Quality Assurance Project Plan: Water Quality and Sediment Chemistry, and Bioassessment Monitoring of the North Branch Chicago River Watershed

Lake County and Cook County, Illinois

North Branch Chicago River Watershed Workgroup
500 W. Winchester Rd.
Libertyville, IL 60048

A1. Title and Approval Page

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Effective Date: May 1, 2018

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500 W. Winchester Rd.
Libertyville, IL 60048

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Michelle Rousey 4/3/19

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List of Acronyms

BCG – biological condition gradient
CFR – Code of Federal Regulations
CPUE – catch per unit effort
CV – coefficient of variation
CWA – Clean Water Act
DC – direct current
DELT – deformities, eroded fins and body parts, lesions, and tumors
DO – dissolved oxygen
EB – Executive Board
EDD – Electronic Data Deliverable
GPP – Generator Powered Pulsator
GPS – Geographical Positioning System
HDPE - High Density Polyethylene
IBI – Index of Biotic Integrity
ICI – Invertebrate Community Index
IDNR – Illinois Department of Natural Resources
Illinois EPA – Illinois Environmental Protection Agency
INHS – Illinois Natural History Survey
LCS – Laboratory Control Spike
LCSMC – Lake County Stormwater Management Commission
LIMS – Laboratory Information Management System
LVI – Low Volume Initiative
MAIS – Macroinvertebrate Aggregated Index for Streams
MBI – Midwest Biodiversity Institute
MDL – Method Detection Limit
MS – Matrix Spike
MSD – Matrix Spike Duplicate
MWRD - Metropolitan Water Reclamation District of Greater Chicago
NBWW – North Branch Chicago River Watershed Workgroup
NELAP – National Environmental Laboratory Accreditation Program
NIST – National Institute for Standards and Technology
NPDES – National Pollutant Discharge Elimination System
NSWRD – North Shore Water Reclamation District
OCDL – Ohio Credible Data Law
Ohio EPA – Ohio Environmental Protection Agency
OSUMB - Ohio State University Museum of Biodiversity
PCBs – Polychlorinated Biphenyls
PI – Principal Investigator
PNAs – Polynuclear Aromatic Hydrocarbons
POTW – Publicly Owned Treatment Works
QAO – Quality Assurance Officer
QAPP – Quality Assurance Project Plan
QA/QC – Quality Assurance and Quality Control

QHEI – Qualitative Habitat Evaluation Index
QMP – Quality Management Plan
RL – Reporting Limits
RPD – Relative Percent Difference
SNAP – Stream Nutrient Assessment Procedure
SOP – Standard Operating Procedure
SU – Standard Unit
TKN – Total Kjeldahl Nitrogen
TMDL – Total Maximum Daily Load
TSS – Total Suspended Solids
U.S. EPA – United States Environmental Protection Agency
UTM – Universal Transverse Mercator
VOCs – Volatile Organic Compounds
TVSS – Total Volatile Suspended Solids
y-o-y – young of the year

Introduction

The Illinois Environmental Protection Agency (Illinois EPA) requires the development of a Quality Assurance Project Plan (QAPP) for any activity involving the collection and analysis of environmental data. A QAPP presents the policies and procedures, organization, objectives, quality assurance requirements and quality control activities designed to achieve the type and quality of environmental data necessary to support project or program objectives. It is the policy of Illinois EPA that no data collection or analyses will occur without an approved QAPP or equivalent documentation, per the agency Quality Management Plan (QMP). All in-house and external environmental data collection activities are subject to this requirement. All contracts must address quality assurance requirements (e.g., data quality and reporting requirements) when those contracts pertain to, or have an impact on, data collection or analysis activities. Additionally, all grants and contracts need to address quality assurance requirements specified in applicable state acquisition or procurement regulations. The North Branch Chicago River Watershed Monitoring QAPP presented herein follows United States Environmental Protection Agency (U.S. EPA) requirements and guidance for the development of a project specific QAPP as detailed in the documents EPA Requirements for Quality Assurance Project Plans (U.S. EPA 2001) and EPA Guidance for Quality Assurance Project Plans (U.S. EPA 2002).

Group A: Project Management Elements

A.3: Distribution List

The proposed project is of interest and potential use to Illinois state agencies and non-governmental organizations, each with specific interests in the protection and restoration of aquatic ecosystems. The following agency staff are recognized as technical advisers given their regional and/or statewide knowledge and expertise and the QAPP will be distributed to them electronically. The original, approved QAPP with signatures will be retained by the Monitoring & Water Quality Impairment Abatement Committee Chair of the North Branch Chicago River Watershed Workgroup (NBWW).

Illinois EPA, Roy Smogor, Springfield
Illinois EPA, Chris Davis, Springfield
Illinois Department of Natural Resources (IDNR), Steve Pescitelli, Plano

In addition, the following entities will also be included in the distribution list as follows:

North Branch Chicago River Watershed Workgroup (all members)
Cook County Forest Preserve District
Illinois Environmental Protection Agency (Illinois EPA), Michelle Rousey
Lake County Forest Preserve District
Lake County Stormwater Management Commission (LCSMC)
Lake County Public Works
Lake County Department of Transportation
Lake County Planning, Building and Development

Metropolitan Water Reclamation District of Greater Chicago (MWRD)
Midwest Biodiversity Institute (MBI), Chris O. Yoder and Peter A. Precario
North Shore Water Reclamation District (NSWRD)
North Branch Chicago River Watershed Municipalities

In order to maintain open communication amongst interested parties, below is a table of contact information.

Table 1: NBWW Contact Information Associated with the Functional Organization for Bioassessment Monitoring

Name	Role	Organization	Phone	Email
Brandon Janes	NBWW President	Village of Deerfield	847-719-7447	bjanes@deerfield.il.us
Rob Flood	Monitoring Committee Chair	NSWRD	847-623-6060	roflood@northshorewrd.org
Mike Warner	Technical Agent	LCSMC	847-377-7716	mwarner@lakecountyil.gov
Chris Yoder	Bioassessment and Analysis Contractor	Midwest Biodiversity Institute	614-457-6000 Ext. 1102	cyoder@mwbinst.com
Toni Favero	Water and Sediment Chemistry Contractor	NSWRD Laboratory	847-623-6060	tofavero@northshorewrd.org
Toni Favero	Quality Assurance Manager	NSWRD Laboratory	847-623-6060	tofavero@northshorewrd.org

A.4: Project/Task Organization

The bioassessment of the North Branch Chicago River Watershed will consist of biologic sampling including fish, macroinvertebrates and habitat, continuous dissolved oxygen (DO) monitoring, benthic periphyton sampling, and water column and sediment sampling to evaluate ecosystem quality and stressors. All phases of the bioassessment will be coordinated and overseen by the Midwest Biodiversity Institute (MBI). The Research Director, MBI will serve as the Principal Investigator (PI) and overall project coordinator. Senior consultant staff will be assigned various aspects of the project under the oversight of the PI. The MBI PI will also be directly responsible for maintenance of the QAPP through the project period of May 2018 through June 2020. Fish, macroinvertebrates, habitat, and sediment collection as well as continuous DO monitoring and benthic periphyton sampling will be completed by MBI. The water column chemistry collection and analysis and sediment analysis will be completed by the NSWRD Laboratory. Toni Favero, the laboratory supervisor, will coordinate and oversee the water column and sediment laboratory analysis. MBI will be responsible for analyzing all data and compiling a final report. A functional table of organization appears in Figure 1.

Advice and assistance with the design of the proposed study has been sought and will continue to be provided by members of the NBWW, Illinois EPA, and DNR. Each agency and organization

will benefit from the data and analyses produced by the proposed study as it affects key water quality management issues such as National Pollutant Discharge Elimination System (NPDES) permitting, stormwater management, Total Maximum Daily Load (TMDL) development and assessment, and standards setting. Users of this study will benefit from the results and how it relates to the development of water quality and biological criteria that are protective of the indigenous aquatic fauna.

The Illinois EPA Bureau of Water Quality Assurance Officer (QAO) will review and approve this QAPP as meeting the QAPP requirements in USEPA's publication QAS/R-5. The QAO will conduct audits if deemed necessary.

A.5: Problem Definition and Background

The proposed study will document the existing status of the rivers and streams in the watersheds of the North Branch Chicago River within Lake County and Cook County, Illinois. The study will emphasize the direct assessment of biological assemblages by sampling fish and macroinvertebrates using standardized sampling and assessment methodologies. In addition to determining aquatic life status, the project will also ascertain the associated causes and sources associated with biological impairments by using paired chemical, physical, and other stressor data and information within a systematic analytical process detailed in a comprehensive Monitoring Strategy (Appendix A).

A.6: Project Description

This study will be performed in the North Branch Chicago River and tributaries within Lake County and Cook County located in the northeastern region of Illinois and in accordance with the Monitoring Strategy (Appendix A including sampling station map and frequency of sampling). Biological sampling will consist of utilizing two assemblages, fish and macroinvertebrates. Water column and sediment chemistry data, continuous DO monitoring and benthic periphyton sampling will also be collected at the same monitoring sites. Sediment data, continuous DO monitoring and benthic periphyton sampling will be collected concurrently with biological sampling, and water column chemistry data will be collected annually with increased frequency. This QAPP specifies the methods and equipment that will be used for all sampling. These estimates are based on the anticipated application of the specific protocols that will likely be used – some adjustments may be required based on the pre-survey reconnaissance and during sampling. A Qualitative Habitat Evaluation Index (QHEI) will be collected at each fish sampling site and will be completed by the crew leader (Appendix B). Field chemical/physical parameters will be collected using a commercially available field meter capable of measuring temperature, dissolved oxygen (DO), conductivity, and pH. Additional chemical parameters will be sampled in the water column and sediment as detailed in Appendix A. Biological laboratory methods will follow the assigned methods and will also include fish voucher verification and macroinvertebrate taxonomy to the lowest practicable level as specified by the scope of work. Habitat, biological, and water column and sediment chemistry methods and their specifications are described in detail in Appendices C-H.

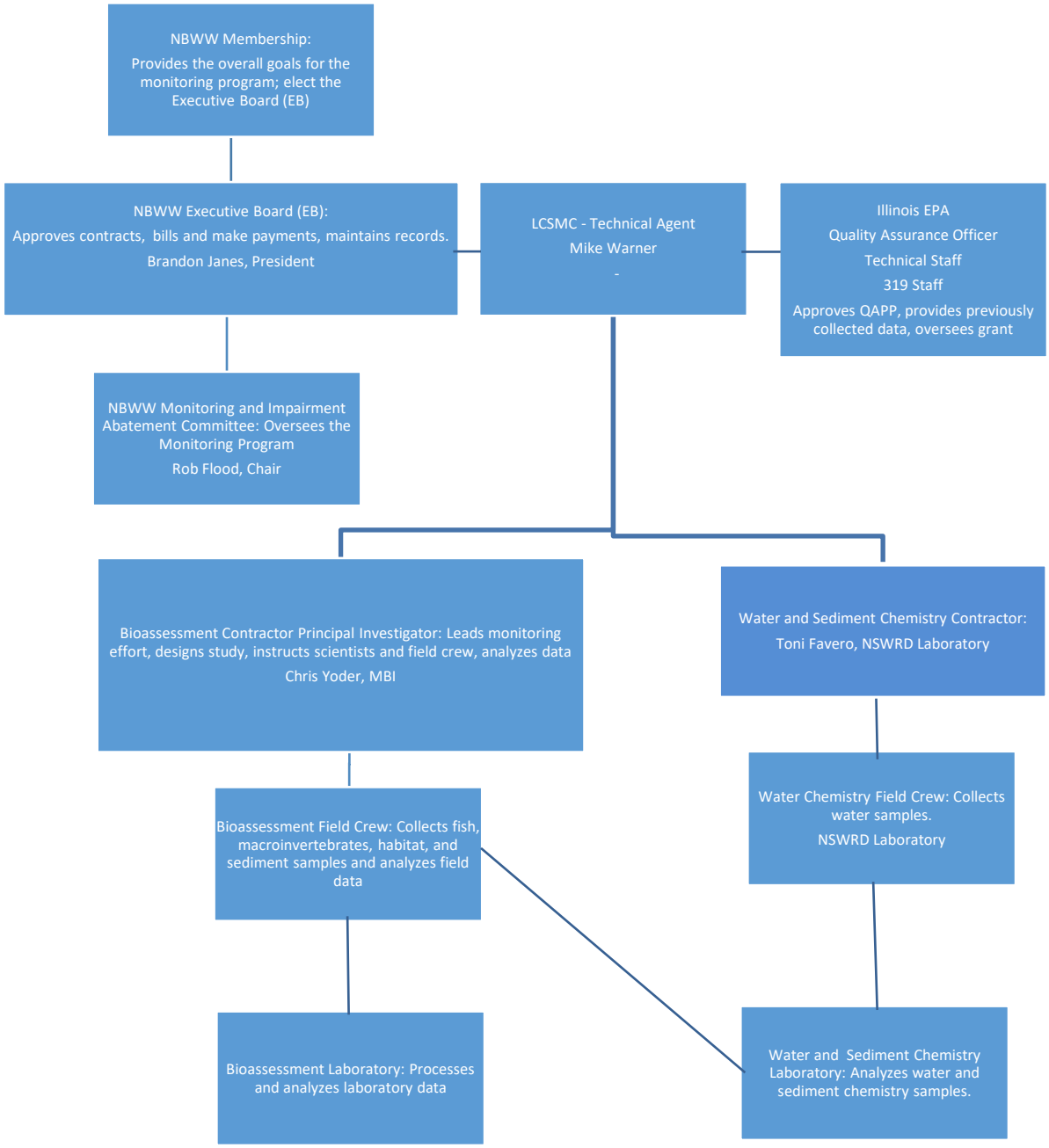


Figure 1: Functional organization for project implementation and management.

A.7: Quality Objectives and Criteria

The accuracy and precision of the biological assessments and water column and sediment investigations is a product of the congruence of the methods and their execution. The properties of the biological assemblage data typified by this study has been previously documented (Ohio EPA 1989, Rankin and Yoder 1990, Fore *et al.* 1993). The methods employed for water column and sediment data collection as well as continuous DO monitoring and benthic periphyton sampling are accepted as common practice by the Illinois EPA due to consistency and replicability.

The types of methods that will be applied in this study have been shown to minimize variability in assessment results, while the sources of variability are known and controlled. This makes the results more useful and reliable for use by Illinois EPA, Illinois DNR, and other organizations. An important goal for bioassessment programs is to employ methods and equipment which are sufficiently effective so as to produce a sufficiently representative sample (accuracy), ensure reproducibility (precision), do so with a reasonable effort (cost-effective), and minimize potential bias from different operators (variability), thus making the results of the assessment comparable. The attributes of each media and the associated data quality objectives and criteria are addressed within this section.

Data Attributes - Fish Assemblage:

The basic attributes of fish data are counts and weights of fish delineated either individually or in the aggregate by species. Species level taxonomy is the minimum data quality objective and identifications to subspecies will be determined when appropriate. Scientific nomenclature will follow that adopted by the American Fisheries Society (AFS; Nelson *et al.* 2004). The historical and spatial distribution of the Illinois ichthyofauna and taxonomy is well described in Smith (1979) and by the Illinois Natural History Survey (INHS)[1]. Information will also be recorded about the occurrence of external anomalies, diseases, parasites, and other abnormalities that are observed on each fish that is weighed and/or counted following the methods used by Ohio EPA (1989) and further described by Sanders *et al.* (1999). Qualitative habitat data will also be produced at each fish sampling location using the methodology originally developed by Rankin (1989, 1995; Ohio EPA 2006; Appendix B) and with the most recent updates by MBI. This includes the characterization and categorization of habitat attributes including substrate types and quality, cover types and extent, channel morphology and modification, riparian and bank composition and condition, pool-run-riffle quality and extent, and local gradient.

Data Attributes - Macroinvertebrate Assemblage:

The basic attributes of the macroinvertebrate data to be produced by the proposed study are counts of each taxa identified to the lowest taxonomic level that is practical for most orders and families. All samples will be processed in the laboratory following Illinois EPA and Ohio EPA (1989) methods. Keys specified in Ohio EPA (1989) and by Illinois EPA will be used to make the identifications.

[1] The Fishes of Illinois maintained by INHS at: http://www.inhs.uiuc.edu/animals_plants/fish/

Data Attributes – Field Water Quality:

The basic attributes of the data to be produced by field measurement are listed in Table 2. The parameters include temperature (°C), dissolved oxygen (DO; mg/l), conductivity (μS/cm^2), and pH (Standard Units) and these will be measured at each biological sampling site at the time of each sampling event. Temperature and conductivity are important for determining what electrofishing equipment is needed and what settings are required. The pH and DO data are intended to supplement the more frequent data provided by the chemical sampling crew. These parameters are useful for conditions to support aquatic life and measuring the secondary effects of nutrient enrichment. The conductivity data is also useful for evaluation of dissolved solids in runoff (e.g., chlorides from road salt, etc.).

Table 2: Precision, accuracy, and measurement range for field parameters.

Parameter	Meter	Precision	Accuracy @20°C	Measurement Range
pH	YSI 556	± 0.2 S.U.	± 0.01 S.U.	0-14 S.U.
Dissolved Oxygen	YSI 556	± 0.01 mg/l	± 0.3 mg/l	0-20 mg/l
Conductivity	YSI 556	± 2%	± 2%	0-4000 μS/cm
Temperature	YSI 556	± 0.5°C	± 0.5°C	0-100°C

Data Attributes – Water Column and Sediment Chemistry:

The basic attributes of water column and sediment chemistry data are measures of concentration by parameter. These sampled parameters are listed in Table 3 and Table 4 for water column and sediment, respectively, and are analyzed by the NSWRD Laboratory and its sub-contractors within a general 20 business days turn-around time. All parameters are analyzed using the laboratory standard operating procedures (SOPs) and analytical methods listed in Tables 3 and 4. SOPs are maintained at the laboratory and are available upon request. The samples are reported based on method detection limits (MDL) and reporting limits (RL) that are specified and are compared to the minimum measurement criteria addressed in the *Sensitivity* section. All data is also compared to data quality objectives including the limit of sensitivity, precision, and accuracy.

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Conventional/Field Parameters									
pH	90% - 110%	10%	0.1 SU	SM 4500-H+ B	42	pH Analysis-Electrometric Method	n/a	0.1	SU
Dissolved Oxygen	90% - 110%	10%	0.1 mg/L	SM 4500-O G	11	Dissolved Oxygen Membrane Electrode	n/a	0.1	mg/L
BOD5	90% - 110%	20%	2 mg/L	SM 5210 B	3B	Biochemical Oxygen Demand	n/a	2	mg/L
Temperature	90% - 110%	10%	0.1 °C	SM 2550 B	QA-4	Temperature	n/a	0.1	°C
Conductivity	90% - 110%	20%	1 µmhos/cm	SM 5210 B	26	Conductivity	n/a	1	umhos/cm
Total Suspended Solids (TSS)	90% - 110%	10%	1 mg/L	SM 2540 D	25B	Total Suspended Solids	n/a	1	mg/L
Volatile Suspended Solids (VSS)	90% - 110%	10%	1 mg/L	SM 2540 G	25B	Total Suspended Solids	n/a	1	mg/L
Metals									
Arsenic	75% - 125%	20%	1 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.23	1.0	ug/L
Barium	75% - 125%	20%	2.5 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.73	2.5	ug/L
Calcium	85% - 115%	20%	0.50 mg/L	SM 3111 B	17B	Metals Analysis by Flame Atomic Absorption	0.027	0.50	mg/L
Cadmium	75% - 125%	20%	0.5 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.17	0.50	ug/L
Chromium	75% - 125%	20%	5 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	1.1	5.0	ug/L
Iron	75% - 125%	20%	100 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	47	100	ug/L
Copper	75% - 125%	20%	2 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.5	2	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Magnesium	85% - 115%	20%	0.50 mg/L	SM 3111 B	17B	Metals Analysis by Flame Atomic Absorption	0.019	0.5	mg/L
Lead	75% - 125%	20%	0.5 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.19	0.5	ug/L
Nickel	75% - 125%	20%	2 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.63	2	ug/L
Sodium	85% - 115%	20%	0.50 mg/L	SM 3111 B	17B	Metals Analysis by Flame Atomic Absorption	0.22	0.5	mg/L
Silver	75% - 125%	20%	0.50 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	0.12	0.5	ug/L
Low Level Mercury	75% - 125%	20%	0.50 ng/L	1631E	n/a	Preparation and Analysis of Mercury in Aqueous and Solid Samples by Cold Vapor Atomic Fluorescence (Method 1631E)	0.14	0.5	ng/L
Zinc	75% - 125%	20%	20 ug/L	200.8	n/a	Metals Analysis –Inductively Coupled Argon Plasma/Mass Spectrometry by EPA 200.8 Rev 5.4	6.9	20	ug/L
Nutrients									
Ammonia (NH_3-N)	90% - 110%	20%	0.10 mg/L	SM 4500-NH3 D	21	Nitrogen Ammonia – Ion Selective Electrode	0.015	0.1	mg/L
Chloride	90% - 110%	20%	2 mg/L	SM 4500-CL-E	7C	Chloride - Automated Ferricyanide Method	0.85	5	mg/L
Total Nitrates (NO_2+NO_3)	90% - 110%	20%	0.10 mg/L	353.2 Rev 2.0	21D	Nitrate-Nitrite Nitrogen - Automated Cadmium Reduction Method	0.046	0.1	mg/L
Total Kjeldahl Nitrogen (TKN)	90% - 110%	20%	0.40 mg/L	SM 4500 N org C, EPA 351.2 Rev 2.0	21F	Total Kjeldahl Nitrogen	0.091	0.4	mg/L
Total Phosphorus	90% - 110%	20%	0.041 mg/L	365.1 Rev2.0	24A	Total Phosphorus – Low Level, Colorimetric Ascorbic Acid - Automated Method	0.011	0.04	mg/L
Chlorophyll a				10200-H	n/a	Determination of Chlorophyll and Periphyton by Chlorophyll a in Water by Spectrophotometry by 10200 H-2001	n/a	n/a	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Bacteria									
E. Coli	N/A	N/A	1 MPN/100 mL	SM 9223 B	56	Colilert	n/a	1	CFU/100mL
Organics									
Pesticides				8081B	n/a	Gas Chromatography– Pesticides by SW-846 Method 8081A and 8081B			
4,4´-DDD	70% - 125%	20%	0.04 µg/L				0.014	0.041	ug/L
4,4´-DDE	70% - 125%	20%	0.04 µg/L				0.0039	0.041	ug/L
4,4´-DDT	70% - 125%	20%	0.04 µg/L				0.0032	0.041	ug/L
Aldrin	70% - 125%	20%	0.04 µg/L				0.0054	0.041	ug/L
alpha-BHC	70% - 125%	20%	0.04 µg/L				0.0026	0.041	ug/L
alpha-Chlordane	70% - 125%	20%	0.04 µg/L				0.0045	0.041	ug/L
beta-BHC	70% - 125%	20%	0.04 µg/L				0.01	0.041	ug/L
Chlordane	70% - 125%	20%	0.08 µg/L				0.041	0.081	ug/L
delta-BHC	70% - 125%	20%	0.04 µg/L				0.01	0.041	ug/L
Dieldrin	70% - 125%	20%	0.04 µg/L				0.013	0.041	ug/L
Endosulfan I	70% - 125%	20%	0.04 µg/L				0.0042	0.041	ug/L
Endosulfan II	70% - 125%	20%	0.04 µg/L				0.0028	0.041	ug/L
Endosulfan sulfate	70% - 125%	20%	0.04 µg/L				0.012	0.041	ug/L
Endrin	70% - 125%	20%	0.04 µg/L				0.014	0.041	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Endrin aldehyde	70% - 125%	20%	0.04 µg/L				0.0083	0.041	ug/L
Endrin ketone	70% - 125%	20%	0.04 µg/L				0.017	0.041	ug/L
gamma-BHC	70% - 125%	20%	0.04 µg/L				0.0057	0.041	ug/L
Heptachlor	70% - 125%	20%	0.04 µg/L				0.014	0.041	ug/L
Heptachlor epoxide	70% - 125%	20%	0.04 µg/L				0.014	0.041	ug/L
Methoxychlor	70% - 125%	20%	0.08 ug/L				0.023	0.081	ug/L
Toxaphene	70% - 125%	20%	0.40 ug/L				0.2	0.41	ug/L
trans-Chlordane	70% - 125%	20%	0.04 ug/L				0.0073	0.041	ug/L
Polychlorinated Biphenyls (PCBs)				8082A	n/a	Gas Chromatography– PCBs by SW-846 Method 8082 and 8082A			
Aroclor 1016	60% - 120%	20%	0.40 µg/L				0.068	0.41	ug/L
Aroclor 1221	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Aroclor 1232	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Aroclor 1242	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Aroclor 1248	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Aroclor 1254	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Aroclor 1260	60% - 120%	20%	0.40 µg/L				0.071	0.41	ug/L
Total PCBs	60% - 120%	20%	0.40 µg/L				0.2	0.41	ug/L
Polynuclear Aromatic Hydrocarbons (PNAs)				8270D	n/a	Gas Chromatography Mass Spectrometry – Volatiles SW-846 Method 8270D			

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Acenaphthene	60%-110%	20%	0.80 µg/L				0.27	0.86	ug/L
Acenaphthylene	60%-110%	20%	0.80 µg/L				0.23	0.86	ug/L
Anthracene	60%-110%	20%	0.80 µg/L				0.29	0.86	ug/L
Benzo(a)anthracene	60%-110%	20%	0.16 µg/L				0.049	0.17	ug/L
Benzo(a)pyrene	60%-110%	20%	0.16 µg/L				0.085	0.17	ug/L
Benzo(b)fluoranthene	60%-110%	20%	0.16µg/L				0.069	0.17	ug/L
Benzo(g,h,i)perylene	60%-110%	20%	0.80 µg/L				0.32	0.86	ug/L
Benzo(k)fluoranthene	60%-110%	20%	0.16 µg/L				0.055	0.17	ug/L
Chrysene	60%-110%	20%	0.16 µg/L				0.058	0.17	ug/L
Dibenzo(a,h)anthracene	60%-110%	20%	0.24 µg/L				0.044	0.26	ug/L
Fluoranthene	60%-110%	20%	0.80 µg/L				0.39	0.86	ug/L
Fluorene	60%-110%	20%	0.80 µg/L				0.21	0.86	ug/L
Indeno(1,2,3-cd)pyrene	60%-110%	20%	0.16 µg/L				0.064	0.17	ug/L
Naphthalene	60%-110%	20%	0.80µg/L				0.27	0.86	ug/L
Phenanthrene	60%-110%	20%	0.80 µg/L				0.26	0.86	ug/L
Pyrene	60%-110%	20%	0.80 µg/L				0.37	0.86	ug/L
Volatile Organic Compounds (VOCs)				8260B	n/a	Gas Chromatography Mass Spectrometry – Volatiles SW-846 Method 8260B			
1,1,1-Trichloroethane	70% - 130%	20%	1 µg/L				0.38	1	ug/L
1,1,2,2-Tetrachloroethane	70% - 130%	20%	1 µg/L				0.4	1	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
1,1,2-Trichloroethane	70% - 130%	20%	1 µg/L				0.35	1	ug/L
1,1-Dichloroethane	70% - 130%	20%	1 µg/L				0.41	1	ug/L
1,1-Dichloroethene	70% - 130%	20%	1 µg/L				0.39	1	ug/L
1,2-Dibromo-3-chloropropane	70% - 130%	20%	5 µg/L				2	5	ug/L
1,2-Dichlorobenzene	70% - 130%	20%	1 µg/L				0.33	1	ug/L
1,2-Dichloroethane	70% - 130%	20%	1 µg/L				0.39	1	ug/L
1,2-Dichloropropane	70% - 130%	20%	1 µg/L				0.43	1	ug/L
1,3-Dichlorobenzene	70% - 130%	20%	1 µg/L				0.4	1	ug/L
1,4-Dichlorobenzene	70% - 130%	20%	1 µg/L				0.36	1	ug/L
2-Butanone	70% - 130%	20%	5 µg/L				2.1	5	ug/L
2-Chloroethyl vinyl ether	70% - 130%	20%	2 µg/L				0.77	2	ug/L
2-Hexanone	70% - 130%	20%	5 µg/L				1.6	5	ug/L
4-Methyl-2-pentanone	70% - 130%	20%	5 µg/L				2.2	5	ug/L
Acetone	70% - 130%	20%	5 µg/L				1.7	5	ug/L
Acrolein	70% - 130%	20%	100 ug/L				23	100	ug/L
Acrylonitrile	70% - 130%	20%	20 µg/L				4.5	20	ug/L
Benzene	70% - 130%	20%	0.50 µg/L				0.15	0.5	ug/L
Bromodichloromethane	70% - 130%	20%	1 µg/L				0.37	1	ug/L
Bromoform	70% - 130%	20%	1 µg/L				0.48	1	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.									
Parameter	**Accuracy (% Recovery)**	**Precision (Relative Percent Difference [RPD])**	**Limit of Sensitivity ***	**Method**	**SOP**	**SOP Title**	**MDL**	**PQL**	**Units**
Bromomethane	70% - 130%	20%	2 µg/L				0.8	2	ug/L
Carbon disulfide	70% - 130%	20%	2 µg/L				0.45	2	ug/L
Carbon tetrachloride	70% - 130%	20%	1 µg/L				0.38	1	ug/L
Chlorobenzene	70% - 130%	20%	1 µg/L				0.39	1	ug/L
Chloroethane	70% - 130%	20%	1 µg/L				0.51	1	ug/L
Chloroform	70% - 130%	20%	2 µg/L				0.37	2	ug/L
Chloromethane	70% - 130%	20%	1 µg/L				0.32	1	ug/L
cis-1,2-Dichloroethene	70% - 130%	20%	1 µg/L				0.41	1	ug/L
cis-1,3-Dichloropropene	70% - 130%	20%	1 µg/L				0.42	1	ug/L
Dibromochloromethane	70% - 130%	20%	1 µg/L				0.49	1	ug/L
Dichlorodifluoromethane	70% - 130%	20%	2 µg/L				0.67	2	ug/L
Ethylbenzene	70% - 130%	20%	0.50 µg/L				0.18	0.5	ug/L
Methyl tert-butyl ether	70% - 130%	20%	1 µg/L				0.39	1	ug/L
Methylene chloride	70% - 130%	20%	5 µg/L				1.6	5	ug/L
Styrene	70% - 130%	20%	1 ug/L				0.39	1	ug/L
Tetrachloroethene	70% - 130%	20%	1 µg/L				0.37	1	ug/L
Toluene	70% - 130%	20%	0.50 µg/L				0.15	0.5	ug/L
trans-1,2-Dichloroethene	70% - 130%	20%	1 µg/L				0.35	1	ug/L
trans-1,3-Dichloropropene	70% - 130%	20%	1 µg/L				0.36	1	ug/L

Table 3: Precision, accuracy, and limit of sensitivity for water column chemistry parameters.

