Add Station, enter location (distance from edge of water), depth at station, and open water for station type. Open Water will be the main station type for the majority of stations in the transect.
   - When Open Water is selected a "Velocity Method" option comes up. Select either 0.6 or the two point option (0.2/0.8) based on < or >2.5 depth. When selecting the two point option, the meter will move on to the second velocity reading automatically after the first is recorded.
   - Select Measure. The screen will show a picture of the top setting wading rod to show correct alignment based on depth and velocity option. Also a level (level bubble should be green), Signal-to-Noise Ratio (SNR) readings, and beam check will be displayed. Check that these readings are normal and sensor is level before selecting Start. Note: SNR readings (signal strength) should ideally be >10 dB and at minimum be 2-3 dB, also the beam check graph will show peaks where both lines are roughly in the same place if probe is reading correctly.
   - Wait for the meter to finish its reading. A "Review Point Measurement" screen will display details about the raw data station readings. At this point you have the option to press Redo to redo the velocity measurement or press Accept. Any QC warnings will be displayed on this screen which will help the user determine whether to redo or accept the reading. If data is acceptable press Accept.
   - The "Review Station" screen will appear. This is a summary of all measurements performed at the station. Pressing Close will go back to the main "Data Collection" screen. At this point, you will press Add Station if continuing across the transect.
   - Repeat this step for each remaining station.
9. At the last station, select Add Station, enter applicable location length, enter depth, and the bank side you finished on (left or right) for station type, then select Done.

10. The "Data Collection" screen will appear after the right bank reading. When finished looking through the stations summaries, select Menu then Complete Measurement and Ok to confirm. Selecting Complete Measurement closes the measurement file and the user will not be able to add additional stations or make any changes to the file.

6.6 FLOW MEASUREMENTS USING MARSH MCBIRNEY 2000

The Marsh McBirney measures velocities across the transect and is used in conjunction with the top-setting wading rod, which measures stream depth, to determine site-specific discharge measurements. Velocity readings are real time and displayed on the meter screen. Data must be manually recorded on the flow datasheet for each transect interval. This meter does not store readings. Flow will be calculated manually when using the Marsh McBirney meter.

The following procedure is an overview. Refer to manual for exact procedures and guidance for this instrument.

After attaching the Model 2000 to the top-setting wading rod, check the display outputs of the Model 2000. The procedure for checking display outputs of the Model 2000 is as follows:

1. Press the "ON/C" button to power the meter on.
2. Check that the velocity units are ft/s. If unit is not ft/s, toggle between velocity units by simultaneously pressing "ON/C" and "OFF" buttons until display reads ft/s.
3. Check the display shows that the meter is using Fixed Point Averaging (FPA) with a period of at least 40 seconds. If it is not, press the up or down arrows until the display reads 40 seconds. FPA averages the velocities read by the meter over a set period of time. At the end of this time, the display shows the averaged velocity. The FPA display shows a time bar underneath the velocity output that indicates the amount of time remaining in the averaging period. To toggle between FPA and time constant filtering (rC), simultaneously press the up and down arrow buttons on the meter.

The procedure to collect a velocity reading:

4. Record the distance from the bank (in feet) at the midpoint of the interval and the depth indicated on the top-setting wading rod (in tenths of a foot) on the flow datasheet (Appendix C).
5. Press the "ON/C" button. The Marsh McBirney will start recording velocity for 40 seconds. When the time bar on the bottom of the Model 2000 display reaches the end of 40 seconds, it will start over and the average velocity will be displayed. Record this velocity measurement number on the flow datasheet. Do not round values when recording flow data. Streamflow is a sum of the velocities across all intervals in a cross-section. Rounding the velocity value at each interval creates error in the final calculation.
6. Repeat for each interval along the transect/cross-section.

6.7 DATA RETRIEVAL AND STORAGE

Data retrieval only applies to the FlowTracker2 for flow recordings. Refer to the FlowTracker2 user's manual for exact procedures on how to upload data files from the handheld to a desktop computer. Upon returning from the field, upload data files to a computer and put into a Microsoft Excel file (or compatible).

6.8 SONTEK FLOWTRACKER2 CALIBRATION

The FlowTracker2 is calibrated at the manufacturer and does not require any calibration by the user. QC is continuously performed on all variables collected at different stages of a measurement and if any values exceed the expected criteria a warning is supplied. To check data accuracy, a Beam Check is performed.

The automated beam check performs a number of QC checks on the data collected to determine if the flow conditions are suitable for discharge measurements. The data that is displayed graphically during the Automated Beam Check is time series of the QC parameters.

To perform an Automated Beam Check:

- The Automated Beam Check functions are available before data collection start and during velocity measurement from the "Data Collection Menu"
- The Automated Beam Check screens will display a list of directions. Select start in the bottom right corner on the screen.

Refer to the SonTek FlowTracker2 manual to evaluate Beam Check Results.

6.9 MARSH MCBIRNEY FLOWMETER CALIBRATION

Calibrate the Model 2000 at the beginning of each field day by performing a zero check and, as necessary, a zero adjustment. The following procedure for the Model 2000 is consistent with the manufacturer's instruction manual (Marsh McBirney 1990).

To calibrate the Model 2000, complete the following steps before each field day:

Figure 7. Set up for calibration of the Model 2000 flow meter. This example shows the probe suspended from the rebar, and stabilized by the hammer.

1. Check to make sure that the sensors on the Model 2000 probe are clean. Oil films on the electrodes may cause noise and interfere with readings.
2. Suspend the probe in a five gallon bucket of water. Ensure that the probe is at least three inches away from the sides and bottom of the bucket. Figure 5 represents an example set up for calibration.
3. Wait five to ten minutes before continuing the check/calibration process. The water in the bucket and the probe must be completely still before proceeding to Step 4.
4. When the water is still, power the meter on and let the meter run through one complete cycle of velocity measurements. The minimum cycle time is 40 seconds; refer to Marsh McBirney (1990) or Section 5.6 for cycle time adjustment as needed. The velocity reading should be 0. If it reads 0, the meter is reading properly and calibration check is complete. If not, then proceed to Step 5.
5. Depress the "STO" (store) and "RCL" (recall) buttons at the same time to begin the zeroing process. Make sure the probe does not move when the buttons are pressed.

6. Depress the down arrow on the meter three times to set the display to 0. Note: The down arrow must be pressed within 5 seconds or the display will report "err" (error). Repeat the process from Step 3 if an error code appears.
7. The Model 2000 will immediately count down on the display from 32 to 0. The meter will automatically turn off after the zero adjust process is complete.
8. Turn the meter back on without moving the probe. Allow the meter to run through one complete velocity cycle. Zero stability of this instrument is +/- 0.05 ft/sec. If a velocity of greater than +/- 0.05 ft/second is read, repeat the zero adjust process from Step 3. Consider cleaning the sensors or giving more time for the water to stabilize inside the bucket before repeating the process.

7. Habitat

7.1 OVERVIEW

The goal of this section is to describe the protocols for habitat assessment. Not all habitat evaluation procedures are appropriate for all projects. Individual project work plans will specify which habitat evaluation procedures are needed.

A two tier approach is used for evaluating stream habitat quality. Both tiers should be completed at all sites where habitat measurements are needed. Tier one combines both a qualitative (visual estimates) and quantitative (in-stream measurements) approach to developing a habitat profile for each sample reach. In tier one, wetted width, depth profile, bank angle, bank stability, substrate, canopy cover, instream cover, and riparian cover are assessed at each transect (Sections 7.4-7.12).

Tier two (Section 7.13) is an observational (qualitative) approach assessing various habitat parameters along the entire reach and assigning a numeric score (0-20) to each parameter. Scores are separated into four broad categories/conditions consisting of poor, 0-5; marginal, 6-10; sub-optimal, 11-15; and optimal, 16-20. Habitat parameters assessed in both high and low gradient streams are epifaunal substrate/available cover, sediment deposition, channel flow status, channel alteration, bank stability, vegetative protection, and riparian vegetative zone width. Channel sinuosity, pool variability, and pool substrate characterization are assessed in low gradient streams. Frequency of riffles (or bends), velocity/depth regime, and embeddedness are assessed in high gradient streams (Barbour et al. 1999).

7.2 EQUIPMENT

- Chest waders
- Wading boots
- Wading belt
- Throw rope
- First aid kit
- 100 m measuring tape
- Flagging tape
- Permanent marker, pens, pencils
- Depth rod
- Densiometer
- Compass
- Clinometer
- Rangefinder
- Camera with extra memory card
- Calculator
- GPS

- One Tier 1 datasheet per site on water resistant paper (Appendix F)
- One Tier 2 datasheet per site on water resistant paper (Appendix G or H)

7.3 SAMPLE REACH LENGTH, DELINEATION AND POOL/RIFFLE/WOOD COUNTS

1. Preferably, reaches should be at least 100 meters upstream of any road or bridge crossing to minimize influence upon velocity, depth, and overall habitat quality. There should be no major tributaries, springs, municipal or industrial discharges directly to the stream in the study reach. From the given site location/bridge crossing, measure at least 100 m upstream before selecting a location as the starting point for the study reach. The starting point should be easily identifiable such as the base of a pool or riffle. Clearly mark this point with flagging tape. This point will represent the bottom of the reach (Transect 1). If sampling fish, macroinvertebrates or periphyton in conjunction with habitat, take care to walk on the edge of the stream so as not to disturb sample areas. Any deviations from this reach selection method must be documented and preferably discussed with DEQ beforehand.
2. With a measuring tape, measure the wetted width at the bottom of the reach. Move 25 m upstream and measure wetted width again. Continue measuring wetted width at 25 m increments until a total of 5 width measurements are collected.
3. Record the 5 measurements and compute reach length using the following rules:
    - Reach length should be 20 times the average of the 5 wetted width measurements.
    - If the average wetted width is ≤ 7.5 m, use 150 m for the minimum reach length.
    - If the average wetted width is > 25 m, use 500 m for the maximum reach length.
4. Starting at Transect 1, measure upstream, along the thalweg, the length of 1/10 the total calculated reach length.
5. As you proceed, record the number of pools, riffles and large woody debris (See Table 6) that occur throughout the reach.
6. Place flagging tape above the bankfull height at the location and clearly label the piece of tape using a permanent marker with the respective transect number. Underline each number, (e.g. 6, 9,).
7. Repeat this process upstream for a total of 10 equally spaced transects.

