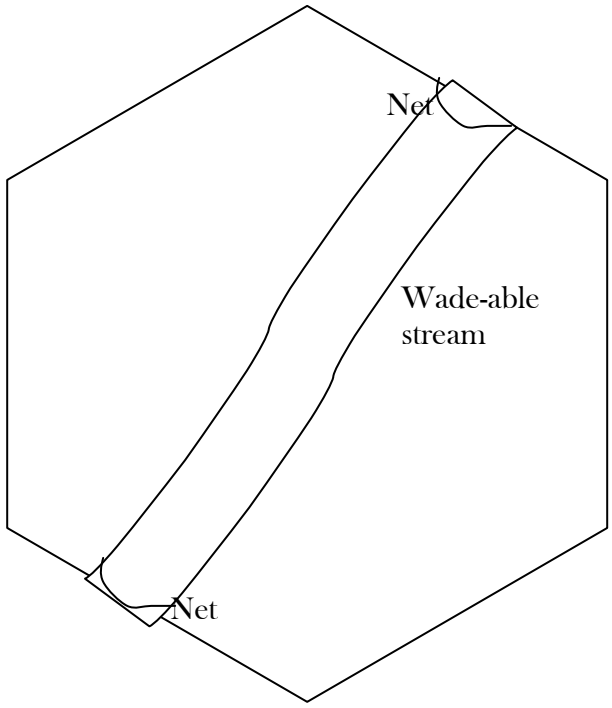
Chapter Fifteen
Fish Monitoring
Wade-able Streams & Rivers

The Fisheries Bureau of the Iowa DNR has been monitoring fish for many years and has protocols for different wetland habitats. The following is an adaptation of the "Biological Sampling Procedures for Wadeable Streams and Rivers in Iowa" (Iowa DNR 2001). Few changes have been made to the original protocol.

IOWA FISH MONITORING IN WADEABLE STREAMS AND RIVERS:

This protocol is completely based upon the "Biological Sampling Procedures for Wadeable Streams and Rivers in Iowa" (Iowa DNR 2001) protocol first drafted in 1994. In addition to recording fish species, information is also collected on benthic macroinvertebrates. A few modifications are suggested in this section, mostly in regard to the length of area to be sampled. The design includes electrofishing to determine fish species and numbers, in addition to collecting benthic macroinvertebrates and habitat data.

Within the permanent sampling plot, any wadeable stream or river should be searched for all fish species using this protocol. In some of these plots a water habitat will be the focal point, meaning the hexagon will be centered on a stream, river, lake, creek, etc. In these plots, it is anticipated that a stream reach of up to 400 meters or more may need to be sampled.



Regardless of the amount of stream occurring within the plot, a 150 meter reach is the minimum that should be sampled. So, if only 50 meters of suitable habitat is found within the plot, then 50 m beyond each of the 2 boundaries should be surveyed as well. Five hundred meters should be the maximum stream reach surveyed due to time considerations.

SURVEY METHODS:
Sampling in wadeable streams and rivers should occur between June 15 and September 30 (15 weeks and 2 days). In general, sampling will occur during daylight hours for active sampling gears.

Stream flow levels should be similar to base flow conditions. Sampling should be halted when the stream flow is elevated or there are high turbidity levels; when the stream flow is extremely low; or when there has been a minor runoff event within the last week. A runoff event could disrupt the aquatic community. Surveys are also halted during inclement weather (extreme wind, lightning, or rain).

The IDNR wadeable streams protocol also suggests that no sampling should be done within one year after a major flood event or within one year of a severe drought. For the purposes of this monitoring program, however, community changes associated with these events also provide important information. Therefore, these two events are not considered valid reasons to disrupt the sampling regime. It should be noted on the data sheets or in the database, however, if and which of these 2 events had occurred and the date(s) of occurrence.

The IDNR wadeable streams protocol further clarifies that within each sampling reach, there should be 2 distinguishable pool/riffle sequences or 2 well defined channel bends. If neither of these is present, then there are specifications as to the length which should be surveyed. These include that waters $\leq$40 feet (12.2 m) in width should be surveyed to a length 30x the width, and waters >40 feet (12.2 m) in width should be surveyed 20x the mean width. For simplicity, this protocol advocates sampling 30x the width of the stream regardless of other considerations. Ideally, this will result in a distance of between 300 & 400 m being surveyed.

The first step in the sampling protocol is to collect information from the GIS database as to the location of roads, trail, and other disturbances near the sampling area. Notes should also be made as to the best (apparent) location for entering the water. See Chapter 3 (Landscape Characteristics) for further information. Sampling each reach is expected to take 8 hours or less. Sampling may only stretch over 2 days if stream conditions do not change overnight.

Data should be collected in the following sequence:
1). Measure stream width, delineate sampling reach, and place block nets.
2). Collect water samples for physicochemical water quality parameters.
3). Collect semi-quantitative benthic macroinvertebrate samples.
4). Collect qualitative, multi-habitat benthic macroinvertebrate sample.
5). Conduct fish sampling.
6). Complete habitat measurements.

Water Sample Collection

Water samples should be taken from the stream or river with the use of clean, glass jars that are labeled with a Sharpie marker. Water samples should be stored following recommendations outlined by the University of Iowa Hygenics Laboratory.