Parameter	Accuracy (% Recovery)	Precision (Relative Percent Difference [RPD])	Limit of Sensitivity *	Method	SOP	SOP Title	MDL	PQL	Units
Trichloroethene	70% - 130%	20%	0.50 µg/L				0.16	0.5	ug/L
Trichlorofluoromethane	70% - 130%	20%	1 µg/L				0.43	1	ug/L
Vinyl chloride	70% - 130%	20%	0.50 µg/L				0.2	1	ug/L
Total Xylenes	70% - 130%	20%	1 µg/L				0.22	1	ug/L

* The limit of sensitivity is defined by the MDL. All laboratory instruments utilize a calibration defining instrument response. Any measurement falling above the upper calibration range is diluted to fall within range.

The limit of sensitivity, and precision and accuracy measurements for sediment data are detailed in Table 4.

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
Metals									
Metals (21)									
Aluminum (Al)	80% - 120%	20%	20 mg/Kg	6010B	n/a	Metals Analysis – Trace Inductively Coupled Argon Plasma by SW-846 6010B	8.2	20.0	mg/kg, dry
Arsenic (As)	80% - 120%	20%	1.0 mg/Kg				0.34	1.0	mg/kg, dry
Barium (Ba)	80% - 120%	20%	1.0 mg/Kg				0.11	1.0	mg/kg, dry
Beryllium (Be)	80% - 120%	20%	0.40 mg/Kg				0.093	0.40	mg/kg, dry
Boron (B)	80% - 120%	20%	5.0 mg/Kg				0.47	5.0	mg/kg, dry
Cadmium (Cd)	80% - 120%	20%	0.20 mg/Kg				0.036	0.20	mg/kg, dry
Chromium (Cr)	80% - 120%	20%	1.0 mg/Kg				0.50	1.0	mg/kg, dry
Cobalt (Co)	80% - 120%	20%	0.50 mg/Kg				0.13	0.50	mg/kg, dry
Copper (Cu)	80% - 120%	20%	1.0 mg/Kg				0.28	1.0	mg/kg, dry
Iron (Fe)	80% - 120%	20%	20 mg/Kg				10.4	20.0	mg/kg, dry
Lead (Pb)	80% - 120%	20%	0.50 mg/Kg				0.23	0.5	mg/kg, dry
Manganese (Mn)	80% - 120%	20%	1.0 mg/Kg				0.15	1.0	mg/kg, dry
Nickel (Ni)	80% - 120%	20%	1.0 mg/Kg				0.29	1.0	mg/kg, dry
Potassium (K)	80% - 120%	20%	50 mg/Kg				17.7	50.0	mg/kg, dry
Silver (Ag)	80% - 120%	20%	0.50 mg/Kg				0.13	0.50	mg/kg, dry
Sodium (Na)	80% - 120%	20%	100 mg/Kg				14.8	100	mg/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
Strontium (Sr)	80% - 120%	20%	0.50 mg/Kg				0.020	0.50	mg/kg, dry
Vanadium (V)	80% - 120%	20%	0.50 mg/Kg				0.12	0.50	mg/kg, dry
Zinc (Zn)	80% - 120%	20%	2.0 mg/Kg				0.88	2.00	mg/kg, dry
Mercury (Hg)	80% - 120%	20%	0.02 mg/Kg	7471B	n/a	Metals Analysis: Mercury by SW-846 7471A/7471B (Modified) – Determination of Mercury in Stones	0.006	0.02	mg/kg, dry
Organics									
Pesticides				8081B	n/a	Gas Chromatography– Pesticides by SW-846 Method 8081A and 8081B			
4,4´-DDD	50% - 130%	30%	1.7 µg/Kg				1.2	6.3	ug/kg, dry
4,4´-DDE	50% - 130%	30%	1.7 µg/Kg				1	6.3	ug/kg, dry
4,4´-DDT	50% - 130%	30%	1.7 µg/Kg				3.3	6.3	ug/kg, dry
Aldrin	50% - 130%	30%	1.7 µg/Kg				2.6	6.3	ug/kg, dry
alpha-BHC	50% - 130%	30%	1.7 µg/Kg				1.6	6.3	ug/kg, dry
alpha-Chlordane	50% - 130%	30%	1.7 µg/Kg				3.2	6.3	ug/kg, dry
beta-BHC	50% - 130%	30%	1.7 µg/Kg				1.9	6.3	ug/kg, dry
Chlordane, technical	50% - 130%	30%	6.7 µg/Kg				12	25	ug/kg, dry
delta-BHC	50% - 130%	30%	1.7 µg/Kg				2	6.3	ug/kg, dry
Dieldrin	50% - 130%	30%	1.7 µg/Kg				0.85	6.3	ug/kg, dry
Endosulfan I	50% - 130%	30%	1.7 µg/Kg				2.7	6.3	ug/kg, dry
Endosulfan II	50% - 130%	30%	1.7 µg/Kg				1	6.3	ug/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
Endosulfan sulfate	50% - 130%	30%	1.7 µg/Kg				1.1	6.3	ug/kg, dry
Endrin	50% - 130%	30%	1.7 µg/Kg				0.86	6.3	ug/kg, dry
Endrin aldehyde	50% - 130%	30%	1.7 µg/Kg				1	6.3	ug/kg, dry
Endrin ketone	50% - 130%	30%	1.7 µg/Kg				1.4	6.3	ug/kg, dry
gamma-BHC	50% - 130%	30%	1.7 µg/Kg				1.3	6.3	ug/kg, dry
Heptachlor	50% - 130%	30%	1.7 µg/Kg				2.6	6.3	ug/kg, dry
Heptachlor epoxide	50% - 130%	30%	1.7 µg/Kg				2.2	6.3	ug/kg, dry
Methoxychlor	50% - 130%	30%	8.3 ug/Kg				1.2	31	ug/kg, dry
Toxaphene	50% - 130%	30%	16.7 ug/Kg				26	62	ug/kg, dry
trans-Chlordane	50% - 130%	30%	1.7 ug/kg				1.6	6.3	ug/kg, dry
PCBs				8082A	n/a	Gas Chromatography– PCBs by SW-846 Method 8082 and 8082A			
Aroclor 1016	60% - 120%	30%	16.7 µg/Kg				22	62	ug/kg, dry
Aroclor 1221	60% - 120%	30%	16.7 µg/Kg				27	62	ug/kg, dry
Aroclor 1232	60% - 120%	30%	16.7 µg/Kg				27	62	ug/kg, dry
Aroclor 1242	60% - 120%	30%	16.7 µg/Kg				20	62	ug/kg, dry
Aroclor 1248	60% - 120%	30%	16.7 µg/Kg				24	62	ug/kg, dry
Aroclor 1254	60% - 120%	30%	16.7 µg/Kg				13	62	ug/kg, dry
Aroclor 1260	60% - 120%	30%	16.7 µg/Kg				31	62	ug/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
PNAs				8270D	n/a	Gas Chromatography Mass Spectrometry – Volatiles SW-846 Method 8270D			
Acenaphthene	60%-110%	30%	33 µg/Kg				22	120	ug/kg, dry
Acenaphthylene	60%-110%	30%	33 µg/Kg				16	120	ug/kg, dry
Anthracene	60%-110%	30%	33 µg/Kg				20	120	ug/kg, dry
Benzo(a)anthracene	60%-110%	30%	33 µg/Kg				16	120	ug/kg, dry
Benzo(a)pyrene	60%-110%	30%	33 µg/Kg				24	120	ug/kg, dry
Benzo(b)fluoranthene	60%-110%	30%	33 µg/Kg				26	120	ug/kg, dry
Benzo(g,h,i)perylene	60%-110%	30%	33 µg/Kg				39	120	ug/kg, dry
Benzo(k)fluoranthene	60%-110%	30%	33 µg/Kg				36	120	ug/kg, dry
Chrysene	60%-110%	30%	33 µg/Kg				33	120	ug/kg, dry
Dibenzo(a,h)anthracene	60%-110%	30%	33 µg/Kg				24	120	ug/kg, dry
Fluoranthene	60%-110%	30%	33 µg/Kg				23	120	ug/kg, dry
Fluorene	60%-110%	30%	33 µg/Kg				17	120	ug/kg, dry
Hexachlorobenzene	60%-110%	30%	67 µg/Kg				28	250	ug/kg, dry
Indeno(1,2,3-cd)pyrene	60%-110%	30%	33 µg/Kg				32	120	ug/kg, dry
Naphthalene	60%-110%	30%	33 µg/Kg				19	120	ug/kg, dry
Phenanthrene	60%-110%	30%	33 µg/Kg				17	120	ug/kg, dry
Pyrene	60%-110%	30%	33 µg/Kg				24	120	ug/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
VOCs				8260B	n/a	Gas Chromatography Mass Spectrometry – Volatiles SW-846 Method 8260B			
1,1,1-Trichloroethane	70% - 130%	30%	2 µg/Kg				2.5	7.5	ug/kg, dry
1,1,2,2-Tetrachloroethane	70% - 130%	30%	2 µg/Kg				2.4	7.5	ug/kg, dry
1,1,2-Trichloroethane	70% - 130%	30%	2 µg/Kg				3.2	7.5	ug/kg, dry
1,1-Dichloroethane	70% - 130%	30%	2 µg/Kg				2.6	7.5	ug/kg, dry
1,1-Dichloroethene	70% - 130%	30%	2 µg/Kg				2.6	7.5	ug/kg, dry
1,2-Dibromo-3-chloropropane	70% - 130%	30%	5 µg/Kg				7.5	19	ug/kg, dry
1,2-Dichlorobenzene	70% - 130%	30%	2 µg/Kg				2.8	7.5	ug/kg, dry
1,2-Dichloroethane	70% - 130%	30%	5 µg/Kg				5.8	19	ug/kg, dry
1,2-Dichloropropane	70% - 130%	30%	2 µg/Kg				1.9	7.5	ug/kg, dry
1,3-Dichlorobenzene	70% - 130%	30%	2 µg/Kg				2.7	7.5	ug/kg, dry
1,4-Dichlorobenzene	70% - 130%	30%	2 µg/Kg				2.9	7.5	ug/kg, dry
2-Butanone	70% - 130%	30%	5 µg/Kg				8.3	19	ug/kg, dry
2-Chloroethyl vinyl ether	70% - 130%	30%	5 µg/Kg				3.5	19	ug/kg, dry
2-Hexanone	70% - 130%	30%	5 µg/Kg				5.8	19	ug/kg, dry
4-Methyl-2-pentanone	70% - 130%	30%	5 µg/Kg				5.5	19	ug/kg, dry
Acetone	70% - 130%	30%	20 µg/Kg				33	75	ug/kg, dry
Acrolein	70% - 130%	30%	400 µg/Kg				460	1500	ug/kg, dry
Acrylonitrile	70% - 130%	30%	80 µg/Kg				53	300	ug/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
Benzene	70% - 130%	30%	2 µg/Kg				1.9	7.5	ug/kg, dry
Bromodichloromethane	70% - 130%	30%	2 µg/Kg				1.5	7.5	ug/kg, dry
Bromoform	70% - 130%	30%	2 µg/Kg				2.2	7.5	ug/kg, dry
Bromomethane	70% - 130%	30%	5 µg/Kg				7.1	19	ug/kg, dry
Carbon disulfide	70% - 130%	30%	5 µg/Kg				3.9	19	ug/kg, dry
Carbon tetrachloride	70% - 130%	30%	2 µg/Kg				2.2	7.5	ug/kg, dry
Chlorobenzene	70% - 130%	30%	2 µg/Kg				2.8	7.5	ug/kg, dry
Chloroethane	70% - 130%	30%	5 µg/Kg				5.5	19	ug/kg, dry
Chloroform	70% - 130%	30%	2 µg/Kg				2.6	7.5	ug/kg, dry
Chloromethane	70% - 130%	30%	5 µg/Kg				7.5	19	ug/kg, dry
cis-1,2-Dichloroethene	70% - 130%	30%	2 µg/Kg				2.1	7.5	ug/kg, dry
cis-1,3-Dichloropropene	70% - 130%	30%	2 µg/Kg				2.3	7.5	ug/kg, dry
Dibromochloromethane	70% - 130%	30%	2 µg/Kg				2.4	7.5	ug/kg, dry
Dichlorodifluoromethane	70% - 130%	30%	5 µg/Kg				4.4	19	ug/kg, dry
Ethylbenzene	70% - 130%	30%	2 µg/Kg				3.6	7.5	ug/kg, dry
Methyl tert-butyl ether	70% - 130%	30%	2 µg/Kg				2.2	7.5	ug/kg, dry
Methylene chloride	70% - 130%	30%	5 µg/Kg				7.4	19	ug/kg, dry
Styrene	70% - 130%	30%	2 µg/Kg				2.3	7.5	ug/kg, dry
Tetrachloroethene	70% - 130%	30%	2 µg/Kg				2.5	7.5	ug/kg, dry

Table 4: Precision, accuracy, and limit of sensitivity for sediment parameters.

Parameter	Accuracy (% Recovery)	Precision (RPD)	Limit of Sensitivity *	Method	NSWRD SOP	Title	MDL	PQL	Units
Toluene	70% - 130%	30%	2 µg/Kg				1.9	7.5	ug/kg, dry
trans-1,2-Dichloroethene	70% - 130%	30%	2 µg/Kg				3.3	7.5	ug/kg, dry
trans-1,3-Dichloropropene	70% - 130%	30%	2 µg/Kg				2.6	7.5	ug/kg, dry
Trichloroethene	70% - 130%	30%	2 µg/Kg				2.5	7.5	ug/kg, dry
Trichlorofluoromethane	70% - 130%	30%	5 µg/Kg				7.5	19	ug/kg, dry
Vinyl chloride	70% - 130%	30%	2 µg/Kg				3.3	7.5	ug/kg, dry
Total Xylenes	70% - 130%	30%	4 µg/Kg				2.4	15	ug/kg, dry
Herbicides				8151A	n/a	Gas Chromatography– Semivolatiles – Herbicides by SW-846 Method 8151A			
2,4-D	30 – 115 %	30%	330 ug/Kg				93.7	330	ug/kg, dry
2,4,5-TP	30 – 115 %	30%	330 ug/Kg				84.9	330	ug/kg, dry
Inorganics									
TKN	80% - 120%	20%	40 mg/Kg	SM 4500 N org C, SM 4500 NH3 H	n/a	Wet Chemistry – Total Nitrogen by the Kjeldahl Method(TKN) by Automated Phenate	105	140	mg/kg, dry
Phosphorus	80% - 120%	20%	10 mg/Kg	SM 4500 P E	n/a	Phosphorus and Ortho-Phosphorus	13	61	mg/kg, dry
Cyanide (low)	80% - 120%	20%	0.5 mg/Kg	9014	n/a	Cyanide (Total/Weak Acid Dissocaible/Amenable)	0.6	1.7	mg/kg, dry
Phenols	90% - 110%	20%	0.5 mg/Kg	9066	n/a	Wet Chemistry - Phenolics	1.5	1.8	mg/kg, dry

* The limit of sensitivity is defined by the MDL. All laboratory instruments utilize a calibration defining instrument response. Any measurement falling above the upper calibration range is diluted to fall within range.

Representativeness:

Representativeness is the measure of the degree to which data accurately and precisely represent a characteristic of a population, parameter variations at a sampling point, a process condition, or an environmental condition. Representativeness of data will be ensured using established field and laboratory procedures and their consistent application. To aid in the evaluation of the representativeness of the sample data, field and laboratory blank samples will be evaluated for the presence of the sampled parameters.

Representativeness – Reference Sites

Data (fish, macroinvertebrate, habitat, water column chemistry, and sediment chemistry) will be collected from selected regional reference sites in northeastern Illinois preferably to include existing Illinois EPA and Illinois DNR reference sites, potentially being supplemented with other sites that meet the Illinois EPA criteria for reference conditions. One purpose of this data will be to index the methods used in this study to the reference condition as defined by Illinois EPA. In addition, the current Illinois EPA reference network does not yet include smaller headwater streams; hence reference data is needed to accomplish an assessment of that data.

Representativeness of Fish Data

Pulsed direct current (D.C.) electrofishing is a widely used methodology for collecting data on stream fish assemblages in the Midwestern U.S. While electrofishing does not collect all of the species present in a stream, it can collect more than 75-80% of the species that are present and approximate their relative abundances (Yoder and Smith 1999). This meets the purposes and requirements for biological assessments and biological criteria in that sufficiently representative data is produced to provide reliable signal about the health and well-being of the entire resource without the need to accomplish an exhaustive faunal inventory. The collection of relative abundance data includes the use of a standardized sampling procedure designed to produce a sufficiently representative sample of the fish assemblage at a site with a reasonable expenditure of effort (i.e., 2-3 hours/site; Yoder and Smith 1999).

Representativeness of Macroinvertebrate Data

The multi-habitat methodology of Illinois EPA (Appendix D) will be the primary method employed in this study. It produces a 300-organism subsample that represents all habitat types present at a site. A minimum 300 feet long reach is established and is intended to meet qualitative criteria of representativeness and including one stream habitat cycle (i.e., pool-run-riffle sequence). Wider width sites will be extended to 600 feet.

Representativeness of Water Chemistry Data

Water column chemistry samples will be collected using a grab sample method. The collection bucket will be washed with phosphate-free detergent and blank water, and then rinsed with river water to remove any potential sources of contamination in addition to sample decontamination between sampling locations. Samples will be collected from the center or point of the river or stream that appear under most uniform conditions.

Representativeness of Sediment Chemistry Data

Surficial sediment samples will be collected as composites in the method appropriate to the waterbody as specified in Appendix F. Samples will be collected in areas that were not previously disturbed by earlier sampling or wading. Composite samples will be collected from downstream to upstream to minimize potential disturbances. Care will also be taken to prevent material loss in the collection method and sampling equipment will be rinsed and decontaminated between sampling locations.

Precision and Accuracy:

Precision is a measure of agreement among repeated measurements of the same property under identical, or substantially similar, conditions; calculated as either the range or as the standard deviation. Precision may also be expressed as a percentage of the mean of the measurements, such as relative range or relative standard deviation (coefficient of variation).

Precision will be measured in the laboratory during the analysis of laboratory matrix spike (MS) and matrix spike duplicate (MSD) samples which are analyzed once per batch or at a rate of one per twenty samples. The analyses of the duplicate samples are considered acceptable if the calculated relative percent difference (RPD) of the measurements is within the acceptance limits listed in Table 3 and Table 4 for water column and sediment samples, respectively. For this study, the results of the duplicate analyses are used to calculate the RPD for evaluating precision using the following formula:

$$RPD = [(A - B) / (A + B)/2] * 100$$

where
A = Original sample concentration
B = Duplicate sample concentration

Accuracy is a measure of the overall agreement of a measurement to a known value. Accuracy includes a combination of random error (precision) and systematic error (bias) components that are due to sampling and analytical operations. Accuracy will be measured during the analysis of environmental water by using laboratory control spike (LCS) samples. In the laboratory, samples of deionized water will be fortified (or spiked) with the analytes of interest. These LCS samples will be analyzed with each batch of samples. The analyses of the LCS samples are considered acceptable if the calculated concentrations for all analytes of interest are within the acceptance limits listed in Tables 3 and 4 for the water column and sediment samples, respectively.

The results of the spiked samples are used to calculate the percent recovery for evaluating accuracy using the following formula:

$$\text{Percent Recovery} = [(S - U) / T] * 100$$

where
S = Spiked sample concentration
U = Unspiked sample concentration
T = True spike concentration

Precision and Accuracy - Fish and Macroinvertebrate Assemblages:
The Consultant employs fish and macroinvertebrate methods of which the precision and accuracy of the resulting data are known. Ohio EPA (1987) extensively tested the reproducibility, accuracy, and precision of the electrofishing sampling protocols in both wadeable streams and non-wadeable rivers and of the macroinvertebrate field methods. Based on a combination of data analyses from specially designed methods testing studies and the aggregate Ohio database, the reproducibility of an Ohio Index of Biotic Integrity (IBI) and Invertebrate Community Index (ICI) score was determined to be 4 units out of a 0-60 (12-60 for IBI) scoring scale (Rankin and Yoder 1999). Rankin and Yoder (1990) showed coefficient of variations (CV) were on the order of 8-10% at least impacted and high-quality sites. CVs increased at sites with lower IBI and ICI scores, presumably due to the effect of stressors at increasingly impacted sites. Fore *et al.* (1993) performed more extensive statistical analyses of the Ohio database and determined that IBI scores were reproducible to an error margin of 2-3 units when fish numbers were >200/0.3 km. Their power analysis confirmed that the Ohio IBI was capable of distinguishing 6 discrete scoring ranges that approximate the delineations of the IBI scale into the qualitative descriptions of exceptional, good, fair, poor, and very poor. Angermier and Karr (1986) analyzed other statistical properties of the IBI focusing on the extent of redundancy among metrics. The results of their analysis showed that careful construction and derivation of an IBI following the original guidance of Karr *et al.* (1986) should produce a robust and non-redundant set of metrics.

Accuracy can also be examined in terms of the assessment produced by the subject method. Biological assessments are viewed as a direct measure of the aquatic life protection goals of the Clean Water Act (CWA) and State water quality standards (as opposed to the surrogate assessment provided by chemical water quality criteria). This has given rise to the concept and interest in biological criteria and adoption by U.S. EPA of a national program, methods, and the development of formal implementation procedures. The issue at stake here is the accuracy of the delineation of waters as impaired or unimpaired for CWA purposes (e.g., TMDLs, NPDES). Historically, States and U.S. EPA based these decisions on chemical water quality data and comparison to State and national water quality criteria. However, studies that compared the relative performance of chemical and biological data and their respective abilities to detect impairment showed that biological data was far superior in its ability to detect impairment and minimize type II assessment error (Rankin and Yoder 1990; Yoder and Rankin 1998). It is implicit in these studies that the better standardized and calibrated the biological assessment method and assessment criteria, the more capable the method of detecting impairment and establishing a relative degree of departure from a baseline criterion and a measurement of biological condition that is continuous along the Biological Condition Gradient (BCG).

Precision and Accuracy – Water Column and Sediment Data:
The in-stream and analytical water column and sediment data will be recorded and analyzed based on the precision and accuracy parameters highlighted in the *Data Attributes – Water Column and Sediment Chemistry* section.

Measurement Range and Comparability:

Comparability is a measure of the confidence with which one data set or method can be compared to another. Comparability will be maximized by using standard analytical methods and standardized, documented sampling techniques. Documentation will include all sampling locations, conditions, and field sampling methods. All results will be reported in standard units or, for field parameters, as defined in the method. All laboratory calibrations will be performed using standards traceable to the National Institute for Standards and Technology (NIST) or another certified reference standard source.

Theoretically there is no upper limit to most of the raw data parameters that comprise the baseline biological data that will be produced by this study. The practical range of these parameters is dependent on the natural attributes of the regional fish and macroinvertebrate assemblages and the effectiveness of the sampling gear and procedure. For example, in the North Branch Chicago River sub basins we expect a wading electrofishing sample to produce 15-25 species and several hundred fish among those species. In higher quality areas, the number of species might increase to more than 30 with thousands of individuals. However, in terms of regional reference condition and potential, the resulting biological assessment should rate a biological assemblage the same with respect to its similarity to or departure from a regional reference condition. This is critical to establishing biological assessments that are comparable across the U.S. Thus, the derivation of reference condition is a critical step in the bioassessment process and is one of the factors that influence comparability.

The resulting assessments and biological indices have discrete scoring ranges, within which the raw data is stratified and compressed. For example, the original IBI and many of its contemporary applications use a scoring range of 12-60, i.e., metric scores of 5, 3, and 1 are assigned to each of 12 metrics. Newly developed IBIs have employed a scoring range of 0-100, which is intuitively more meaningful as a theoretical scoring range and communication tool. The rigor, adequacy of the method, development, and calibration ultimately determines the accuracy, precision, and reproducibility of the index, its statistical rigor, and its resulting assessment.

The water column and sediment samples will be collected as grab and composite samples at specified locations within the assemblage range. Sampling protocols specified within this QAPP will minimize external factors and allow for consistency and comparability between samples. Collection of duplicate samples will also be used to confirm consistency in technique and reproducibility of sample collection to further support data comparisons.

Bias:

Bias is the systematic or persistent distortion of a measurement process that causes errors in one direction. The accuracy of the water column and sediment samples will be assessed in order to identify and address any bias encountered.

Completeness:

Completeness is a measure of the amount of valid data needed to be obtained from a measurement system. The percent completeness is calculated by dividing the number of valid

sample results by the total number of samples planned, and multiplying the result by 100 percent. Completeness will be reported as the percentage of all measurements judged valid. The following equation will be used to determine completeness:

Percent Completeness = (V/T) * 100

where

V = Valid number of sample results

T = Total number of samples planned

For this project, the QA objective for degree of completeness for both field and laboratory data is 90 percent. If completeness is less than the target of 90 percent, representatives at the NBWW will evaluate the data to determine whether there are enough data to complete the study or if additional data collection is necessary.

It is expected that all of the data collected by the proposed study will be used for multiple purposes. The collection of the biological, habitat, and water column and sediment chemistry will be spatially integrated. This will provide enough information to compare the biological responses exhibited by the fish and macroinvertebrate assemblages with the exposure suggested by the habitat and water quality data at the same sampling sites. Sediment data integrates conditions over time more so than the water column grab sample data. All sampling protocols are designed to control the conditions under which sampling takes place so as to minimize external and confounding influences (e.g., high flows) and to ensure the data is comparable and representative.

Sensitivity:

Sensitivity is the capability of a method or instrument to discriminate between measurement responses representing different levels of the variable of interest. Many analytes measured for this project are present in analytically low concentrations throughout the biota, surface water, and sediment. All analytes are subject to chemical, biological, and physical processes that will alter their presence in the rivers and tributaries. It is the intent of this project to employ methods of measurements that will detect and quantify all analytes of interest wherever possible.

Although there are many intended and potential uses of the data, minimum measurement criteria will be established at the lowest analyte concentration required for planned uses of the measurement data. Minimum measurement criteria are State of Illinois water quality standards for general use waters where applicable. Where no minimum measurement criteria can be identified, the water column samples will be analyzed to the lowest concentration readily achievable by the NSWRD Laboratory. The monitored parameters and the established minimum measurement criteria are shown in Table 5. No table is constructed for sediment samples due to limited State of Illinois sediment standards.

Table 5 also gives the minimum measurement objectives for the project. The minimum measurement objectives will be set at approximately one-fifth of the minimum measurement criteria shown to ensure that parameters will be measured with reasonable accuracy at the

minimum measurement criteria concentrations, and measured to reasonable levels below the minimum measurement criteria.

The minimum measurement objective for any analyte will be achieved when the analytical procedure selected for sample analysis can be shown to have a MDL at or below the minimum measurement objective. Table 5 compares the minimum measurement objective against the reporting limit achieved by the NSWRD Laboratory. Most analytes meet the minimum measurement objective.

Analyte MDLs shall be determined by the USEPA method given in the Code of Federal Regulations (CFR), Volume 40, Part 136, Appendix B. The MDL is defined as "the minimum concentration of a substance that can be reported with 99% confidence that the measured concentration is distinguishable from method blank results." Since the MDL procedure is based upon precision obtained for a standard greater than the MDL, it also is a measure of method sensitivity at concentrations near the MDL.

For analytes without minimum measurements criteria, the minimum measurement objectives will be understood to be the MDL level that is readily achievable using analytical methods generally employed at the NSWRD Laboratory. For parameters where MDLs are not applicable such as pH and dissolved oxygen, the minimum measurement objectives shown in Table 5 are the sensitivity to be obtained by the measurement method. The accuracy and precision completeness for each parameter are also indicated in Tables 3 and 4 for the water column and sediment, respectively.

Table 5: Minimum measurement criteria, minimum measurement objectives, method MDLs, and laboratory RLs for regulated, sampled parameters.

Parameter	Minimum Measurement Criteria	Minimum Measurement Objectives	Method* MDL	RL
Total Chloride	500 mg/L[C]	100 mg/L	NA	5 mg/L
pH, field	6.5 - 9.0 pH unit	0.1 pH unit[S]	NA	NA
Dissolved Oxygen	5.0 mg/L (min)[C]	0.1 mg/L[S]	NA	N/A
Arsenic	190 µg/L[C]	38 µg/L	NA	1.0 µg/L
Copper	32 µg/L[C]	6.4 µg/L	NA	2.0 µg/L
Iron	1.0 mg/L[C]	0.2 mg/L	NA	100 ug/L
Total Manganese	152 µg/L[C]	30.4 µg/L	NA	50 µg/L
Total Mercury	1.2 ng/L[C]	0.24 ng/L	NA	0.50 ng/L
Nickel	14 µg/L[C]	2.8 µg/L	NA	2.0 µg/L
Total Zinc	61 µg/L[C]	12.2 µg/L	NA	20 µg/L
Ammonia (NH3-N)	15.0 mg/L[G]	3.0 mg/L	NA	0.10 mg/L
Phosphorus, Total	0.05 mg/L[G]	0.01 mg/L	NA	0.04 mg/L

NA = Not applicable
* Limits are current and subject to change
[S] = Required sensitivity

[G] = State of Illinois General Use Water Quality Standard
[C] = State of Illinois General Use Water Quality Standard that is based on acute or chronic standards and in some cases, variable based on the hardness of the water body.
[M] = Method Detection Limit based on the Selection Ion Monitoring Mode for direct aqueous analysis.
The General Use Water Quality Standards can be found at IAC Title 35 Section 302.208.

A.8: Training and Certification

The methods and protocols used in the proposed study require implementation by adequately trained and skilled biologists, field technicians, and laboratory staff. For the bioassessment, the lead biologist(s) must be well trained and experienced in all aspects of conducting the sampling, making decisions that affect quality in the field, being familiar with the study area, and knowing how to identify all species of fish and taxa of macroinvertebrates that will be encountered. Biological crew leaders must also be knowledgeable about safety procedures for boat electrofishing and boat and water safety. All crew leaders will be certified as Level 3 Qualified Data Collectors under the Ohio Credible Data Law (OCDL) or equivalent.

Field personnel assigned to this project will be directly supervised by the principal investigator and will have been trained by the principal investigator in an apprenticeship format (training documentation provided upon request). Of particular importance will be training in the electrofishing procedure, use of the modified QHEI, and the identification of external anomalies on fish. Each will follow the procedures outlined in Ohio EPA (1989) and Rankin (1989). Bioassessment laboratory personnel will adhere to the laboratory's internal protocols.

For the water column and sediment investigation, all laboratory staff utilizing the methods and protocols addressed in this study meet or exceed the educational requirements outlined in the NSWRD Quality Assurance Project Plan (QAPP). For each analysis, the chemist must demonstrate proficiency for each individual analysis. The proficiency requirements are typically defined in the specific method, within the U.S. EPA program for which the work is performed, within the National Environmental Laboratory Accreditation Program (NELAP) requirements and NSWRD's QAPP. The NSWRD Laboratory and its sub-contractors are NELAP accredited.