7.4 WETTED WIDTH

Record the wetted width at each transect.

1. Starting at either bank, identify the wetted edge of the stream and stretch a tape measure to the wetted edge of the opposite bank, perpendicular to the thalweg. Record measurement on the field sheet.
    - If a transect is dry, record the wetted width as zero
    - If a transect has a dry bar within the wetted channel, individually record the width of each wetted section and the width of the dry bar.

7.5 DEPTH PROFILE

Record a depth profile at each transect.

1. Starting at the left bank of each transect, use the depth rod to measure stream depth at 4 locations: left quarter, center, right quarter, and thalweg.
2. When depth is greater than what can be safely sampled, provide a best estimate of depth using the following steps.
    - Stand in shallower water and extend the depth rod at an angle to reach the thalweg.
    - Determine the angle by resting the clinometer on the upper surface of the rod and reading the angle on the external scale of the clinometer.
    - Record the water level from the depth rod and the rod angle.
    - Calculate thalweg depth by multiplying the height of the water level on depth rod by the sine of the clinometer angle measurement.

7.6 BANK ANGLE

Measure and record bank angle on the left and right banks at each transect.

1. Place the base of the depth rod at bed-meets-bank (Refer to Appendix D for examples of where bed meets bank) and lay the depth rod along the bank, perpendicular to the channel (Figure 8).
2. Measure the bank angle as follows:
    - For a non-undercut bank:
        - > Lay a depth rod along the bank perpendicular to the channel. Banks that rise in a stair-step fashion can be difficult to measure this way; instead, measure the average angle by laying the depth rod along the outer corner of the steps (Figure 8a).
        - > Place a clinometer on top of the depth rod (not on the sides) and record the angle displayed to the nearest degree.
    - For undercut banks:
        - > Lay a depth rod from the bottom of the bank to the tip of overhang, perpendicular to the channel (Figure 8b).
        - > Place a clinometer on top of the depth rod (not on the sides). Subtract the angle displayed from 180 to obtain the angle (e.g. the clinometer reading is 40; 180-40=140°).

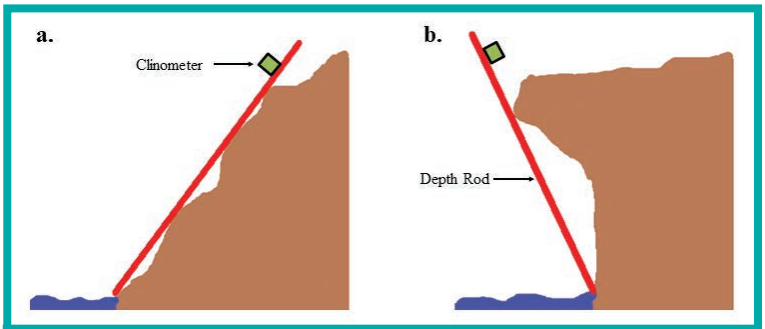


Figure 8. How to measure the (a.) non-undercut or (b.) undercut bank angle.

7.7 SUBSTRATE

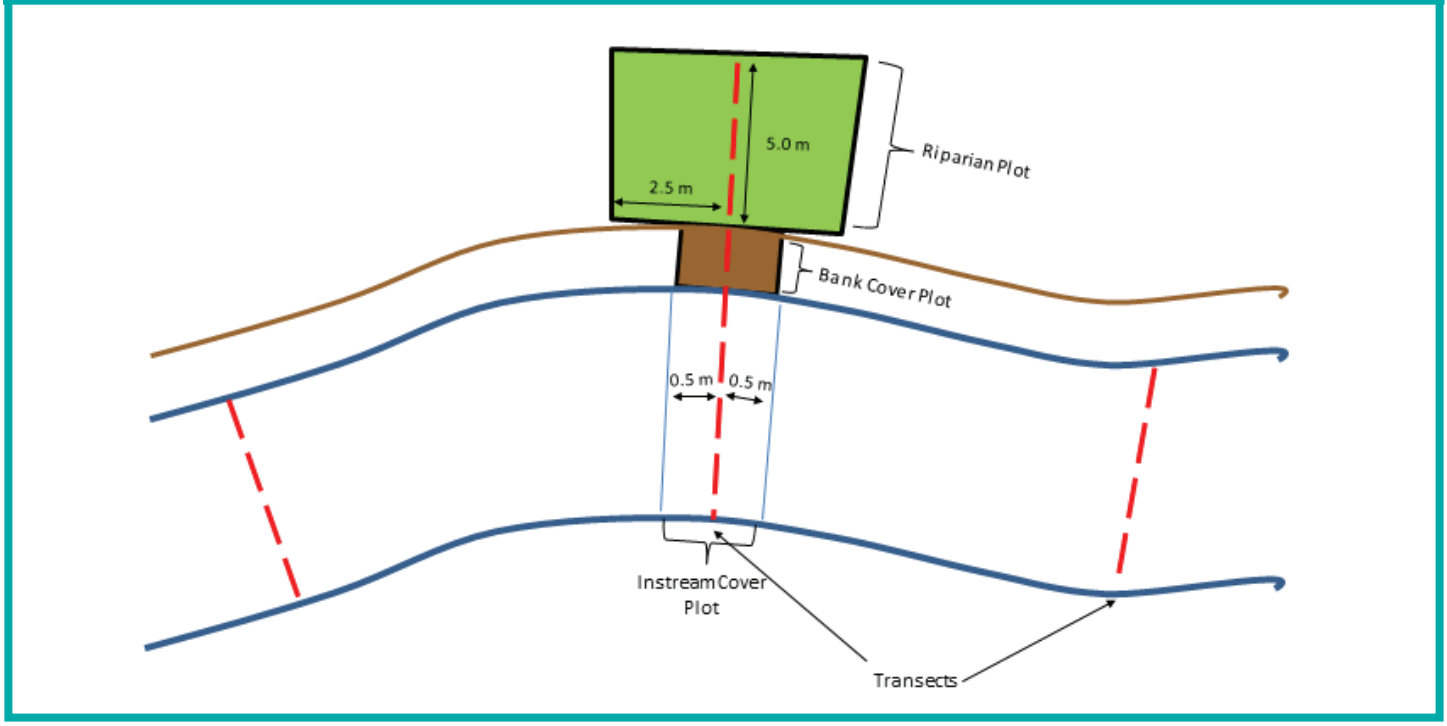
Assess substrate composition at each transect.

1. Record if the transect occurs at a pool, riffle or run.
2. Starting at the left bank of each transect, record substrates at three locations: left quarter, center, and right quarter.
3. Place the end of the depth rod at each location. Without looking at the substrate, reach down and pick up a substrate particle at the base of the depth rod.
4. Classify substrates according to the scale shown below in Table 5 and record on the data sheet.
5. Finally, observe the entire transect from left bank to right bank and record which of the substrate types in Table 5 makes up the majority of the substrate present.

Table 5. Modified Wentworth scale

CLASSIFICATION	PARTICLE SIZE (DIAMETER)
Clay	<0.05 mm fine sediment forming sticky or hard packed stream substrate
Silt	<0.05 mm soft, fine sediment forming loose stream substrate
Sand	0.05-3 mm
Gravel	2-50 mm
Pebble	50-150 mm
Cobble	150-500 mm
Boulder	>500 mm
Other	Root mat, snags, leaf pack, manmade structures that are too dense to reach through to determine underlying substrate

Figure 9. Diagram of plot spaces assessed at each transect and both banks along a stream reach.



7.8 IN-CHANNEL HABITAT

Assess in-channel habitat at each transect.

1. At each transect, define the plot area needed to estimate total in-channel cover.
   - The plot begins at left wetted edge and extends to right wetted edge.
   - The plot width extends parallel to the stream 0.5 m upstream and downstream of the transect line (1 m wide total) (Figure 9).
2. Record the total percentage of the plot covered by complex in-channel habitat. In-channel cover types are listed below in Table 6.
3. Note all in-channel cover types and record approximate percentages of each to the nearest 10%.

Table 6. In channel cover types and definitions

CLASSIFICATION	DEFINITION
Large woody debris	Larger pieces of wood that can influence cover and stream morphology (> 1.5 m in length and 10 cm in diameter at the large end).
Filamentous algae	Long streaming algae that often occur in slow moving waters.
Aquatic macrophytes	Aquatic plants growing in the stream that could provide cover for fish or macroinvertebrates. This includes mosses and wetland grasses.