Benthic Macroinvertebrate Sampling

These data are qualitative and semi-quantitative, providing a list of macroinvertebrate species as well as an abundance index to the taxa observed. These techniques will not allow for the estimation of density or biomass. For the semi-quantitative data, triplicate samples should be made of either 1) rock substrates in riffle or shallow run habitat, or 2) multi-plate, artificial substrates deployed in moderately swift run habitat.

To do this, a modified-Hess sampler, a Surber sampler, or modified Hester-Dendy (multi-plate artificial) substrates, is used, depending upon the habitat characteristics of the stream being monitored. If it is necessary to use the multi-plate artificial substrate device, this must first be deployed for 4-6 weeks to allow for colonization before data can be collected. The IDNR routinely deploy these substrates during reconnaissance visits to the site or during sampling of nearby sites in order to minimize travel costs.

The modified-Hess sampler is an open-ended, mesh enclosed cylinder. Photos of this can be seen in INDR (2001). The following is copied verbatim from the INDR (2001) sampling protocol, pages 6-14: The upstream side is a mesh window that allows water to flow through the sampler while keeping all drifting macroinvertebrates out of the sampler. The downstream side of the cylinder has a funnel-shaped mesh collection bag and collection container for capturing macroinvertebrates dislodged as substrates inside the sampler are agitated. The modified-Hess sampler is most effective in shallow riffles and runs (< 1.5 feet or 45.7 cm) with abundant rock substrates. This sampling device performs well in streams where there is a mixture of substrate particle sizes and the sampler can be penetrated 2-4 inches (5-10 cm) into the stream bottom.

Whenever possible, collect the triplicate samples from the same riffle or run. If the riffle or run is too small to obtain 3 samples, collect the remaining samples from another suitable riffle or run in the sampling reach. Record observations on the amount and type of periphyton growing on the substrates, the amount of embeddedness of coarse substrates, and the amount of macroinvertebrate colonization on the field data sheet (see appendix). Apply the following protocol when collecting the modified-Hess samples:

1. Approach the riffle or sampling area from downstream to minimize disturbance.
2. Select the area to place the sampler and push the sampler 2-4 inches in to the substrate, with the funnel collection bag downstream.

3. Carefully wash all cobbles and large gravel particles within the cylinder and remove all clinging organisms before discarding.
4. Vigorously agitate the remaining substrate to approximately the same depth as the base of the sampler.
5. Try to rinse as many macroinvertebrates as possible off of the sampler and funnel net, down into the collection container.
6. Transfer the contents of the collection container and all remaining organisms on the sampler into the sample container.

Process the triplicate modified-Hess samples individually and do not composite them in the field. Add a 10% formalin solution to the sample containers to field preserve them for later analysis. Buffer the sample by adding 3 grams of borax to one liter of solution to neutralize the pH of formalin and prevent shrinkage and damage to the tissue of preserved organisms (USGS 1993).

Label the sample containers with indelible ink. The information on the label must include stream name, site identification number, sampling date, collector, and a unique sample identification number. Complete a sampling documentation form for each sample according to University of Iowa Hygienic Laboratory (UHL) Limnology field sampling protocols. Record the sample identification numbers on the field observation data sheet.

Artificial Substrates

In streams that lack productive riffle or run habitat, use the modified Hester-Dendy artificial substrates to obtain the semi-quantitative samples. Deployment of 4 multi-plate artificial substrates occurs at each sampling site. The colonization period lasts a minimum of 4 weeks and must not exceed 6 weeks. The advantages of artificial substrates, which include habitat standardization and macroinvertebrate productivity, seem to outweigh their disadvantages that include habitat artificiality and taxa selectivity.

Each artificial substrate consists of 8 - $^1/_8$" x 4" x 4" (or 20.6 cm x 10.2 cm x 10.2 cm) wood plates and 12 - $^1/_8$" thick and 1" diameter (or 30.8 cm thick and 2.5 cm diameter) cylindrical PVC spacers. The total surface area of the multi-plate unit is 145.6 in^2 (0.094 m^2) (OEPA 1989). Placement of the spacers between the wood plates on a $\frac{1}{4}$" (0.64 cm) threaded steel rod is as follows: 3 single spacers on top, 3 double spacers in the middle, and 1 triple spacer on the bottom.

Artificial substrate placement – Try to deploy the artificial substrates in moderately swift run habitat with firm substrate (sand or sand/gravel, not silt or muck). Apply the following deployment criteria to ensure consistent artificial substrate placement across sampling sites and ecoregions:

1. Deploy the artificial substrates in flowing water having a current velocity of 0.5 to 1.5 feet per second (15.2 cm to 45.7 cm per second).
2. Deploy the artificial substrates in runs with depths of 1 to 3 feet (30.8 to 91.4 cm). Consider the anticipated flow stability when determining the appropriate distance from the top plate to the surface of the water. Ideally, deploy the sampling unit in the photic zone of the water column and sufficiently deep to ensure that the top plates remain submersed throughout the 4 – 6 week colonization period if flow levels decline. The distance from the top plate to the surface of the water is normally between 4 and 8 inches (10.2 to 20.3 cm). The bottom plate should be at least 3 inches (7.62 cm) above the bottom to prevent sedimentation of the sampling unit.
3. Deploy the artificial substrate units in the main axis of flow and at least 3 feet (0.91 m) from shore. Place the 4 sampling units in a diamond configuration approximately 3 to 5 feet (0.91 to 1.5 m) apart.

