Chapter Sixteen
Fish Monitoring
Lakes

The Fisheries Section 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 "Statewide Biological Sampling Plan" which was co-written by J. Larscheid and L. Mitzner with input from M. Conover, D. Bonneau, K. Hill, J. Hudson, S. Grummer, M. Flammang, J. Wahl, L. Miller, M. McGhee, S. Waters, and D. McWilliams.

IOWA FISH MONITORING IN LAKES:

Within the permanent sampling plot, any non-wadeable pond or lake should be searched for all fish species using this protocol. In some of these plots a water habitat will be the focal point. In these plots, it is anticipated that a large water body will need to be sampled. In other plots, it may be that only a small water body will need to be surveyed. Regardless of primary habitat classification, some wetlands on the property may need to be surveyed using this protocol depending on the size and type of the wetland. For example, a large lake within 500 m of the center point chosen using the protocol in chapter 3 (Landscape Characteristics) in the forested habitat class would still be surveyed using this protocol. Water bodies that are shallow enough to be surveyed using a back-pack shocker should be examined following the protocol in Chapter 15 (Fish Monitoring in Wade-able Streams). The protocol described in the current chapter is for deeper water bodies.

SURVEY METHODS:

Sampling in lakes and deeper ponds will occur between September and October to allow for cooler surface waters so fish are more likely to be found using the techniques. In general, sampling will occur between 8 am and 5 pm. By electroshocking only during these hours, surveys will be standardized to allow comparisons on a capture-per-unit-effort basis. Trends as to fish abundance are usually evaluated based upon the number of fish sampled per minute of actual shocking time. Lake and pond water bodies should be visited 3 times during these 2 months.

<u>Electrofishing</u>

DC electrofishing boats will be the primary sampling tool on ponds and lakes. Each DC shocking boat will have 16, ½ inch droppers. Dropper exposure will be based on the measured conductivity (umhos) such that increasing conductivity will result in decreasing dropper exposure as outlined in Reynolds (1996). Electrofishing is most effective in shallow water and selects species associated with shoreline or shallow water habitats. Once sampling locations have been chosen, they should be georeferenced and diagramed on a map to ensure that future sampling occurs in the same area. Within the water body, areas should be chosen (and mapped for future data collection) for searches that contain a variety of structure and habitat types. Bays, points, stumps, aquatic vegetation, and the faces of dams work well for black bass, bluegill, crappie, and several other species depending on the lake being surveyed.

The total amount of time spent shocking (meaning the amount of time that electricity is sent into the water, not including times when the current is stopped), will vary with the size of the water body as follows:

Lake or pond size	Effort in minutes
< 100 acres (40.5 ha)	30-90
100 – 500 acres (40.5-202 ha)	60-120
> 500 acres (202 ha)	>90

Shocking runs are to be conducted in 15 minute segments (Pearson 1993, NYSBF 1989) and divided into at least 3 runs of similar distances so that variability can be calculated. This will allow a minimum of 3 runs using 45 minutes per lake. The track taken should be recorded using a GPS unit.

Dip-nets with a small mesh size (3/16 inch (4.76 mm) or smaller) should be used.

Trawling

Recent work from Missouri has indicated that a trawling device will be effective for catching small bodied fish in a variety of habitats (Herzog et al. 2005). This method entails using a modified two-seam balloon trawl, also called a Missouri trawl. As of October 2006, Missouri Department of Conservation staff (who designed the system) was advocating Innovative Net Systems (http://www.innovativenetsystems.com/) for the supplier of the trawl (David Herzog, personal communication). The company has several designs, but MDC recommends either the Missouri trawl or the Armadillo-Herzog (AH) trawl. The primary difference in the 2 trawls appears to be that the AH trawl is made of more durable materials (and is therefore more expensive).

The trawls should be pulled through the water moving downstream. The trawl should just barely move faster than the current. It can be pulled by 2 people in shallow water or by a boat. If pulled by a boat, it should be attached to the front of the boat and the boat should move backwards downstream at a speed slightly greater than that of the current. Be sure to GPS the locations of each haul's start and stop (or, alternatively to record the track taken as the boat moves. Each haul should take between 3 and 5 minutes before the net is pulled aboard and emptied into the holding buckets. These data will be quantified by time as in fish captured per unit of time.

Fish Handling

All fish captured with either of the above methods will be placed into holding tanks or buckets. Make sure the fish in the holding buckets or tanks have fresh water and an air bubbler to limit mortality. These data should be collected (and identified as such on the data sheet) for each electrofishing run and net-haul. At pre-determined stopping points, 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 captured fish, regardless of size (i.e. those less than 1 inch in size should also be identified 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 at the end of this protocol. A minimum of 50 fish should be measured for each species captured. Lengths should be measured to the nearest 1 mm. The rest of the captured fish must be counted to obtain valid catch per unit effort information in the data set. These counts should be grouped by length class. Ideally the first 10 fish in each length class will be measured for exact length with the remaining grouped together (e.g., 53, 54, 51, 52, 55, 53, 53, 57, 59, 53 and 120 additional fish in the 50-60 mm group).