CLASSIFICATION	DEFINITION
Brush and small woody debris	Smaller wood pieces that primarily affect cover but not morphology.
In-channel live trees or roots	Living portions of trees that are within the channel.
Overhanging vegetation	Tree branches, brush, twigs, or other small debris that is not in the water but is close to the stream (within 1 m of the surface) and provides potential cover.
Boulders or bedrock shelves	Basketball to car-sized rock pieces, or shelved bedrock providing under-shelf refugia space.
Undercut banks	Any bank that has an undercut covering > 5 cm of water surface.
Artificial structures	Structures designed for fish habitat enhancement, as well as in-channel structures that have been discarded (e.g., concrete, asphalt, cars, or tires) or deliberately placed for diversion, impoundment, channel stabilization, or other purposes.

7.9 BANK COVER

Assess bank cover at each transect on both banks.

1. At each transect, define the plot area needed to estimate bank stability (Figure 9).
   - Plot height extends perpendicular to the stream between the water line and the first flat or depositional feature at or above bankfull.
   - Plot width extends parallel to the stream 0.5 m upstream and downstream of the transect flag (1 m wide total).
2. Assess bank coverage within the plot space and designate banks as either:
   - Covered: This applies to banks with at least 50% cover within the plot of the following types or combination thereof (Refer to Appendix E for examples):
     - > Perennial vegetation (including roots)
     - > Cobbles 15 cm or larger
     - > Anchored trees and large woody debris with a diameter >10 cm
     - > Solid bedrock banks
   - Uncovered: This applies to all banks that do not meet the "Covered" criteria (Refer to Appendix E for examples).

7.10 CANOPY COVER

Assess canopy cover at each transect.

1. Place a depth rod at the left wetted edge of the stream.
2. Standing on left bank and facing the center of the stream, rest the densitometer against the side of the depth rod to maintain it at a consistent height of 0.3 m above the surface of the stream.
3. Level the densiometer and hold in front of you so your face is just out of sight.
4. Count and record the number of grid intersection points (17 total) that are covered by any form of vegetation or anything else that creates shade such as an undercut bank (Figure 10).
5. Repeat densiometer measurements three more times in the following locations:
   - In the center of the stream facing left bank
   - In the center of the stream facing right bank
   - On the right bank facing the stream center
6. You will end up with four total measurements: left bank, center left, center right, and right bank.

7.11 RIPARIAN COVER

Assess riparian cover at each transect on each bank.

1. Stand at the center of the stream facing the bank and define the plot area needed to estimate total riparian cover (Figure 9). If bank height or dense vegetation impairs plot visibility, you may move to an area of the stream that allows for better visualization of the plot area.
   - The plot begins at the first flat depositional surface and extends to 5 m outside of the stream.
   - The plot width extends parallel to the stream 2.5 m upstream and downstream of the transect line (5 m wide total).
2. Within this 5 m X 5 m area, conceptually divide the riparian vegetation into 3 layers:
   - Tree layer (>5 m high)
   - Shrub layer (0.5 to 5 m high)
   - Ground cover layer (<0.5 m high)
3. Estimate the percent aerial cover that each subcategory would make up of the total cover within the respective canopy, understory, or ground cover layer. The aerial cover is the amount of shadow that would be cast by a particular sub-category if the sun were directly over the plot area.
4. Record percent aerial cover estimates for each of the three layers.

Make a note of any man-made structures that affect riparian cover estimates or quality such as buildings, roads, fences, trash piles, etc.

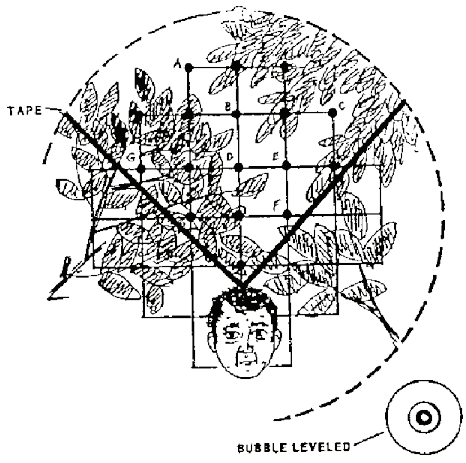


Figure 10. Modified spherical densitometer. In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and face reflected at the apex of the "V" (USEPA 2019).

7.12 PHOTOS

Take photos at each transect.

1. At each site, first photograph the top of the data sheet that includes the Site ID.
2. At each transect, take a picture of the transect number. Stand in the center of the stream, take pictures of the upstream, downstream, left bank, and right bank for a total of four (4) photos per transect. Always take transect photos in this order. Check the photos box on the habitat datasheet when all 4 photos have been completed at each transect.
3. Take additional photos of any other noteworthy features including problematic features for protocol implementation (e.g. waterfalls) and manmade features that might influence the stream (e.g. impoundments, large pieces of waste, etc.).

7.13 TIER 2 ASSESSMENT

1. Determine if the stream is low gradient or high gradient.
2. Walk the entire reach paying attention to the nine habitat parameters listed on the RBP Habitat Assessment Field Form (Appendix G & H).
3. Record a score for each of the nine habitat parameters (0=Poor – 20=Optimal) that best characterizes the stream reach as a whole.

8. Fish

8.1 OVERVIEW

Biological communities reflect ecological integrity and provide insight into pollutant stressors in stream ecosystems. Fish sampling is a useful technique for assessing the ecological condition of aquatic ecosystems. This section describes standard operating procedures for fish collection in an effort to effectively and consistently characterize fish communities in aquatic habitats, including procedures for tote barge electrofishing, backpack electrofishing, and seining.

All fish sampling should be conducted under a scientific collection permit from the Arkansas Game and Fish Commission (AGFC). At least one permit holder and the permit should be present at the time of sampling. DEQ should carry this permit when conducting fish sampling. Additionally, adherence to the guidelines established by the United States Fish and Wildlife Service should be attained (**https://www.fws.gov/permits/ltr/ltr.html**).

8.2 ECOLOGICAL CONSIDERATIONS

Presence of federally threatened or endangered species or Species of Greatest Conservation Need (SGCN), as listed in the most recent "Arkansas Wildlife Action Plan", www.wildlifearkansas.com) may alter preferred sampling methods. When these species are likely or known to exist within the sample area, take caution to reduce specimen loss and research any considerations or permission needed through the US Fish and Wildlife Service (USFWS) (**https://www.fws.gov/permits/ltr/ltr.html**). If necessary, do not employ electrofishing techniques and only sample using nets/seines.

In the event sampling overlaps in known areas of federally listed threatened or endangered species or SGCN, adhere to USFWS or other relevant established sampling protocols for each species (**https://www.fws.gov/permits/ltr/ltr.html**).

Report any federally or state listed, threatened, or endangered species collected during sampling to the USFWS and the AGFC. Return these species to the waterbody as quickly as possible outside of the electrical field. If mortality occurs, they need to be retained and a report sent to the AGFC and/or the USFWS.

8.3 EQUIPMENT

General

- AGFC scientific collection permit
- Buckets
- Clipboards
- Collection containers
- Data sheets printed on Rite in the Rain® paper
- Camera for photo vouchers
- Dipnets
- Block net (seine)
- Formalin
- Plastic graduated cylinder
- Polarized sunglasses (recommended)
- Rite in the Rain® pen
- Pencil
- Permanent markers
- Collection container labels
- Non-breathable waders
- Wader belts (recommended)
- Lineman gloves
- Wading boots
- Flow-through fish nets/holding nets

Tote barge electrofishing

- Tote barge electrofisher (non-metal boat used in wadeable streams)
- Electrofishing control unit (5.0 Generated Powered Pulsator (GPP) Electrofisher) or equivalent equipment
- Generator
- Electrodes (Anode(s))
- Livewell (large plastic tub)
- Fuel

Backpack electrofishing

- Model Smith-Root LR-20B and/or LR-24 or equivalent equipment
- Backpack electrofisher batteries and adaptor (if necessary)
- Electrodes (Cathode and Anode(s))
- Battery charger

Seining

- 6 ft tall, 15 – 30 ft seine with 3/16 or 1/4 inch mesh or equivalent equipment
- AGFC scientific collection permit
- Buckets
- Clipboards
- Collection containers
- Data sheets printed on Rite in the Rain® paper
- Camera for photo vouchers
- Dipnets
- Block net (seine)
- Formalin
- Plastic graduated cylinder
- Polarized sunglasses (recommended)
- Rite in the Rain® pen
- Pencil
- Permanent markers

8.4 GENERAL FISH SAMPLING PROCEDURES

DEQ utilizes either of two electrofishing methods (backpack or tote barge) for collecting fishes in wadeable streams, contingent upon stream size (width and depth), instream flow, and known presence of federally threatened or endangered species or SGCN. In addition to backpack and barge electrofishing, DEQ utilizes seining whenever possible to capture a broader range of species. Electrofishing and seining methods are described in this document following general sampling procedures.

- Sampling is generally conducted from June through early October when streams are at or near base flow.
- Number of crew members required is determined on a case by case basis depending upon equipment used and stream size.
- Crew members are required to wear non-breathable waders and lineman gloves when electrofishing in wadeable streams.
- Polarized sunglasses are recommended to maximize capture efficiency.
- Select and delineate the reach using the methodology from Section 7.3. Sample reaches are selected to represent local instream characteristics and all geomorphic and instream microhabitat features within the sampling reach that will be sampled. The starting point should be easily identifiable such as the base of a pool or riffle.
- Determine mean wetted width (MWW) from five transect widths approximately 25 meters apart.
- Determine sampling reach length by multiplying MWW by 20. Use 150 meters as a minimum when the mean wetted width in less than 7.5 meters.
- Use the MWW to determine the type of sampling gear to use.
  - One backpack should be used in streams with an average width of less than 5 meters.
  - Two backpacks should be used in streams with an average width between 5 and 10 meters.
  - A barge electrofisher should be used in streams with an average stream width that is greater than 10 meters and enough depth to float the tote barge. Many times there will not be enough depth to float a tote barge in the riffle habitat of the reach, but a tote barge is still needed for the pools. When this occurs, sample the reach as described in Section 8.6.