Whenever possible, locate the sampling units near the downstream boundary of the sampling reach to enable the benthic macroinvertebrates residing on natural substrates in the sampling reach an opportunity to colonize the artificial substrates via drift. Careful consideration of the susceptibility to vandalism and damage from high flows in critical in the placement of artificial substrates.

Illustrate on a hand-sketched map, the location of the substrates with distances to at least 2 landmarks on the shore indicated. Attachment of brightly colored nylon flagging tape to the artificial substrate units may make them easier to find after colonization. Using wooden survey stakes or flagging tape to mark the approximate locations of artificial substrates is also accepted.

Field sample processing – Retrieve the artificial substrates in a downstream to upstream manner. Remove all artificial substrate units present after the colonization period from the stream. Evaluate the status of the substrates and choose the 3 'best' substrates to process. 'Best' are those substrates that are still completely submersed at time of retrieval and free from an extraordinary amount of silt or debris. Samples obtained from heavily damaged or silted units

are discarded only after determining that 3 acceptable samples, containing at least 100 organisms per sample, are available.

Examine each artificial substrate during removal and record the following observations on the field data sheet (see appendix):

1. Amount and type of periphyton growth on the plates.
2. Amount of sedimentation and/or other damages to the plates.
3. Amount of benthic macroinvertebrate colonization.

Remove the artificial substrates from the streambed with care to minimize the loss of macroinvertebrates. Carefully remove any extraneous debris, such as leaves or sticks, residing against the sampling unit before removing the unit from the stream bottom. Place a 500 μm mesh collection bag over the sampling unit and draw tightly at the base to insure that any dislodged organisms are not lost while the artificial substrate is pulled from the stream bed.

Empty the artificial substrate unit and other contents of the collection bag into a white enamel pan containing a small amount of clean water. Remove all clinging organisms from the collection bag with forceps and place in the pan. Disassemble the artificial substrate unit and remove the macroinvertebrates from the plates by gentle scraping each plate surface with a single-edge razor blade or pocketknife. Rinse and examine all extraneous debris (e.g., leaves and sticks) for macroinvertebrates and then discard. Transfer the pan contents to a labeled sample jar containing 10% formalin solution. Use separate labeled containers for the artificial substrate samples and do not composite the samples in the field or laboratory.

Label the sample jars with the following information: stream name, site identification number, sampling date, collector, and the unique sample identification number. Fill out a sample documentation form for each sample according to UHL Limnology field sampling protocols. Record the sample identification numbers on the field observation data sheet.

Multi-Habitat Sampling Procedures

The purpose of sampling multi-habitat is to increase the number of macroinvertebrate taxa represented on the qualitative list of taxa for the sampling site. Habitat-specific sampling (e.g., riffle-only sampling) is known to result in an underestimate of taxa richness for an entire reach of stream compared to multi-habitat sampling methods (Lenat 1988, Mackey 1984).

Multi-habitat sampling is preferably conducted on the same day but after, or simultaneous to, the retrieval of artificial substrates of natural substrate sampling. The multi-habitat sampling requires 2 or 3 crew members. Before initiating the sampling, crewmembers must review sampling procedures and divide-up tasks. Time allocation for natural substrate multi-habitat sampling and processing is approximately 1.5 person hours. In stream reaches that have complex benthic habitat and/or high biological diversity, extend the sampling time to ensure adequate sampling of the reach. Indicate the amount of extra sampling time on the field data sheet.

Sampling approach – Subdivide the sampling reach into 3 areas: upper, middle, and lower reach. One crewmember is responsible for each of the areas. Typically, crewmembers use standard No. 30 brass sieves to collect and concentrate organisms; however, wash buckets, kick-nets, or other sampling gear are also accepted. The mesh size of all nets, sieves, wash buckets, or other sampling gear used in multi-habitat sampling ranges from 500-600 m. Collect macroinvertebrates from all accessible types of benthic substrates by handpicking or sieving. Common techniques used to collect insects include:

- Sieving the gravel, fine substrate, clay hardpan, and overhanging vegetation
- Disturbing the rocky riffle and run areas by foot and using the sieve as a drift capture tool
- Handpicking macroinvertebrates from large cobbles and boulders, woody debris, and any other large substrates found in the stream.

It is important to sample as many different substrates as possible by not lingering in 1 area too long. When 3 crewmembers conduct the sampling, each crewmember should try to collect approximately 40-50 organisms. When 2 crewmembers are sampling, each should try to collect 60-75 organisms. It is important to collect as many different types of organisms as possible. However, if during sampling, it appears the taxa richness is minimal, the number of organisms per crewmember mentioned above still applies.