Field technicians assigned to this project, for the purpose of collecting samples and performing the analyses that are required to be completed in the field, have received adequate training from trained and experienced personnel. Field technicians will operate under the guidance and supervision of the Laboratory Supervisor. The field technicians are trained to be compliant with the requirements set forth by NELAP, U.S. EPA, and NSWRD's QAPP where applicable.

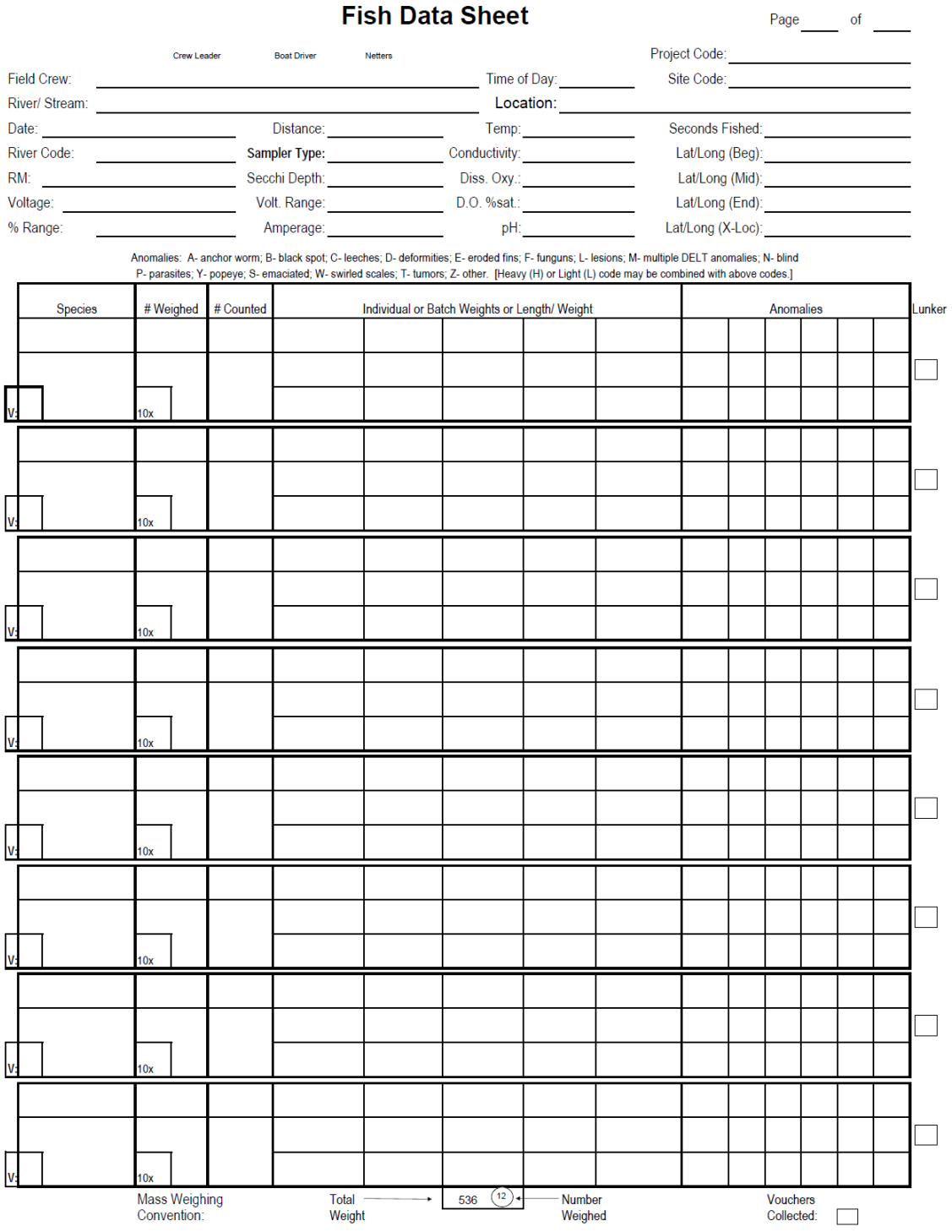
A.9: Documents and Records

The Quality Assurance Project Plan (QAPP) and all updates will be maintained by the NBWW in a secure location for five years. Revisions to the QAPP will be noted as to version and date and signed by the lead signatories. The revised QAPP document will be submitted to the distribution list via email. A detailed plan of study will be used to guide the execution of the annual field sampling. Laboratory data will be retained as hard copy or as electronic files for a minimum of 5 years in a secure location.

Field Data Recording:

Field data and observations will be recorded using standard data forms and field sheets. Fish data is recorded using the data sheet in Figure 2. Habitat data will be recorded using the QHEI data sheet in Figure 3. Initial water column chemistry data when grab samples are collected will be recorded using the data sheet in Figure 4. Grab samples for laboratory analysis will be accompanied by a laboratory supplied sample chain of custody (COC) in Appendix F. Sample times will be recorded for sediment sampling in conjunction with the bioassessment record keeping of Figure 2. All data will be entered into a relational database. All biological, chemical, and habitat data is initially managed by the consultants, transformed, analyzed, and then transferred to NBWW. All raw field data will be retained for minimum of five years in a secure file cabinet.

Figure 2. Field data sheet for recording electrofishing collection data and for entry into the database.



Fish Data Sheet

Page ____ of ____

Crew Leader Boat Driver Netters

Project Code: _____

Field Crew: _____ Time of Day: _____ Site Code: _____

River/ Stream: _____ Location: _____

Date: _____ Distance: _____ Temp: _____ Seconds Fished: _____

River Code: _____ **Sampler Type:** _____ Conductivity: _____ Lat/Long (Beg): _____

RM: _____ Secchi Depth: _____ Diss. Oxy.: _____ Lat/Long (Mid): _____

Voltage: _____ Volt. Range: _____ D.O. %sat.: _____ Lat/Long (End): _____

% Range: _____ Amperage: _____ pH: _____ Lat/Long (X-Loc): _____

Anomalies: A- anchor worm; B- black spot; C- leeches; D- deformities; E- eroded fins; F- funguns; L- lesions; M- multiple DELT anomalies; N- blind
P- parasites; Y- popeye; S- emaciated; W- swirled scales; T- tumors; Z- other. [Heavy (H) or Light (L) code may be combined with above codes.]

Species	# Weighed	# Counted	Individual or Batch Weights or Length/ Weight					Anomalies						Lunker
V:	10x													
V:	10x													
V:	10x													
V:	10x													
V:	10x													
V:	10x													
V:	10x													
V:	10x													

Mass Weighing Convention: Total Weight → 536 (12) ← Number Weighed Vouchers Collected: ☐

Figure 2. Fish data sheet (continued)

Page ______ of ______

Species	# Weighed	# Counted	Individual or Batch Weights or Length/ Weight					Anomalies						Lunker
														☐
V:	10x													
														☐
V:	10x													
														☐
V:	10x													
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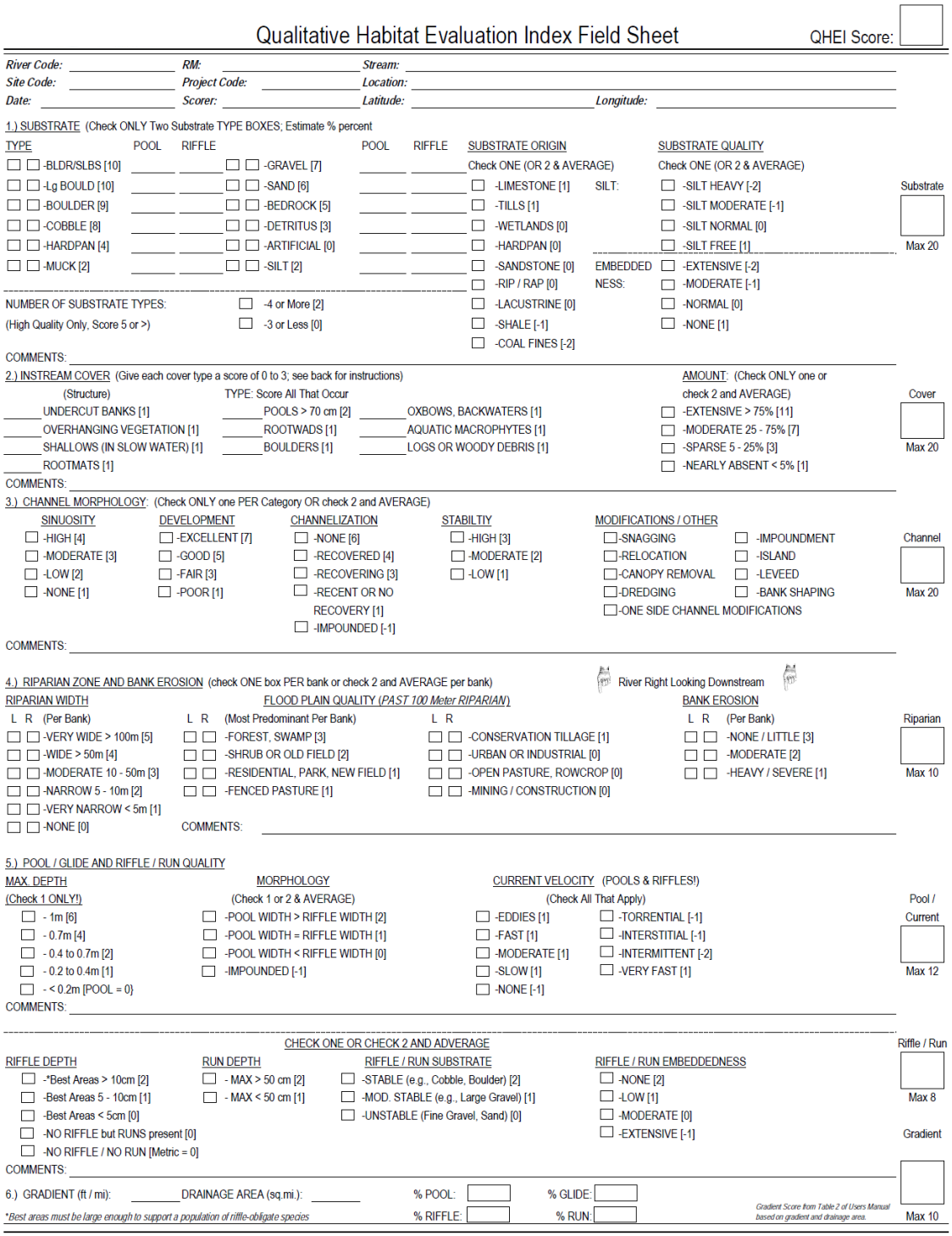
V:	10x													

Figure 3. QHEI field sheet.



Qualitative Habitat Evaluation Index Field Sheet — QHEI Score:

River Code: ____ RM: ____ Stream: ____
Site Code: ____ Project Code: ____ Location: ____
Date: ____ Scorer: ____ Latitude: ____ Longitude: ____

1.) SUBSTRATE (Check ONLY Two Substrate TYPE BOXES; Estimate % percent

TYPE	POOL	RIFFLE	TYPE	POOL	RIFFLE
☐ ☐ -BLDR/SLBS [10]			☐ ☐ -GRAVEL [7]		
☐ ☐ -Lg BOULD [10]			☐ ☐ -SAND [6]		
☐ ☐ -BOULDER [9]			☐ ☐ -BEDROCK [5]		
☐ ☐ -COBBLE [8]			☐ ☐ -DETRITUS [3]		
☐ ☐ -HARDPAN [4]			☐ ☐ -ARTIFICIAL [0]		
☐ ☐ -MUCK [2]			☐ ☐ -SILT [2]		

SUBSTRATE ORIGIN — Check ONE (OR 2 & AVERAGE)
☐ -LIMESTONE [1]
☐ -TILLS [1]
☐ -WETLANDS [0]
☐ -HARDPAN [0]
☐ -SANDSTONE [0]
☐ -RIP / RAP [0]
☐ -LACUSTRINE [0]
☐ -SHALE [-1]
☐ -COAL FINES [-2]

SUBSTRATE QUALITY — Check ONE (OR 2 & AVERAGE)
SILT:
☐ -SILT HEAVY [-2]
☐ -SILT MODERATE [-1]
☐ -SILT NORMAL [0]
☐ -SILT FREE [1]
EMBEDDED NESS:
☐ -EXTENSIVE [-2]
☐ -MODERATE [-1]
☐ -NORMAL [0]
☐ -NONE [1]

Substrate ____ Max 20

NUMBER OF SUBSTRATE TYPES: ☐ -4 or More [2] ☐ -3 or Less [0]
(High Quality Only, Score 5 or >)

COMMENTS: ____

2.) INSTREAM COVER (Give each cover type a score of 0 to 3; see back for instructions)

(Structure)
____ UNDERCUT BANKS [1]
____ OVERHANGING VEGETATION [1]
____ SHALLOWS (IN SLOW WATER) [1]
____ ROOTMATS [1]

TYPE: Score All That Occur
____ POOLS > 70 cm [2]
____ ROOTWADS [1]
____ BOULDERS [1]
____ OXBOWS, BACKWATERS [1]
____ AQUATIC MACROPHYTES [1]
____ LOGS OR WOODY DEBRIS [1]

AMOUNT: (Check ONLY one or check 2 and AVERAGE)
☐ -EXTENSIVE > 75% [11]
☐ -MODERATE 25 - 75% [7]
☐ -SPARSE 5 - 25% [3]
☐ -NEARLY ABSENT < 5% [1]

Cover ____ Max 20

COMMENTS: ____

3.) CHANNEL MORPHOLOGY: (Check ONLY one PER Category OR check 2 and AVERAGE)

SINUOSITY
☐ -HIGH [4]
☐ -MODERATE [3]
☐ -LOW [2]
☐ -NONE [1]

DEVELOPMENT
☐ -EXCELLENT [7]
☐ -GOOD [5]
☐ -FAIR [3]
☐ -POOR [1]

CHANNELIZATION
☐ -NONE [6]
☐ -RECOVERED [4]
☐ -RECOVERING [3]
☐ -RECENT OR NO RECOVERY [1]
☐ -IMPOUNDED [-1]

STABILTIY
☐ -HIGH [3]
☐ -MODERATE [2]
☐ -LOW [1]

MODIFICATIONS / OTHER
☐ -SNAGGING
☐ -RELOCATION
☐ -CANOPY REMOVAL
☐ -DREDGING
☐ -ONE SIDE CHANNEL MODIFICATIONS
☐ -IMPOUNDMENT
☐ -ISLAND
☐ -LEVEED
☐ -BANK SHAPING

Channel ____ Max 20

COMMENTS: ____

4.) RIPARIAN ZONE AND BANK EROSION (check ONE box PER bank or check 2 and AVERAGE per bank) — River Right Looking Downstream

RIPARIAN WIDTH
L R (Per Bank)
☐ ☐ -VERY WIDE > 100m [5]
☐ ☐ -WIDE > 50m [4]
☐ ☐ -MODERATE 10 - 50m [3]
☐ ☐ -NARROW 5 - 10m [2]
☐ ☐ -VERY NARROW < 5m [1]
☐ ☐ -NONE [0]

FLOOD PLAIN QUALITY (*PAST 100 Meter RIPARIAN*)
L R (Most Predominant Per Bank)
☐ ☐ -FOREST, SWAMP [3]
☐ ☐ -SHRUB OR OLD FIELD [2]
☐ ☐ -RESIDENTIAL, PARK, NEW FIELD [1]
☐ ☐ -FENCED PASTURE [1]
L R
☐ ☐ -CONSERVATION TILLAGE [1]
☐ ☐ -URBAN OR INDUSTRIAL [0]
☐ ☐ -OPEN PASTURE, ROWCROP [0]
☐ ☐ -MINING / CONSTRUCTION [0]

BANK EROSION
L R (Per Bank)
☐ ☐ -NONE / LITTLE [3]
☐ ☐ -MODERATE [2]
☐ ☐ -HEAVY / SEVERE [1]

Riparian ____ Max 10

COMMENTS: ____

5.) POOL / GLIDE AND RIFFLE / RUN QUALITY

MAX. DEPTH (Check 1 ONLY!)
☐ - 1m [6]
☐ - 0.7m [4]
☐ - 0.4 to 0.7m [2]
☐ - 0.2 to 0.4m [1]
☐ - < 0.2m [POOL = 0]

MORPHOLOGY (Check 1 or 2 & AVERAGE)
☐ -POOL WIDTH > RIFFLE WIDTH [2]
☐ -POOL WIDTH = RIFFLE WIDTH [1]
☐ -POOL WIDTH < RIFFLE WIDTH [0]
☐ -IMPOUNDED [-1]

CURRENT VELOCITY (POOLS & RIFFLES!) (Check All That Apply)
☐ -EDDIES [1]
☐ -FAST [1]
☐ -MODERATE [1]
☐ -SLOW [1]
☐ -NONE [-1]
☐ -TORRENTIAL [-1]
☐ -INTERSTITIAL [-1]
☐ -INTERMITTENT [-2]
☐ -VERY FAST [1]

Pool / Current ____ Max 12

COMMENTS: ____

CHECK ONE OR CHECK 2 AND ADVERAGE

RIFFLE DEPTH
☐ -*Best Areas > 10cm [2]
☐ -Best Areas 5 - 10cm [1]
☐ -Best Areas < 5cm [0]
☐ -NO RIFFLE but RUNS present [0]
☐ -NO RIFFLE / NO RUN [Metric = 0]

RUN DEPTH
☐ - MAX > 50 cm [2]
☐ - MAX < 50 cm [1]

RIFFLE / RUN SUBSTRATE
☐ -STABLE (e.g., Cobble, Boulder) [2]
☐ -MOD. STABLE (e.g., Large Gravel) [1]
☐ -UNSTABLE (Fine Gravel, Sand) [0]

RIFFLE / RUN EMBEDDEDNESS
☐ -NONE [2]
☐ -LOW [1]
☐ -MODERATE [0]
☐ -EXTENSIVE [-1]

Riffle / Run ____ Max 8

Gradient ____ Max 10

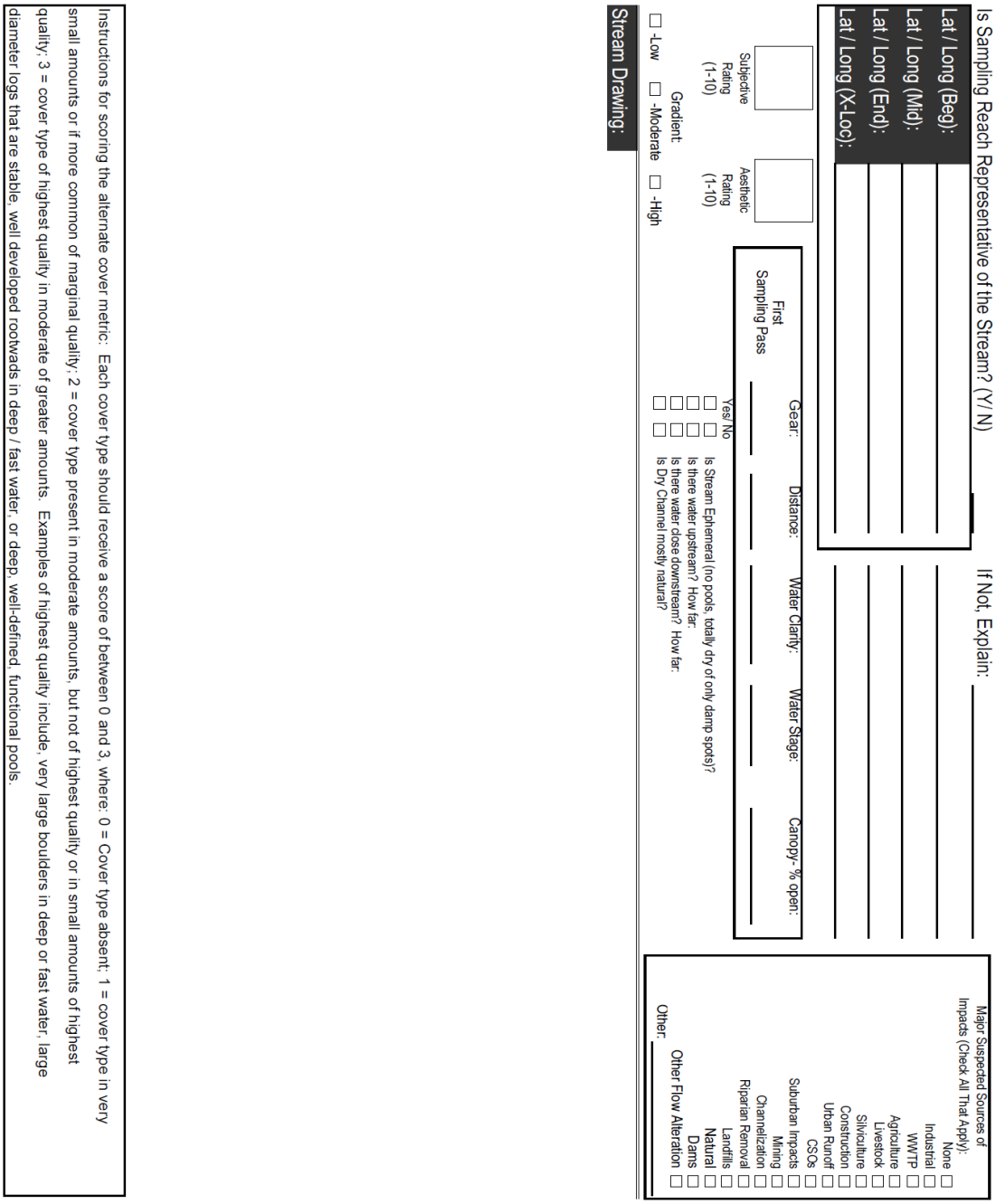
COMMENTS: ____

6.) GRADIENT (ft / mi): ____ DRAINAGE AREA (sq.mi.): ____ % POOL: ____ % GLIDE: ____ % RIFFLE: ____ % RUN: ____

Gradient Score from Table 2 of Users Manual based on gradient and drainage area.

**Best areas must be large enough to support a population of riffle-obligate species*

Figure 3. QHEI field data sheet (continued).



Instructions for scoring the alternate cover metric: Each cover type should receive a score of between 0 and 3, where: 0 = Cover type absent; 1 = cover type in very small amounts or if more common of marginal quality; 2 = cover type present in moderate amounts, but not of highest quality or in small amounts of highest quality; 3 = cover type of highest quality in moderate of greater amounts. Examples of highest quality include, very large boulders in deep or fast water, large diameter logs that are stable, well developed rootwads in deep / fast water, or deep, well-defined, functional pools.

Figure 4. Water column chemistry field data sheet.

NSWRD Field Sampling Water Chemistry

Location: _____ **Investigator(s):** _____
Date: _____ **Time:** _____ **Remarks:** _____

Conductivity: _____
Meter: _____ Calibration: _____

Dissolved Oxygen: _____
Meter: _____ Calibration: _____

Temperature (°C): _____

pH: _____
Meter: _____ Calibration: _____

Average Depth (ft.): _____

Flow (low, mod., high): _____

Velocity (ft./sec.) Meter: _____

Water Clarity: _____

Notes _____
_____

Reporting:

Progress reports will be made on a periodic basis and in accordance with the contract that supports the survey by the contractor. These will be distributed by the NBWW (see A.3). A final report will be produced by the contractor in accordance with the requirements of the original bioassessment plan.

Group B: Data Generation and Acquisition

B.1: Sampling Process Design

Monitoring sites are located on the main stem, tributaries, and within the headwaters, as shown in Appendix A, Figure 1 and Table 1. Publicly Owned Treatment Works (POTWs) are sampled above and below to determine to what effect effluent impacts the receiving waters. Sites are also located to determine the influences of tributary streams and other features, for example, the impact of online lakes (Appendix A).

B.2: Sampling Methods

Biological sampling for fish and macroinvertebrate assemblage data, as well as sampling methods for habitat, water column chemistry, and sediment chemistry will follow established protocols of the Illinois DNR (2001) and Illinois EPA (1997, 2005) *and* be capable of producing comparable data and assessments. In some cases, the applicable protocol will need to be determined in the field, thus the best two candidates are listed in these instances. The specifications for the different equipment and methods are described in Table 6 for fish assemblage and Table 7 for macroinvertebrates.

Fish Assemblage Methods:

Methods for the collection of fish at wadeable sites will be performed using a tow-barge or long-line pulsed D.C. electrofishing equipment based on a T&J 1736 DCV electrofishing unit described by Ohio EPA (1989) and as used by the consultant. A Wisconsin DNR battery powered backpack electrofishing unit will be used as an alternative to the long line and in accordance with the channel dimension restrictions described by Ohio EPA (1989). Generally, a three-person crew is required to execute the sampling protocol for each type of wading equipment. Sampling effort is determined by distance and ranges from 150-200 meters in length.

Table 6. Fish assemblage sampling method and gear specifications for the North Branch Chicago River biological assessment by geometric site level.

Parameter	Site Levels[2] Levels 6-7	Levels 2-6	Levels 1-2
Waterbody Size[3]	<1.0-5.0 mi^2	5.0-75 mi^2	75-150 mi^2
Channel Dimensions:[4]	<0.3-0.5m depth; 1-2m width	0.5-1.0m depth; 2-10m width	>1.0m depth; 10-100m width
Platform:	Backpack or Bank set/long line	Tow boat or Bank set/long line	12' boat 14' raft
Power Source:[5]	12v battery or 300W alternator;[6] 1750 W alternator[7]	1750-2500W alternator	2500 or 5000 W alternator
Amperage Output:	1.5-2A; 2-4A	4-8A	8-20A
Volts D.C. Output:	100-200; 150-300	150-300; 300-1000	500-1000
Anode Location:	Net ring w/assist netters	Net ring w/assist netters	Boom w/droppers; bow netter
Sampling Direction:	Upstream	Upstream	Downstream
Distance Sampled:	0.10-0.15km	0.15-0.20km	0.5km
Catch Per Unit Effort Basis (CPUE):[8]	per 0.3km	per 0.3km	per 1.0km
Time Sampled	1800-3600 sec	1800-3600 sec	2500-3500 sec
Time of Sampling:	Daylight	Daylight	Daylight
Crew Size[9]	2-3	3	2

[2] Site levels described under Watershed Monitoring Design and described for each site in Tables 7-9.
[3] Watershed size upstream from the sampling site.
[4] Size dimensions are approximate and may vary by site – these should not be used as primary criteria.
[5] Wattage (W) is sustained output (not peak output).
[6] Back pack units can be either battery or generator powered.
[7] This is used with the long line sampling method.
[8] Basis for determining relative abundance parameters.
[9] Crew consists of a qualified crew leader and field technicians.

Non-wadeable sites will be sampled with either a boat or raft-mounted pulsed D.C. electrofishing device. A Smith-Root 5.0 GPP unit will be used on a 12-foot john boat following the design of Ohio EPA. A Smith-Root 2.5 Generator Powered Pulsator (GPP) unit is used on a 14-foot raft. Sampling effort for this method is 500 meters. A summary of the key aspects of each method appear in Table 6.

Fish Sampling Reach Selection and Delineation

Sampling distance will be measured with a Geographic Positioning System (GPS) unit or laser range finder. When using the GPS unit each zone is measured by determining cumulative lineal distance based on waypoints established by the GPS unit. When using the laser range finder, measurements are taken in increments of 50-100 meters using fixed objects as focal points. Sampling site locations are delineated using the GPS mechanism and indexed to latitude/longitude and Universal Transverse Mercator (UTM) coordinates at the beginning, end, and mid-point of each site. Range finders are calibrated prior to being used in the field on a marked course and adjusted as necessary. The boundaries of each electrofishing zone are clearly marked on stationary objects (e.g. trees, bridge piers, etc.) with trail flagging or spray marking and fixed landmarks are referenced. This enables accurate relocation of sites in the event repeat visits are made. The location of each sampling site will be indexed by river mile (using river mile zero as the mouth of the river). A description of the sampling location shall also include proximity to a fixed local landmark such as a bridge, road, discharge outfall, railroad crossing, park, tributary, dam, etc. The field crew involved with the sampling is noted on the field sheet with crew duties listed (driver, netters, primary identifier, etc.).

Sampling Procedure

The tow-barge or longline pulsed D.C. electrofishing apparatus will be the preferred gear employed at wadeable sites. Electric current is converted, controlled, and regulated by a T&J 1736DCV alternator-pulsator that produces up to 1750 Watts at 100-300 volts DC at 2-7 amperes. The electrode anode array consists of the metal net ring hoop. The cathode consists of a woven steel cable strand on the front of the towboat or trailing the longline behind the sampler. A wading electrofishing crew consists of a primary netter who operates the anode, an assist netter and a third member who pulls the tow barge or attends to the long-line and keeps the collected fish cool and oxygenated by frequently changing water in a live well, bucket, or floating live well.

At wadeable sites, the accepted procedure is to slowly and methodically sample *upstream* sampling the best available habitat along the shoreline and/or midstream and sampling in and around submerged cover (undercut banks, log jams, root wads, emergent beds of vegetation, etc.) to advantageously position the netters to capture stunned and immobilized fish. Riffle/run areas are sampled using strategies that include “riffle raking”, which consists of “casting” the primary net ring (anode) upstream and allowing it to “float” downstream into the assist net. The assist netter also looks for fish attempting to swim downstream around the anode. Backwater and other margin habitats are sampled if present. Although sampling effort is measured by distance, the time fished is an important indicator of adequate effort. Time fished can legitimately vary over the same distance as dictated by cover and current conditions and

the number of fish encountered. In all cases, there is a minimum time that shall be spent sampling each zone regardless of the catch. This is generally in the range of 1200-1500 seconds for 150-200 meters and upwards to 2500 seconds where there is extensive instream cover and diverse current flows. Safety features include easily accessible toggle switches on the electrofishing unit and positive pressure thumb switches operated by the netter. All crew members wear rubber gloves and chest waders. Sampling will be conducted during a June 15-October 15 seasonal index period.

Boat sites will be sampled using a boat-rigged, pulsed D.C. electrofishing apparatus. This consists of a 12'-foot john boat that is specifically constructed and modified for electrofishing. Electric current is converted, controlled, and regulated by Smith-Root 5.0 GPP alternator-pulsator that produces up to 1000 volts DC at 2-20 amperes depending on the relative conductivity. The pulse configuration consists of a fast rise, slow decay wave that can be adjusted to 30, 60, or 120 Hz (pulses per second). Generally, electrofishing is conducted at 120 Hz, depending on which selection is producing the optimum combination of voltage and amperage output and most effectively stunning fish. This is determined on a trial and error basis at the beginning of each boat electrofishing zone and the settings will generally hold for all similar rivers and reaches. The voltage range is selected based on what percentage of the power range produces the highest amperage readings. Generally, the high range is used at conductivity readings less than 50-100 $\mu s/m^2$ and the low range is used at higher conductivities up to 1200 $\mu s/m^2$. Lower conductivities usually produce lower amperage readings.