- Consider the depth of the pools, flow velocity and instream habitat. On occasion a smaller stream may need two backpacks to effectively sample large deep pools. Two or three backpacks may be preferred when there is not a way to put a barge electrofisher in the water safely or if the instream habitat is too dense to move the unit upstream.

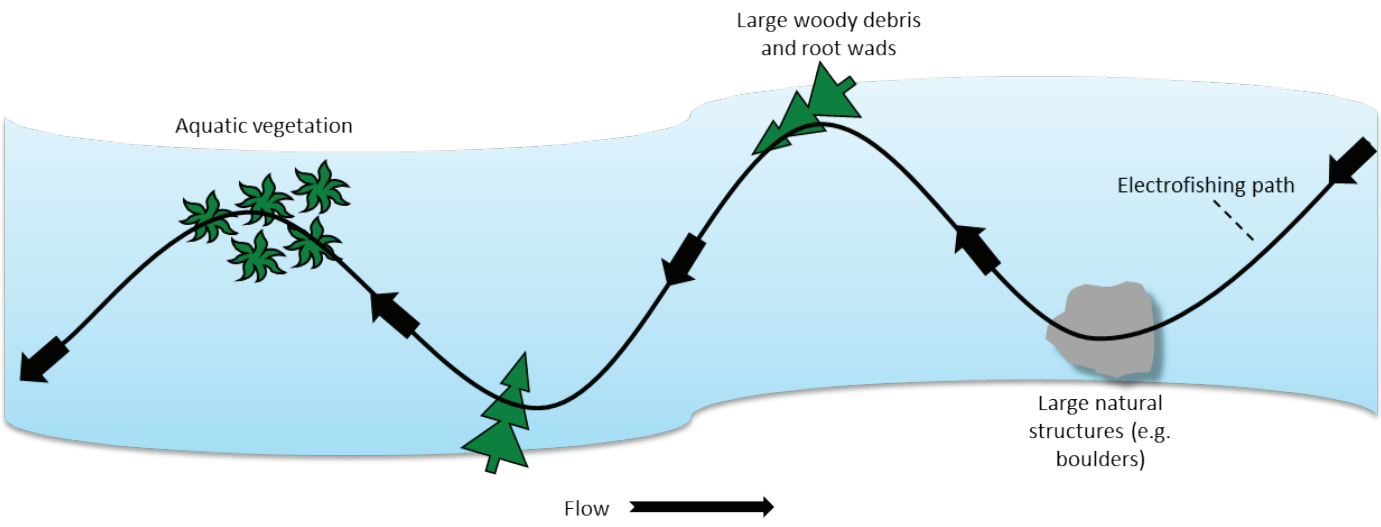
- Electrofishing may cease at the end of the delineated reach if no new species have been encountered in the last 25 meters. Otherwise, continue sampling for another 25 meters until no new species have been encountered.
- Conduct three seine hauls approximately 10 meters in length. If a new species is encountered in the last seine haul, conduct additional hauls until no new species are encountered.
- After all sampling activities have ceased, enumerate and release sensitive species (e.g., Catostomidae, Percidae) that can be easily identified. Additionally, fish such as stonerollers (Campostoma spp.) and game species (e.g., Centrarchidae) that can be easily identified may also be enumerated and released in the field to reduce biomass being taken from each site.
- Collect voucher specimens for rare and uncommon species to a particular geographic range. If there is any question about the proper identification of any specimen, return it to the laboratory for identification.
- Do not enumerate or list voucher species on the field sheet.
- Be sure to indicate on the field sheet that vouchers specimens were kept or no voucher specimens were kept.
- Place specimens being taken back to the laboratory in a container filled with 10% formalin. Samples should loosely fill the container so that all fish are completely covered with the formalin solution and placed so specimens will not be bent or preserved in a misshapen manner that makes identification difficult. For larger specimens, make a slit in the abdominal cavity to ensure proper preservation. The samples should then be stored in a cooler with ice until delivered to the laboratory.
- Do not store electrofished and seine hauled vouchers in the same container.
- Ensure containers are stored upright and in a way to prevent rolling or spilling.
- Write labels for fish sample jars or buckets in pencil on Rite in the Rain® paper. If more than one jar is required for a sample site, label jars with the following format: ## of ##. Write and place one label inside each sample container and write a second label on the side of each sample bucket. Do not label lids as they can be easily misapplied to sample containers. Place the following information on each label:
  - Sample site ID
  - Location/stream name
  - Date
  - Collector's initials
  - Project
  - Sampling method used
- Document any additional information in field datasheets with the same information above.

8.5 BACKPACK ELECTROFISHING

- Sample entire reach using one pass, moving from downstream to upstream, to minimize streambed disturbance which could decrease visibility and reduce catch. Use a zig-zag pattern (Figure 11) to sample as many different types of habitat as possible. The reach should not be shocked continuously; instead, use a pulse ambush technique focusing on fish habitat.

- Three crew members are usually required for each sampling event per backpack electrofisher used.
  - One crew member dedicated to carry and operate backpack electrofisher.
  - Two netters to flank each backpack.
  - Utilize additional netters if necessary and personnel are available.
  - Additional personnel may assist with transporting livewell of collected fish.
  - Fish in livewell should be monitored and water changed/refreshed periodically to reduce fish stress and mortality.
- In smaller streams, the electrofisher operator should move back and forth to sample both stream banks. However, it is important to sample mid-stream to collect those species that prefer deeper water.
- In larger streams that require two backpack electrofishers, the electrofisher operators should be along the same cross-section within the stream covering the area from bank-to-bank. When the width of the stream cannot be fully covered from bank-to-bank, one electrofisher should be used near one stream bank, sampling available fish habitat, while the other electrofisher is located mid-stream and upstream of the near bank electrofisher. This will help to funnel any deeper water species toward the downstream electrofisher and enhance the capture of these species. Large woody debris and other optimal fish habitat should be surrounded to limit the ability of fish to escape the electric field and avoid capture. This procedure should be repeated on the opposite bank if suitable habitat is present.
- Always sample riffle habitat upstream to downstream when possible. Do not disturb the riffle before shocking. If approaching from downstream, walk upstream along the bank to the head of the riffle with electrodes. Dependent on the riffle length, the riffle may have to be divided into multiple sections for sampling. Additional personnel will place a block net (seine or dip nets placed next to one another for small riffles) at the base of riffle. Once the block net is set, electrofishing crewmembers will simultaneously electrofish and kick substrate to dislodge fishes, taking care to move downstream at a pace slow enough for stunned fishes to make it to the block net before crewmembers arrive at the seine. The pace is generally just less than the flow velocity in the riffle. Allow the sampled volume of water to flow through the seine before lifting. Lift the seine out of the water for retrieval of stunned fishes by quickly lifting the lead line in a single motion to prevent the escape of fish.
- Retain fishes in buckets/flow-through containers while electrofishing. One netter generally will carry a bucket to deposit netted fish. These buckets should be emptied frequently into flow-through holding containers placed along the reach to reduce fish stress, especially when biomass is high. Secure flow-through holding containers so that fish cannot escape (i.e. rocks placed in bottom, tied to a tree) and place out of the area being electrofished so that additional electric shock will be avoided, preferably in a shaded area with water depth enough to submerge about half of the flow-through container. If holding fish in non flow-through buckets, frequently replenish fresh stream water and/or aerate to not cause stress to fishes due to elevated temperatures or decreased oxygen or ensure low fish density.

Figure 11. Single pass sampling technique for wadeable streams. Zig-zag electrofishing path encounters all available microhabitats within sampling reach. Figure modified from Petersen et al. (2008).



8.6 TOTE BARGE ELECTROFISHING

- Use a tote barge electrofisher if the site to be sampled maintains a minimum depth that does not require the barge to be dragged over an excessive distance.
- Check the battery in the GPP that powers the counter. Replace if needed.
- Six to eight crew members are required for each sampling event requiring tote barge electrofishing.
  - One crew member dedicated to push the barge and operate the electrofisher.
  - Two or three probe operators with capture nets.
  - One netter to flank each probe operator.
  - One netter following behind the barge.
  - Utilize additional netters if necessary and personnel are available.
  - Additional personnel may assist with pushing barge and processing collected fish.
- Fish in livewell should be monitored and water changed/refreshed periodically to reduce fish stress and mortality.
- Additional crew members may be necessary to transport the specimens from the netters to the livewell in the tote barge. These crew members also help to provide safety and backup to the rest of the crew.
- Sample entire site using one pass, moving (ideally) from downstream to upstream, to minimize streambed disturbance which could decrease visibility and reduce catch, using a zig-zag pattern (Figure 11).