Each crewmember carries a plastic sampling container that serves as a temporary receptacle during sampling. At the end of the allotted sampling time (1.5 combined person-hours), combine the sample containers into one labeled

sample jar containing a 10% formalin solution. Label the sample container with the following information: stream name, site identification number, sampling date, collector(s), and a unique sample identification number. Complete the sample documentation for the multi-habitat sample according to UHL Limnology field sampling protocol. Record the unique sample identification number on the field observation data sheet.

Laboratory Macroinvertebrate Sample Processing

Field preserved benthic macroinvertebrate samples are transported to UHL, transferred into 85% ethanol solution, and stored until identification. Obtain (pick) a random subsample of 100 organisms from each triplicate semi-quantitative sample (Modified-Hess or artificial substrate). Sort and identify every organism in the composited qualitative multi-habitat samples (all picks). Initially sort all organisms by order in preparation of the more detailed taxonomic analysis.

Identify the macroinvertebrates in the samples to the "lowest practical taxonomic level". The lowest practical taxonomic level varies between and within invertebrate orders depending on the availability of appropriate taxonomic keys and the amount of time and expertise needed to attain precise determinations. The lowest practical taxonomic level is usually genus or species, however, in certain problematic taxa (e.g., Chronomidae and Oligochaetes) it is family level. If desired, retain several representative individuals of each problematic taxon for a more precise taxonomic analysis later. Follow UHL protocol for taxonomic verification and laboratory QA/QC procedures.

Record the totals of each taxon in the subsample on laboratory bench sheets. The data will eventually reside in the STORET/EDAS database. Following data storage, compare data printouts against laboratory bench sheets similarly to the verification process of the DNR/UHL ambient stream monitoring data in STORET.

Fish Community Sampling

Electrofishing

For small streams (average base-flow widths of less than 15 feet or 4.6 m) a single backpack unit is sufficient. In wider streams, it may be necessary to use 2 backpack units simultaneously. For other streams which may be too deep or wide to cover with backpack units, a towboat electro-fishing unit (with a generator, electrical control box, retractable electrodes, and a live well) is used.

Both the downstream and upstream ends of the sampling area should be blocked using 3/16" block nets. Beginning at the downstream starting point, a single pass is made upstream to capture all fish in the water. Sample all habitats thoroughly by methodically sweeping the anode from side to side. All stunned fish are captured in 3/16" dip nets and transferred into buckets or tanks until processed.

Additional data collected include the type of equipment used to stun the fish, the beginning and ending times for the use of the backpack shocker, and stream reach length and average width.

Seining

Seining may be the most efficient method to sample small fishes (e.g. redfin shiner *Lythrurus umbratilis*). However, recent research in northwest Iowa appears to indicate that seining does not add additional information when electrofishing is also used (Clay Pierce, personal communication). This issue can be addressed during the first few years of the monitoring program. The seine should be of 3/16 inch mesh size, and have floats attached at the top and weights attached at the bottom. For most wadeable streams and rivers in Iowa a haul or bag seine should be sufficient. If not performed correctly, fish could escape from under the net. If available, the same equipment could be used in wadeable streams as in the larger systems, but in the wadeable streams, the trawling net would be drawn through the water by hand (Herzog et al. 2005). The mesh size on the inner trawling net used in the larger systems is also 3/16 inch (4.76-mm).

Two technicians should pull the seine from a downstream to upstream direction, taking care that the net stays on the bottom of the channel bed. The seine should be removed from the water every 50 meters. Fish should be removed from the net and can be processed by another technician as the seine technicians continue upstream, or they can be placed in a holding bucket until processing.

The entire reach should be sampled with the electroshock technique moving from downstream to the upstream blocking net. This same area should also be sampled with between 1 and 3 seine hauls (Quist et al. 2003).

Make sure the fish in the holding buckets or tanks have fresh water to limit mortality. At pre-determined stopping points (which can be blocked by additional nets prior to beginning the sampling), identify and count the fish. If fish are to be marked at that site, mark the fish and record the mark. Release all fish.

Collect information on all captured fish, regardless of size (i.e. those less than 1 inch in size should also be identified if possible, and counted). In addition, examine all collected fish for external abnormalities [skeletal deformities, eroding fins, lesions, and tumors (DELTs)]. Record this information on the data sheet. The DELT coding procedures have been adapted from the Ohio EPA fish sampling procedures (OEPA 1989). These guidelines are listed in the appendix.

For any un-identifiable species, a voucher may be collected by preserving 1 or more specimen in 10% formalin.

HABITAT AND PLANT COMPOSITION DATA COLLECTION:
It is expected that the Aquatic Habitat Monitoring Protocol (Chapter 20) will acquire all necessary habitat data. That chapter includes information on collecting data on the habitats stratified into a wetland classification (i.e. river, stream, creek, impoundment, lake, etc.), as well as wetlands which occur on sites classified as terrestrial habitats. As the same areas will be searched for multiple species, no additional habitat data is expected to be collected under the fish in wadeable streams protocol. However, fisheries technicians should coordinate with other crews to ensure that all needed habitat data is collected.

Environmental data collected the day of sampling should include: surface water temperature, ambient air temperature, flow level, secchi disk reading (in tenths of feet), conductivity (uhmos), weather conditions, sampling effort (in minutes), and any relevant comments. In addition, be sure to record the number of people in the crew and their names, the name of the site, and sketch a map of the area sampled.