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

In some situations it may be necessary to collect tissue for age-growth calculations. Most likely, this will be rare for the MSIM program and will only be done at the request of a scientist willing to do the lab work and analysis. All aging structures/tissues collected should be placed into scale envelopes on which the following information has been recorded: site name, sampling gear used, date of sampling, species, length, weight, and any comments. At the end of each day the scale envelopes should be spread out and allowed to dry completely. This is especially important for spines which can go rancid quickly if not allowed to dry.

HABITAT AND PLANT COMPOSITION DATA COLLECTION:
Environmental data collected the day of sampling should include: surface water temperature, 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.

See chapter 20 for information on aquatic habitat measurements. As the same areas will be searched for amphibians, fish, dragonflies & damselflies, and/or mussels, no additional habitat data is expected to be collected under the fish in lakes protocol. However, fisheries technicians should coordinate with other crews to ensure that all needed habitat data is collected.

EQUIPMENT NEEDED:
Water collection jars
Dip nets
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
DC Electroshocking boat
Trawling equipment

STAFF & TRAINING:
Two weeks of training (beginning on August 15) is recommended and should include 1) field guide use and id, 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 trawling 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 monitored through 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 trapping day, field crew pairs should review data sheets to ensure all information present. At the end of the week, the field crew leader should review the data sheets for ID, escape and mortality rates, and legibility. Be sure to keep data collected by different methods separate. Also be sure to keep the locations of the data collection labeled.

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. For additional information on the PAO techniques, see Chapter 5 (Data Analysis).

The data collected should allow the estimate of the following community parameters of the fish sample:

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

The methods employed do not provide quantitative information suitable for fish population 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.

Electro-fishing can be dangerous. All personnel need to be trained in the use of this equipment. Working in aquatic situations can be dangerous. Technicians should be cautious of slippery substrates and be aware of the speed of water flow. Sampling should be suspended during inclement weather, including heavy rain or lightning storms. If a person is swept into the water when wearing chest waders, it is possible that the air trapped in the bottom of the waders will force the person to travel 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 to dry for 3-4 days between sites. This may be impractical given the short time frame available for fish surveying in Iowa. As an alternative, it may be best to rinse the waders and 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). These animals are those that may be potentially encountered along an aquatic environment. 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 additional, more common, species 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
Silver chub	*Macrybopsis storeriana*	Larger interior rivers statewide

Target species continued:

Common Name	Scientific Name	Habitat
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	*Annicrypta 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:

Fyke Nets

Fyke nets are passive gear that sample fish by entrapment. Fyke nets tend to be selective for cover seeking, mobile species (Neilson and Johnson 1983, McWilliams et al. 1974). Nets used in this procedure should be standardized by size to ensure continuity across areas. All sampling will be conducted using 2 ft x 4 ft (60.96 cm x 121.92 cm) frames with 7 hoops of 2 ft (60.92 cm) diameters enclosed with ¾ inch (1.91 cm) bar mesh netting for larger fish or 3/16 inch (4.79 mm) mesh for smaller fish.

Fyke nets are typically deployed in shoreline habitats where the water is about 4 feet (1.22 m) deep at the frame. Sampling sites should be geo-referenced and mapped to ensure the same areas are sampled through time. The number of nets set should vary with the size of the water body as follows:

Waterbody size	Effort (nets/night)
< 100 acres (40.5 ha)	3-15
100 – 500 acres (40.5-202 ha)	5-20
> 500 acres (202 ha)	7-28

Typically, nets are set for just one night, meaning that up to 28 net sets may be needed per wetland. Fyke nets are set overnight and emptied each day. The time of setting and raising should be recorded.

Spring Sampling

It should be left to the biologist's discretion to decide if supplemental sampling for fish should be conducted in the spring for certain water bodies.

Minnow Traps:

Minnow traps may be an effective way to find additional fish. They 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 breathe. 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 in common to fish in Ohio 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 other 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.

Lake or pond Fish Community Data Sheet: DATE:__________OBS:________________
LOCATION:_______________________ Water body name:______________________
Sampling method: ___shock boat; _____fyke net; ________________Other
Actual shock time:____sec; Volts:____; Amps:____; Waveform: (AC)(DC)(Pulsed DC)
Fyke net: net length:_____(ft); Mesh size:____(in); % clouds:____; Secchi dp:_______;
Flow level:_______; Start time:_____Start temp:_____; End time_____ End temp:_____
Comments:____
____

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

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 :_____

Back of Lake or pond Fish Community Data Sheet:
DATE:__________OBS:________________ LOCATION:_______________________

Fish tags used here:

Species	Tag numbers:

Species	Length (cm)

Species	Length (cm)

Lengths and weights for subsample of fish at LOCATION:________________

DATE:______________
OBS:____________________________

Species	Length (cm)

Species	Length (cm)