- In smaller streams, electrode operators can work independently working both stream banks simultaneously. However, it is important to sample mid-stream to collect those species that prefer deeper water. The barge should be located at an equal distance between the two electrodes if possible. However, the electrode operators should work together to surround large woody debris and other optimal instream cover to limit the ability of fish to escape the electric field and not be captured. In most instances it is better to halt shocking, surround the habitat then resume shocking using a pulse ambush technique.
- In larger streams, the electrode operators should work together when the width of the stream cannot be fully covered from bank-to-bank. One electrode should be used near one stream bank sampling available habitat. The other electrode should be located mid-stream and upstream of the near bank electrode. The barge should be located equidistance between the two electrodes as possible. This will help to funnel any deeper water species toward the downstream electrode and enhance the capture of these species. This procedure should be repeated on the opposite bank if suitable habitat is present. Large woody debris and other optimal fish habitat should be surrounded to limit the ability of fish to escape the electric field and avoid capture. In some instances it is better to halt shocking, surround the habitat then resume shocking using a pulse ambush technique.
- Always sample riffle habitat upstream to downstream when possible. Do not disturb riffle before shocking. If approaching from downstream, two to three crew members will walk upstream along the bank to head of riffle with electrodes while leaving the barge downstream of the blocknet. Dependent on the riffle length, the riffle may have to be divided into multiple sections for sampling. Additional personnel places a block net (seine or dip nets placed next to one another for small riffles) at the base of riffle. Once the block net is set, electrofishing crew members will simultaneously electrofish and kick substrate to dislodge fishes, taking care to move downstream at a pace slow enough for stunned fishes to make it to the block net before crewmembers arrive to the seine. The pace is generally just less than the flow velocity in the riffle. Allow the sampled volume of water to flow through the seine before lifting. Lift seine out of water for retrieval of stunned fishes by quickly lifting the lead line in a single motion to prevent the escape of fish.
- Retain fishes in a livewell while electrofishing. If the livewell does not contain a bubbler, frequently replenish fresh stream water to not cause stress to fishes due to elevated temperatures or decreased oxygen.

8.7 SEINING

- During the single pass through electrofishing, samplers should note areas of the streambed with few snags or obstructions. These areas will be used for seine hauls after electrofishing.
- Pull the seine with two crew members.
- Follow behind the seine with a third crew member to free the net from snags.
- Seining is a required technique in addition to electrofishing in all streams.
- Seining is generally conducted after electrofishing to prevent the disturbance of sediment and decreased visibility for electrofishing.
- Seining is typically most effective in runs or pools and conducted in a downstream direction slightly faster than current velocity.
- The seine should be pulled in a manner that allows the lead line to remain on the bottom to prevent fish from escaping under the net.

- When seining, one crew member should slow to allow the other crew member to swing around and move toward the take out area.
- The crew member closer to the bank should be slightly behind the other to form a slight "U" shape. This will help to trap the fish between the seine and the stream bank.
- The seine should be beached on a gently sloping bank if possible with the lead line remaining on the bottom until it is out of the water. If no such bank is available, the lead line should be pulled to the bank and lifted in a single motion to prevent the escape of fish. This may require assistance from additional crew members.
- Three seine hauls should be conducted on each reach for approximately 10 meters per haul.
- If the seine gets caught on woody debris and cannot be freed easily without fish escaping, it may be considered an ineffective seine haul and should be repeated. If a seine haul is determined to be ineffective, release fish, and repeat the haul.

8.8 TAXONOMIC IDENTIFICATION AND VERIFICATION

- Since it is preferable that fish identification takes place in the field to prevent unnecessary take of fishes, staff should be familiarized with fishes of Arkansas prior to field sampling. Staff should review what species are expected to occur in the drainage basin they are sampling.
- Robison and Buchanan (1988 & 2020) and Pflieger (1997) are the primary keys for fish identification. Supplemental keys as well as new editions of currently used keys must be approved by the senior staff before use in fish identification.
- Identify and record fishes on Fish Collection Identification Forms (Appendix I). Note number of specimens with deformities, erosions, lesions, or tumors (DELT) and note number of specimens with blackspot on the field form. Include a note on the form to indicate that either vouchers were taken, or no vouchers were taken. In addition, if the entire collection was vouchered, this information should be recorded on the field form.
- Regardless of previous experience, identifications by all new staff members are reviewed by senior staff until they satisfactorily complete taxonomic QC. Once a staff member has satisfactorily completed their QC, 10% of their identified samples will be randomly checked by senior staff who will identify all fishes in the sample without consulting the original taxonomist's list, and the two staff members will review and discuss discrepancies. If a consensus cannot be reached by the original taxonomist and senior staff, the specimen will be sent to an independent third party for verification.
- Staff should work side-by-side when identifying and enumerating specimens in the field. Staff with limited fish identification experience should only identify and enumerate the more easily identifiable species, centrarchids, stonerollers, and game fishes after initial training and verification by senior staff. Smaller specimens and species that are more difficult to field-identify, minnows and darters with similar characteristics, and those with only a minimal number of specimens, should be vouchered.
- Take a photo voucher for each species that is identified in the field. Additionally, use photo vouchers for fish too large for collection containers or for threatened or endangered species. Take notes on the characteristics of the specimens to aid in identification. Ensure while photo vouchering that all characteristics are captured that allow for the identification or verification of the species. Include in the voucher photo a small label with information identifying the location, date, sample type, and scientific name, if known. Put a note on the field sheet that a photo voucher was taken.

- Leave fishes taken to the laboratory for identification in 10% formalin for at least 5-7 days for preservation and then soak in water for three days, changing the water each day. Transfer specimens to 50% isopropyl or 75% percent ethanol before identification.

9. Macroinvertebrates

9.1 OVERVIEW

Many aquatic macroinvertebrates require specific physical and chemical conditions for growth, feeding, and reproduction. As such, changes in occurrence, taxa richness, abundance, and/or behavior of these organisms can indicate harmful alterations to natural conditions. Because of this differential sensitivity to biotic and abiotic factors and their abundance and relative ease of collection, invertebrate sampling is a useful technique for assessing the ecological condition of aquatic ecosystems. This section describes standard operating procedures for macroinvertebrate collection in an effort to effectively and consistently characterize macroinvertebrate communities in aquatic habitats, including procedures for single habitat and multi-habitat macroinvertebrate collection methods.

9.2 ECOLOGICAL CONSIDERATIONS

Presence of federally threatened or endangered species, SGCN, or endemic species may alter preferred sampling methods. When these species are likely or known to exist within the sample area, take caution to reduce specimen loss.

9.3 EQUIPMENT

Macroinvertebrate field sampling supplies:

- Pencil or Rite in the Rain® pen (only use pencil or Rite in the Rain® pens on labels that will be stored in ethanol)
- Rite in the Rain® Labels
- D-frame dip net with 500-micron mesh (plus a spare)
- Sieve bucket with 500-micron mesh bottom
- Stop watch
- Wash bottle/squirt bottle
- Scoopula
- 95% ethanol
- Plastic sample containers
- Forceps
- Multi tool (for net repair)
- Toothbrush or other small soft bristled brush

9.4 PRE-SAMPLING CONSIDERATIONS

Select study reaches that represent local instream characteristics. Reaches should be wadeable and at least 100 meters upstream or downstream (preferably upstream) of the area of influence at any road or bridge crossing to minimize effects on velocity, depth, and overall habitat quality. Unless study design necessitates, the study reach should not include direct inputs from major tributaries, springs, or municipal or industrial discharges. If summer fish samples were collected at the site, utilize the same reach used for fish sampling.

Allow a minimum of two weeks for recolonization following major scouring events prior to collecting macroinvertebrate samples. Scouring events are determined by best professional judgment using flow, local weather reports, or other local resources. Avoid sampling during excessive deviations from base flow.

Select the appropriate method(s) for collecting a benthic macroinvertebrate sample by walking the reach to determine available habitat. DEQ generally uses the single habitat method for collecting macroinvertebrates in high gradient, wadeable streams with abundant clearly defined riffles or runs with cobble/gravel/pebble substrates. Use the multi-habitat sampling method in low gradient streams without well-defined riffles or runs with cobble/gravel/pebble substrate.

- **Single Habitat Method:**
  20 riffle kicks
- **Multi Habitat Method:**
  20 kick/jabs in snags, root wads, and leaf packs

As a general rule, conduct benthic macroinvertebrate sampling during fall (October through early December). Conditions generally represent non-summer low flow conditions, increased leaf packs, and cooler water temperatures.

9.5 MACROINVERTEBRATE FIELD SAMPLING PROCEDURES

Collect macroinvertebrate samples according to the following specific protocols for the applicable sampling method.

If measuring in situ water quality parameters such as temperature, pH, dissolved oxygen, flow etc., complete this task prior to collecting macroinvertebrate samples. Take care to minimize disturbance of the sample reach. If you must walk through the stream, do so just downstream of the area to be sampled for macroinvertebrates. Sample reach for macroinvertebrates from downstream to upstream.

9.5.1 SINGLE HABITAT METHOD

Complete 20 substrate kicks when riffle/run habitat with cobble/gravel/pebble substrate is available to sample. Cobble/gravel/pebble is the dominant substrate in riffle and run habitats for most Arkansas high gradient streams (typically in Ozark Highlands, Boston Mountains, and Ouachita Mountains ecoregions). Macroinvertebrates inhabit the surfaces of these substrate particles as well as the interstitial spaces between the particles. Diversity and abundance are usually highest in cobble substrate habitats and this approach should provide a representative sample (Barbour et al. 1999).

1. A "kick" is a stationary sampling technique accomplished by positioning the net on the stream bed and disturbing the substrate for 15 seconds. Disturb a 0.3 m^2 (the width of the net) area of substrate upstream of the net. Using the toe or heel of the boot, dislodge and disturb the upper layer of substrate down to the underlying bed, being careful not to kick large substrate material into the collection net or out of the sample area. Use water current and boot to guide dislodged invertebrates into the net. Take caution not to grind substrate with boots, damaging invertebrates (Barbour et al. 1999). The sampler will use the kick technique on substrates such as cobble/gravel/pebble, within riffle or run hydraulic units. The sampler should conduct kicks such that dislodged organisms are directed toward the center of the frame and carried into the mouth of the net. In addition, the sampler should apply the same effort to each kick.