EQUIPMENT NEEDED:
GPS unit
Water collection jars
Binoculars
Dip nets
Block nets
Twine for repairs to blocknets and seine nets
Backpack electrofishing units
Extra batteries and gas:oil mix for Backpack units
Tow boat if needed
Buckets or holding tanks
Non-breathable chest waders
Inflatable life preservers
Plastic calipers
Standard field kit: Clip board, pencils, ruler, small scissors, Sharpie markers, hand sanitizer, & data sheets.
Field guides
Rubber gloves
Benthic macroinvertebrate surveys:
Modified-Hess sampler or Surber sampler, or 4 Modified Hester-Dendy artificial substrate Samplers
Collection jars
Jar labels
10% formalin with Borax solution

STAFF & TRAINING:
Two weeks of training is recommended and should include 1) field guide use and identification, 2) trips to University museums to discuss defining species characteristics, 3) field practice with an experienced observer, 4) safely using the sampling equipment, 5) proficiency testing, and 6) habitat data collection. The crew leader should review duties and safety precautions with the sampling crew before each survey.

DATA QUALITY & MANAGEMENT:
Electroshocking and seining data can be affected by:

- Incorrect use of equipment: Should be checked periodically by supervisor.
- Observer handling care: Fish should not be left in holding buckets any longer than necessary. Mortalities can be assessed by examining the data, and should be <1%.
- Error in species ID: Difficult to monitor, therefore, could switch observers between crews or collect voucher specimen.

At the end of each sampling day, field crews should review data sheets to ensure all information is present. At the end of the week, the field crew leader should review the data sheets for ID, escape and mortality rates, and legibility.

DATA ANALYSIS:
The basic information should allow the creation of a species list for each site, and data should at least be used to estimate the proportion of points occupied using program PRESENCE or program MARK. This is the only protocol where sites are visited only once per year. Both of the other 2 fisheries protocols (rivers and lakes) visit each site 3 times per year. The sampling design for fish in wadeable streams may affect the potential analysis of the data. For additional information on the PAO techniques, see Chapter 5 (Data Analysis).

Following the methods are outlined in the IDNR (2001) protocol: The data collected allow the estimate of the following community parameters of the fish sample:

1. Species composition (i.e., the number of fish of each species as a percentage of the total number of captured fish)
2. Fish species relative abundance (i.e., catch per unit effort)
3. Proportion of fish with external abnormalities.

The methods employed do not provide quantitative information suitable for fish population density or biomass estimates.

SAFETY CONSIDERATIONS:
As with all other protocols, basic hygiene, including washing hands prior to eating or face touching should be followed by all personnel.

Electrofishing can be dangerous. All personnel need to be trained in the use of this equipment. Working in wadeable streams is also physically challenging. Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of the river flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. If a person is swept off their feet when wearing chest waders, it is possible that the air trapped in the bottom of the waders will force the person to travel down the channel upside down with their head below water. Therefore, it is recommended that chest waders have release snaps in the front of the bib to allow the technician to escape in that situation. It would also be advisable to wear an inflatable life jacket underneath the bib of the chest waders.

Care should be taken in order to lessen the probability of spreading an infectious agent, such as a fungus or virus, between wetlands. One way to reduce the chance of spreading an infectious agent between wetlands is to allow the waders and equipment to dry for 3-4 days between sites. This may be impractical given the short time frame available for aquatic surveying in Iowa. It may be best to rinse the waders, gloves, and other equipment with a solution of hot water and bleach.

TARGET SPECIES:
The following list of fish species represents the 67 species of greatest conservation need as chosen by the Steering committee for the Iowa Wildlife Action Plan (Zohrer et al. 2005) and may be encountered during a survey. Distribution maps for these species can be found in "Iowa Fish & Fishing" (Harlan et al. 1987) and also in Iowa AQUATIC GAP (http://www.cfwru.iastate.edu/IAGAP_final_report.pdf). Appendix 1 contains a list of all fish species known to occur in Iowa which may also be encountered during the monitoring efforts.

Target species:

Common Name	Scientific Name	Habitat
Chestnut lamprey	*Ichthyomyzon castaneus*	Mississippi and Chariton rivers
Silver lamprey	*Ichthyomyzon unicuspis*	Mississippi River
American brook lamprey	*Lampetra appendix*	Northeast 1/4
Lake sturgeon	*Acipenser fulvescens*	Mississippi River
Pallid sturgeon	*Scaphirhynchus albus*	Missouri River
Shovelnose sturgeon	*Scaphirhynchus platorynchus*	Mississippi and Missouri Rivers
Paddlefish	*Polydon spathula*	Mississippi, Missouri, Des Moines, Iowa, Cedar, and Skunk rivers
Bowfin	*Amia calva*	Mississippi River
Longnose gar	*Lepisosteus osseus*	Mississippi and Missouri Rivers & larger tributaries
American eel	*Anguilla rostrata*	Mississippi and Missouri Rivers & larger tributaries
Skipjack herring	*Alosa chrysochloris*	Mississippi and Missouri Rivers
Mooneye	*Hiodon tergisus*	Larger interior rivers statewide
Goldeye	*Hiodon alosoides*	Missouri River & large streams in W, S, and SE
Brook trout	*Salvelinus fontinalis*	NE corner
Grass pickerel	*Esox americanus*	Missouri River & tributaries
Central mudminnow	*Umbra limi*	N 1/3
Largescale stoneroller	*Campostoma oligolepsis*	NE 2/3
Western silvery minnow	*Hybognathus agryritis*	Missouri drainage
Mississippi silvery minnow	*Hybognathus nuchalis*	Mississippi drainage
Plains minnow	*Hybognathus placitus*	Missouri drainage
Speckled chub	*Macrhybopsis aestivalis*	Large interior rivers statewide
Flathead chub	*Platygobio gracillis*	Missouri drainage
Sicklefin chub	*Macrybopsis meeki*	Missouri River