The electrode array consists of four 8-10 foot long cathodes (negative polarity; 1" diameter flexible steel conduit) which are suspended from the bow and 4 anodes (positive polarity) suspended from a retractable boom, the number used to be dependent on the conductivity of the water. Each anode consists of a 3/8" woven steel cable strand 4-feet in length that are spaced equally on the boom cross member. Gangs of anodes can be added or detached as conductivity conditions change; anodes are increased at low conductivity and reduced at high conductivity. The anodes are suspended from a retractable boom that extends 2.75 meters in front of the bow. The width of the array is 0.9 meters. Anodes and cathodes are replaced when they are lost, damaged, or become worn.

A 12-foot boat or 14-foot raft electrofishing crew consists of a boat driver and one netter. Limited access to free-flowing segments may necessitate launching at an upstream location and recovering at a downstream location. Put-in and take-out sampling is conducted where navigational barriers preclude contiguous navigation. The accepted sampling procedure is to slowly and methodically maneuver the electrofishing boat/raft in a *down current* direction along the shoreline maneuvering in and around submerged cover to advantageously position the netter(s) to pick up stunned and immobilized fish. This may require frequent turning, backing, shifting between forward and reverse, changing speed, etc. depending on current velocity and cover density and variability. The driver's task is to maneuver the electrofishing boat/raft in a manner that advantageously positions the netter to pick up stunned and immobilized fish. The driver also monitors and adjusts the 2.5/5.0 GPP pulsator to provide the maximum, yet safe operational mode in terms of voltage range, pulse setting, and amperage.

In areas with extensive woody debris and submergent aquatic macrophytes, it is necessary to maneuver the boat/raft in and out of these "pockets" of habitat and wait for fish to appear within the netters field of view. In moderately swift to fast current the procedure is to electrofish with or slightly faster than the current through the fast water sections and then return upstream to more thoroughly sample the eddies and edges of the faster water. It is often necessary to pass over these swift water areas twice to ensure an adequate sample. Electrofishing efficiency is enhanced by keeping the boat/raft and electric field moving with or at a slightly faster rate than the prevailing current velocity. Fish are usually oriented into the current and must turn sideways or swim into the approaching electric field to escape. As such they present an increased voltage gradient making the fish more susceptible to being immobilized by the electric current. Sampling in an upstream direction is prohibited as this compresses the electrical field towards the surface, which significantly diminishes sampling effectiveness. Although sampling effort is measured by distance, the time fished is an important indicator of adequate effort. Time fished can legitimately vary over the same distance as dictated by cover and current conditions and the number of fish encountered. In all cases, there is a minimum time that shall be spent sampling each zone regardless of the catch. This is generally in the range of 2000-2500 seconds for a 0.5 km site, but could range higher where there is extensive instream cover and slack flows.

Safety features include easily accessible toggle switches on the pulsator unit and next to the driver and a foot pedal switch operated by the primary netter. The netters wear jacket style life preservers, rubber gloves, and all crew members wear chest waders.

All netters for both the wadeable and non-wadeable methods are required to wear polarized sunglasses to facilitate seeing stunned fish in the water during each daytime electrofishing run. The nets in the anode and in the assist net each consist of 7.62mm Atlas mesh knotless netting. A concerted effort is made to capture every fish sighted by all crew members. Since the ability of the netters to see stunned and immobilized fish is partly dependent on water clarity, sampling is conducted only during periods of "normal" water clarity and flows. Periods of high turbidity and high flows are avoided due to their negative influence on sampling efficiency and site access. If high flow conditions prevail, sampling will be delayed until flows and water clarity return to seasonal, low flow norms.

General Cautions Concerning Field Conditions

Electrofishing shall be conducted only during "normal" summer-fall water flow and clarity conditions. What constitutes normal can vary considerably from region to region. Generally normal water conditions in the Midwest occur during below annual average river flows. Under these conditions the surface of the water generally will have a placid appearance. Abnormally turbid conditions are to be avoided as are high water levels and elevated current velocities. In addition to safety concerns, any of these conditions can adversely affect sampling efficiency and may rule out data applicability for bioassessment purposes. Since the ability of the netter to see and capture stunned fish is crucial, sampling shall take place only during periods of normal water clarity and flow. Floating debris such as twigs, tree limbs, flotsam, and other trash are usually visible on the surface during elevated flow events. Such conditions shall be avoided and sampling

delayed until the water returns to a "normal" flow and clarity. High flows shall also be avoided for obvious safety reasons in addition to the reductions in sampling efficiency. Boat mounted methods are particularly susceptible as it becomes more difficult to maneuver the boat into areas of cover and the fish assemblage is locally displaced by the elevated flow events. It may take several days or even weeks for the assemblage to return to their normal summer-fall distribution patterns. Thus, sampling may need to be delayed by a similar time period if necessary. Knowing this requires local knowledge and a familiarity with flow gage readings and conditions. Generally, these conditions coincide with low flow durations of approximately 80% or greater, i.e., flows that are exceeded 80% of the time for the period of record. These statistics are available for most Midwest rivers from the U.S. Geological Survey at: http://waterdata.usgs.gov/.

Field Sample Processing Procedures

Captured fish are immediately placed in a live well, bucket, or live net for processing. Water is replaced and/or aerated regularly to maintain adequate dissolved oxygen levels in the water and to minimize mortality. Special handling procedures may be necessary for species of special concern. Fish not retained for voucher or other purposes are released back into the water after they have been identified to species, examined for external anomalies, and weighed, except at level 6 and 7 sites where only numbers are recorded. Every effort is made to minimize holding and handling times. The majority of captured fish are identified to species in the field; however, any uncertainty about the field identification of individual fish requires their preservation for later laboratory identification. Fish are preserved for future identification in borax buffered 10% formalin and labeled by date, river or stream, and geographic identifier (e.g., river mile). Identification is required to the species level at a minimum and may be necessary to the sub-specific level in certain instances. A number of regional ichthyology keys will be used and include the Fishes of Illinois (Smith 1979). Assistance will be solicited from Illinois DNR and the Field Museum of Natural History.

The sample from each zone is processed by enumerating and recording weights by species. Weights will be recorded at main stem sites. Fish weighing less than 1000 grams will be weighed to the nearest gram on a spring dial scale (1000 g x 2g) with those weighing more than 1000 grams weighed to the nearest 25 grams on a 12 kg spring dial scale (12 kg x 50 g) or a hand held spring scale for fish larger than 12 kg. Scales are checked before each sampling run with National Bureau of Standards check weights and adjusted accordingly. Samples that are comprised of two or more distinct size classes of fish (e.g., young of the year, juveniles, and adults) are processed as separate size groupings. These are recorded separately on the field data sheet by adding an A, B, or Y to the species code, A for adults, B for juveniles, and Y for young-of-year (y-o-y). For example, if both adult and juvenile white suckers occur in the same sample the adult numbers and weights are recorded as family-species code 40-016A with juvenile numbers and weights recorded as 40-016B. Although each is listed separately on the fish data sheet they are treated in the aggregate as a single sample of the same species in any subsequent data analyses. The data management programs used are designed to calculate relative numbers and weight data based on the input of the weighted subsample data. Larval fish will not be included in the data, as these are difficult to identify and offer questionable

information to an assemblage assessment (Angermier and Karr 1986). Fish measuring less than 15-20 mm in length are generally not included in the data recording as a matter of practice.

The incidence of external anomalies will be recorded following procedures outlined by Ohio EPA (1989) and refinements made by Sanders *et al.* (1999). The frequency of DELT anomalies (deformities, eroded fins and body parts, lesions, and tumors) is a good indication of stress caused by chronic agents, intermittent stresses, and chemically contaminated sediments. The percent DELT anomalies is a metric of most fish assemblage assessments that have been developed across the U.S.

A qualitative habitat assessment using an appropriate and updated modification of the QHEI (Ohio EPA 1989, 2006; Rankin 1989) will be completed by the fish crew leader. The QHEI is a physical habitat index designed to provide an empirical, quantified evaluation of the lotic macrohabitat characteristics that are important to fish assemblages. The QHEI was developed within several constraints associated with the practicalities of conducting a large-scale monitoring program, i.e., the need for a rapid assessment tool that yields meaningful information and which takes advantage of the knowledge and insights of experienced field biologists who are conducting biological assessments. This index has been used widely outside of Ohio and parallel habitat evaluation techniques are in widespread existence throughout the U.S. The QHEI incorporates the types and quality substrate, the types and amounts of instream cover, several characteristics of channel morphology, riparian zone extent and quality, bank stability and condition, and pool-run-riffle quality and characteristics. Slope or gradient is also factored into the QHEI score. We will follow the specific guidance and scoring procedures outlined in Ohio EPA (1989, 2006) and Rankin (1989) with more recent modifications made by MBI that may not appear in the 2006 manual. The QHEI users guide (Ohio EPA 2006) appears in Appendix B.

Macroinvertebrate Assemblage Methods:

The macroinvertebrate assemblage will be sampled using three principal methods. The attributes of each are summarized in Table 6. The Illinois EPA multihabitat method (Appendix D) will be used as a matter of preference at all sites where it is feasible. The Macroinvertebrate Aggregated Index for Streams (MAIS) method (Appendix D) adapted for application to Illinois streams will be used in lieu of the multihabitat method in small headwater streams and artificial substrates (Ohio EPA 1989) will be used at larger sites. This will be determined during sampling.

Illinois EPA Multi-Habitat Sampling Procedure

The Illinois EPA multi-habitat method for sampling stream macroinvertebrates provides information useful for determining the biological integrity of a stream, as reflected in selected attributes of the macroinvertebrate assemblage living in a stream. These biological attributes represent how macroinvertebrates respond to and integrate the chemical, physical, and biological effects of human-induced impacts (both negative and positive) on streams and their watersheds, e.g., point- or nonpoint-source impacts, stream-restoration efforts. The multi-

habitat approach allocates sampling effort based on the relative amounts of several predefined macroinvertebrate habitat types that occur in the sampling reach.

Table 7. Macroinvertebrate assemblage sampling method and gear specifications for the North Branch Chicago River biological assessment by geometric site level.

	Site Levels[10]		
Parameter	Levels 6-7	Levels 2-6	Levels 1-2
Waterbody Size[11]	<1.0-5.0 mi^2	5.0-75 mi^2	75-150 mi^2
Channel Dimensions:[12]	<0.3-0.5m depth; 1-2m width	0.5-1.0m depth; 2-10m width	>1.0m depth; 10-100m width
Protocol:	Qualitative Dip-Net, handpick	Multi-habitat Illinois EPA Method	Multi-habitat or Artificial Substrate
Collection device:	D-frame dip net	D-frame dip net	D-frame dip net;
Effort:	20 sweeps habitat defined 300 feet	20 sweeps; habitat defined 300 feet	20 sweeps; habitat defined 300-600 feet
CPUE Basis:[13]	No. individuals per site	No. individuals per site	No. ind./site; No./m^2
Subsample:	300 organisms	300 organisms	300 organisms;
Taxonomic Resolution:	Lowest Practicable	Lowest practicable	Lowest practicable
Crew Size[14]	2	2	2

[10] Site levels described under Watershed Monitoring Design and described for each site in Tables 7-9.
[11] Watershed size upstream from the sampling site.
[12] Size dimensions are approximate and may vary by site – these should not be used as primary criteria.
[13] Basis for determining relative abundance parameters.
[14] Crew consists of a qualified crew leader and one field technician.

The Illinois EPA multi-habitat method specifies the selection of a sampling reach that has instream and riparian habitat conditions typical of the entire assessment reach, has flow conditions that approximate typical summer base flow, has no highly influential tributary streams, contains one riffle/pool sequence or analog (i.e., run/bend meander or alternate point-bar sequence), if present, and, where the multi-habitat method is applicable, is at least 300 feet long and up to 800 feet long in order to meet the qualitative criteria for a site. The method is applicable if conditions allow the sampler to collect macroinvertebrates (i.e., to take samples with a dip net) in all bottom-zone and bank-zone habitat types that occur in a sampling reach. The habitat types are defined explicitly in Appendix D. Conditions must also allow the sampler to apply the 11-transect multi-habitat sampling method, as described in *"Standard Operating Procedure for Methods to Collect Aquatic Macroinvertebrates with Grab Samplers"* in *Appendix B: Methods Utilized to Determine the Types and Amounts of Pertinent Macroinvertebrate Habitats in Perennial Wadeable Streams for 20-Jab Allocation* (Illinois EPA 2011b) or to estimate with reasonable accuracy via visual or tactile cues the amount of each of several bottom-zone and bank-zone habitat types. If conditions (e.g., inaccessibility, water turbidity, or excessive water depths) prohibit the sampler from estimating with reasonable accuracy the composition of the bottom zone or bank zone throughout the entire sampling reach, then the multi-habitat method is not applicable. In most cases, if more than one-half of the wetted stream channel cannot be seen, touched, or otherwise reliably characterized by the sampler, it is unlikely that reasonably accurate estimates of the bottom-zone and bank-zone habitat types are attainable; thus, the multi-habitat method is not applicable.

Water Column Chemistry Methods:

The water column will be sampled annually at the locations and frequency specified in Appendix A. The sampling will consist of on-site field measurements and water samples which will be analyzed within the laboratory. On-site monitoring will be conducted using a multi-parameter datasonde to measure dissolved oxygen, pH, specific conductance, and temperature. The datasonde will be deployed as specified in Appendix G.

Analytical samples will be collected following the methods listed in Table 8. For sample collection, all sample bottles shall be kept closed until they are filled. Additionally, all sampling equipment shall be decontaminated between each sample collection site. The sampler will wear a new pair of gloves for decontamination and a new pair for sample collection at each collection site sampled. Samples will be collected in the manner appropriate for the waterbody with care to not disturb sediments during sample collection. Laboratory analytical sample bottles will then be placed on ice to maintain analytical temperature requirements described in Table 8. All samples will be recorded on the laboratory supplied chain of custody that is maintained from sample collection to sample acceptance at the laboratory and shown in Appendix F.

Table 8: Water column sampling containers, parameters, preservation, and holding time.				
Sample Container	**Parameters**	**Chemical Preservation**	**Thermal Preservation**	**Analytical Holding Time**
Stainless Steel Bucket	pH	Unpreserved	Sample readings collected in the field	15 minutes
	DO			
	Conductivity			
	Temperature			
1/2 gallon HDPE Bottle	TSS/TVSS	Unpreserved	Must be received at or below 6° C or delivered on ice	7 days
	Chlorophyll a			Keep from light, filter as soon as possible after collection, keep frozen until analysis, analyze within 3 ½ weeks
	Chloride			28 days
	BOD5			48 hours
	Total Nitrates (NO_3 + NO_2as N)	H_2SO_4 to pH < 2		28 days
250 mL HDPE Bottle	Metals	HNO_3 to pH < 2	No requirement for temperature	6 months
40 mL Glass vial (2)	Hg via Method 1631	BrCl	No requirement for temperature	28 days, must be preserved within 48 hours
500 mL HDPE Bottle	NH_3-N TKN	H_2SO_4 to pH < 2	Must be received at or below 6° C or delivered on ice	28 days
200 mL Glass Bottle	Total Phosphorus, TPO_4	H_2SO_4 to pH < 2	Must be received at or below 6° C or delivered on ice	28 days
120 mL Polystyrene Bottle	E.coli	Unpreserved, $Na_2S_2O_3$ is added only if chlorine is expected to be present	< 10° C	8 hours
250 mL LVI Amber Glass Bottle (2 ea)	Pesticides/PCB	Unpreserved	Must be received at or below 6° C or delivered on ice	7 days from collection to preparation (extraction), then 40 days from preparation to analysis
	PNAs			
40 mL Glass Vials (3)	VOCs	Unpreserved with zero headspace	Must be received at or below 6° C or delivered on ice	7 days

Benthic Periphyton Sampling:

Benthic periphyton samples will be collected during a representative low flow period between early July and late August and to coincide with datasonde deployment. Field data is recorded on a periphyton sample collection form (Appendix H). Field sampling procedures are based on substrate characteristics. Three separate subsamples of the slurry (defined volume) are field filtered thru three separate filters. The filters are then placed in individual zip lock bags wrapped in aluminum foil and placed on ice for shipping. Each sample is labeled by site code, date, time of collection, and sample collector. Samples received at the lab are stored at -20°C for a maximum of 28 days until analysis.

Care must be taken that all required equipment is properly cleaned prior to the preparation of equipment blanks and collection of samples. All non-metal equipment shall be cleaned with dilute HCl acid rinse. Soap (non-phosphate) and tap water shall be used on all equipment followed by a distilled water rinse. In the field, where such cleaning is not possible, a distilled water rinse shall be done before collecting a sample at each site.

Sediment Chemistry Methods:

Surficial sediment samples will be collected concurrently with the bioassessment as composites across locations specified in Appendix A. All samples will be collected in the manner appropriate to the waterbody as dictated in Appendix F with the bottleware and analytes specified in Table 9. Surface sediment samples will be collected in locations of minimal disturbance from other sampling efforts. For sample collection, all sample bottles shall be kept closed until they are filled and all sampling equipment shall be decontaminated between each sample collection site. The sampler will wear a new pair of gloves for decontamination and a new pair for sample collection at each collection site sampled. Sample bottles will then be placed on ice to maintain analytical temperature requirements described in Table 9. All samples will be recorded on the log sheet/ chain of custody that is maintained from sample collection to sample acceptance at the laboratory.

Table 9: Sediment sampling containers, parameters, preservation, and holding time.

Sample bottle	Parameters	Chemical Preservation	Thermal Preservation	Analytical Holding Time
9 oz. Glass Jar	Metals (Al, As, Ba, Be, B, Cd, Cr, Co, Cu, Fe, Pb, Mn, Hg, Ni, K, Ag, Na, Sr, V, Zn)	Unpreserved	Must be received at or below 6°C or delivered on ice	6 months
	Pesticides			14 days from collection to preparation (extraction), then 40 days from preparation to analysis
	PCBs			
	PNAs			
	TKN			28 days
	Phosphorus			28 days
	Cyanide (low)			14 days
	Phenols			28 days

4 oz. Glass Jar (zero headspace)	VOCs	Unpreserved	Must be received at or below 6°C or delivered on ice	7 days
4 oz. Glass Jar	Herbicides	Unpreserved	Must be received at or below 6°C or delivered on ice	40 days

B.3: Sample Handling and Custody

The sample products produced by this project will be fish and macroinvertebrate assemblage data, habitat assessments, and water column and sediment data. All data will be collected and managed by the associated consultant. All samples will be documented with appropriate data sheets and notations of the primary collectors and constitute a documentation of the chain-of-custody process. Completed field forms, laboratory forms, and the qualitative habitat assessment data sheets will comprise the hard copy documentation. Any subsequent changes that are made to the field and lab sheets are initialed and dated. Samples will be transported to the laboratory by the field sampling contractor. The laboratory will retain samples for a month after analysis.

For the fish, macroinvertebrate, and habitat data collection, all field data sheets are logged by the field crew leader (back-up copies are made to prevent loss) and assure that all sites are sampled according to the bioassessment plan. Data is entered from the field and laboratory sheets into the data management system in the format presented in the field data sheets (Figures 2 and 3). Each entry is logged by basin-river code, date of entry, river mile or other site locator, and date of sampling. The data sheets are assembled in a notebook along with site description sheets, maps of the sampling sites, the QHEI field sheet, and the bioassessment plan. After the data have been entered into the database the entries are proofread by the lead biologist for accuracy. All corrections or updates are then entered into the database.

Fish voucher specimens and macroinvertebrate samples will be archived for the purpose of confirming identifications and to serve as a permanent record. Photographs will also be used to record fish species occurrence, particularly larger species that are not easily preserved and stored. Fish will be transferred from 10% formalin to wash water and then to a series of ethyl alcohol washes from 35% to 50% to 70%. Voucher specimens will be deposited in the vertebrate collection at The Ohio State University Museum of Biodiversity (OSUMB)[15]. All photographs will be maintained by the consultant in an archived electronic file. Macroinvertebrates are transferred from 10% formalin to 70% ethyl alcohol for processing and permanent storage at MBI in Hilliard, Ohio. All samples are archived by the consultant for a minimum of 10 years.

For the water column and sediment investigation, all field forms, including documentation of sampling and calibration, and laboratory forms will be managed by the consultant. Laboratory

[15] Fish collections are taken to the OSUMB where they are given an accession number and then catalogued into the permanent collections.

and method protocols will be employed to ensure that samples are preserved, maintained at required temperatures, and analyzed within required hold times that are listed in Table 8 and Table 9. The consultant will also follow laboratory protocols concerning data quality assurance and quality control (QA/QC) and reporting as referenced in B.10. All reported data and metadata will be submitted to MBI who will utilize the data in analyses.

B.4: Analytical Methods

The principal analytical tools used for the biological data are those associated with basic data analysis. Data manipulation will be performed on personal computers using relational databases such as FoxPro, Access, and Excel. Appropriate modifications to those routines are initiated as needed to satisfy project objectives. Data will also be exported to various statistical and graphic packages such as Kaliedagraph for presentation graphics and S-Plus for statistical analyses. Habitat will be assessed using the QHEI following the methods in Appendix B.

Fish and macroinvertebrate data will be reduced to standard relative abundance and species/taxa richness and composition metrics. The Illinois EPA fish Index of Biotic Integrity (IBI) will be calculated with the fish data. The macroinvertebrate data will be analyzed using existing and developing indices of Illinois EPA.

Water column and sediment data will be analyzed in the laboratory based on the analyte-specific methods and SOPs specified in Tables 3 and 4. Reported laboratory data shall then be analyzed using data manipulation on personal computers in relational databases such as FoxPro, Access, and Excel. The data will be assessed for temporal and spatial variability, when appropriate. Additionally, the water column and sediment data will be integrated into the analysis of the bioassessment data and other stressor data. Datasonde DO data, water column and benthic periphyton sampling data along with the fish and macroinvertebrate data will be used to perform the Stream Nutrient Assessment Procedure (SNAP, Appendix I).

Products of this study will include a determination of biological status for all flowing waters, identification of stressor variables associated with biological impairments, spatial analyses of patterns in biological response variables, and recommendations for management actions as appropriate.

B.5: Quality Control

Quality control consists of ensuring that the data collected are the result of the proper execution of the sampling protocols and that the data are reproducible and precise. The precautions taken for each assemblage group and media in the field and laboratory are different, but the objective remains the same, to produce data that is of a sufficient quality so as to reduce type I and type II assessment errors.

Field Quality Control:

Fish Assemblage

Quality control of electrofishing includes adhering to sampling protocols and monitoring the power output variables. Other important measures of adequate effort include time

electrofished and the effort made by the netters to capture stunned and immobilized fish. There is an inherent degree of judgment involved in the assessment of individual crew member performance and this will be performed by the crew leader and the principal investigator. The quality of identifications made in the field will be evaluated by the principal investigator and also based on the retention of voucher specimens that will be verified independent of the field crew. Selected field audits of crew performance will be performed by the principal investigator. The field crew will be responsible for reporting any quality control issues, including deviations in procedure, to the principal investigator. In turn, the principal investigator will be responsible for reporting any issues to the NBWW representatives, the LCSMC Administrative Agent, and the Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the principal investigator will be responsible for performing this correction action and writing the corrective action reports.

Habitat Assessment

Annual crew leader training in using the QHEI is a requirement that assures consistent interpretation of QHEI variables and the resulting QHEI score and to make users aware of any recent modifications and updates. Visual identity is the key to being able to properly use the QHEI and this is reinforced by the required training, the annual refresher, and in the QHEI field guide which contains ample photographs and illustrations. Each QHEI is re-examined at all two pass fish sampling sites. The field crew will be responsible for reporting any quality control issues, including deviation in procedure, to the principal investigator. In turn, the principal investigator will be responsible for reporting any issues to the NBWW representatives, the LCSMC Administrative Agent, and the Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the principal investigator will be responsible for performing this correction action and writing the corrective action reports.

Macroinvertebrate Assemblage

The quality of macroinvertebrate sample collection and processing involves strict adherence to the specific protocols, re-sampling selected sites, and independent identification and enumeration of selected samples. A 10% subset of all sites will be re-sampled with the Illinois EPA multi-habitat method. This will allow the establishment of baseline variability within a seasonal index period to be established. A 10% subset of laboratory processed samples will also be identified and enumerated by an independent taxonomist. The results of this process will be used to reconcile the data prior to its use in the bioassessment. The field crew will be responsible for reporting any quality control issues, including deviations in procedure, to the principal investigator. In turn, the principal investigator will be responsible for reporting any issues to the NBWW representatives, the LCSMC Administrative Agent, and the Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the principal investigator will be responsible for performing this correction action and writing the corrective action reports.

Water Column Sampling

Quality control of water column sampling involves the adherence to multiple sampling protocols and collection of quality control samples. Sampling protocols include sample location

selection under representative flow conditions, care to minimize any disturbances during collection, efficient decontamination, proper application and calibration of field meters, and proper sample handling. Quality control samples will be collected and analyzed to ensure reproducibility and quality of sampling, transportation, and decontamination processes. These quality control samples will include an annual field blank for VOCs and metals, monthly duplicate samples for each analyte, and trip blanks for VOCs which are utilized with each set of samples. Analysis of water column parameters must fall within the methodology, reporting limits, quantification limits, and precision and accuracy as detailed in Table 2 and Table 3. The field crew will be responsible for reporting any quality control issues, including deviation in procedure, to the laboratory project manager. In turn, the project manager will be responsible for reporting any issues to the NBWW representatives, the LCSMC Administrative Agent, and the Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the project manager will be responsible for performing this correction action and writing the corrective action reports.

Sediment Sampling

Quality control of sediment is based in the adherence to sampling protocols and collection of quality control samples. This protocol includes sample location selection, sampling methodology to minimize losses, sample preparation, and sample handling. One field duplicate sample will be collected per sample event to assess consistent sampling techniques and data reproducibility. Analysis of sediment parameters must fall within the methodology, reporting limits, quantification limits, and precision and accuracy as detailed in Table 4. The field crew will be responsible for reporting any quality control issues, including deviation in procedure, to the principal investigator. In turn, the principal investigator will be responsible for reporting any issues to the NBWW representatives, the LCSMC Administrative Agent, and the Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the principal investigator will be responsible for performing this correction action and writing the corrective action reports.

Laboratory Quality Control:

Quality control will be maintained at the laboratory based on strict adherence to analytical methods and SOPs listed in Tables 3 and 4 and the NSWRD Laboratory Quality Assurance Plan. Under the methods specified in the manual, the laboratory will run internal QA/QC samples including blanks, MS, MSD, and LCS samples. The laboratory will also calibrate instruments as specified in B.7 to ensure quality control. When appropriate, the data will be qualified as specified in D.1 in order to report any analysis quality control issues. Additionally, any variances in procedure will be reported to the laboratory supervisor. The laboratory supervisor will be responsible for reporting any issues to the NBWW representatives, the LCSMC Technical Agent. The NBWW representatives will determine the course of correction action that is required, and the laboratory supervisor will be responsible for performing this correction action and writing the corrective action reports.

B.6: Instrument/Equipment Testing, Inspection, and Maintenance

All equipment is used and maintained in accordance with manufacturer's specifications. Safety logs are maintained for major pieces of sampling equipment (boats, trailers, vehicles). The electrofishing equipment is evaluated for performance during all phases of sampling as described previously in B.2 and performance characteristics are recorded on the field data sheets. All connections and switches must be in good condition to ensure acceptable performance and are inspected several times each day by the sampling crew. Malfunctioning and worn parts are replaced immediately with spare parts carried by each sampling crew. All engines undergo maintenance as prescribed by the manufacturer for intensive use. All water column and sediment sampling equipment are inspected and repaired or replaced as necessary prior to use. Analytical field meters used by the sampling crew are maintained in accordance with the manufacturer's specifications and calibration logs are maintained.

B.7: Instrument/Equipment Calibration and Frequency

Field meters used by the field crews are calibrated at the beginning of each day of use by the crew leader in accordance with the manufacturer's recommendations and specifications and in accordance with the parameter tolerances in Table 2. Standard calibration solutions are used from the manufacturer and within expiration limits. Equipment is adjusted as needed following B.6. Daily calibrations of the field meters will be recorded and the records will be maintained in field log books by the NSWRD Laboratory. Field log sheets will be stored in a secure location in the NSWRD Laboratory. In addition, calibration checks will be conducted on field meters after the completion of calibration.

B.8: Inspection/Acceptance of Supplies and Consumables

All supplies used in this project undergo an initial inspection for usability and suitability by the project lead at MBI. Outdated solutions and standards are replaced. Any open or broken sample containers are decontaminated or replaced. No hazardous reagents or sensitive supplies will be used in the field during this project.

B.9: Non-direct Measurements

We will make an effort to access historical information about the water column and sediment chemistry and fish and macroinvertebrate fauna of the study area. This will be especially valuable in evaluating the historical trends through time. Some expert judgment may be necessary to evaluate the quality and accuracy of this information.

B.10: Data Management

Fish and macroinvertebrate assemblage data and habitat data are entered directly via the electronic data entry routine from the field sheets (Figures 2 and 3). All data entry codes follow those specified in Ohio EPA (1987) and those added by the consultant for non-Ohio fish species. All entries are proofread by the data entry analyst and corrections are made in the electronic database. All corrections are noted and initialed by the data entry operator and confirmed by the project manager. Other checks on data entry accuracy are made via the routine processing and analysis of the data. Both desktop and laptop personal computers are used to manage

data, which is processed in software including Excel, Access, and FoxPro. Data is stored on the consultant's secure server.

Water column and sediment chemistry data are processed through various laboratory equipment, which is then entered into Laboratory Information Management System (LIMS) software. Access to LIMS is granted to authorized users with a specific password. The data is secure within this software and retained indefinitely. Data will be reported to Illinois EPA using the "Electronic Data Deliverable (EDD) Master Structure and Format".

All bioassessment, water column, and sediment data is reported to the NBWW, the LCSMC Administrative Agent, and the Technical Agent through electronic data deliverables (EDDs). Both desktop and laptop personal computers are used to manage data. Additionally, the data is maintained by the NBWW's secure County network drive and is backed-up daily in two locations. The procedure for retaining and filing of data sheets and field notes was described in B.2.

Group C: Assessment and Oversight

C.1: Assessments and Response Actions

Due to the well-defined and relatively localized scope of the project, assessment and oversight will be the joint responsibility of the NBWW monitoring committee and the principal investigator. However, the stakeholder agencies and organizations will be afforded an opportunity to make inspections and audits of the field sampling, the equipment, laboratory procedures, and the results if they so wish. This will be coordinated by the NBWW monitoring committee and the principal investigator. The principal investigator will be responsible for documenting any corrective action taken as based on the assessments.

C.2: Reports to Management

The principal investigator will file periodic verbal and/or written reports with the NBWW monitoring committee. The reports may concern but are not limited to project status, the results of performance evaluations and audits, the results of periodic data quality assessments, and significant QA/QC problems. Recipients may comment directly to the project sponsor lead and the principal investigator.

Group D: Data Validation and Usability

D.1: Data Review, Validation, and Verification

Data acceptance will initially be evaluated in the field and post-hoc during data management using the processes described in B.2 and B.5. However, later inspection of the data may also raise issues of acceptance. A systematic process will be used to reconcile any inconsistencies or issues prior to conditioning or disqualifying already collected data. Analytical data will be

qualified by the NSWRD Laboratory, and the qualifiers will be reported with the data. The defined list of qualifiers can be seen in Table 10.