2. Select areas within the reach that contain riffles with cobble/gravel/pebble substrate. If multiple riffles exist within the reach, distribute total kicks among at least two representative riffles.
3. Start at the downstream corner of the riffle and kick the substrate at multiple locations, following a zig zag path upstream through the riffle and attempting to sample as many microhabitats in the riffle as possible.
4. Allot about 15 seconds of effort to each kick for a total of five minutes kicking effort over six square meters of sampling area.
5. As often as necessary, empty contents of net into sieve bucket with 500-micron mesh bottom to retain organisms >500 microns in size. Dislodge stray organisms and debris by shaking the net and rinsing with water into the sieve bucket. If needed, remove organisms and debris from the net with forceps or scoopula and place into the sieve bucket. Empty the net often to ensure no organisms are lost during collection and to avoid clogging net mesh and diverting flow away from the net mouth.
6. If samples are collected from multiple riffle/runs, combine contents of all samples into the sieve bucket.
7. Once all 20 kicks have been complete, return to the bank to process the sample. Brush (with your hand or soft bristled brush) and rinse large debris (rocks, leaves, and sticks) of any attached organisms and discard debris outside of the sample area, using caution in order to prevent damaging specimens.
8. Rinse sieve bucket contents into a plastic wide-mouth container and preserve with 95% ethanol. Samples should loosely fill a container 75% full or less with at least five centimeters (two inches) of free ethanol above the sample.
9. Complete labels for interior and exterior of macroinvertebrate sample container. If a sample contains a large amount of algae, leaves, or other material that will decay rapidly, it may be necessary to drain the liquid from the sample and add fresh ethanol within two days of initial collection and in subsequent months in order to maintain adequate ethanol concentrations and preserve the morphological integrity of the invertebrates and aid in taxonomic identification.

9.5.2 MULTI-HABITAT METHOD

The multi-habitat sampling method consists of performing 20 kicks/jabs in productive habitat types described below. Habitat types are ranked from most productive to least productive (Table 8). Use the multi-habitat method when not enough cobble/gravel/pebble bottom riffle/run habitats exist to complete the single habitat method within a representative reach (e.g., in low-gradient streams characteristic of the Mississippi Alluvial Plain, South Central Plain and Arkansas Valley ecoregions). Sampling techniques for individual habitat types are described below.

Table 7. Sampling techniques for non-riffle and riffle habitats. Non-riffle habitat technique modified from Georgia DNR 2007.

IF NO RIFFLES HABITAT IS PRESENT:	IF RIFFLE HABITAT EXISTS:
• Conduct sampling from downstream to upstream by kicking/jabbing/sweeping the D-frame net into productive habitats at 20 different locations. • Collect specified number of samples from habitats 1-4 in Table 8. • If there are missing habitats in the stream, reallocate jabs equally among available habitat types using the priority list in Table 9. For example, if no leaf packs are available, use the three kicks allotted to leaf packs for one woody debris/snag, one undercut bank, and one soft sediment kick. • If vegetation is present add three kicks in addition to the 20 kicks for habitat priorities 1-4 for a possible total of 23 kicks.	• Collect as many kicks as possible from the riffle(s) for a maximum of 20 kicks. Ensure no kicks overlap with previously kicked areas. • For each kick, follow kick procedure from the single habitat methodology. • If all 20 kicks cannot be completed within available riffle habitat, refer to Table 9 for distribution of remaining kicks among habitat types. • If vegetation is present, add three kicks in addition to the 20 kicks for habitat priorities 1-4 for a possible total of 23 kicks.

- A single jab/sweep consists of forcefully thrusting the net into a productive habitat for a linear distance of 0.5-meter (MACS, 1997).
- A kick consists of "kicking" or disturbing a 0.3 m^2 area of the substrate upstream of a stationary D-frame net and sweeping the net through the water column to capture loose invertebrates.
- Snags and woody debris which have been submerged in the stream for an extended period of time can be sampled by jabbing small to medium piles of sticks and branches. Snag habitat may be kicked first to help dislodge organisms, but only after placing the net downstream of the snag. Large pieces of woody debris can be rubbed clean into the net using hands or soft bristle brushes.
- Sample root wads and undercut banks by jabbing into the habitat and then sweeping the net through the water column to capture dislodged invertebrates. Bank habitat can be kicked first to help dislodge organisms, but only after placing the net downstream.
- Sample leaf packs by looking for accumulations of deciduous leaf material on snags, root wads, or large substrate particles. One large handful of well-conditioned (i.e., partially broken down) leaves consist of one sample. Avoid new leaf fall that has not become conditioned if possible (Georgia DNR, 2007).
- Sand and fine sediments require caution to avoid overloading sample with debris and sediment. Disturb sediment with boot to dislodge macroinvertebrates into the water column. Sweep net back and forth to capture macroinvertebrates dislodged and floating within the water column.

- Sample aquatic vegetation by jabbing and sweeping the net within the vegetation. In deep water, the net should be swept through standing vegetation. In shallow water, jab and sweep net through the vegetation making sure to sample the roots but not dig into the substrate. This habitat is generally not present. If present, add a maximum of three jabs to total number (i.e., total number of jabs could potentially be 23) (Georgia DNR 2007).

As habitats are sampled:

1. Record the number of jabs/kicks/sweeps in each habitat on field data sheets (Appendix J). Use a tally counter if necessary.
2. Empty contents of net into sieve bucket with 500-micron mesh bottom to retain organisms >500 microns in size. Dislodge stray organisms and debris by shaking the net and rinsing with water into the sieve bucket. If needed, remove organisms and debris from the net with forceps or scoopula and place into the sieve bucket.
3. Brush (with your hand or soft bristled brush) and rinse large debris (rocks, leaves, and sticks) of any attached organisms and discard debris outside of the sample area, using caution in order to prevent damaging specimens.
4. Rinse sieve bucket contents into a plastic wide-mouth container and preserve with 95% ethanol. Samples should loosely fill a container 75% full or less with at least five centimeters (two inches) of free ethanol above the sample.
5. Complete labels for interior and exterior of macroinvertebrate sample container. If a sample contains a large amount of algae or other material that will decay rapidly, it may be necessary to occasionally drain the liquid from the sample and add fresh ethanol in order to help preserve the morphological integrity of the invertebrates and aid in taxonomic identification.

Table 8. Prioritized list of habitat types. Modified from Georgia DNR 2007.

Priority	Habitat Type	Number of Samples
1	Woody Debris/Snags	8
2	Root Wads/Undercut Banks	6
3	Leaf packs	3
4	Soft Sediment/Sand	3
	Vegetation	3

Table 9. Sample distribution guide for use with multi-habitat sampling method in low gradient streams with cobble/gravel/pebble riffle habitat.

RIFFLE KICKS	WOODY DEBRIS/SNAGS	ROOT WADS/ UNDERCUT BANKS	LEAF PACKS	SOFT SEDIMENT/SAND	TOTAL
0	8	6	3	3	20
1	8	6	3	2	20
2	8	6	2	2	20
3	8	5	2	2	20
4	7	5	2	2	20
5	6	5	2	2	20
6	6	5	2	1	20
7	6	5	1	1	20
8	6	4	1	1	20
9	5	4	1	1	20
10	5	4	1		20
11	5	3	1		20
12	5	2	1		20
13	5	2			20
14	5	1			20
15	5				20
16	4				20
17	3				20
18	2				20
19	1				20

9.6 FIELD NOTES AND SAMPLE LABELING

Complete appropriate field forms and attach to any associated field notes, if applicable.

Annotate and record supplementary information in field notes. Appropriate information for field notes includes comments on weather, instream flow, recent channel or riparian modifications, or other features that could affect the sample. It is often helpful to sketch the site.

Write labels for sample jars in pencil on Rite in the Rain® paper. Write and place one label inside each sample container and write a second label on the side of each sample bucket. Ensure ethanol does not erase the label on the bucket. Do not label lids as they can be easily misapplied to sample containers. Both labels will contain the following information:

- Sample site ID
- Location/Stream
- Date and time
- Collectors' initials
- Sampling method used
- ## of ## (If only one sample container is needed, write "1 of 1.")