Target species continued:

Common Name	Scientific Name	Habitat
Silver chub	*Macrybopsis storeriana*	Larger interior rivers statewide
Gravel chub	*Erimytax x-punctatus*	Central & NE
Pallid shiner	*Hybopsis amnis*	Upper Mississippi River
Pugnose minnow	*Opsopoeodus emiliae*	Mississippi River
Pugnose shiner	*Notropis anogenus*	West Lake Okojobi
River shiner	*Notropis blennius*	Mississippi and Missouri Rivers & larger tributaries
Ghost shiner	*Notropis buchanani*	Mississippi River
Blacknose shiner	*Notropis heterolepis*	NW
Spottail shiner	*Notropis hudsonius*	Natural lakes, Mississippi River
Ozark minnow	*Notropis nubilus*	NE ¼
Weed shiner	*Notropis texanus*	Cedar & Mississippi Rivers
Topeka shiner	*Notropis Topeka*	W ¾
Channel mimic shiner	*Notropis volucellus*	Upper Mississippi River
Longnose dace	*Rhinichthys cataractae*	NE corner
Pearl dace	*Margariscus margarita*	Worth county
Blue sucker	*Cycleptus elongates*	Mississippi and Missouri Rivers & larger tributaries
Black buffalo	*Ictiobus niger*	Mississippi River & large tributaries
Black redhorse	*Moxostoma duquesnei*	Turkey & upper Iowa river drainages
Golden redhorse	*Moxostoma erythrurum*	Small & medium streams statewide
River redhorse	*Moxostoma carinatum*	Upper pools of Mississippi
Greater redhorse	*Moxostoma valenciennesi*	Upper Mississippi River
Spotted sucker	*Minytrema melanops*	Mississippi River
Brown bullhead	*Ameiurus nebulosus*	N 1/3
Slender madtom	*Noturus exilis*	Mississippi River tributaries
Tadpole madtom	*Noturus gyrinus*	Statewide
Freckled madtom	*Noturus gyrinus*	Mississippi River & large tributaries
Pirate perch	*Aphredoderus sayanus*	Mississippi River & large tributaries
Trout perch	*Percopsis omiscomycus*	NW ¼; Upper Mississippi River, Grand & Chariton Rivers
Burbot	*Lota lota*	Missouri River, Mississippi River & tributaries
Banded killifish	*Fundulus diaphanous*	Natural lakes in NW; Missouri River
Blackstripe topminnow	*Fundulus notatus*	E 1/3
Mottled sculpin	*Cottus bairdi*	Lower Bear Creek
Slimy sculpin	*Cottus cognatus*	NE corner
Warmouth	*Lepomis gulosus*	S ½; Mississippi River
Pumpkinseed	*Lepomis gibbosus*	Mississippi River & natural lakes
Slenderhead darter	*Percina phoxocephala*	Mississippi drainage
Blackside darter	*Percina maculate*	Mississippi River
River darter	*Percina shumardi*	Mississippi River
Northern logperch	*Percina caprodes*	Mississippi drainage, Clear Lake

Target species continued:

Common Name	Scientific Name	Habitat
Crystal darter	*Crystallaria asprella*	Mississippi & Turkey Rivers
Western sand darter	*Ammicrypta clara*	Mississippi River
Banded darter	*Etheostoma zonale*	NE ¼
Mud darter	*Etheostoma asprigene*	Mississippi River & tributaries
Orangethroat darter	*Etheostoma spectabile*	SE ¼
Least darter	*Etheostoma microperca*	Maquoketa, tributary to Otter Creek

ADDITIONAL METHODS FOR SPECIAL LOCATIONS:

Minnow traps

Minnow traps may be an effective way to find additional fish. These are used as part of the Amphibian protocol for capturing tadpoles. Minnow traps should be deployed in water at least deep enough to cover the trap opening but with an empty plastic bottle or other floatation device to ensure part of the trap stays above water to allow non-gilled captures to breath. Traps should be checked daily and left in the water for 3 to 5 days.

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APPENDIX. Methods for Examinations of Fish External Abnormalities – Adopted from the Ohio EPA, *copied verbatim from IDNR 2001*.

External Abnormalities – All fish that are captured are examined for the presence of gross external anomalies and their occurrence is recorded in the fish data sheet and subsequently entered into the FINV database. In order to standardize the procedure for counting and identifying anomalies the following criteria should be followed.