Table 10: NSWRD or Subcontract Laboratory reportable qualifiers for water column and sediment samples.

Qualifier	Definition
B	Analyte detected in the associated Method Blank
J	Estimated, Analyte detected below quantitation limit (QL) but above the MDL
In case narrative	Discussion of sample receipt, analytical and QC issues
TIC	Tentatively identified compound
*	RPD outside accepted recovery limits
*	Spike Recovery outside accepted recovery limits

D.2: Verification and Validation of Methods

Most of the raw data will be field validated in accordance with the processes described in B.2, B.3, B.4, and B.10. Post-sampling validation will entail verification of identifications made in the field and later in the laboratory. Laboratory generated data will follow established procedures detailed in Appendices E, F, and G.

D.3: Reconciliation with User Requirements

The sampling and analytical approach proposed for this project are designed to provide the opportunity to adjust and modify methods as appropriate to obtain results that meet the project goals and objectives. Initial methods scoping may be done to assure comparability and making adjustments, modifications, and refinements to the methods described in B.2. Other changes and modifications may not be apparent until the project is completed and the data is fully analyzed and discussed. These changes will be documented in progress reports and the final report and will include a detailed description of all data analyses used. The QAPP will be reviewed annually and updated as necessary by the NBWW monitoring committee. All changes will be recorded and dated. After these changes have been approved by the monitoring committee, the updated QAPP will be redistributed electronically to the contacts listed under A.3 distribution List.

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APPENDIX A

MONITORING STRATEGY FOR THE NORTH BRANCH CHICAGO RIVER WATERSHED

Bioassessment 2-Year Monitoring Strategy for the North Branch Chicago River Watershed

March 19, 2018 (Revised April 2, 2019)

FAA#3191812

Purpose

The North Branch Watershed Workgroup (NBWW) will undertake a comprehensive monitoring program to document the existing water quality status of the rivers and streams in the sub-watersheds of the North Branch Chicago River watershed within Lake County and Cook County, Illinois. The monitoring program will emphasize the direct assessment of biological assemblages by sampling fish and macroinvertebrates using standardized sampling and assessment methodologies. In addition to determining aquatic life status, the monitoring program will also ascertain the related causes and sources associated with biological impairments by using paired chemical, physical and other stressor data and information within a systematic analytical process detailed in a comprehensive plan of study, specifically monitoring habitat and water and sediment chemistry.

The Monitoring Strategy is considered a living document. The NBWW Monitoring & Water Quality Impairment Abatement Committee will use adaptive management to review the results of the monitoring program and will revise and update the Monitoring Strategy if changes are needed. After the first two years, all of the sites will have been sampled for biological attributes and water chemistry. At that time, the Monitoring Strategy will be reviewed before the next full assessment begins.

Introduction and Background

The project area (see Figure 1) consists of the North Branch Chicago River watershed, covering approximately 50 square miles in Lake County, Illinois and 44 square miles in Cook County, Illinois. Three tributary subwatersheds made up of 55 miles of rivers and streams make up the watershed: West Fork North Branch Chicago River (West Fork, HUC 12: 071200030103), Middle Fork North Branch Chicago River (Middle Fork, HUC 12: 071200030102 and upper portions of 071200030105) and Skokie River (HUC 12: 071200030101). Each of the three subwatersheds originate in Lake County and flow south into Cook County where they converge in Morton Grove to form the mainstem of the North Branch of the Chicago River. The North Branch Chicago River flows south through the Chicago River, South Branch and Sanitary and Ship Canal to join with the Des Plaines River, which is a tributary of the Illinois and Mississippi Rivers.

Portions of the North Branch Chicago River, tributaries and lakes within the watershed in Lake County and Cook County are listed as impaired by the Illinois EPA and do not meet their designated uses under the Clean Water Act. Segments are listed as impaired for pollutants including aldrin, cadmium, chromium, hexochlorobenzene, nickel, barium, chloride, copper, endrin, lead, mercury, silver, sediment/siltation, total phosphorus, bottom deposits, chlordane, Dichlorodiphenyltrichloroethane (DDT), fecal coliform, total suspended solids, dissolved oxygen (DO), and temperature. Most of the impairments are being directly monitored (see Tables 2 & 3). However, E. coli is being monitored for the fecal coliform impairment and Total Suspended Solids is being monitored for the sediment/siltation and bottom deposits impairment, as well as including a robust sediment chemistry analysis in the program. The parameter “pesticides” includes analysis of aldrin, endrin, chlordane and DDT.

A North Branch Chicago River Watershed Stage 3 Total Maximum Daily Load (TMDL) Report is under development for some stream segments within the watershed for fecal coliform, DO and chloride. However, it is unclear as to whether implementation of the TMDL recommendations and the existing regulatory mechanisms will ultimately allow for the impaired waterbodies to meet Clean Water Act standards. The NBWW brings together local stakeholders to better determine stressors to the aquatic system through a long-term water quality monitoring program to work together to preserve and enhance water quality in the North Branch Chicago River and its tributaries. The preliminary monitoring strategy was developed by the NBWW Monitoring & Water Quality Impairment Abatement Committee.

NBWW Program Goals

The NBWW will undertake a comprehensive monitoring program to fulfill the following goals:

- Develop and implement a comprehensive monitoring program that will include chemical, physical and biological components that will more accurately identify the quality of stream and river ecosystems as well as stressors associated with non-attainment of water quality standards and designated uses. The NBWW monitoring program will establish baseline conditions, and then measure progress towards meeting water quality standards.
- Provide a secondary benefit to NPDES permittees by meeting certain monitoring permit requirements, including monitoring requirements for upstream and downstream of Publicly Owned Treatment Works (POTWs) and Municipal Separate Storm Sewer Systems (MS4s).

Budget and Timeline

The NBWW will use annual membership dues to support the comprehensive monitoring program. Qualified contractors will be thoroughly screened. Preliminary annual budget (based on 2019 budget):

- Annual dues: $122,478
    - $6,000 Technical Support
    - $19,000 Administration/Management
    - $87,964 Monitoring Program
        - Monitoring Compilation and Statistics
        - Water column Chemistry Monitoring
        - Sediment Chemistry Analysis
        - Bioassessment Monitoring/Sediment Collection
        - Benthic chlorophyll a Analysis

Monitoring Program

Monitoring of the North Branch Chicago River Watershed will consist of a bioassessment program (sampling of fish, macroinvertebrates and habitat), continuous dissolved oxygen (DO) monitoring, benthic periphyton sampling, and water column and sediment sampling to evaluate ecosystem quality and stressors. The monitoring program will be conducted at 25 sites throughout the North Branch Chicago River watershed within Lake County and Cook County, Illinois as shown in Figure 1 and listed in Table 1. The water column chemistry monitoring will be completed six times annually throughout the watershed with a tiered site design, as shown in Table 2. Tier 1 and 2 sites (all 25 sites) are sampled 4 (four) times per year and stay the same year after year. Tier 3 sites are a subset of the Tier 1 and 2 sites and receive an additional 2 (two) sampling events each year to complement the bioassessment. The Tier 3 sites change each year to follow the bioassessment locations. The bioassessment program, consisting of monitoring of fish, macroinvertebrates and habitat will be completed on a two-year

rotating basis. Water column and sediment sampling, sediment chemistry, continuous DO monitoring, benthic periphyton sampling and two additional nutrient water chemistry sample events will be completed on a two-year rotating basis concurrent with the bioassessment program.

Training and Certification

The methods and protocols used in the proposed study require implementation by adequately trained and skilled biologists, field technicians, and laboratory staff. For the bioassessment, the lead biologist(s) must be well trained and experienced in all aspects of conducting the sampling, making decisions that affect quality in the field, being familiar with the study area, and knowing how to identify all species of fish and taxa of macroinvertebrates that will be encountered. Biological crew leaders must also be knowledgeable about safety procedures for boat electrofishing and boat and water safety. All crew leaders will be certified as Level 3 Qualified Data Collectors under the Ohio Credible Data Law (OCDL) or equivalent.

Field personnel assigned to this project will be directly supervised by the principal investigator and will have been trained by the principal investigator in an apprenticeship format (training documentation provided upon request). Of particular importance will be training in the electrofishing procedure, use of the modified QHEI, and the identification of external anomalies on fish. Each will follow the procedures outlined in Ohio EPA (1989) and Rankin (1989). Bioassessment laboratory personnel will adhere to the laboratory's internal protocols.

For the water column and sediment investigation, all laboratory staff utilizing the methods and protocols addressed in this study meet or exceed the educational requirements outlined in the NSWRD Quality Assurance Project Plan (QAPP). For each analysis, the chemist must demonstrate proficiency for each individual analysis. The proficiency requirements are typically defined in the specific method, within the U.S. EPA program for which the work is performed, within the National Environmental Laboratory Accreditation Program (NELAP) requirements and NSWRD's QAPP. The NSWRD Laboratory and its sub-contractors are NELAP accredited.

Field technicians assigned to this project, for the purpose of collecting samples and performing the analyses that are required to be completed in the field, have received adequate training from trained and experienced personnel. Field technicians will operate under the guidance and supervision of the Laboratory Supervisor. The field technicians are trained to be compliant with the requirements set forth by NELAP, U.S. EPA, and NSWRD's QAPP where applicable.

Monitoring Site

Monitoring sites are located on the North Branch of the Chicago River and the three branches that flow into and form the North Branch Chicago River. The Skokie River, the Middle Fork of the Chicago River and the West Fork of the Chicago River. The two POTWs located in the watershed (Village of Deerfield POTW and North Shore Water Reclamation District POTW) are bracketed to determine to what effect effluent impacts the receiving waters. Sites are also located to determine the influences of tributary streams.

Water Column Chemistry Monitoring

The sampling will consist of on-site field measurements and water samples which will be analyzed within the laboratory. On-site monitoring will be conducted using water quality instruments to measure dissolved oxygen, pH, specific conductance, and temperature. Detection and measurement of additional parameters for water and sediment testing will be conducted in the laboratory. The sampling parameters are listed in the Quality Assurance Project Plan (QAPP). Water chemistry will be monitored 6 (six) times

January through October. A tiered site design will allow for more frequent monitoring of sites while keeping within budget and allowing for comprehensive coverage of the watershed. Samples will be collected using grab samples at the monitoring station unless otherwise noted in site description maps. If high pollutant loads are detected, follow up sampling at a refined scale may be undertaken to further determine the cause. Table 2 shows the parameters and summarizes the frequency of sampling described below for water column chemistry monitoring.

- **Tier 1:** 8 (eight) sites monitored 4 (four) times per year for common water quality parameters including nutrients and bacteria; and once annually under low flow conditions for metals and water organics.
- **Tier 2:** 17 (seventeen) sites monitored 4 (four) times per year for the majority of common water quality parameters including nutrients and bacteria.
- **Tier 3:** 2 (two) additional monitoring events per year at each bioassessment site for common water quality parameters including nutrients and bacteria concurrent with the bioassessment sampling period.

Equipment necessary to complete the water column chemistry monitoring will be provided by the contractor and may include buckets, collection bottles and gloves. Water column chemistry monitoring began in 2018 at eleven (11) Tier 1 and 2 sites on the Skokie River. Any monitoring completed prior to approval of the QAPP is not eligible to receive 319 grant funds or be used as match for a 319 grant.

Bioassessment and Sediment Chemistry Monitoring

The bioassessment and sediment chemistry monitoring will be conducted on all of the sites, with approximately half of the sites being monitored each year on an every other year rotating basis. Biological sampling for fish and macroinvertebrate assemblage data, habitat and sediment chemistry shall follow established protocols of the Illinois Department of Natural Resources (Illinois DNR; 2001) and Illinois EPA (1997, 2005) and be capable of producing comparable data and assessments. Sampling methods will be determined based on whether the stream is non-wadeable or wadeable. Ultimately, methods will be determined by the contractor and documented in the QAPP. Equipment necessary to complete the bioassessment will be provided by the contractor and may include electrofishing equipment, nets and an analytical field meter. Table 3 shows the parameters and summarizes the frequency of sampling for sediment chemistry monitoring. Bioassessment and sediment chemistry monitoring began in 2018 at 11 (eleven) sites on the Skokie River and will begin at 14 (fourteen) sites on the Middle Fork and West Fork in 2019.

Continuous DO Monitoring

Datasondes will be used in the North Branch Chicago River watershed to record continuous water quality data for dissolved oxygen (DO) over 3-5 day consecutive periods. Approximately 7 (seven) datasondes will be deployed in late summer each year under low flow conditions. The location of the datasondes will be concurrent with the biological sample locations for any given year. In 2018, 7 (seven) datasondes were deployed on the Skokie River branch where the biological monitoring occurred. In 2019, 7 (seven) datasondes will be deployed in the Middle Fork and West Fork.

The instruments will consist of either YSI 6-Series V2 model or EXO2 model units and used in accordance with the manufacturer specifications (YSI 2017). Each monitoring crew is required to maintain a calibration and maintenance log for each Datasonde Unit. The log will have consecutively numbered pages and include the following information at a minimum: date, Datasonde Model, Datasonde I.D. Number, description of monitoring (survey name), calibration comments, maintenance performed, and

crew leader name. Each instrument will be clearly identified (*e.g.,* the make, model, serial and/or I.D. number) to differentiate among multiple units. The appropriate calibration procedure must be followed and if the instrumentation does not have an electronic program that maintains a running calibration log, the results will be recorded in the logbook each time that unit is used, along with the date and name/initials of the person performing the calibration. If any difficulties are encountered during calibration or if the instrument will not hold calibration, this information will also be recorded. Malfunctioning equipment will not be used to collect data and will be scheduled for maintenance and/or repair and recorded in the log indicating what was done to correct the problem, along with the date and initials of the person that identified the problem. Continuous DO monitoring began in 2018 on the Skokie River and will begin on the Middle Fork and West Fork in 2019.

Benthic Periphyton Sampling

Benthic periphyton is collected to provide data on chlorophyll a content in support of the determination of the effect of nutrients as part of a combined nutrient approach that includes the diel DO flux as measured by a datasonde continuous monitor deployed at the same location. Benthic periphyton samples will be collected during a representative low flow period between early July and late August and to coincide with datasonde deployment and bioassessment. The results of the biological assemblage assessment (fish and macroinvertebrates) and concentrations of total phosphorus and nitrogen are also part of the combined assessment.

Benthic periphyton sampling began in 2018 on the Skokie River and will begin on the Middle Fork and West Fork in 2019.

Quality Assurance Project Plan (QAPP)

All monitoring will be conducted under an Illinois EPA approved QAPP. Illinois EPA requires the development of a QAPP for any grant activity involving the collection and analysis of environmental data. A QAPP presents the policies and procedures, organization, objectives, quality assurance requirements and quality control activities designed to achieve the type and quality of environmental data necessary to support project or program objectives. It is the policy of Illinois EPA that no data collection or analyses will occur without an approved QAPP. All in-house and external environmental data collection activities are subject to this requirement. All contracts must address quality assurance requirements (e.g., data quality and reporting requirements) when those contracts pertain to, or have an impact on, data collection or analysis activities. Additionally, all grants and contracts need to address quality assurance requirements specified in applicable state acquisition or procurement regulations. The NBWW QAPP will follow U.S. and Illinois EPA guidance for the development of a project specific QAPP.

Data and Reporting

Following analysis, the laboratory contractor will send all data via email to the NBWW in one final report in .pdf format. In addition, the laboratory will send an Excel spreadsheet summarizing all sites and parameters after each sampling event. NBWW staff will take this data and format it to fit the STOrage and RETrieval Data Warehouse (STORET) preferred by the Illinois EPA.

The Bioassessment contractor will send a monthly status report on bioassessment activities to the NBWW. This report will be provided electronically and as a hard copy, with chain-of-custody forms and laboratory reports attached. The consultant will use the datasonde DO data, water column and benthic chlorophyll a data along with the fish and macroinvertebrate data to perform the Stream Nutrient Assessment Procedure (SNAP).

The bioassessment contractor will also complete a final report, analyzing the results of the water column and sediment chemistry as well as the fish, macroinvertebrate, habitat and field water chemistry data. Interpretative statistics, such as long-term central tendencies, will be based on all available data within the database, developed over time, including past data collection efforts. This final report will be due on October 31, 2020. Data will be submitted annually to Illinois EPA.

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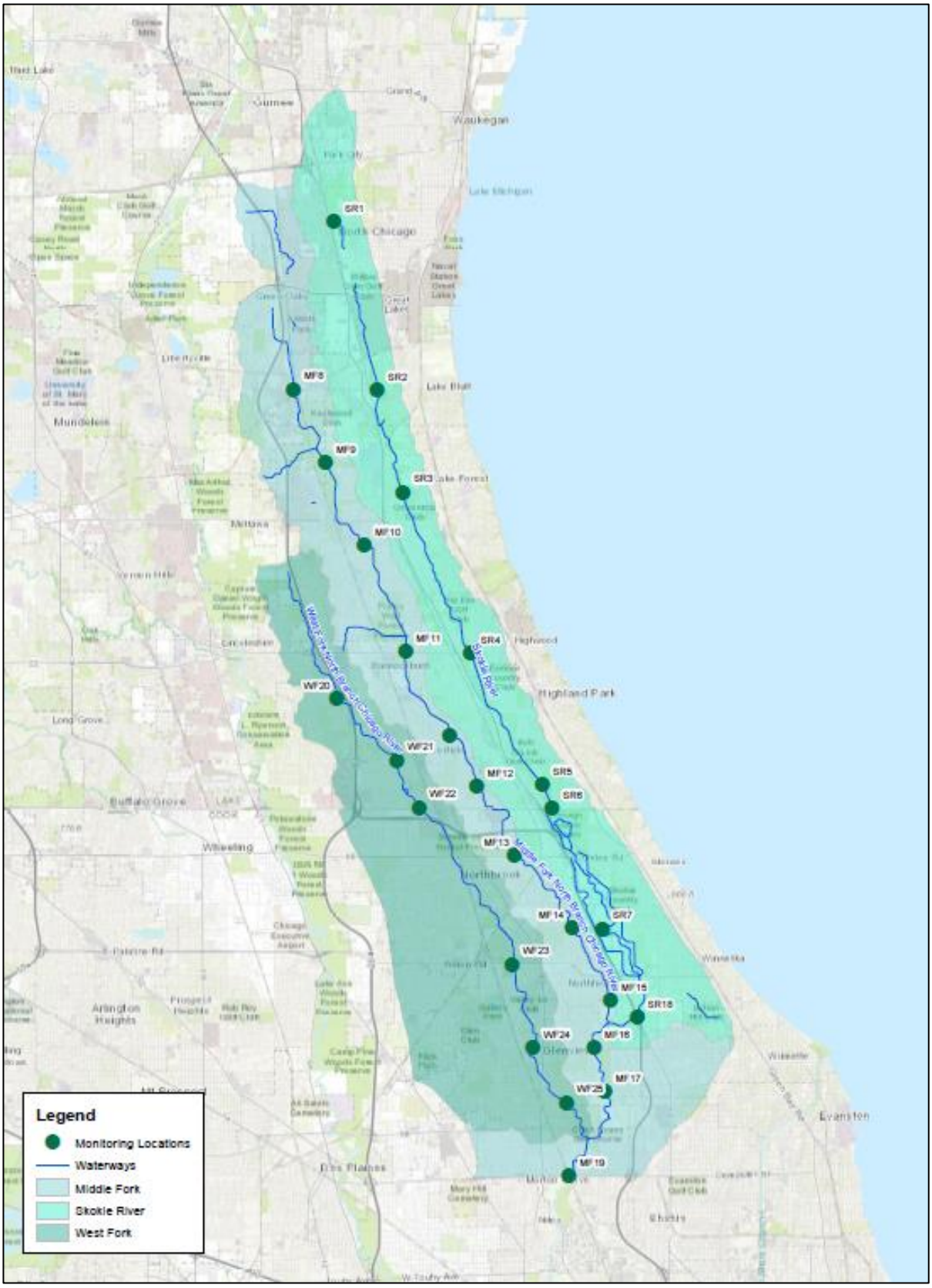
Figure

Figure 1: NBWW Sampling Locations



Tables

Table 1: Sampling Locations

North Branch Sample Locations	Street	NBWW Station IDs	Illinois EPA Station IDs	Illinois EPA AUIDs	Tier Designation		
Skokie River	Northern Boundary of the Foss Park Golf Course	SR1	HCCD-12	IL_HCCD	1		3
Skokie River	Rockland Road, Lake Bluff	SR2	HCCD-07	IL_HCCD-01		2	3
Skokie River	W. Deerpath Road, Lake Forest	SR3	HCCD-11	IL_HCCD-01		2	3
Skokie River	Half Day Road, Highland Park	SR4	HCCD-06	IL_HCCD-01		2	3
Skokie River	Clavey Road, Highland Park	SR5	HCCD-02	IL_HCCD-01		2	3
Skokie River	Lake Cook Road, north of Skokie Lagoons	SR6	HCCD-01	IL_HCCD-01		2	3
Skokie River	Tower Road, crosses the Skokie Lagoons, Winnetka	SR7	HCCD-04	IL_RHJ		2	3
Middle Fork	Route 176, Green Oaks	MF8	HCCC-16	IL_HCCC-02	1		3
Middle Fork	Middlefork Savanna Forest Preserve	MF9	HCCC-15	IL_HCCC-02		2	3
Middle Fork	Middlefork Trail & Greenway, W. Westleigh Road	MF10	HCCC-14	IL_HCCC-02		2	3
Middle Fork	Half Day Road, near Del Mar Woods	MF11	HCCC-13	IL_HCCC-02		2	3
Middle Fork	Carriage Way, south of Tea Tree Park, north of Briarwood Nature Area	MF12	HCCC-12	IL_HCCC-02		2	3
Middle Fork	Dundee Road, South of Somme Woods Forest Preserve	MF13	HCCC-11	IL_HCCC-02		2	3
Middle Fork	Sunset Drive, Northfield	MF14	HCCC-10	IL_HCCC-02		2	3
Middle Fork	Winnetka Road, Northfield	MF15	HCCC-03	IL_HCCC-02	1		3
Middle Fork	E. Lake Ave., Glenview/North of Blue Star Memorial Woods	MF16	HCCC-08	IL_HCCC-04		2	3
Middle Fork	South of Glenview Road, on the Forest Preserve Trail	MF17	HCCC-09	IL_HCCC-02		2	3
Skokie River	W. Frontage, west of I-94	SR18	HCCD-10	IL_RHJ	1		3
North Branch Chicago River	Dempster St., southernmost point of the watershed	MF19	HCC-10	IL_HCC-07	1		3
West Fork	South of Duffy Lane Bridge, off Saunders	WF20	HCCB-07	IL_HCCB-05	1		3
West Fork	South of Deerfield Road - Central Ave. in Deerfield	WF21	HCCB-06	IL_HCCB-05		2	3
West Fork	Lake Cook Road, Deerfield	WF22	HCCB-03	IL_HCCB-05	1		3
West Fork	Willow Road, southern end of Willow Hill Golf Course	WF23	HCCB – 12	IL_HCCB-05		2	3
West Fork	E. Lake Ave., Glenview	WF24	HCCB-11	IL_HCCB-05		2	3
West Fork	Long Valley Road, North of Glen View Club	WF25	HCCB-01	IL_HCCB-05	1		3

**All sites will be included in Tier 3 sampling. Sites SR1-7, 18 and MF16, 17 and 19 were sampled in 2018 twice during the summer concurrent with the biological sampling. The remaining sites in the Middle and West Fork will be sampled twice in 2019 concurrent with biological sampling.*

Table 2: Water Column Sampling Parameters and Frequency

Parameter	NBWW Frequency	Tier 1 (Four Times per Year)	Tier 2 (Four Times per Year)	Tier 3 (Two additional sampling events concurrently with bioassessment locations)
		Number of Sample Events*		
General Water Quality Parameters				
Chloride	six times a year	4	4	2
Conductivity	six times a year	4	4	2
pH	six times a year	4	4	2
TSS	four times a year	4	4	
Volatile Suspended Solids	four times a year	4	4	
DO	six times a year	4	4	2
Temperature	Six times a year	4	4	2
BOD5	Six times a year	4	4	2
Metals				
Arsenic	annually under low flow conditions	1	0	
Iron	annually under low flow conditions	1	0	
Calcium	annually under low flow conditions	1	0	
Magnesium	annually under low flow conditions	1	0	
Sodium	annually under low flow conditions	1	0	
Barium	annually under low flow conditions	1	0	
Cadmium	annually under low flow conditions	1	0	
Chromium	annually under low flow conditions	1	0	
Lead	annually under low flow conditions	1	0	
Mercury low level	annually under low flow conditions	1	0	
Copper	annually under low flow conditions	1	0	
Nickel	annually under low flow conditions	1	0	
Silver	annually under low flow conditions	1	0	
Zinc	annually under low flow conditions	1	0	
Nutrients				
Ammonia	four times a year	4	4	
Total Nitrates (NO2+NO3)	six times a year	4	4	2
TKN	four times a year	4	4	
Total phosphorus	six times a year	4	4	2
Chlorophyll a	five times a year	3	3	2
Bacteria				
E. coli	six times a year	4	4	2
Water Organics				
PCBs	annually under low flow conditions	1	0	
Pesticides	annually under low flow conditions	1	0	
PNAs	annually under low flow conditions	1	0	
VOCs	annually under low flow conditions	1	0	

**Tier 1 and Tier 2 sites are sampled 4 times per year. The only difference between Tier 1 and Tier 2 sites is that Tier 1 sites get the additional organics and metals analysis during the low flow August sample event. Table 1 shows all of the Tier 1 and Tier 2 sites. Then any of the sites that are scheduled for the bioassessment (11 in 2018 and 14 in 2019) would be considered Tier 3 and they would be sampled in July and September. Therefore, all of the sites are considered Tier 3 but only sampled when the bioassessment scheduled.*

Table 3: Sediment Sampling Parameters and Frequency

Parameter	NBWW Frequency	Tier 1	Tier 2
		Number of Sample Events (Annually)	
Sediment Metals			
Aluminum	Concurrent with bioassessment	1	1
Arsenic	Concurrent with bioassessment	1	1
Barium	Concurrent with bioassessment	1	1
Beryllium	Concurrent with bioassessment	1	1
Boron	Concurrent with bioassessment	1	1
Cadmium	Concurrent with bioassessment	1	1
Chromium	Concurrent with bioassessment	1	1
Cobalt	Concurrent with bioassessment	1	1
Copper	Concurrent with bioassessment	1	1
Iron	Concurrent with bioassessment	1	1
Lead	Concurrent with bioassessment	1	1
Manganese	Concurrent with bioassessment	1	1
Mercury	Concurrent with bioassessment	1	1
Nickel	Concurrent with bioassessment	1	1
Potassium	Concurrent with bioassessment	1	1
Silver	Concurrent with bioassessment	1	1
Sodium	Concurrent with bioassessment	1	1
Strontium	Concurrent with bioassessment	1	1
Vanadium	Concurrent with bioassessment	1	1
Zinc	Concurrent with bioassessment	1	1
Sediment Organics			
PCBs	Concurrent with bioassessment	1	1
Pesticides	Concurrent with bioassessment	1	1
VOCs/Hexachlorobenzene	Concurrent with bioassessment	1	1
PNAs	Concurrent with bioassessment	1	1
TKN	Concurrent with bioassessment	1	1
Phosphorus	Concurrent with bioassessment	1	1
Cyanide (low level)	Concurrent with bioassessment	1	1
Herbicide (2, 4, D, 2, 4, 5, TP)	Concurrent with bioassessment	1	1
Phenols	Concurrent with bioassessment	1	1

Please note: Sediment sampling does not include a Tier 3. Tier 3 for the water column sampling was included to conduct 2 additional monitoring events per year at bioassessment sites for general water quality parameters including nutrients and bacteria concurrent with the bioassessment sampling period.

Appendix B:

Methods for Assessing Habitat in Flowing Waters: Using the Qualitative Habitat Evaluation Index (QHEI)

Methods for Assessing Habitat in Flowing Waters: Using the Qualitative Habitat Evaluation Index (QHEI)

Introduction

This document summarizes the methodology for completing a general evaluation of macrohabitat, generally done by the fish field crew leader while sampling each location using the Ohio EPA Site Description Sheet - Fish (Appendix 1). This form is used to tabulate data and information for calculating the Qualitative Habitat Evaluation Index (QHEI). The following guidance should be used when completing the site evaluation form.

Header/Geographical Information

Complete site identification information is critical to making field data useful. Figure 1 illustrates the location information required for the QHEI.

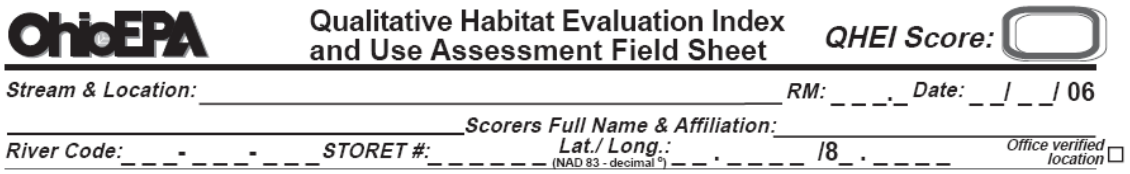


Figure 1. Header of Ohio EPA QHEI Sheet

1) Stream & Location, River Mile (RM), Date. The official stream name may be found in the Gazetteer of Ohio Streams (Ohio DNR 2001) or on USGS 7.5 minute topographic maps. If the stream is unnamed, a name and stream code is assigned by the Ohio ECOS Database Coordinator. Usually the name of a nearby landmark is used for the stream name. The River Mile (RM) designations used are found on 7.5 minute topo maps stored at the Ohio EPA, Division of Surface Water, Lazarus Government Center, Front Street (PEMSO RMI maps), one of five Ohio EPA District offices (maps for that district), and the Ohio EPA, Ecological Assessment Section at Grove City. These maps should soon be available as Adobe PDF files. A brief description of the sampling location should include proximity to a local landmark such as a bridge, road, discharge outfall, railroad crossing, park, tributary, dam, etc.

2) QHEI Scorers Full Name/Institution. The full name of the person who filled out the sheet are listed, along with the institution, company etc. QHEI information is to be completed someone who has successfully completed the QHEI training (e.g., crew leader). Ohio EPA will track the level of qualifications for each scorer. Level 2 QHEI practitioners have completed the two day training and successfully scored an additional site in a manner similar to EPA staff; Level 3 practitioners have additional training and have submitted three sites scored independently which will be verified as similar to EPA staff.

3) River Code, STORET, and Lat/Long. The River Code is Ohio EPA river code (PEMSO system) and the STORET # is the official unique Station Identifier used to link all data collected at a given "site" or "station" deemed to be similar for assessment purposes within a certain spatial area.