Complete applicable fields on Macroinvertebrate Sampling Datasheet (Appendix J) or in a project-specific field notebook. Parameters to be measured may include but are not limited to:

- Reach length (Section 7.3)
- Substrate type (Section 7.7, 9.5.2)
- Flow (Section 6)

10. Definitions

Aliquot	to portion a larger whole, especially a sample taken for chemical analysis
Aliquot Container	specified bottle or other apparatus used when aliquoting
Bankfull	the lowest bank height at which the stream over tops its banks and spills out onto the active floodplain
Bed-Meets-Bank	the location where the streambed begins to become constrained by its streambanks, often characterized by a dramatic change in slope or particle size
Calibration	the process of testing an instrument's accuracy by using a known measurement
Collection vessel	bucket with plastic handle, pitcher, or other non-metallic sampling device used to collect water
Cross-section	(or transect) a vertical plane oriented perpendicular to the stream flow direction that extends from bank to bank and from the channel substrate to the water surface
D-frame net	net with a "D" shaped metal frame, 500 micron mesh bottom, and canvas sides specifically designed to sample macroinvertebrates
Discharge	the volume of water moving down a stream per unit of time, commonly expressed in cubic feet per second
Electrofisher	fishing that employs a direct electric current from the anode to the cathode that causes galvanotaxis in fish and temporarily immobilizes fish for easy capture
Field Blank	a sample of analyte-free water poured into a sample container in the field, preserved, and shipped to the laboratory with field samples
Flow meter	instrument for measuring flow rate of a liquid
High gradient stream	streams typical of the Boston Mountains, Ouachita Mountains, and Ozark Highlands, generally characterized by moderate to high gradients and stream flows, larger cobble or boulder substrate, and a mix of pools, riffles, and runs
Interval	(sometimes referred to as stations, sections, cell, segment) distance between 2 adjacent vertical measurements along the transect/cross-section
Left bank	looking downstream, the bank to the left of the thalweg
Low gradient stream	streams typical of the South Central Plains, the Mississippi Alluvial Plains, and parts of the Arkansas Valley, generally characterized by little elevation change and low flow velocity, with smaller substrate materials and few to no riffles
Mean Wetted Width (MWW)	the average wetted width of the waterbody determined by measuring wetted widths at five locations approximately 25 meters apart that are typical for the waterbody

Pools	bowl-shaped areas of the stream, usually deep compared to other parts of the channel and containing still or low velocity water with a smooth glassy surface
Riffle	a shallow landform in a flowing channel typically exhibiting an area of higher flow than the surrounding hydrology, often marked by a section of agitated water where rocks break the surface
Right bank	looking downstream, the bank to the right of the thalweg
Run	area of consistent depth where water is noticeably flowing and moderately deep. Generally between a riffle (shallow, fast flow) and pool (deep, low flow)
Sample container	smaller vessels that water is aliquoted into from a collection vessel
Sampling site	a location in which water samples and/or in situ data are collected
Seine	a net of varying lengths and mesh sizes fixed between two brails for capturing smaller species and juveniles
Sestonic	suspended within the water column
Species of Greatest Conservation Need (SGCN)	a list of plant and animal species that occur in Arkansas that is listed as either globally and/or state threatened or endangered. The list is maintained in the Arkansas Wildlife Action Plan and can be downloaded at **https://www.agfc.com/en/wildlife-management/awap/the-plan**
Streamflow (or discharge)	volumetric rate of flow of water (volume per unit time) in an open channel
Type I DI Water	ultrapure deionized water, as standardized by the American Society for Testing and Materials (ASTM)
Unexploded Ordnance (UXO)	explosive weapons (bombs, bullets, shells, grenades, land mines, naval mines, etc.) that did not explode when they were employed and still pose a risk of detonation
USGS top setting wading rod	a hexagonal rod used to measure depth in shallow streams that has a vernier scale on top with a sliding rod attached for use with a flow meter to measure flow
Velocity	the speed at which water is moving
Vernier scale	a small, movable auxiliary graduated scale attached parallel to a main graduated scale and calibrated to indicate fractional parts of the subdivisions of the larger scale
Wadeable stream	typically first to fourth order streams, that can be easily navigated by foot (in terms of depth and flow velocity) for most of the sample area length and that do not require a boat to adequately sample
Wetted width	the horizontal width of the channel that contains water at the time of sampling
1:1 Preservative	an acid preservative added to sample containers in a ratio of one part Type I DI water to one part acid

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APPENDIX A.

SONDE DEPLOYMENT DATASHEET

Arkansas - Division of Environmental Quality
Short-term Deployment Field Sheet

Deployment Notes

Site Code: _____ Stream Name: _____

Technician(s): _____

Date: _____ Instrument ID: _____

Time logging set to start:_____ Time sonde placed in stream: _____

Deployment # (circle 1): 1 2

File name: _____ Logging interval:_____

Deployment Lat/Long: _____

Deployment location description (including what was used as an anchor and general water depth):

_____

_____

Upstream or downstream of nearest crossing: Upstream Downstream

Flow Code:_____

Water conditions (flowing, stagnant or in pools, water level low or filled bank to bank, water clarity):

_____

Weather conditions:_____

Pickup Notes

Technician(s):_____

Date:_____ Time sonde removed from stream:_____

Logging stopped at pickup? Y N

Water conditions during pickup:_____

Notable weather events during deployment: _____

Notes (e.g. mud on sensors, fish in sonde guard, evidence sonde was disturbed):_____

_____

APPENDIX B. USING A USGS TOP-SETTING WADING ROD WHEN COLLECTING FLOW DATA

The vernier scale on the handle of the top-setting wading rod is graduated in tenths of feet (see below). When the scale on the sliding suspension rod is aligned with the vernier scale on the handle to the water depth observed on the hexagonal rod, the attached instrument probe is automatically set to six-tenths depth from water surface. The vernier scale can be read to the nearest 0.01 feet by visually interpolating between the 0.10 graduations. This is particularly important when measuring shallow depths.

- For depths measuring 0.20 ft (minimum depth the rod can measure) to 2.5 ft: set probe to six-tenths of the total depth from the water surface by lining up the foot scale on the suspension rod with the vernier scale on the top-setting wading rod handle. For example, if the depth is 0.9 ft, line up the 0 on the suspension rod foot scale with the 9 on the top-setting wading rod handle's vernier scale (See below).

- For depths equal to or greater than 2.5 ft: set the probe to two-tenths and eight-tenths of the total depth. For example, if the depth is 2.8 ft, set the top-setting wading rod as follows:

  To measure two-tenths depth from water surface, double the depth observed on the hexagonal rod (i.e., 2.8 ft x 2 = 5.6) and set the scale to 5.6 by aligning the 5 on the suspension rod foot scale with the 6 on the vernier scale of the top-setting wading rod handle.

  To measure eight-tenths depth from surface, divide the depth by two (i.e., 2.8 ft / 2 = 1.4) and set the scale to 1.4 by aligning the 1 on the suspension rod foot scale with the 4 on the vernier scale of the top-setting wading rod handle.

  USGS Top-Setting Wading Rod handle showing vernier scale set at 0.9 ft (CDFW 2013)

APPENDIX C.

FLOW FIELD DATASHEET

Streamflow Measurement Form

Stream:_____ Date:_____ Time:_____

Station ID:_____ Crew members:_____

Description:_____

Lat/Long:_____ Meter type:_____

Observations:_____ *# of intervals used_____

Total wetted width at transect:_____ (ft) Total Q (ft/s)_____

Distance from initial point (ft)	Width of interval (ft) (W)	Depth (ft) (D)	**Sensor Depth (0.2, 0.6, or 0.8)	Velocity (V)		Obstruction (log, rock, other) or QC comment	Flow =Q (ft/s) Q=(W)(D)(V)
				At point (ft/s)	Average (ft/s)		
						Total Discharge/Flow:	

*Stream width < or = 10 feet wide: 10 intervals; Stream width >10 feet wide: 20-30 intervals (intervals no less than 0.5 ft or greater than 3 ft)

**If depth is greater than 2.5 ft then switch to two point method of readings at 0.2 and 0.8 depth. Calculate 0.2 (depth x 2), Calculate 0.8 (depth / 2)

Distance from initial point (ft)	Width of interval (ft) (W)	Depth (ft) (D)	**Sensor Depth (0.2, 0.6, or 0.8)	Velocity (V)		Obstruction (log, rock, other) or QC comment	Flow =Q (ft/s) Q=(W)(D)(V)
				At point (ft/s)	Average (ft/s)		
						Total Discharge/Flow:	

APPENDIX D. EXAMPLES OF BED-MEETS-BANK IDENTIFICATION

Figure D1. Bed-meets-bank is indicated by a distinct change in slope and vegetation.

Figure D2. Bed-meets-bank is below the waterline, indicated by a dramatic change in slope.

Figure D3. Bed-meets-bank is indicated by a dramatic change in slope

Figure D4. Bed-meets-bank is indicated by a dramatic change in slope

APPENDIX E. EXAMPLES OF COVERED AND UNCOVERED BANKS

Figure E1. Examples of covered banks. Banks are considered covered if more than half of the plot surface is covered by perennial vegetation (including roots), cobbles 15 cm or larger, anchored trees, and large woody debris with a diameter >10 cm, or if the banks are solid bedrock.

Figure E2. Examples of uncovered banks. Banks are considered uncovered if less than half of the plot surface is covered by materials that would help the bank resist erosion. Several of the banks pictured have some perennial vegetation, cobbles, or large woody debris, but these materials do not cover over 50% of the total bank area.

APPENDIX F.

TIER 1 HABITAT DATASHEET

Arkansas Division of Environmental Quality Stream Habitat Profile: Tier 1							
Date (mm/dd/yyyy):		Start Time:				Crew:	
Stream Name:		Site Code:				Reach Length:	
5 Stream Widths:	1.	2.	3.	4.	5.	Average Width:	Flow:
Temperature:		pH:		DO:		Conductivity:	

Stream Physical Measurements												
	Wetted Width (m)	Depth Profile (m)				Bank Angle		Substrate				
Transect	Wetted Width (m)	Left Quarter	Center	Right Quarter	Thalweg	Left Bank	Right Bank	Pool/Riffle/Run	Left Quarter	Center	Right Quarter	Dominant Substrate
1												
2												
3												
4												
5												
6												
7												
8												
9												
10												

Measure wetted width perpendicular to thalweg from left wetted edge to right wetted edge.

Measure thalweg at point in stream carrying majority of discharge. Often, but not always the deepest point or mid channel. If thalweg and center are the same, still record the depth in both fields.

If measuring an undercut bank, subtract the clinometer reading from 180, then record.