All fish are examined for gross external anomalies. These are anomalies that are visible to the naked eye when the fish are captured, identified, and counted. Table 1 lists the types of anomalies which are recorded on the fish data sheet and subsequently entered into FINV. Exact counts of anomalies present (i.e. the number of tumors, lesions, etc. per fish) are not made; however, light and heavy infestations are noted for certain types of anomalies (Table 1). An external anomaly is defined as the presence of an externally visible skin or subcutaneous disorder. Ultimately, the number and percentage of DELTs and non-DELTs are computed and recorded in the FINV database. Then the total percent anomalies for a specific type of anomaly or group of anomalies can be calculated for 1 or more sites.

The following is a review of some anomalies commonly encountered in freshwater fishes. These characteristics should be used in determining the types of external anomalies present and in coding the fish data sheets.

1. Deformities – These can affect the head, spinal vertebrae, fins, stomach shape, and have a variety of causes including toxic chemicals, viruses, bacteria, (e.g. *Mycobacterium* spp.), infections, and protozoan parasites (e.g. *Myxosoma carebaiis*, Post 1983). Fish with extruded eyes (see Popeye disease) or obvious injuries should not be included.
2. Eroded fins – These are the result of a chronic disease principally caused by flexibacteria invading the fins and causing a necrosis of the tissue (Post 1983). Necrosis of the fins may also be caused by gryodactylids, a small trematode parasite. When necrosis occurs in the tissue at the base of the caudal fin, it is referred to as peduncle disease. Erosions also occur on the preopercle and operculum and these should be included. In Ohio streams and rivers this anomaly is generally absent in least impacted fish communities, but can have a high incidence in polluted areas. It occurs most frequently in areas

with multiple stresses, particularly low or marginal dissolved oxygen (D.O.) or high temperatures in combination with chronic toxicity (Pippy and Hare 1969, Sniezko 1962).

3. Lesions and ulcers – These appear as open sores or exposed tissue and can be caused by viral (e.g. *Lymphocystis* sp.) and bacterial (e.g. *Flexibacter columnaris*, *Aeromonas* spp., *Vibrio* sp.) infections. Prominent bloody areas on fish should also be included. Small, uncharacteristic sores left by anchor worms and leeches should not be included unless they too, are likewise infected. As with eroded fins, lesions often times appear in areas impacted by multiple stresses, particularly marginal D.O. in combination with sublethal levels of toxics.
4. Tumors – These result from the loss of carefully regulated cellular proliferative growth in tissue and are generally referred to as neoplasia (Post 1983). In wild fish populations, tumors can be the result of exposure to toxic chemicals. Baumann et al. (1987) identified polynuclear aromatic hydrocarbons (PAHs) as the cause of hepatic tumors in brown bullheads in the Black River (Ohio). Viral infections (e.g. *Lymphocystis*) can also cause tumors. Parasites (e.g. *Glugea anomala*, and *Ceratomyxa hasta*; Post 1983) may cause tumor like masses, but these should not be considered as tumors. Parasite masses can be squeezed and broken between thumb and forefinger; whereas true tumors are firm and not easily broken (P. Baumann, personal communication).
5. Anchor worm (*Lernaea cyprinacea*) – This is a common parasitic copepod and can be identified by the presence of an adult female which appears as a slender worm-like body with the head attached (buried) in the flesh of the fish. A small, characteristic sore is left after the anchor worm detaches. Attachment sites are included in the determination of light and heavy infestations. If the formed attachment site becomes infected and enlarged as the result of an infection, it should be recorded as a lesion.
6. Black spot – This disease is common to fish and is caused by the larval stage of a trematode parasite (e.g. *Uvulifer ambloplitis* and *Crassiphiala bulboglossa*). They are easily identified as small black cysts (approximately the size of a pin head) on the skin and fins. Black spot has been reported as being most prevalent on fish inhabiting relatively shallow stream and lake habitats which have an

abundance of aquatic vegetation with snails and fish eating birds, 2 of its intermediate animal hosts. It may also increase in frequency in mildly polluted streams or where fish are crowded due to intermittent pooling.

7. Leeches – These parasites belong to the family Piscicolidae and are usually greenish brown in color and 5-25 mm long (Allison et al. 1977). Leeches can be identified by the presence of 2 suckers (one on each end) and the ability to contact or elongate their body. They may occur almost anywhere on the external surface of the fish, but are most frequently seen on the anterioventral surface of bullheads (*Ictaluras* spp.). Field investigators should become familiar with the small sores or scars left by leeches as these are included in the determination of light and heavy infestations. If these sores become enlarged and infected they are also regarded as lesions. Leeches are seldom harmful to fish unless the infestation is very heavy.
8. Fungus – There is a growth that can appear on a fish's body as a white cottony growth and is most frequently caused by *Saprolegnia parasitica*. This fungus usually attacks an injured or open area of the fish and can eventually cause further disease or death.
9. Ich or *Icthyophthirus multifilis* – This is a protozoan that manifests itself on a fish's skin and fins as a white spotting. This disease rarely occurs in wild fish populations.
10. Popeye – This disease is generally identified by bulging eyes and can be caused by gas accumulation in areas where the water is gas supersaturated. It occurs most frequently in Ohio as the result of fluid accumulation from viral infection, nematodes (*Philometra* sp.), or certain trematode larvae (Rogers and Plumb 1977).