Habitat Characteristics: QHEI Metrics

The Qualitative Habitat Evaluation Index (QHEI) is a physical habitat index designed to provide an empirical, quantified evaluation of the general lotic macrohabitat characteristics that are important to fish communities. A detailed analysis of the development and use of the QHEI is available in Rankin (1989) and Rankin (1995). The QHEI is composed of six principal metrics each of which are described below. The maximum possible QHEI site score is 100. Each of the metrics are scored individually and then summed to provide the total QHEI site score. This is completed at least once for each sampling site during each year of sampling. An exception to this convention would be when substantial changes to the macrohabitat have occurred between sampling passes. Standardized definitions for pool, run, and riffle habitats, for which a

variety of existing definitions and perceptions exist, are essential for accurately using the QHEI. For consistency the following definitions are taken from Platts et al. (1983). It is recommended that this reference also be consulted prior to scoring individual sites.

Riffle and Run Habitats:

Riffle - areas of the stream with fast current velocity and shallow depth; the water surface is visibly broken.

Figure 2. Riffle cross-section.

Figure 3. Run cross-section.

Run - areas of the stream that have a rapid, non-turbulent flow; runs are deeper than riffles with a faster current velocity than pools and are generally located downstream from riffles where the stream narrows; the stream bed is often flat beneath a run and the water surface is not visibly broken.

Pool and Glide Habitats:

Pool - an area of the stream with slow current velocity and a depth greater than riffle and run areas; the stream bed is often concave and stream width frequently is the greatest; the water surface slope is nearly zero.

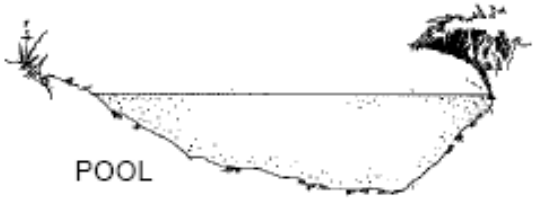


Figure 4. Pool cross-section.

Figure 5. Glide cross-section.

Glide - this is an area common to most modified stream channels that do not have distinguishable pool, run, and riffle habitats; the current and flow is similar to that of a canal; the water surface gradient is nearly zero. HINT: These habitat types typically grade into one another. For example a run gradually changes into a pool. When measuring typical depths of these features take measurements where the feature is clearly of that type, not where they are grading from one type to another. The following is a description of each of the six QHEI metrics and the individual metric components. Guidelines on how to score each is presented. Generally, metrics are scored by checking boxes. In certain cases the biologist completing the QHEI sheet may interpret a habitat characteristic as being intermediate between the possible choices; in cases where this is allowed (denoted by the term "Double-Checking") two boxes may be checked and their scores averaged.

Metric 1: Substrate (Figure 6).

This metric includes two components, substrate type[1] and substrate quality. **Substrate type** Check the two most common substrate types in the stream reach. If one substrate type predominates (greater than approximately 75- 80% of the bottom area OR what is clearly the most functionally predominant substrate) then this substrate type should be checked twice. **DO NOT CHECK MORE THAN TWO BOXES**. Note the category for artificial substrates. Spaces are provided to note the presence (by check marks, or estimates of % if time allows) of all substrate types present in pools (includes pools and glides) and riffles (includes riffles and runs) that each comprise sufficient quantity to support species that may commonly be associated with

[1] We suggest that QHEI practitioners should conduct some pebble count assessments which help calibrate an investigators ability to identify predominant substrates.

that substrate type. This section must be filled out completely to permit future analyses of this metric. If there are more than four or more high quality substrate types in the zone that are present in sufficient amounts (see above) then check the appropriate box for number of best types. This metrics award points to those sites with a diversity of high quality substrate types. Substrate origin refers to the parent material from which the substrate type(s) originated. This can be double-checked if two origin types are common (e.g., tills & limestone). See end of this section for some definitions.

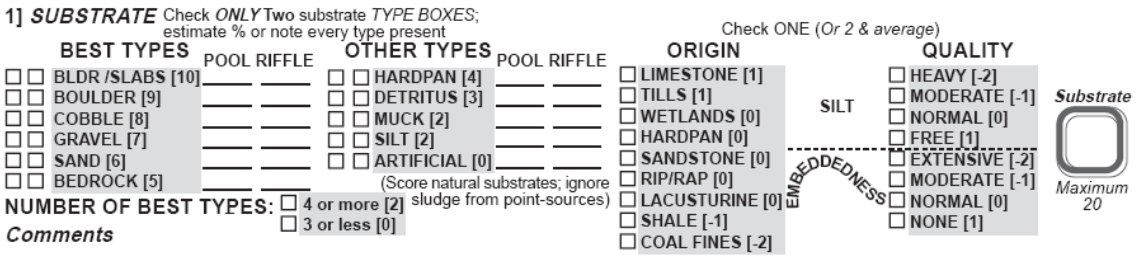


Figure 6. QHEI substrate metric.

Substrate quality.

Substrate origin refers to the "parent" material that the stream substrate is derived from. Check ONE box under the substrate origin column unless the parent material is from multiple sources (e.g., limestone and tills).

Embeddedness is the degree that cobble, gravel, and boulder substrates are surrounded, impacted in, or covered by fine materials (sand and silt). Substrates should be considered embedded if >50% of surface of the substrates are embedded in fine material. Embedded substrates cannot be easily dislodged. This also includes substrates that are concreted or "armor-plated". Naturally sandy streams are not considered embedded; however, a sand predominated stream that is the result of anthropogenic activities that have buried the natural coarse substrates is considered embedded.

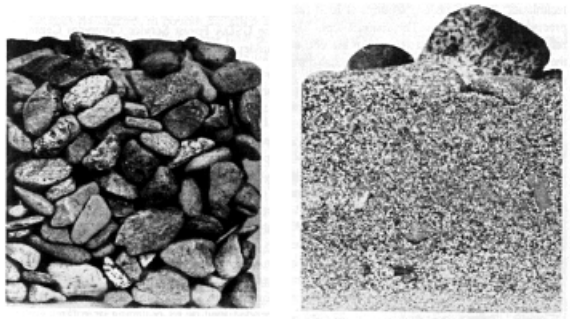


Figure 7. Side view of clearly un-embedded and embedded substrates.

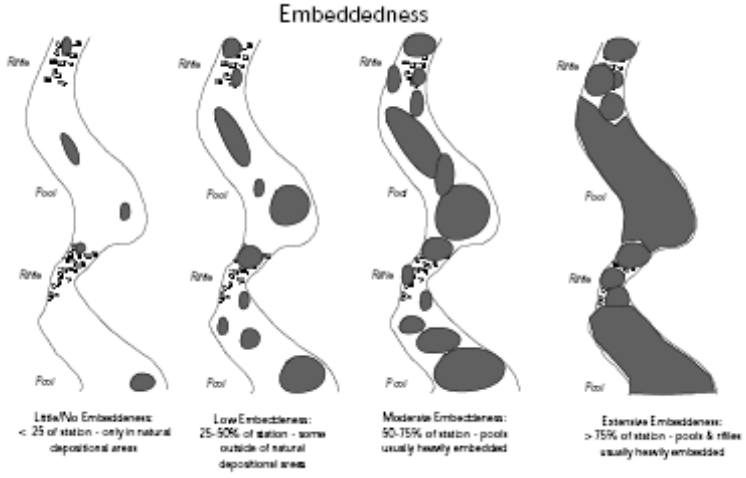


Figure 8. Illustration of example of degrees of pervasiveness of embeddedness for this QHEI component.

This can be very difficult to perceive. One help is to examine fresh point bars and look at the most common large materials that have been recently moved. According to Kappesser (1993), for gravel-bed rivers, the median of these large pieces should be equivalent to the median of the pieces on a riffle (based on a Wolman pebble count). If the riffles are finer than this, then sediment is aggrading in the reach and is evidence of embedded conditions. In some cases one can dig though the fine surface materials and fine coarser materials buried below. In this metric we are estimating the

pervasiveness of embedded conditions through-out a station. Boxes are checked for extensiveness (*i.e.*, pervasiveness throughout the area of the sampling zone) of the embedded substrates as follows: Extensive – > 75% of site area, Moderate – 50-75%, Normal[2] – 25-50%, None[3] – < 25%.

Silt Cover is the extent that substrates are covered by a silt layer (*i.e.*, a 1 inch thick or obviously affecting aquatic habitats). Silt cover differs from the embeddedness metric in that it only considers the fine silt size particles whereas fine gravels, sands, and other fines are considered in assessing embedded conditions. **Silt Heavy** means that nearly the entire stream bottom is layered with a deep covering of silt. (pool/glides and all but the fastest areas of riffle/runs). **Moderate** means extensive covering by silts, but with some areas of cleaner substrate (*e.g.*, riffles). **Normal** silt cover includes areas where silt is deposited in small amounts along the stream margin or is present as a "dusting" that appears to have little functional significance. If substrates are exceptionally clean the **Silt Free** box should be checked.

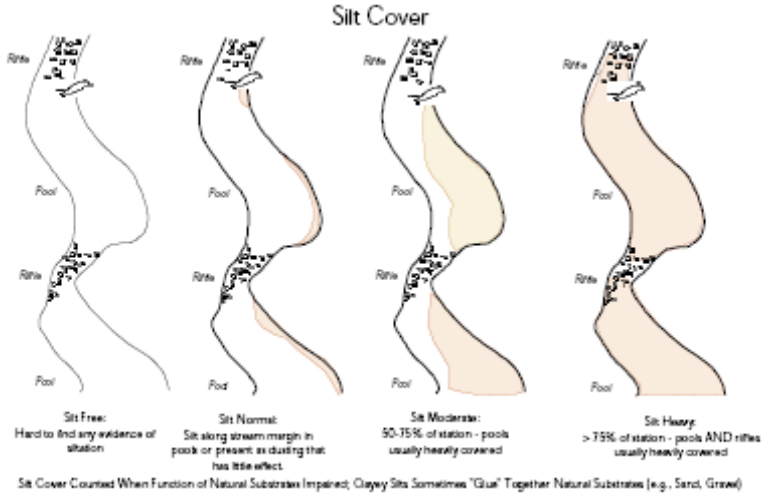


Figure 9. Illustration of example of degrees of pervasiveness of silt cover.

Substrate types are defined as:
a) **Bedrock** - solid rock forming a continuous surface.
b) **Boulder** - rounded stones over 256 mm in diameter (10 in.) or large "slabs" more than 256 mm in length (Boulder slabs)[4].
c) **Cobble** - stones from 64- 256 mm (2 1/2 - 10 in.) in diameter.
d) **Gravel** - mixture of rounded course material from 2-64 mm (1/12 - 2 1/2 in.) in diameter. Note the wide range of sizes included under gravel. In the riffle metric we distinguish between large and fine gravels
e) **Sand** - materials 0.06 - 2.0 mm in diameter, gritty texture when rubbed between fingers.
f) **Silt** - 0.004 - 0.06 mm in diameter, generally this is fine material which feels "greasy" when rubbed between fingers.
g) **Hardpan** - particles less than 0.004 mm in diameter, usually clay, which forms a dense, gummy surface that is difficult to penetrate.
h) **Marl** - calcium carbonate; usually grayish-white; often contains fragments of mollusk shells.
i) **Detritus** - dead, unconsolidated organic material covering the bottom which could include sticks, wood and other partially or un-decayed coarse plant material.
j) **Muck** - black, fine, flocculent, completely decomposed organic matter (does not include sewage sludge).
k) **Artificial** - substrates such as rock baskets, gabions, bricks, trash, concrete etc., placed in the stream for reasons OTHER than habitat mitigation.

Sludge is defined as a thick layer of organic matter that is decidedly of human or animal origin. NOTE: SLUDGE THAT ORIGINATES FROM POINT SOURCES IS NOT INCLUDED; THE SUBSTRATE SCORE IS BASED ON THE UNDERLYING MATERIAL. This scenario is rare today and was done to prevent underestimating stream habitat potential affect by discharges.

Substrate Metric Score: Although the sum of the individual metric scores can be greater than 20 the maximum substrate core allowed for this metric is 20 points.

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[2] In some earlier training materials "normal" was described as "low" (e.g., see Figure 7).
[3] In some earlier training materials "None" was described as "little-no" (e.g., see Figure 7).
[4] A version of the QHEI used in Maine distinguishes large boulders.

Example of stream with heavily embedded substrates.

Example of spongy deposits of fine gravels and sands from recent erosion activities.

Substrate Origin Identification Tips:

- Limestone: Often contains fossils, easily scratched with knife, usually bedrock or flat boulders and cobbles
- Tills: Sediments deposited by glaciers; particles often rounded. Can be carried into non-glaciated areas
- Wetlands: Usually organic muck and detritus
- Hardpan: Clay – smooth, usually slippery
- Sandstone: Contains rounded fragment of sand "cemented" together
- Rip/Rap: Artificial boulders
- Lacustrine: Old lake bed sediments
- Shale: "Claystone," sedimentary rock made of silt/clay, soft and cleaves easily
- Coal Fines: Black fragments of coal, generally SE Ohio only

We suggest that QHEI practitioners gain some experience in pebble count procedures. Conducting Wolman or Zig-Zag pebble counts helps to improve the ability to visually estimate predominant substrate sizes and size categories.

Stream characterized by cobble and boulder-size substrates.

2] *INSTREAM COVER* Indicate presence 0 to 3: **0**-Absent; **1**-Very small amounts or if more common of marginal quality; **2**-Moderate amounts, but not of highest quality or in small amounts of highest quality; **3**-Highest quality in moderate or greater amounts (e.g., very large boulders in deep or fast water, large diameter log that is stable, well developed rootwad in deep / fast water, or deep, well-defined, functional pools.

AMOUNT
Check ONE (*Or 2 & average*)
☐ EXTENSIVE >75% [11]
☐ MODERATE 25-75% [7]
☐ SPARSE 5-<25% [3]
☐ NEARLY ABSENT <5% [1]

____ UNDERCUT BANKS [1]
____ OVERHANGING VEGETATION [1]
____ SHALLOWS (IN SLOW WATER) [1]
____ ROOTMATS [1]
____ POOLS > 70cm [2]
____ ROOTWADS [1]
____ BOULDERS [1]
____ OXBOWS, BACKWATERS [1]
____ AQUATIC MACROPHYTES [1]
____ LOGS OR WOODY DEBRIS [1]

Comments

Cover Maximum 20

Figure 10. Instream cover (structure) metric.

Metric 2: Instream Cover (Figure 10).

This metric scores presence of instream cover types and amount of overall instream cover. Ohio EPA has been phasing in an alternative scoring system for this metric, but for this 2006, the total scoring still follows the existing methods. The changes will be discussed later.

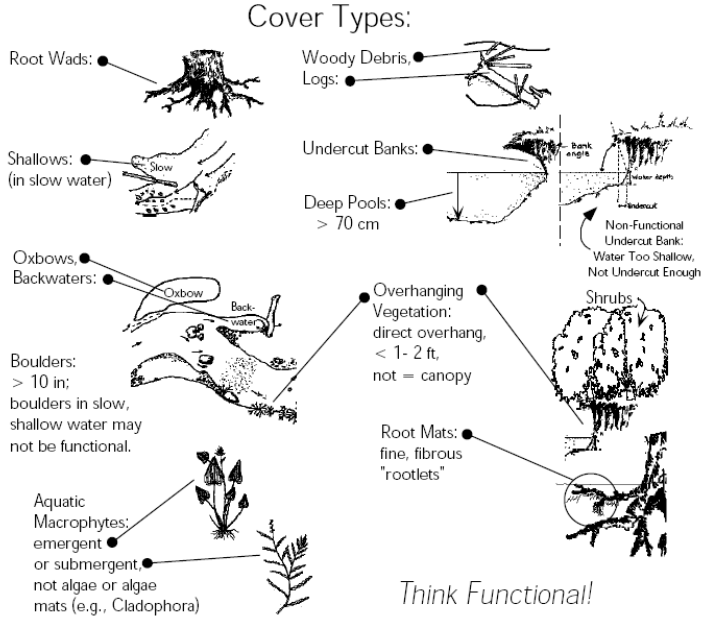


Figure 11. Examples of major cover/structure types measured with QHEI.

Existing Scoring Method:
Each cover type that is present in an amount occurs in sufficient quantity to support species that may commonly be associated with the habitat type should be scored.[5] Cover should not be counted when it is in areas of the stream with insufficient depth (usually < 20 cm) to make it useful. For example a logjam in 5 cm of water contributes very little, if any cover, and at low flow may be dry. Other cover types with limited function in shallow water include undercut banks and overhanging vegetation, boulders, and rootwads. Under amount, one or two boxes may be checked. Extensive cover is that which is present throughout the sampling area, generally greater than about 75% of the stream reach sampled. Cover is moderate when it occurs over 25- 75% of the sampling area. Cover is sparse when it is present in less than 25% of the stream margins (sparse cover usually exists in one or more isolated patches). Cover is nearly absent when no large patch of any type of cover exists anywhere in the sampling area. This situation is usually found in recently channelized streams or other highly modified reaches (e.g. ship channels). If cover is thought to be intermediate in amount between two categories, check two boxes and average their scores. For wide streams cover amount is estimated along the swath of stream sampled (or that would be sampled) with an electrofisher. In smaller streams

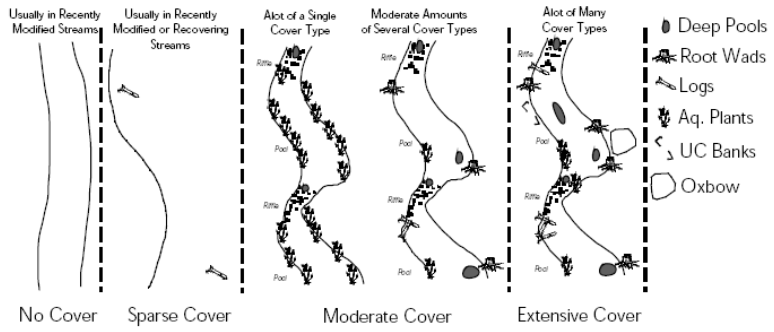


Figure 12. Illustration of the four categories of cover amounts.

[5] We had mentioned a 5% rule of thumb for an amount threshold if biological experience is low – this would be as a linear, not an areal amount.

(smaller wadeable and headwater streams) this generally covers most of the stream width. If a single type of cover is extensive and others are absent or uncommon then the total is scored as moderate because of the low diversity of types.

A desire to investigate and measure variation in amount and quality of individual cover types lead to a change in scoring of this metric. Over the next year or so the existing scoring method (each cover type scored on an presence/absence rating and a cumulative cover amount score) will be replaced with the following scoring method that focuses on scoring each cover type on a gradient of amount and quality. Each cover type would receive a score of 0-3 where:

> 0 - Absent;
> 1 - Very small amounts or if more common of marginal quality;
> 2 - Moderate amounts, but not of highest quality or in small amounts of highest quality;
> 3 - Highest quality in moderate or greater amounts (e.g., very large boulders in deep or fast water, large diameter logs that are stable, well developed rootwads in deep/fast water, or deep, well-defined, functional pools.

The cover ratings have been collected for about the last five years and an assessment of their relation to biological measures will be used to adjust a final scoring for this metric. At present, continue scoring these as present/absent and use the overall cover metric score. Cover types include: 1) undercut banks, 2) overhanging vegetation, 3) shallows (in slow water)[6], 4) logs or woody debris, 5) deep pools (> 70 cm), 6) oxbows, backwaters, or side channels, 7) boulders, 8) aquatic macrophytes, and 9) rootwads (tree roots that extend into stream). Do not check undercut banks AND rootwads unless undercut banks exist along with rootwads as a major component. Although the theoretical maximum score is > 20 the maximum score assigned for the QHEI for the instream cover metric is limited to 20 points.

High quality logs and woody debris in deep water.

High quality rootwad in deep, fast water.

[6] Shallows are habitats that provide nursery areas for small fish.

Example of good quality shallow habitat with aquatic macrophyte bed that acts as nursery habitat.

High quality boulder in fast water

Root Mats

Importance of logs and woody debris in large rivers.

Functional overhanging vegetation

Metric 3: Channel Morphology (Figure 13)

This metric emphasizes the quality of the stream channel that relates to the creation and stability of macrohabitat. It includes channel sinuosity (i.e. the degree to which the stream meanders), channel development, channelization, and channel stability. One box under each should be checked unless conditions are considered to be intermediate between two categories; in these cases check two boxes and average their scores.

3] *CHANNEL MORPHOLOGY* Check ONE in each category (*Or 2 & average*)

SINUOSITY	DEVELOPMENT	CHANNELIZATION	STABILITY
☐ HIGH [4]	☐ EXCELLENT [7]	☐ NONE [6]	☐ HIGH [3]
☐ MODERATE [3]	☐ GOOD [5]	☐ RECOVERED [4]	☐ MODERATE [2]
☐ LOW [2]	☐ FAIR [3]	☐ RECOVERING [3]	☐ LOW [1]
☐ NONE [1]	☐ POOR [1]	☐ RECENT OR NO RECOVERY [1]	

Comments

Channel
Maximum
20

Figure 13. Channel morphology metric.

a) *Sinuosity* - **No sinuosity** is a straight channel. **Low sinuosity** is a channel with only 1 or 2 poorly defined outside bends in a sampling reach, or perhaps slight meandering within modified banks. **Moderate sinuosity** is more than 2 outside bends, with at least one bend well defined. **High sinuosity** is more than 2 or 3 well defined outside bends with deep areas outside and shallow areas inside. Sinuosity may be more conceptually described by the ratio of the stream distance between two points on the channel of a stream and the straight-line distance between these same two points, taken from a topographic map. This metric measures the formation of pools and increased habitat area as the primary "functions" of sinuosity as related to aquatic life. Check one box or select two and average.

b) *Development* - This refers to the development of riffle/pool complexes. **Poor** means riffles are absent, or if present, shallow with sand and fine gravel substrates; pools, if present are shallow. Glide habitats, if predominant, receive a **Poor** rating. **Fair** means riffles are poorly developed or absent; however, pools are more developed with greater variation in depth. **Good** means better defined riffles present with larger substrates (gravel, rubble or boulder); pools have variation in depth and there is a distinct transition between pools and riffles. **Excellent** means development is similar to the **Good** category except the following characteristics must be present: pools must have a maximum depth of >1 m and deep riffles and runs (>0.5 m) must also be present. In streams sampled with wading methods, a sequence of riffles, runs, and pools must occur more than once in a sampling zone. Check one box or check two and average.

Table 1: Scoring criteria for pool/riffle development metric.

	Excellent	Good	Fair	Poor
Pool	> 1 m deep, well defined	0.7-1.0 m deep, well defined	Some depth variation	Shallow, if present
Glide	Not common	Not common	Common	Predominant
Riffle	Deep, well defined riffles, large substrates	Defined riffles, large substrates	Poorly defined riffles or riffles absent	Absent of shallow with fine substrates
Run	> 0.5 m deep, well defined	Deep, well defined	Usually absent	Absent

This metric can be double-checked. For situations, for example where riffles are excellent and pools are only fair, it is advantageous to check the excellent and the fair box rather than checking the good box as an average to keep information on the variance in quality.

Note how well defined (i.e., distinct) the riffle and pool are in this high quality headwater stream pictured on the left. Also note the large tree in the riparian

c) *Channelization* - This refers to anthropogenic channel modifications. **Natural** refers to no obvious direct moving or alteration of the channel and a natural appearance. **Recovered** refers to streams that have been channelized in the past, but which have recovered most of their natural channel characteristics. **Recovering** refers to channelized streams which are still in the process of regaining their former, natural however, these habitats are still degraded. This category also applies to those streams, especially in the Huron/ Erie Lake Plain ecoregion (NW Ohio), that were channelized long ago and have a riparian border of mature trees, but still have Poor channel characteristics. **Recent** or **No Recovery** refers to streams that were recently channelized or those that show no significant recovery of habitats (e.g. drainage ditches, grass lined or rock rip-rap banks, etc.). The specific type of habitat modification is checked in the last two columns but not scored.

A channelized stream channel starting to revert towards more natural channel features.

Unstable channel features and low stability.

d) *Stability* - This refers to channel stability. Artificially stable (concrete) stream channels receive a High score. Even though they generally have a negative influence on fish assemblages, the negative effects are related to features other than their stability. Channels with **Low stability** are usually characterized by fine substrates in riffles that often change location, have unstable and severely eroding banks, and a high bedload that slowly creeps downstream. Sometimes these unstable riffles form diagonally across the channel (see figure, right). Channels with **Moderate stability** are those that appear to maintain stable riffle/ pool and channel characteristics, but which exhibit some symptoms of instability, e.g. high bedload, eroding or false banks, or shows the effects of wide fluctuations in water level. Channels with **High stability** have stable banks and substrates, and little or no erosion and bedload. e) *Modifications/Other* - Check the appropriate box if impounded, islands present, or leveed (these are not included in the QHEI scoring) as well as the appropriate source of habitat modifications. The maximum QHEI metric score for Channel Morphology is 20 points.

Metric 4: Riparian Zone and Bank Erosion (Figure 14)

This metric emphasizes the quality of the riparian buffer zone and quality of the floodplain vegetation. This includes riparian zone width, floodplain quality, and extent of bank erosion. Each of the three components requires scoring the left and right banks (looking downstream). The average of the left and right banks is taken to derive the component value. One box per bank should be checked unless conditions are considered to be intermediate between two categories; in these cases check two boxes and average their scores.

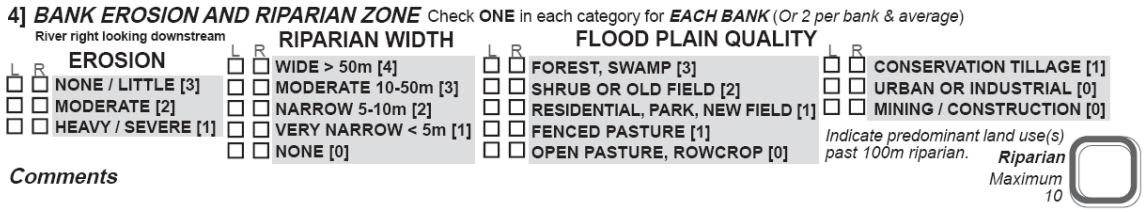


Figure 14. Bank erosion and riparian zone metric.

Severe bank erosion.

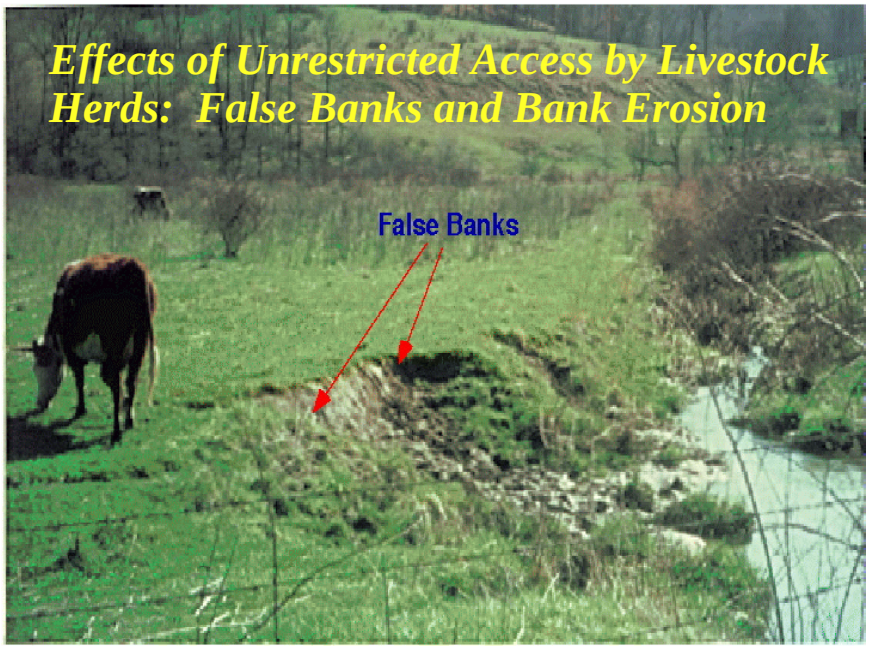
a) *Bank Erosion* – A modified Streambank Soil Alteration Ratings from Platts et al. (1983) is used here; check one box for each side of the stream and average the scores. False banks are used in the sense of Platts et al. (1983) to mean banks that are no longer adjacent to the normal flow of the channel but have been moved back into the floodplain most commonly as a result of livestock trampling. 1) **None** - streambanks are stable and not being altered by water flows or animals (e.g. livestock) - Score 3. 2) **Little** - streambanks are stable, but are being lightly altered along the transect line; less than 25% of the streambank is receiving any kind of stress, and if stress is being received it is very light; less than 25% of the streambank is false, broken down or eroding - Score 3. 3) **Moderate** - streambanks are receiving moderate alteration along the transect line; at least 50 percent of the streambank is in a natural stable condition; less than 50% of the streambank is false, broken down or eroding; false banks are rated as altered - Score 2. 4) **Heavy** - streambanks have received major alterations along the transect line; less than 50% of the streambank is in a stable condition; over 50% of the streambank is false, broken down, or eroding - Score 1. 5) **Severe** - streambanks along the transect line are severely altered; less than 25% of the streambank is in a stable condition; over 75% of the streambank is false, broken down, or eroding - Score 1

b) *Riparian Width* - This is the width of the riparian (stream side) vegetation. Width estimates are only done for **forest, shrub, swamp,** and **old field vegetation** if it has **woody** components (*e.g.*, willows). Old field refers to a fairly mature successional field that has stable, woody plant growth; this generally does not include weedy urban or industrial lots that often still have high runoff potential. Two boxes, one each for the left and right bank (looking downstream), should be checked and then averaged.

c) *Floodplain Quality* - The two most predominant floodplain quality types should be checked, one each for the left and right banks (includes urban, residential, etc.), and then averaged. By floodplain we mean the areas immediately outside of the riparian zone or greater than 100 meters from the stream, whichever is wider on each side of the stream. The concept is to identify land uses that might deliver harmful runoff to the stream. These are areas adjacent to the stream that can have direct runoff and erosion effects during normal wet weather. This is considered a ground truthing exercise and we suggest those interested in estimating of the effects of adjacent or riparian land uses use now well-developed GIS approaches. We do not limit it to the riparian zone and it is much less encompassing than the stream basin.

The maximum score for Riparian Zone and Erosion metric is 10 points.

Estimating riparian zone width.



Example of un-restricted livestock access and the formation of "false" banks.

Metric 5: Pool/Glide and Riffle-Run Quality (Figure 15)

This metric emphasizes the quality of the pool, glide and/or riffle-run habitats. This includes pool depth, overall diversity of current velocities (in pools and riffles), pool morphology, riffle-run depth, riffle-run substrate, and riffle-run substrate quality.