Classification	Particle Size
Clay	<0.5 mm sticky or hardpacked
Silt	<0.5 mm loose, soft fines
Sand	0.5-3 mm
Gravel	2-50 mm
Pebble	50-150 mm
Cobble	150-500 mm
Boulder	>500 mm
Other	Dense root mat, leaf pack, woody debris piles

Notes:

Reach-wide Habitat Tallies:

Total Pools	Total Riffles	Total Large Woody Debris

In-Channel Habitat										
Transect	Large Woody Debris	Small Woody Debris	Live Trees/Roots	Leaf Pack	Filamentous Algae	Macrophytes	Boulders	Undercut Banks	Artificial Structures	Note approximate cover of each type to the nearest 10%.
1										
2										
3										
4										
5										
6										
7										
8										
9										
10										

Terrestrial Cover													Photos
	Bank Cover (C=Covered B=Bare)		Canopy Cover				Riparian Cover						
							Left Bank			Right Bank			
Transect	Left Bank	Right Bank	Left Bank	Center Left	Center Right	Right Bank	Ground Cover (<0.5m)	Shrub Layer (0.5-5 m)	Tree Layer (>5 m)	Ground Cover (<0.5m)	Shrub Layer (0.5-5 m)	Tree Layer (>5 m)	Photographs Collected?
1													
2													
3													
4													
5													
6													
7													
8													
9													
10													
Banks are considered covered if >50% of bank plot covered by vegetation, cobble, anchored woody debris or bedrock.			Assess canopy 0.3 m from water's surface on left and right bank while facing stream center, and in stream center facing left and right bank.				The plot for riparian cover begins at the wetted edge and extends to 5 m outside of the stream. The plot width extends parallel to the stream 2.5 m upstream and downstream of the transect line (5 m wide total).						Take photos of upstream, downstream, left bank, right bank

APPENDIX G.

TIER 2 LOW GRADIENT DATASHEET

Arkansas Division of Environmental Quality Stream Habitat Profile: Tier 2 Low Gradient	
Stream Name:	Station ID:
Date:_____Time:_____ am pm	Lat:_____ Long:_____
Sampling Crew:	Reach Length:
Form Completed By:	Mean Velocity:_____Flow:_____

Parameters to be evaluated in sampling reach	Low Gradient Stream Habitat Parameters	Low Gradient Streams: Condition Category			
		Optimal	**Suboptimal**	**Marginal**	**Poor**
	1. Epifaunal Substrate/ Available Cover	Greater than 50% of substrate favorable for epifaunal colonization; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e. logs/snags that are not new fall and not transient)	30-50% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale)	10-30% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed	Less than 10% stable habitat; lack of habitat is obvious; substrate unstable or lacking
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	2. Pool Substrate Characterization	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present	All mud or clay or sand bottom; little or no root mat; no submerged vegetation	Hard-pan clay or bedrock; no root mat of vegetation
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	3. Pool Variability	Even mix of large-shallow, large-deep, small-shallow, small-deep pools present	Majority of pools large-deep; very few shallow	Shallow pools much more prevalent than deep pools	Majority of pools small-shallow or pools absent
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 20% of the bottom affected by sediment deposition	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 20-50% of the bottom affected; slight deposition in pools	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 50-80% of the bottom affected; sediment deposits at obstructions, constrictions and bends; moderate deposition of pools prevalent	Heavy deposits of fine material, increased bar development; more than 80% of the bottom changing frequently; pools almost absent due to substantial sediment deposition
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed	Water fills >75% of the available channel; or <25% of channel substrate is exposed	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed	Very little water in channel and mostly present as standing pools
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

	Low Gradient Stream Habitat Parameters	Low Gradient Streams: Condition Category			
		Optimal	**Suboptimal**	**Marginal**	**Poor**
Parameters to be evalusted broader than sampling reach	**6. Channel Alteration**	Channelization or dredging absent or minimal; stream with normal pattern	Some channelization present, usually in areas of bridge abutements; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	7. Channel Sinuosity	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (Note - channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas)	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line	The bends in the stream increase the stream <1 time longer than if it was in a straight line	Channel straight; waterway has been channelized for a long distance
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	8. Bank Stability (Score each bank)	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars
	SCORE LB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	SCORE RB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	9.Riparian Vegetative Zone Width (Score each bank riparian zone)	Width of riparian zone >18 m; human activities (i.e. parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone	Width of riparian zone 12-18 m; human activities have impacted zone only minimally	Width of riparian zone 6-12 m; human activities have impacted zone a great deal	Width of riparian zone <6 m; little or no riparian vegetation due to human activities
	SCORE LB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	SCORE RB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

TOTAL SCORE _____

NOTES:

APPENDIX H.

TIER 2 HIGH GRADIENT DATASHEET

Arkansas Division of Environmental Quality Stream Habitat Profile: Tier 2 High Gradient

Stream Name:	Station ID:
Date:_____Time:_____ am pm	Lat:_____ Long:_____
Sampling Crew:	Reach Length:
Form Completed By:	Mean Velocity:_____Flow:_____

	High Gradient Stream Habitat Parameters	High Gradient Streams: Condition Category			
		Optimal	**Suboptimal**	**Marginal**	**Poor**
Parameters to be evaluated in sampling reach	**1. Epifaunal Substrate/Available Cover**	Greater than 70% of substrate favorable for epifaunal colonization; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e. logs/snags that are not new fall and not transient)	40-70% mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale)	20-40% mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed	Less than 20% stable habitat; lack of habitat is obvious; substrate unstable or lacking
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	2. Embeddedness	Gravel, cobble and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space	Gravel, cobble and boulder particles are 25-50% surrounded by fine sediment	Gravel, cobble and boulder particles are 50-75% surrounded by fine sediment	Gravel, cobble and boulder particles are more than 75% surrounded by fine sediment
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	3. Velocity/Depth Regime	All four velocity/depth regimes present (slow-deep; slow-shallow; fast-deep; fast-shallow). (Slow is <0.3 m/s, deep is >0.5 m)	Only 3 or 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes)	Only 2 or 4 regimes present (if fast-shallow or slow-shallow are missing, score low)	Dominated by 1 regime (usually slow-deep)
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	4. Sediment Deposition	Little or no enlargement of islands or point bars and less than 5% of the bottom affected by sediment deposition	Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5-30% of the bottom affected; slight deposition in pools	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% of the bottom affected; sediment deposits at obstructions, constrictions and bends; moderate deposition of pools prevalent	Heavy deposits of fine material, increased bar development; more than 50% of the bottom changing frequently; pools almost absent due to substantial sediment deposition
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	5. Channel Flow Status	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed	Water fills >75% of the available channel; or <25% of channel substrate is exposed	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed	Very little water in channel and mostly present as standing pools
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

	High Gradient Stream Habitat Parameters	High Gradient Streams: Condition Category			
		Optimal	**Suboptimal**	**Marginal**	**Poor**
Parameters to be evaluated broader than sampling reach	**6. Channel Alteration**	Channelization or dredging absent or minimal; stream with normal pattern	Some channelization present, usually in areas of bridge abutements; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	7. Frequency of Riffles (or bends)	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream <7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important	Occurrence of riffles infrequent; distance between riffles divided by the width of the stream is between 7 to 15	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is a ration of >25
	SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	8.Bank Stability (Score each bank)	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected	Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion	Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars
	SCORE LB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	SCORE RB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	9. Riparian Vegetative Zone Width (Score each bank riparian zone)	Width of riparian zone >18 m; human activities (i.e. parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone	Width of riparian zone 12-18 m; human activities have impacted zone only minimally	Width of riparian zone 6-12 m; human activities have impacted zone a great deal	Width of riparian zone <6 m; little or no riparian vegetation due to human activities
	SCORE LB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0
	SCORE RB	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

TOTAL SCORE _____

NOTES:

APPENDIX I.

FISH COLLECTION DATASHEET

Fish Electroshocking Collection – Field Identification		
Stream:	**WQMS Code:**	**Collection Date:**
County:	**Ecoregion:**	**Collectors:**
Total Shocking Time:	**ID'd By:**	
1 Backpack	**2 Backpack**	**Barge**
Duty Cycle:	**Volts:**	**Frequency:**
Approximate Seining Distance:		
Notes:		

Species	Count Box	Total	DELT	Blackspot

Species	Count Box	Total	DELT	Blackspot

APPENDIX J.

MACROINVERTEBRATE FIELD DATASHEET

Arkansas Division of Environmental Quality Stream Macroinvertebrate Collection		
Date (mm/dd/yyyy):	Start Time:	Crew:
Stream Name:	Site Code:	Reach Length:

Temperature:	pH:	DO:

Single Habitat Method		
Total Kicks		Perform 20 separate 15-second riffle kicks. When available, kick two separate riffles and allocate 10 kicks to each. Note any deviations from this protocol.
Total Riffles Kicked		

Multi-Habitat Method		
Habitat Type	**Kick Totals**	**Target Kick Numbers**
Riffle		Aim for as many kicks as possible if riffle is present (maximum 20 kicks) and distribute any remaining kicks among other habitat types as specified in the protocol
Woody Debris/Snags		8
Rootwads/ Undercut Banks		6
Leaf packs		3
Soft Sediment/Sand		3
Vegetation		3 additional kicks if present

Notes:

ARKANSAS
ENERGY & ENVIRONMENT

CONTACT INFORMATION:

501.682.0744

info@adeq.state.ar.us

MAILING ADDRESS:

5301 Northshore Dr.
North Little Rock, AR 72118