Information on external anomalies is recorded because many are either caused or exacerbated by environmental factors and often times indicate the presence of multiple, sublethal stresses. Komanda (1980) found that morphological abnormalities are uncommon in unimpacted, natural fish populations. The effects of temperature, salinity, dissolved oxygen, diet, chemicals, organic wastes, etc, especially during the ontogeny and larval stages of fished can be the cause of many types of anomalies (Berra and Au 1981). The presence of anomalies on fish may act as an index of pollution stress. A high frequency of DELT anomalies (deformities, eroded fins, lesions, and tumors) is a good indication of stress caused by sublethal

stresses, intermittent stresses, and chemically contaminated substrates. The percent DELT anomalies is a metric of the IBI (Ohio EPA 1987). Field investigators are urges to refer to texts on fish health for further information and pictures of specific anomalies. If necessary, affected fish should be preserved for laboratory examination.

Table 1. Anomaly codes utilized to record external anomalies on fish.

Anomaly code	Description of the anomaly
D	Deformities of the head, skeleton, fins, and any body parts.
E	Eroded fins.
L	Lesions, ulcers.
T	Tumors.
M	Multiple DELT anomalies (e.g. lesions, tumors, etc.) on the same individual fish.
AL	Anchor worm – light infestation: fish with 5 or fewer attached worms and/or previous attachment sites.
AH	Anchor worm – heavy infestation: fish with 6 or more attached worms and/or previous attachment sites.
BL	Black spot – light infestation: spots do not cover most of the body with the average distance between spots greater than the diameter of the eye.
BH	Black spot – heavy infestation: Spots cover most of the body and fins with the average distance between spots less than or equal to the eye diameter.
CL	Leeches – light infestation: Fish with 5 or fewer attached leeches and/or previous attachment sites.
CH	Leeches – heavy infestation: Fish with 6 or more attached leeches and/or previous attachment sites.
F	Fungus.
I	Ich (*Icthyophthirus multifilis*).
N	Blind – one or both eyes; includes missing and grown over eyes (does not include eyes missing due to Popeye disease).
S	Emaciated (poor condition, thin, lacking form).
P	External parasites (other than those already specified).
W	Swirled scales.
Y	Popeye disease.
Z	Wound, other, not included above.

Stream Fish Community Data Sheet: DATE:_____ OBS:_____
LOCATION:_____ Water body name:_____
Sampling method: ___Backpack; ____Tow boat; _____Seine; _____Other
Actual shock time:____sec; Volts:____; Amps:____; Waveform: (AC)(DC)(Pulsed DC)
Seine: __Bag; __Straight line; Seine length:_____(ft); Mesh size:____(in): # hauls:____
% clouds:_____; Secchi depth:_____; Flow level:_____
Start time:_____Start temp:_____; End time_____ End temp:_____
Comments:_____
_____

Species	Count (tally) 0-3"	4-6"	7-9"	10-12"	13-15"	16-18"	19-21"	22+"	Anomaly code	# affect.

Anomaly codes: **D**=deformities, **E**=eroded or frayed fins, **L**=lesions or ulcers, **T**=tumors, **M**=multiple DELTS on same fish**, AL**=anchor worm-light, **AH**=anchor worm-heavy, **BL**=black spot-light, **BH**=black spot-heavy, **CL**=leeches-light, **CH**=leeches-heavy, **F**-fungus**, I**=Ich, **N**=blind, **S**=emaciated, **P**=external parasites, **Y**=popeye, **W**=swirled scales, **Z**=wound, other (describe)

Date data entered:_____ by:____ Corresponding record #s:_____ Date data checked:_____ by:_____

Stream Benthic Macroinvertebrate Community survey data sheet.

DATE:_____ OBS:_____ Water Body name:_____

LOCATION:_____ START TEMP:_____END TEMP:_____

Rain:_____ GPS Coordinates of downstream starting point:_____ % CLOUDS:____

Turbidity:_____ Overall sampling effectiveness:_____ Flow level:_____

Semi-Quantitative (Modified-Hess / Surber / Artificial Substrate) Sampling:

Sampling gear used:_____

Preservative used:_____

Replicate sample ID #	#1	#2	#3
Unique sample ID #			
Dominant form of periphyton growth*			
Amount of periphyton growth**			
Amount of sedimentation/embeddedness**			
Amount of macroinvertebrate colonization**			
Other comments			

* FA=Filamentous Algae Growth; NF=Non-filamentous Algae Growth.

** LT (light) < 25% of substrate surface effected; MD (moderate) 25-50% effected; MH (moderately heavy) 51-75% effected; & HV (heavy) > 75% effected.

Qualitative, Multi-Habitat Sampling

Sampling gear used:_____

Begin time:_____ End time:_____ Total sampling minutes:_____

Date data entered:_____ by:____ Corresponding record #s:_____ Date data checked:_____ by:_____