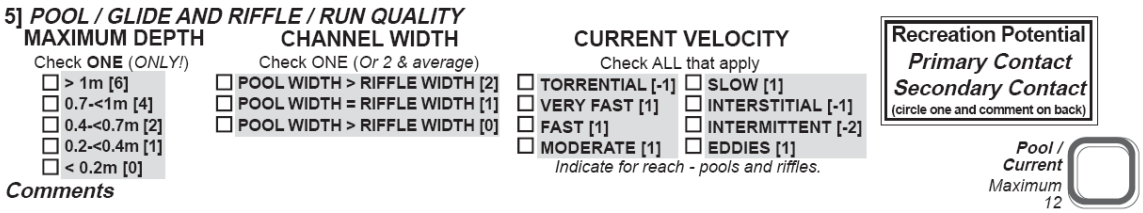


Figure 15. Pool/glide and riffle/run metric

A) Pool/Glide Quality

1) *Maximum depth* of pool or glide; check one box only (Score 0 to 6). Pools or glides with maximum depths of less than 20 cm are considered to have lost their function and the total metric is scored a 0. No other characteristics need be scored in this case.

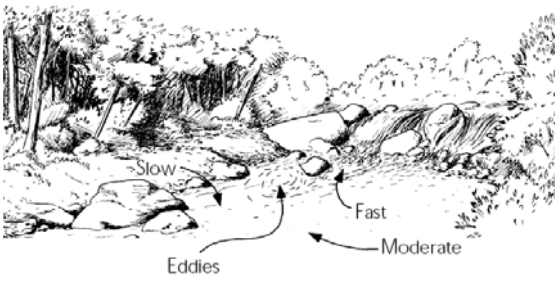


Figure 16. Typical locations of various current velocity types in a stream.

2) *Current Types* - check each current type that is present in the stream (including riffles and runs; score -2 to 4), definitions are: **Torrential** - extremely turbulent and fast flow with large standing waves; water surface is very broken with no definable, connected surface; usually limited to gorges and dam spillway tailwaters. **Very Fast** – turbulent flow that may make it difficult to stand and creates pulsating effect again leg. **Fast** - mostly non-turbulent flow with small standing waves in riffle/run areas; water surface may be partially broken, but there is a visibly connected surface. Fast current has sufficient energy to flow forcefully <u>over</u> objects. Sharp drop evident on depth rod. **Moderate** - non-turbulent flow that is detectable and visible (i.e. floating objects are readily transported downstream); water surface is visibly connected. With moderate current water flows around rather than over objects. Little drop around depth rod. **Slow** - water flow is perceptible, but very sluggish. **Eddies** - small areas of circular current motion usually formed in pools immediately downstream from riffle-run areas. **Interstitial** - water flow that is perceptible only in the interstitial spaces between substrate particles in riffle-run areas. **Intermittent** - no flow is evident anywhere leaving standing pools that are separated by dry areas. The role of bank erosion in sediment delivery to streams is often underestimated. Higher gradient stream showing typical locations of fast, moderate, and slow areas and eddies.

4) *Morphology* - Check **Wide** if pools are wider than riffles, **Equal** if pools and riffles are the same width, and **Narrow** if the riffles are wider than the pools (Score 0 to 2, see Figure 17). If the morphology varies throughout the site average the types. If the entire stream area (including areas outside of the sampling zone) is pool or riffle, then check riffle = pool.

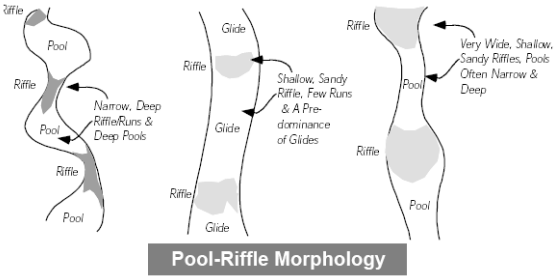


Figure 17. Pool morphology metric categories.

Although the theoretical maximum score for the pool metric is greater than 12 the maximum score assigned for the QHEI for the Pool Quality metric is limited to 12 points.

Illustration of the importance of pool depth to aquatic life

Estimating current velocity, Sharp drop from front to back of rod and boot indicates fast current velocities.

B) Riffle-Run Quality (Figure 18)

This entire metric is scored 0 if no riffles are present.

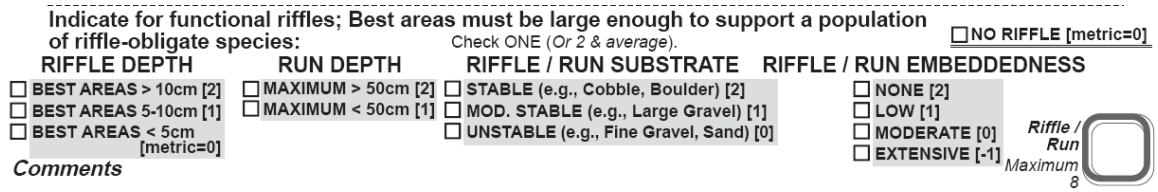


Figure 18. Riffle-run metric.

1)Riffle - select one box that most closely describes the depth characteristics of the best riffle in the zone (Score 0 to 2). The best riffle is selected because we want to identify bottlenecks during harsh periods (*e.g.*, drought). Estimate depths in areas that are clearly riffle, not transitional between a riffle and a run. If the riffle is generally less than 5 cm in depth, riffles are considered to have loss their function and the entire riffle metric is scored a 0.

2) Run Depth - select one box that most closely describes the depth characteristics of the runs (Score 0 to 2). Estimate depth in areas that are clearly run, not transitional between a pool and a run or a riffle and a run.

3) *Riffle/Run Substrate Stability*— select one box from each that best describes the substrate type and stability of the riffle habitats (Score 0 to 2).

4) *Riffle/Run Embeddedness*— Embeddedness is the degree that cobble, gravel, and boulder substrates are surrounded or covered by fine material (sand, silt); here in the riffle/runs only. We consider substrates embedded if >50% of surface of the substrates are embedded in fine material—these substrates cannot be easily dislodged. This also includes substrates that are concreted. Boxes are checked for pervasiveness of (riffle/ run area of sampling zone) embedded substrates: **Extensive** — > 75% of stream area, **Moderate** — 50-75%, **Sparse** — 25- 50%, **Low** — < 25%. The maximum score assigned for the QHEI for the Riffle/Run Quality metric is 8 points.

Metric 6: Map Gradient

Local or map gradient is calculated from USGS 7.5 minute topographic maps by measuring the elevation drop through the sampling area. This is done by measuring the stream length between the first contour line upstream and the first contour line downstream of the sampling site and dividing the distance by the contour interval. If the contour lines are closely "packed" a minimum distance of at least one mile should be used. Some judgment may need to be exercised in certain anomalous areas (e.g. in the vicinity of waterfalls, impounded areas, etc.) and this can be compared to an infield, visual estimate which is recorded next to the gradient metric on the front of the sheet. Scoring for ranges of stream gradient takes into account the varying influence of gradient with stream size, preferably measured as drainage area in square miles or stream width. Gradient classifications (Table V-4-3) were modified from Trautman (p 139, 1981) and scores were assigned, by stream size category, after examining scatter plots of IBI vs. natural log of gradient in feet/mile (see Rankin 1989). Scores are listed in Table 2. The maximum QHEI metric score for Gradient is 10 points

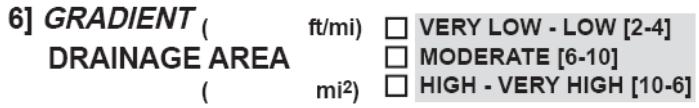


Figure 19. QHEI Stream gradient metric.

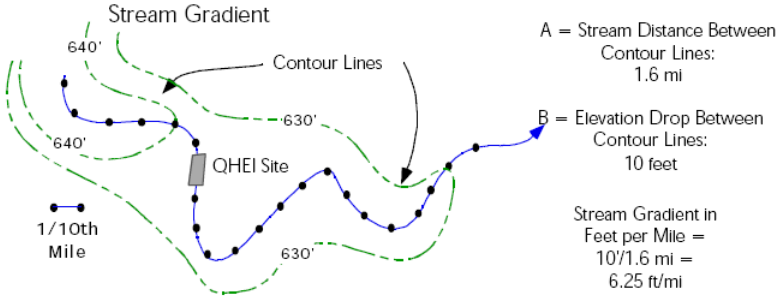


Figure 20. Illustration of methodology for determining stream gradient from topographic maps.

Table 2 Classification of stream gradients for Ohio by stream size. Modified from Trautman (p 139, 1981). Scores were derived from plots of IBI versus stream gradient for each stream size category.

Stream Width	Drainage Area (sq mi)	Gradient (feet/mile)						
		Very Low	Low	Low-Moderate	Moderate	Moderate-High	High	Very High[1]
≤ 4.7	< 9.2	0 - 1.0 **2**	1.1 - 5.0 **4**	5.1 - 10.0 **6**	10.1 - 15.0 **8**	15.1 - 20 **10**	20.1 - 30 **10**	30.1 - 40 **8**
4.8 - 9.2	9.2 - 41.6	0 - 1.0 **2**	1.1 - 3.0 **4**	3.1 - 6.0 **6**	6.1 - 12.0 **10**	12.1 - 18 **10**	18.1 - 30 **8**	30.1 - 40 **6**
9.3 - 13.8	41.7 - 103.7	0 - 1.0 **2**	1.1 - 2.5 **4**	2.6 - 5.0 **6**	5.1 - 7.5 **8**	7.6 - 12 **10**	12.1 - 20 **8**	20.1 - 30 **6**
13.9 - 30.6	103.8 - 622.9	0 - 1.0 **4**	1.1 - 2.0 **6**	2.1 - 4.0 **8**	4.1 - 6.0 **10**	6.1 - 10 **10**	10.1 - 15 **8**	15.1 - 25 **6**
> 30.6	> 622.9		0 - 0.5 **6**	0.6 - 1.0 **8**	1.1 - 2.5 **10**	2.6 - 4.0 **10**	4.1 - 9 **10**	> 9 **8**

[1]Any site with a gradient greater than the upper bound of the "very high" gradient classification is assigned a score of 4.

Computing the Total QHEI Score: To compute the total QHEI score, add the components of each metric to obtain the metric scores and then sum the metric scores to obtain the total QHEI score. The QHEI metric scores cannot exceed the Metric Maximum Score indicated below.

Narrative ranges of QHEI scores

For communicating general habitat quality to the public general narrative categories have been assigned to QHEI scores. Habitat influences on aquatic life, however, occur at multiple spatial scales and these narrative ranges are general and not always definitely predictable of aquatic assemblages are any given site.

Table 2. General narrative ranges assigned to QHEI scores. Ranges vary slightly in headwater (≤ 20 sq mi) vs. larger waters.

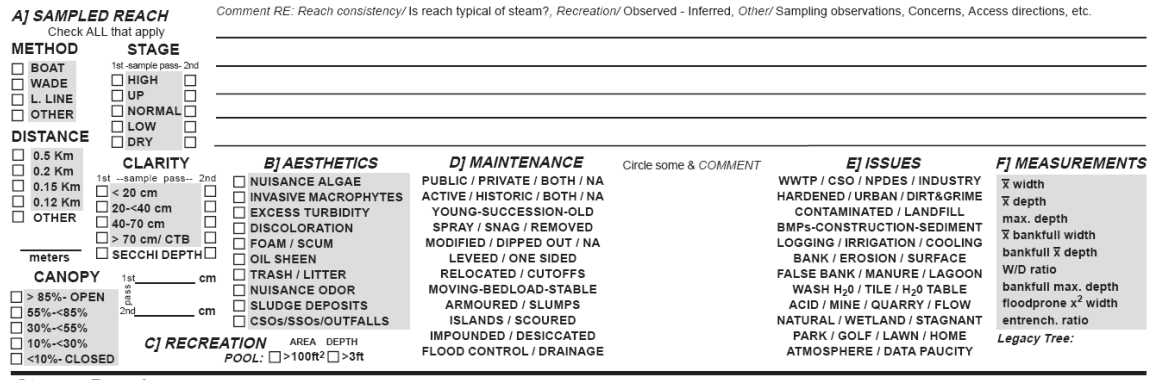
Narrative Rating	QHEI Range	
	Headwaters	Larger Streams
Excellent	≥ 70	≥ 75
Good	55- to 69	60 to 74
Fair	43 to 54	45 to 59
Poor	30 to 42	30 to 44
Very Poor	< 30	< 30

QHEI SCORING (Maximum = 100)

QHEI Metric	Metric Component	Component Scoring Range	Metric Max. Score
1) Substrate	a) Type b) Quality	0 to 21 -5 to 3	20
2) Instream Cover	a) Type b) Amount	0 to 10 1 to 11	20
3) Channel Morphology	a) Sinuosity b) Development c) Channelization d) Stability	1 to 4 1 to 7 1 to 6 1 to 3	20
4) Riparian Zone	a) Width b) Quality c) Bank Erosion	0 to 4 0 to 3 1 to 3	10
5a) Pool Quality	a) Max. Depth b) Current c) Morphology	0 to 6 -2 to 4 0 to 2	12
5b) Riffle Quality	a) Depth b) Substr Stab. c) Substr Embd.	0 to 4 0 to 2 -1 to 2	8
6) Gradient		2 to 10	10

Additional Information/Back of QHEI Sheet

Additional information is recorded on the reverse side of the Site Description Sheet. Several versions of the reverse of the QHEI sheet have been produced over the past 10 years, but this description is based on the most recent revision of the Ohio EPA sheet (Figure 21).



A – Sampling Characteristics

1) *Methods Used* – A series of check boxes to record the type of sampling completed in the reach.

2) *Distance* – Distance assessed for the QHEI and/or fish assessment.

3) *Stage* – Estimate of flow stage during assessment. Since some sites are sampled twice, a box is included for each sampling effort.

4) *Clarity* – Estimate of water clarity during assessment. Since some sites are sampled twice, a box is included for each sampling effort. There are also two places to record Secchi depths, if taken.

5) *Canopy* – Estimate of average width of canopy

B. Aesthetics

1) Check all of the boxes that apply in terms of aesthetic characteristics of the site

C. Recreation
1) Record whether there exists, within the area, greater than 100 ft^2 of water greater than three feet in depth. This is used to estimate whether full body immersion is possible or likely.

D. Maintenance
1) Record what types of stream maintenance activities or special features occur in the sampling zone. Some of this information was previously on the front of the sheet and is used as an aid when determining aquatic life uses (e.g., existing on ongoing channel maintenance).

E. Issues
1) Record various potential sources of impact that may occur in or near the site.

F. Measurements
1) If some quantitative measurements of stream channel characteristics are collected they may be recorded here. It is likely, however, that more detailed stream measurements (e.g., geomorphic assessment) will be recorded on separate forms.

G) *Stream Maps and Diagram*
Stream maps for each site can be very important. The act of drawing a map usually helps to identify habitat types scored with the QHEI. It can also help later samples identify sampling sites and determine whether changes have occurred. The level of detail of the drawings will likely vary with the objective. For example, sites assessed for 401 purposes should have as much detail as possible to help in later decisions of habitat limitations or high potential. Two or three cross-sections of the stream can provide useful information on the stream bank, stream bottom, stream channel, and floodplain characteristics.

QHEI Pool/Riffle Development Metric

Excellent Pool/Riffle Development:

Pools - > 1 m Deep
Glides - Only Transitional Habitats
Runs - > 0.5 m Deep
Riffles - Deep, Large Substrates
Morphology - All Habitats Easily Definable, Riffles Narrow and Deep, Pools Wide with Deep and Shallow Sections

Good Pool/Riffle Development:

Pools - > 0.7 m Deep
Glides - Mostly Transitional Habitats
Runs - Deep, but < 0.5 m
Riffles - Some Deep Areas, Large Substrates (At Least Large Gravels)
Morphology - All Habitats Fairly Well Definable, Riffles Typically Narrower Than Most Pools

Fair Pool/Riffle Development:

Pools - Show Some Depth Variation
Glides - Common
Runs - Typically Absent
Riffles - Poorly Defined, Shallow
Morphology - Habitat Types Not As Distinct, Glides Typically Difficult to
Separate From Pools and Riffles

Poor Pool/Riffle Development:

Pools - Shallow if Present
Glides - Predominant
Runs - Absent
Riffles - Absent, Or if Present Unstable and Shallow With Fine Substrates
Morphology - Mostly Glide Characteristics, Riffles Ephemeral if Present

Qualitative Habitat Evaluation Index and Use Assessment Field Sheet

OhioEPA

QHEI Score: ____

Stream & Location: _____ RM: _ _ _._ Date: _ _/ _ _/ 06

Scorers Full Name & Affiliation: _____

River Code:_ _ _- _ _ _- _ _ _ STORET #:_ _ _ _ _ _ _ Lat./ Long.: (NAD 83 - decimal °) _ _ . _ _ _ _ _ /8_ . _ _ _ _ _ Office verified location ☐

1] *SUBSTRATE* Check *ONLY* **Two** substrate *TYPE BOXES*; estimate % or note every type present

BEST TYPES	POOL	RIFFLE	OTHER TYPES	POOL	RIFFLE
☐ ☐ BLDR /SLABS [10]	____	____	☐ ☐ HARDPAN [4]	____	____
☐ ☐ BOULDER [9]	____	____	☐ ☐ DETRITUS [3]	____	____
☐ ☐ COBBLE [8]	____	____	☐ ☐ MUCK [2]	____	____
☐ ☐ GRAVEL [7]	____	____	☐ ☐ SILT [2]	____	____
☐ ☐ SAND [6]	____	____	☐ ☐ ARTIFICIAL [0]	____	____
☐ ☐ BEDROCK [5]	____	____			

(Score natural substrates; ignore sludge from point-sources)

NUMBER OF BEST TYPES: ☐ 4 or more [2] ☐ 3 or less [0]

Check ONE (*Or 2 & average*)

ORIGIN
- ☐ LIMESTONE [1]
- ☐ TILLS [1]
- ☐ WETLANDS [0]
- ☐ HARDPAN [0]
- ☐ SANDSTONE [0]
- ☐ RIP/RAP [0]
- ☐ LACUSTURINE [0]
- ☐ SHALE [-1]
- ☐ COAL FINES [-2]

QUALITY

SILT
- ☐ HEAVY [-2]
- ☐ MODERATE [-1]
- ☐ NORMAL [0]
- ☐ FREE [1]

EMBEDDEDNESS
- ☐ EXTENSIVE [-2]
- ☐ MODERATE [-1]
- ☐ NORMAL [0]
- ☐ NONE [1]

Substrate ____ *Maximum 20*

2] *INSTREAM COVER* Indicate presence 0 to 3: **0**-Absent; **1**-Very small amounts or if more common of marginal quality; **2**-Moderate amounts, but not of highest quality or in small amounts of highest quality; **3**-Highest quality in moderate or greater amounts (e.g., very large boulders in deep or fast water, large diameter log that is stable, well developed rootwad in deep / fast water, or deep, well-defined, functional pools.

- ____ UNDERCUT BANKS [1]
- ____ OVERHANGING VEGETATION [1]
- ____ SHALLOWS (IN SLOW WATER) [1]
- ____ ROOTMATS [1]
- ____ POOLS > 70cm [2]
- ____ ROOTWADS [1]
- ____ BOULDERS [1]
- ____ OXBOWS, BACKWATERS [1]
- ____ AQUATIC MACROPHYTES [1]
- ____ LOGS OR WOODY DEBRIS [1]

AMOUNT

Check ONE (*Or 2 & average*)
- ☐ EXTENSIVE >75% [11]
- ☐ MODERATE 25-75% [7]
- ☐ SPARSE 5-<25% [3]
- ☐ NEARLY ABSENT <5% [1]

Cover Maximum 20 ____

3] *CHANNEL MORPHOLOGY* Check ONE in each category (*Or 2 & average*)

SINUOSITY
- ☐ HIGH [4]
- ☐ MODERATE [3]
- ☐ LOW [2]
- ☐ NONE [1]

DEVELOPMENT
- ☐ EXCELLENT [7]
- ☐ GOOD [5]
- ☐ FAIR [3]
- ☐ POOR [1]

CHANNELIZATION
- ☐ NONE [6]
- ☐ RECOVERED [4]
- ☐ RECOVERING [3]
- ☐ RECENT OR NO RECOVERY [1]

STABILITY
- ☐ HIGH [3]
- ☐ MODERATE [2]
- ☐ LOW [1]

Channel Maximum 20 ____

4] *BANK EROSION AND RIPARIAN ZONE* Check **ONE** in each category for *EACH BANK* (*Or 2 per bank & average*)

River right looking downstream

EROSION

L	R	
☐	☐	NONE / LITTLE [3]
☐	☐	MODERATE [2]
☐	☐	HEAVY / SEVERE [1]

RIPARIAN WIDTH

L	R	
☐	☐	WIDE > 50m [4]
☐	☐	MODERATE 10-50m [3]
☐	☐	NARROW 5-10m [2]
☐	☐	VERY NARROW < 5m [1]
☐	☐	NONE [0]

FLOOD PLAIN QUALITY

L	R	
☐	☐	FOREST, SWAMP [3]
☐	☐	SHRUB OR OLD FIELD [2]
☐	☐	RESIDENTIAL, PARK, NEW FIELD [1]
☐	☐	FENCED PASTURE [1]
☐	☐	OPEN PASTURE, ROWCROP [0]

L	R	
☐	☐	CONSERVATION TILLAGE [1]
☐	☐	URBAN OR INDUSTRIAL [0]
☐	☐	MINING / CONSTRUCTION [0]

Indicate predominant land use(s) past 100m riparian.

Riparian Maximum 10 ____

5] *POOL / GLIDE AND RIFFLE / RUN QUALITY*

MAXIMUM DEPTH

Check **ONE** (*ONLY!*)
- ☐ > 1m [6]
- ☐ 0.7-<1m [4]
- ☐ 0.4-<0.7m [2]
- ☐ 0.2-<0.4m [1]
- ☐ < 0.2m [0]

CHANNEL WIDTH

Check ONE (*Or 2 & average*)
- ☐ POOL WIDTH > RIFFLE WIDTH [2]
- ☐ POOL WIDTH = RIFFLE WIDTH [1]
- ☐ POOL WIDTH > RIFFLE WIDTH [0]

CURRENT VELOCITY

Check ALL that apply
- ☐ TORRENTIAL [-1]
- ☐ VERY FAST [1]
- ☐ FAST [1]
- ☐ MODERATE [1]
- ☐ SLOW [1]
- ☐ INTERSTITIAL [-1]
- ☐ INTERMITTENT [-2]
- ☐ EDDIES [1]

Indicate for reach - pools and riffles.

Recreation Potential
Primary Contact
Secondary Contact
(circle one and comment on back)

Pool / Current Maximum 12 ____

Indicate for functional riffles; Best areas must be large enough to support a population of riffle-obligate species: Check ONE (*Or 2 & average*). ☐ **NO RIFFLE [metric=0]**

RIFFLE DEPTH
- ☐ BEST AREAS > 10cm [2]
- ☐ BEST AREAS 5-10cm [1]
- ☐ BEST AREAS < 5cm [metric=0]

RUN DEPTH
- ☐ MAXIMUM > 50cm [2]
- ☐ MAXIMUM < 50cm [1]

RIFFLE / RUN SUBSTRATE
- ☐ STABLE (e.g., Cobble, Boulder) [2]
- ☐ MOD. STABLE (e.g., Large Gravel) [1]
- ☐ UNSTABLE (e.g., Fine Gravel, Sand) [0]

RIFFLE / RUN EMBEDDEDNESS
- ☐ NONE [2]
- ☐ LOW [1]
- ☐ MODERATE [0]
- ☐ EXTENSIVE [-1]

Riffle / Run Maximum 8 ____

6] *GRADIENT* (ft/mi) ☐ VERY LOW - LOW [2-4] ☐ MODERATE [6-10] ☐ HIGH - VERY HIGH [10-6]

DRAINAGE AREA (mi²)

%POOL: ____ **%GLIDE:** ____ **%RUN:** ____ **%RIFFLE:** ____

Gradient Maximum 10 ____

A] SAMPLED REACH

Check ALL that apply

METHOD

- ☐ BOAT
- ☐ WADE
- ☐ L. LINE
- ☐ OTHER

STAGE

1st -sample pass- 2nd	
☐ HIGH	☐
☐ UP	☐
☐ NORMAL	☐
☐ LOW	☐
☐ DRY	☐

DISTANCE

- ☐ 0.5 Km
- ☐ 0.2 Km
- ☐ 0.15 Km
- ☐ 0.12 Km
- ☐ OTHER

_____ meters

CLARITY

1st --sample pass-- 2nd	
☐ < 20 cm	☐
☐ 20-<40 cm	☐
☐ 40-70 cm	☐
☐ > 70 cm/ CTB	☐
☐ SECCHI DEPTH	☐

CANOPY

- ☐ > 85%- OPEN
- ☐ 55%-<85%
- ☐ 30%-<55%
- ☐ 10%-<30%
- ☐ <10%- CLOSED

pass 1st _____ cm

pass 2nd _____ cm

Comment RE: Reach consistency/ Is reach typical of steam?*, Recreation/* Observed - Inferred, *Other/* Sampling observations, Concerns, Access directions, etc.

B] AESTHETICS

- ☐ NUISANCE ALGAE
- ☐ INVASIVE MACROPHYTES
- ☐ EXCESS TURBIDITY
- ☐ DISCOLORATION
- ☐ FOAM / SCUM
- ☐ OIL SHEEN
- ☐ TRASH / LITTER
- ☐ NUISANCE ODOR
- ☐ SLUDGE DEPOSITS
- ☐ CSOs/SSOs/OUTFALLS

C] RECREATION

POOL: ☐ >100ft² ☐ >3ft (AREA / DEPTH)

D] MAINTENANCE

Circle some & *COMMENT*

PUBLIC / PRIVATE / BOTH / NA
ACTIVE / HISTORIC / BOTH / NA
YOUNG-SUCCESSION-OLD
SPRAY / SNAG / REMOVED
MODIFIED / DIPPED OUT / NA
LEVEED / ONE SIDED
RELOCATED / CUTOFFS
MOVING-BEDLOAD-STABLE
ARMOURED / SLUMPS
ISLANDS / SCOURED
IMPOUNDED / DESICCATED
FLOOD CONTROL / DRAINAGE

E] ISSUES

WWTP / CSO / NPDES / INDUSTRY
HARDENED / URBAN / DIRT&GRIME
CONTAMINATED / LANDFILL
BMPs-CONSTRUCTION-SEDIMENT
LOGGING / IRRIGATION / COOLING
BANK / EROSION / SURFACE
FALSE BANK / MANURE / LAGOON
WASH H_20 / TILE / H_20 TABLE
ACID / MINE / QUARRY / FLOW
NATURAL / WETLAND / STAGNANT
PARK / GOLF / LAWN / HOME
ATMOSPHERE / DATA PAUCITY

F] MEASUREMENTS

- $\bar{x}$ width
- $\bar{x}$ depth
- max. depth
- $\bar{x}$ bankfull width
- bankfull $\bar{x}$ depth
- W/D ratio
- bankfull max. depth
- floodprone x^2 width
- entrench. ratio

Legacy Tree:

Stream Drawing:

Appendix C:

IDNR Fisheries Stream Sampling Guidelines (2001)

IDNR Fisheries Stream Sampling Guidelines (2001)

IDNR fisheries managers and others involved with the management of Illinois streams need accurate and consistent data on which to base their decisions. Guidelines for IDNR stream sampling help standardize the collection of stream-fish information. Standardized collection allows valid comparisons among sites by minimizing variability in sampling technique. Such comparisons are necessary for effective management and stewardship of stream resources throughout the state. Because Illinois streams differ greatly in physical and biological characteristics, statewide sampling guidelines must be flexible enough to accommodate this variability. These guidelines are intended to optimize data standardization while also accommodating the practical need to adjust sampling procedures to particular situations.

These guidelines were developed for professional, experienced fishery biologists, thoroughly acquainted with the operation, handling and maintenance of the sampling equipment; use of this equipment by inexperienced or uninitiated personnel could result in serious injury.

Background

The baseline and monitoring data collected by the Division of Fisheries provide sport fish population assessments which are important to stream fisheries management and protection (e.g., Sallee et al. 1991, Putman et al. 1995). Additionally, the sampling conducted by Fisheries biologists assists with delineating threatened and endangered species distributions (e.g., Burr et al. 1996) and fish community assessments. As part of the fish community assessments, fisheries data are used for characterizing stream health through the use of the Index of Biotic Integrity (Karr 1981, Karr et al. 1986). Subsequently, the IBI was revised by Hite and Bertrand (1989) and adapted for use in Illinois through the Biological Stream Characterization (BSC) Work Group. The IBI is a major component of the BSC rating of streams (Illinois EPA 1996a) and is used in the Aquatic Life Use-Assessment of the IEPA 305(b) (Illinois EPA 1996b) report to the US EPA, which rates the water quality of Illinois streams. The BSC is also incorporated into the Illinois EPA Targeted Watershed Approach to stream protection and restoration (Illinois EPA 1997).

Stream Sampling Guidelines address the three main objectives of the Division's stream fish sampling. These objectives are: 1) Fish community composition, 2) Sport fishery characterization and 3) Special (targeted) fish studies.

The goal of fish community sampling is to determine the identity and number of fish species present (species richness) and the relative number of individuals of each species (relative abundance) in a stream segment. Because length and weight of individual fish are routinely measured, estimates of species-specific population size and age structure can be obtained. Stream segment fish biomass estimates can also be calculated.

The second objective, Sport fishery characterization, is useful to the Fisheries Division in its

strategic planning efforts and for informing the public on sport fishing opportunities in Illinois streams.

Special (targeted) fish studies are conducted to obtain detailed estimates of population size, population age and growth structure, or migration and movement patterns of particular target species. These studies are often conducted with specific management objectives in mind, such as fish stocking assessments, watershed management evaluations or fisheries response to habitat improvement efforts.

Section 1. Station Selection Criteria

Stations should be selected based upon the following criteria:

1. Sites which have been previously sampled (particularly during the 1981 - 1998 cooperative basin survey effort) should receive priority over sites for which no data have been collected.
2. If no historical fisheries data are available, then site selection should be based on general characteristics of stream habitat, location relative to tributaries or point source pollution, relative position within the watershed (e.g., headwaters, middle, mouth). Consideration should be given for ***both*** representative and unique habitats. For example, if a stream is predominantly channelized, then at least one station should be placed in a channelized reach, even if this is not considered the "best" section of the stream.
3. IEPA ambient water quality or macroinvertebrate sampling sites. Typically, IEPA ambient water quality sites have a substantial water chemistry data set and therefore can be supportive for fisheries data.

Section 2. Sampling station selection

A reconnaissance trip is strongly recommended to familiarize the lead biologist with each potential sampling site. During the reconnaissance, the upstream and downstream limits of the sampling station may be determined and noted on the Stream Reconnaissance Form. The information on the reconnaissance forms should be sufficient to allow any IDNR fisheries biologist to lead the sampling. Although stream conditions can change from time of reconnaissance to time of sampling, this information can reduce confusion regarding where the sample is to be collected.

A reasonable attempt must be made to obtain landowner permission prior to sampling. The process of landowner contact can begin during reconnaissance, or by contacting the Natural Resources Conservation Service, in the county in which the stream segment to be sampled is located, to obtain the name, address and telephone number of the landowner in question. Landowners can then be contacted by phone and/or mail for permission to sample. Landowner information should be filed for subsequent sampling efforts.

Stream sampling locations should be chosen based on the physical characteristics, including stream width and depth, that will influence the amount of stream sampled. Stream segments to be sampled should be selected based upon habitat. Habitat diversity will also influence the length of stream sampled.

For non-channelized or old channelized (> 40 years) streams, *at least one* and preferably two to three pool/riffle sequences should be sampled. The number of pool/riffle sequences will depend upon the geological conditions, stream size and other factors, but this should be a minimum goal. No station should be less that 100 meters in length. If the hydraulic habitat is of a homogeneous nature (e.g., channelized), then a minimum of 15-21x normal base-flow width should be sampled. Normal base-flow is that volume of water that occupies the stream channel up to the vegetation (forbs, grasses, shrubs) line.

Setting the Station limits

Using the the habitat criteria listed above, the upstream and downstream limits of the station are blocked with nets. When setting the nets, every effort should be made to avoid disturbing the area to be sampled. Crew members should not enter the area to be sampled until the nets have been secured and should remain downstream of the sampling area to minimize turbidity disturbance. The preferred location for setting the nets are constrictions or upstream limits of riffles. Consideration should be given to the effects of hydraulic modifications to the stream caused by a bridge, because bridges often present anomalous habitat conditions, they should generally be avoided. The nets should be long enouth to block the entire stream width. Net height should be 6 ft and mesh should be 0.25 inch bar measure. Net stakes should be used to prevent the net from collapsing during the sampling. Usually, one stake for every 10 ft of stream width should be used in low flow conditions. More stakes may be required at higher stream discharges. The stakes are to be placed through the lead line and angled upstream. Metal bottom anchors (J-hooks) should be placed through the lead line to minimize fish escape. These may be supplemented with rocks. The float line should be pulled sufficiently taut to keep fish from jumping over the net, but not so tight that the lead line lifts off the stream bottom.

General Stream Conditions for Sampling

To maintain consistency with IDNR historical collections and optimize efficiency, sampling should be conducted during typical summer **low-flow** conditions. This is typically from early July to mid-September, although sampling could be conducted in June in far southern Illinois. Sampling should not be conducted at high flows without sufficient justification. Due to the lack of gauging stations on small-to-intermediate sized streams, it is difficult to develop standardized criteria for determining the range of flows that is acceptable for sampling, rather this is at the discretion of the lead biologist. Fish sampling and habitat data must be collected at the same flow levels, preferably on the same day or contiguous days.

Related to stream flow, water clarity (turbidity) is a critical component to sampling efficiency. Ambient turbidity will vary regionally in Illinois. For example, in south-central Illinois, the presence of clay-ladened soils contributes to high turbidity levels even in low or no-flow conditions. By comparison, northern Illinois streams with rocky substrates, may have very low turbidity even in high flows. Turbidity should be characteristic for low-flow conditions. In eutrophic streams, phytoplankton blooms or floating aquatic macrophytes may also reduce visibility.

General Fish Sampling Procedures

Wadable sampling techniques should be used in streams with a **average depth of 1.5 ft or less.**

Deep pools, up to 3.5 ft may be encountered in these streams, but they should not be common. When flow is present, wadable electrofishing is conducted from downstream to upstream. This is necessary to avoid creating plumes of silt in the area to be sampled. The increased turbidity limits visibility and reduces sampling efficiency. Only in "no-flow" or pooled conditions can sampling in an upstream to downstream direction be considered an option. Boat sampling and minnow seining may be conducted in either direction.

For all electrofishing, the amount of shocking time and length of stream sampled should be recorded. For minnow seining, the number and length of hauls, width of net used for each haul and average depth should be recorded.

When electrofishing, fish should not be kept in the dip nets and repeatedly subjected to the electrical field. Dip net handles must be made of non-conductive fiberglass or similar material and the net mesh should not be larger than 0.25 inch bar measure.
In community sampling, it is extremely important that ALL nettable fish be collected. Every fish is important and could represent another species. To obtain this type of coverage, all representative habitats should be sampled and must be included in the sampling station.

A reasonable effort should be made to keep all fish alive. For most sampling, an oxygen supply is required and to prevent undo stress which may cause mortality, the use of a 0.5% solution (0.04 lbs per gallon) of non-iodized salt is used. For wadable streams an "R" oxygen bottle provides a convenient source. During any electrofishing effort, if it appears that the number of fish is excessive and will result in stressed fish, then fish must either be redistributed to holding containers with adequate oxygen or sampling must be stopped and fish processed. If sampling is stopped, a block net should be placed at the location where sampling is interrupted. Fish should then be processed and released downstream of the station. A floating cage can also be used to hold fish while being processed. Upon completion of fish processing, sampling should then resume upstream of the temporary block net.

Section 3. Fish Sampling Techniques

Gear selection criteria
1) Boat electrofishing, supplemented with minnow seine hauls, is the method of choice when the habitats present within the station can be reasonably sampled with a boat (i.e., motor lower unit does not frequently contact the substrate and there is enough depth to operate the boat).
2) The electric seine (with block nets) should be used when the station is entirely wadeable (average depth is 1.5 ft or less) and narrow enough to block.
3) The backpack shocker (with block nets) is used when conditions won't permit use of boat electrofishing or electric seine (e.g., small headwater streams).

Boat electrofishing
A boat sampling crew should consist of a minimum of two (2) and up to five (5) people. Although only two people (one netter, one motor operator) are able to sample at a time, the additional people can collect water chemistry data and conduct minnow seining. When the electrofishing crew returns to the access site, fish can be processed immediately by two people and electrofishing can continue for the next run.

For small, non-wadable streams a 12'-14' boat is the preferred size as it allows movement over riffles and in confined areas. Dip net mesh size should be .125 to .25 inch. The motor operator and netter must communicate by using a variety of hand signals because generator noise usually precludes verbal communication. The netter and motor operator must watch for underwater obstructions, livestock fences or other potential hazards, and immediately alert one another to their presence. If anglers are encountered, the motor operator should either turn off the electricity to the electrodes or divert course to reduce disturbance.

If sampling is to be conducted upstream and downstream of the access point, then the downstream segment should be sampled first. This will reduce the likelihood of recapturing fish that are processed from the first sampling run. Because the effects of electrofishing differ among fish species, the crew should often check behind the boat for stunned fish. Frequent circling is recommended to assure adequate coverage of the station.

As at wadable sites, the actual length of a boat sampling station will vary with the stream size, habitat diversity and presence of impassable obstructions. Typically, a boat station will cover from 0.25 mile to one (1) mile. The electrofishing crew should sample all available habitats, including open water and midchannel areas, not just shoreline habitats. Electrofishing time must be accurately recorded. The length of stream sampled (combined length along both banks and midchannel) should be estimated (to within 10ft). This can be done on site (with tape measure or rangefinder) or may be measured on USGS topographic 7.5 minute quadrangle. Unlike wadeable sites, boat sampling stations are sampled for a given time (usually 15 or 30 min individual runs), rather than for a pre-determined distance.

When sampling in shallow water it may be necessary to get out of the boat to push the boat or retrieve fish. If this occurs, the power to the electrodes must be turned off before getting out of the boat.

Minnow Seine

The major emphasis of minnow seine sampling is to determine species occurrences. Minnow seine samples are usually collected to supplement boat electrofishing samples. A minnow seine crew should have a minimum of two (2) and optimally three (3) people. Minnow seining should not be the exclusive gear for non-headwater streams (>10 ft) wide. In headwater streams, conditions may be conducive for efficient minnow seining because stream width and depth allow sufficient 'sampling space' for this method. The length of seine used will vary with stream conditions, depth should be 6 ft and mesh should be 0.125 to 0.25 inch (bar measure). For pool or run conditions, an area relatively clear of obstructions should be selected. Sampling may be conducted either in an upstream or downstream direction. Number and length(s) of seine hauls should be recorded with the fish data. Circular sweeps allow sampling where debris or other obstructions restrict linear sampling. Riffles or deep, fast runs can be sampled by placing the net across the riffle and having a crew member kick from upstream towards the net. If it is a large riffle, select an area up to 15 ft in width and place the net across that area. Then, one or more persons should walk upstream approximately 20 ft and begin kicking the substrate; moving downstream toward the net . When they arrive at the net, “kickers” should reach into the water, find the lead line and purse the net.

For all minnow seine sampling, it is very important that the lead line be kept on the bottom. If an impediment is encountered during a haul, attempts should be made to quickly dislodge or bypass the obstruction. When beaching the seine, keep the lead line pressed to the substrate and pull the seine towards shore. Quickly remove all fish from the seine and process (or preserve). The number and length(s) of seine hauls should be recorded with the fish data.

Electric Seine

For electric seine sampling, the crew should consist of a minimum of five (5) persons with an optimum of six (6). One (1) person is responsible for generator operation and assuring that fish are kept oxygenated. Three (3) members of the crew net fish and two (2) members operate the brails of the electric seine. Skilled brail operators may also opt to carry a dip net for maximum efficiency in confined areas. All persons will wear heavy duty (lineman) rubber gloves and either hip boots or chest waders (preferred). Prior to activating the seine, one of the brail operators must indicate verbally that the seine is going to be turned "ON". Similarly, when it is turned "OFF" one of the operators must indicate that the seine is "OFF".

The pace of sampling should accommodate the netters so that when large numbers of fish are present, the operators should reduce forward progress until fish have been netted and placed in live wells. When appropriate, brush, logs, or shoreline cover should be sampled by having one or both of the brail operators wrap around the cover. The netters should keep pace with the brail operators as they surround the object, to collect stunned fish. Using their dip nets, the netters may need to push the electric seine into the brush or deeper pool to assure full coverage. After this process, the seine could briefly be turned off for the crew to regroup.

If the stream is wider than the electric seine the sampling crew should follow the thalweg, concentrating on instream cover and minimizing deep water fish escape routes. If depth is sufficient across the channel, a second pass may be needed to cover the "unsampled" side.

Riffles should be sampled by first carrying the electric seine upstream of the riffle and having the netters place the nets side by side at the downstream end of the riffle. The brail operators, with the seine "ON", should then kick the riffle to dislodge fish. Depending upon the length of the riffle, this could be done multiple times. If time and manpower allows, a minnow seine can be positioned below the riffle instead of or in addition to side by side dip nets.

Backpack Electrofisher

The backpack electrofishing crew will consist of a minimum of two (2) persons, with three (3) optimal. One person will operate the backpack, one person will net fish and carry a bucket to hold stunned fish. Block nets will be set as noted above. All persons will wear rubber gloves and either hip boots or chest waders. Electrofishing settings will be contingent upon water conditions including conductivity and depth, but settings should be sufficient to optimize collection, but to minimize harm to fish.

Backpack shocking is generally done in an upstream direction for reasons noted above. For optimal catch efficiency, the anode probe is thrust into cover (e.g.,undercut bank, log jam) with

the power "OFF", then drawn slowly back to the operator with the power "ON". This minimizes scaring fish and utilizes the galvanotaxic response of fish to DC current.

Section 4. Habitat and Methods Data

Procedures for completion of stream investigation forms and stream methods and habitat form are in the Operations Manual - FDM 6230 and FDM 6230.1, respectively.

Section 5. Fish Workup

1) Small fishes (e.g. minnows, darters and y-o-y sunfishes) and fishes not easily identified should be preserved in 10% formalin as quickly as possible for ease of identification and value as voucher specimens. Make sure preserved samples are clearly labeled with sampling location, method and date.

2) Weigh and measure length of all fishes greater than or equal to 6", measure length of smaller fishes

3) All reasonable effort should be made to return fish alive back to the stream.

4) Dead fish should be buried (preferred) or scattered throughout the surrounding area at least 50 ft from the stream in areas unlikely to cause inconvenience to stream users or landowners.

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Appendix D:

Methods of Collecting Macroinvertebrates in Streams

Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 167

Standard Operating Procedure for Sample Processing for the Macroinvertebrate Index of Biotic Integrity (mIBI)

Surface Water Section
1021 North Grand Avenue East
P.O. Box 19276
Springfield, Illinois 62794-9276
Contact: Bureau of Water, Quality Assurance Officer 217-782-3362

1.0 Scope and Application

The instructions included in this Standard Operating Procedure (SOP) describe how to obtain a 300 ± 20% fixed-count macroinvertebrate subsample and how to enumerate and identify the individual members at the proper taxonomic resolution in order to properly compute and interpret the macroinvertebrate Index of Biotic Integrity (mIBI).

The SOP is part of a larger mIBI protocol that includes specific macroinvertebrate collection, identification, computation, and application instructions. To ensure a correct application of the mIBI, follow the protocols outlined in the module *"Calculation of the macroinvertebrate Index of Biotic Integrity (mIBI)"*. To ensure that samples are appropriate for the mIBI, follow the protocol described in *"Methods to Collect Aquatic Macroinvertebrates from Wadeable Streams for Biotic Integrity Assessments".*

The mIBI represents how macroinvertebrates respond to and integrate the chemical, physical and biological effects of human-caused impacts on streams and their watersheds when compared to a subset of least disturbed locations sampled during 2001.

2.0 Summary of Method(s)

- Collect samples according to the method outlined in "*Methods to Collect Aquatic Macroinvertebrates from Wadeable Streams for Biotic Integrity Assessments*".
- Rinse the sample with water to remove preservatives.
- Assign unique numbers to identify each of the thirty 6x6-cm grid cells in the screened subsample tray.
- Load the rinsed sample materials into the screened subsample tray.
- Evenly distribute the materials over the subsample tray.
- Submerge the loaded subsample tray in water to partially suspend the sample materials.
- Remove the subsample tray from the water to facilitate random distribution of the sample over the entire screen.
- Randomly select four cells from the gridded subsample tray.
- Place the contents of the selected cells into a shallow white pan. Partially fill the pan with liquid to facilitate macroinvertebrate sorting.
- Sort 300 ± 20% aquatic macroinvertebrates using a ring-light magnifier. Exclude macroinvertebrates (e.g., semi-aquatic taxa and others listed in, "*Illinois Environmental Protection Agency Tolerance List and Functional Feeding Group Classification*", in Appendix A.) from the 300 ± 20% count.

- If fewer than 300 ± 20% macroinvertebrate exist in the initial four cells, randomly select and sort additional sample material, one cell at a time, until the target number of organisms is obtained.

- If greater than 300 ± 20% macroinvertebrates exist in the initial four cells then reduce the amount of material and macroinvertebrates sorted. Place all of the materials from the initial four cells into a second gridded screen and conduct a "*Level-2"* subsample. Subsample the second gridded screen as before and obtain materials from the four ("*Level-2"*) cells. Repeat as necessary (e.g., "*Level-3"*, "*Level-4"* etc.) until the subsample contains 300 ± 20% macroinvertebrates.

- The subsample is complete.

- Identify macroinvertebrate with the taxonomic keys listed below in "*10.0 Macroinvertebrate Keys*".

- Refer to the module "*Calculation of the macroinvertebrate Index of Biotic Integrity (mIBI)"*, for information on the required levels of taxonomic resolution for specimen identification.

3.0 Interferences and Corrective Action

Proper application of the mIBI requires properly collected samples. To collect mIBI samples, follow the instructions outlined in the module "*Method to Collect Aquatic Macroinvertebrates from Wadeable Streams for Biotic Integrity Assessments*". In addition, a 300 ± 20% individual fixed-count subsample is required since this sample size was utilized to develop metric score expectations and mIBI interpretation criteria.

4.0 Safety

To reduce exposure to sample preservatives, protective eyewear and latex gloves should be worn. Process samples in a well ventilated area or under a fume hood. Depending on the preservative utilized, several rinsing and soaking steps may be needed to prepare the sample for processing.

5.0 Equipment and Supplies

Standardized gridded screen (595 micron screen, 30 cells, each 6 cm^2), shallow watertight tray for submerging the gridded screen, 6 cm scoop, 6 cm^2 metal dividing frame, forceps, scissors, white plastic or enamel pan for sorting, ring-light, specimen vials with caps or stoppers, sample labels, standard laboratory bench sheets, 95 percent ethanol for storage of specimens, sample log and tracking sheets.

6.0 General Guidelines (in part from Barbour et. al., 1999)

The procedures, outlined below, follow the methods established in the US EPA Rapid Bioassessment Protocol (Barbour et. al. 1999). These procedures use a fixed-count sub-sampling approach for sorting organisms from the materials included in the sample matrix. The reason for the subsample requirement is that the volume of material collected with the proportionally allocated multihabitat collection approach is typically impractical to process in its entirety. Sub-sampling reduces the effort required for the sorting and identification aspects of macroinvertebrate samples and provides a more

expedient expenditure of time (Barbour and Gerritsen 1996).

To facilitate processing and identification, the randomized 300 ± 20% fixed count subsample is sorted and preserved separately from the remaining sample. Document the level-of-effort, or proportion of sample processed by recording this information on the Laboratory Bench Sheet.

The instructions in this SOP describe two phases of macroinvertebrate sorting. The "*Phase-1"* sort is the source of all mIBI information. A "*Phase-1"* sort refers to the sub-sampling process utilized to randomly select 300 ± 20% macroinvertebrates from the sample. A *"Phase-1"* sort can progress in stages and each stage corresponds to a particular "*sub-sample level*". The level of sub-sampling needed to meet the 300 ± 20% target depends on the density of macroinvertebrates in the sample. "*Phase-2"* sort refers to a fifteen minute search conducted on all remaining unsorted material to check for obviously unique taxa not found during "*"Phase-1"* sub-sampling. "*Phase-2"* organisms are placed in a separate vial and are not included as part of the subsample. Exclude taxa obtained during the "*Phase-2"* sort from all mIBI computations. "*Phase-2"* taxa may be utilized, independent of the mIBI, as an aid when making water quality interpretations.

6.1 Specific Guidelines (in part from Barbour et. al., 1999)

- Record sample collection information on the benthic macroinvertebrate tracking sheets. These sheets are located in the sample log-book. Record sample label information plus the information from the Illinois EPA field assessment form on the benthic macroinvertebrate laboratory bench sheet. This information will include sample ID/station code, method of collection, allocation of jabs, number of containers per sample, initials of collector, start time, duration and sample collection date.

- Thoroughly rinse the sample in a No. 30 mesh (595-µm openings) sieve to remove preservative and fine sediment. Depending on the preservative utilized, this step may need to be initiated several days prior to sub-sampling and this step may need to be repeated daily. The log-book containing sample tracking sheets should be posted in the lab or sorting area to track this process. Large organic material (whole leaves, twigs, algal or macrophyte mats, etc.) not removed in the field should be rinsed, visually inspected, and discarded. Use large Rubbermaid type tubs to facilitate this activity. If the samples have been preserved in alcohol, it will be necessary to soak the sample contents in water for about 15 minutes to hydrate the benthic organisms, which will prevent them from floating on the water surface during sorting. If the sample is stored in more than one container, the contents of all containers for a given sample should be combined and homogenized at this time.

- Use Oregon Department of Environmental Quality standardized sub-sampling gear (Caton, 1991) to obtain 300 ± 20% randomly selected macroinvertebrates from the 20-jab sample. The subsample gear has two parts- the first part is a shallow watertight tray and the second part is a 600-micron screen. The screened tray is equally-divided into a grid of 30 clearly marked cells and each cell measures 6x6-cm. Place the screened tray into the watertight tray and spread the entire sample onto the screened grid. Pour enough water into the gear to immerse the sample. This activity will help suspend the sample and facilitate a more even distribution of sample materials over the screened grid. Then lift the screen out of the tray, which will cause the sample materials to settle evenly onto the screen. When samples are too large to be effectively-sorted in a single tray, place half of the homogenized sample in each of two gridded trays.

- Assign a unique number (e.g., 1-30) to each of the 30 cells in the grid of the screened tray. Randomly select four of the thirty gridded tray cells. Use a random number table or computerized random number generator to randomly select the four numbers. If two gridded trays are needed then eight cells should be selected- four from each of the two trays. The material contained in the four, randomly- selected, cells make the subsample.

- Beginning with the first-four randomly-selected cells (eight cells if two trays were used), remove all organisms and debris contained within the boundary of the pertinent cells and place these materials into a white pan to sort. When transferring organisms from the grid to the sorting pan, consider any organisms that straddle adjoining cells to be on the cell containing the head. In those instances where it may not be possible to determine the location of the head (worms for instance), the organism is considered to be in the cell containing most of its body. When a selected cell has organic debris lying over or entangled among the materials in an adjacent cells, it may be necessary to use a pair of scissors to cut materials along the gridline. Cutting will help prevent the unintended transfer of debris-entangled macroinvertebrates from one cell to another.

- After placing the randomly-selected materials into a shallow white pan, add a small amount of liquid to facilitate sorting. Use a ring light (approximately 1.75X) in the sorting process (this provides focused light on the sorting pan and helps standardize the visual acuity among sorters). Sort through the debris, and remove and count all the macroinvertebrates in the pan.

- If it becomes obvious, at any point during the sorting process, that the density of macroinvertebrates contained in the initial four-cells will exceed the subsample target (300 ± 20%), then suspend the sort. Exceeding the target number of organisms before all four cells are sorted essentially amounts to a "*false-start*". If a "*false-start"* occurs, place the just-sorted organisms back in with the unsorted portion of the initial four-cell subsample. Re-homogenize the material in the subsample. At this point, the composition of the re-homogenized material is as if the four cells were never sorted and were just now randomly selected and transferred from the subsample to the sorting tray. Since the "*false-start*" establishes that too many organisms exist in the re-homogenized material, these four cells must undergo a "*Level-2"* subsample before another sort is attempted. To perform the "*Level-2"* subsample, transfer the re-homogenized materials from the initial four cells into a second gridded tray. Distribute these materials evenly over the second tray and randomly select four new cells one at a time. The four newly-selected cells represent the "*Level-2"* subsample. Perform material dispersion and cell selection for the "*Level-2"* subsample in the same manner as for the initial four cells. It is important to note that *if* the target number of organisms is routinely exceeded in the first four cells, the sorter may elect to automatically perform the "*Level-2"* subsample rather than waste time due to "*false-start*" sorting.

- If the density of organisms is large enough that many more than the targeted number of organisms are contained in the four cells, then transfer the contents of the cells to a third gridded pan and continue as before. If the targeted subsample amount of 300 ± 20% organisms is reached, the subsample is finished. If less than the targeted 300 ± 20% organisms exist, continue randomly selecting and sorting cells, one-at-a-time, until the targeted subsample number is obtained. If picking through the entire next cell is likely to result in a subsample with greater than 300 ± 20% organisms, then subsample that single cell in the same manner as before. That is, spread the contents of the last cell into another gridded pan. Pick cells, one-at-a-time, until 300 ± 20% organisms are reached. The total number of cells for each subsample level should be noted on the laboratory bench sheet. Each cell selected for sorting, must be sorted in its entirety.

- Except for one situation, a 300 ± 20% organism fixed count subsample is required for all macroinvertebrate samples utilized for the mIBI. The exception occurs when the entire sample contains fewer than 240 total individuals. In this situation, the subsample will never reach the 300 ± 20% organism target. Use extreme caution when interpreting mIBI results based on fewer than 240 macroinvertebrates. A 300 ± 20% individuals fixed-count subsample is required since this sample size was utilized to develop metric score expectations and mIBI interpretation criteria

- Until the target number of organisms is obtained, be careful not to accidentally disturb the subsample tray between these activities (e.g., removing cell contents or while sorting) because this may cause a redistribution of specimens that could possibly change the probability of selection if additional cells must be sorted.

- It is important to note that organisms, which are damaged beyond recognition, should not be included in the subsample count. Likewise, exclude all Hemiptera and most semi-aquatic Coleoptera from the subsample count. Regardless of the number of semi-aquatic taxa omitted, the subsample procedure continues until the fixed-count target of 300 ± 20% organisms is attained. Refer to the tolerance assignments in Appendix A, to identify ineligible semi-aquatic taxa. The ineligible taxa have tolerance values of 99.9.

- Once this initial phase of sorting is completed and the target number of organisms is obtained, a fifteen minute "*Phase-2"* sort is conducted on all remaining unsorted material to check for obviously unique taxa not found while obtaining the require 300 ± 20% macroinvertebrates. "*Phase-2"* organisms will be placed in a separate vial and will not be included as part of the initial subsample. Also, exclude taxa obtained during the "*Phase-2"* sort from all mIBI computations. "*Phase-2"* taxa may be useful, independent of the m-IBI, as an aid when making site quality interpretations.

- Note the type of sample, sample ID/station code, sorter, date sorted and any comments regarding the sample (e.g., description of organic material in sample -- sand, fine organics), and sorting time on the bench sheet.

- For 10% of the samples, save the unsorted sample residue in a container labeled "sample residue"; this container should include the original sample label. Save the sorted debris in a separate container labeled "sorted residue" in addition to original label information. Length of storage and archival is to be determined.

- Place specimens sorted as the subsample **(**300 ± 20% organisms) into a glass vial, and preserve in 95 percent ethanol. Label the vial inside with the station code, stream name, collection date and sample ID/station code.

- Any entirely-sorted sample that falls below the designated subsample size of 300 ± 20% may provide spurious information. Use extreme caution when interpreting mIBI results based on fewer than 240 macroinvertebrates. A 300 ± 20% individuals fixed-count subsample is required since this sample size was utilized to develop metric score expectations and mIBI interpretation criteria

7.0 Quality Control (QC) for mIBI samples

- Ten percent of the sorted samples in each lot should be examined by laboratory QC personnel or a qualified co-worker. A lot is defined as a special study, basin study, entire index period, or individual sorter. The QC worker will examine the cells chosen and the tray used for sorting and will look for organisms missed by the sorter. Organisms found will be added to the sample vial. If the QC worker finds that less than 10% of the organisms remain in the sorting tray, the sample passes; if more than 10% are found, the sample fails. If 10 percent of the sample lot fails, another 10 percent is randomly selected until failure no longer occurs or the entire lot is checked. Sorters in-training will have their samples 100 percent checked until sorting efficiency reaches 90%. The results of the QC check are recorded on the Benthic Macroinvertebrate Laboratory Bench Sheet.

- After laboratory processing is complete for a given sample, all sieves, pans, trays, etc., that have come in contact with the sample will be rinsed thoroughly, examined carefully, and picked free of organisms or debris.

8.0 Organism Identification for mIBI Calculations

8.1 General Guidelines

- The taxonomic resolution of macroinvertebrate identification varies for mIBI calculations. In general, it is preferred to identify organisms to species level whenever practical. However, for mIBI calculations use generic resolution for insects and non-sphaerid bivalves, class resolution for leeches, worms and flatworms, and family resolution for crayfish, fingernail clams, and mussels. the taxonomic keys listed below for macroinvertebrate identification. Refer to "*Calculation of the macroinvertebrate Index of Biotic Integrity (mIBI)"* for specific information about the level of taxonomic resolution required for a given taxonomic group.

- When there is a question concerning the correct identification of a specimen, send it to other recognized authorities for confirmation.

- A voucher (reference) collection must be maintained to help resolve questions regarding the accuracy of taxonomic identifications. These specimens should be properly labeled, preserved and stored in the laboratory for future reference. Specimen labels should include the name(s) of the verifying person(s). Senior taxonomist must verify 10% of the sample identifications.

- Information on samples completed (through the taxonomy process) is recorded in the sample notebook or bench sheet to track the progress of each sample within the sample lot. The tracking of each sample will be updated as each step is completed (i.e., rinsing, sub-sampling, sorting and taxonomy).

8.1.1 Specific Guidelines for Organism Identification

- For identification and enumeration, examine organisms under a stereoscopic dissecting microscope. The microscope should have a magnification range of 7X to 120X. After they are identified and counted, all organisms are returned to a single sample vial containing 95% ethanol and the original field sample label, OR original field sample label information (see previous page, section 6.1. "For 10% of the samples, save the unsorted sample residue in a container labeled "sample residue"; this container should include the original sample label. Save the sorted debris in a separate container labeled "sorted residue" in addition to original label information."

- Some taxa require slide mounts for proper identification (e.g., chironomid larvae). Before mounting chironomids, the larvae are made more transparent (cleared) to facilitate the observation of internal structures utilized in taxonomic identification. The process of clearing and mounting the larvae may make certain characteristics difficult to see. Membranous structures such as the Lauterborn organs and the antennal blade become very faint after treatment in warm KOH. Overnight clearing in cool (room temperature) KOH is less damaging and will make structures more visible (Simpson and Bode, 1980). The anal and ventral tubules, preanal papillae, eyespots and body coloration should be observed under a dissecting microscope before the specimen is cleared. Another clearing/mounting agent is CMC-10 mounting media from Masters Group, Inc..

- After being picked from the sample, chironomid larvae should be sorted into groups based on location of eyespots, shape of head and possession of ventral tubules.

- Clear the larvae of soft tissue by placing them in cool 10% potassium hydroxide (KOH) overnight;

  Or

- Heat the KOH on a hot plate (low only) until the head capsule clears, approximately 20-30 minutes.

- Rinse out the KOH by soaking the larvae in distilled water approximately 10 minutes.

- Place the larvae through a graded alcohol series (50%, 70% and 95%) to remove all the water. Failure to remove the water may cause bubbles to appear. Minimum time per rinse is 10 minutes.

- Place approximately two drops of Canada balsam (Permount or other suitable mounting medium) on a microscope slide and arrange the midge with the ventral side up.

- Cover with a coverslip and press down on the head capsule to expose the mouthparts.

- Note: On large larvae the abdomen may prevent the depression of the head capsule. In such cases, mount the abdomen under a separate coverslip, but on the same slide.

9.0 Sample Integrity and Disposition

- To ensure the integrity of each sample, a label containing the information discussed in the subsection on labeling is included in each vial. In order to maintain chain-of-custody, samples are the responsibility of the collecting biologist. Keep samples in a secure location at all times (e.g. lock the vehicle whenever vacant). Final disposition of samples is also the responsibility of

the collecting biologist.

10.0 Macroinvertebrate Keys

Use any available literature as an aid to identify the organisms. However, take the final identification and recorded name from the following references.

PLATYHELMINTHES
TURBELLARIA: Family (Pennak, 1989 pgs 124-51)
ANNELIDA
OLIGOCHAETA: Class (Pennak, 1989 pg 290) / (Kathman and Brinkhurst, 1998)
HIRUDINEA: Family (Pennak, 1989 pgs 314-333)
ARTHROPODA
CRUSTACEA
ISOPODA: Genus (Pennak, 1989 pgs 462-73)
AMPHIPODA
Hyalellidae: species (Pennak, 1989 pgs. 474-88)
Gammaridae: Genus (Pennak, 1989 pgs. 474-88)
DECAPODA
Cambaridae: Genus/species (Page, 1985 INHS Bull. 33:4)
Palaemonidae: species (Page, 1985 INHS Bull. 33:4)
INSECTA
EPHEMEROPTERA
Siphlonuridae: Genus (Merritt and Cummins, 1996)
Isonychiidae: Genus (Merritt and Cummins, 1996)
Baetidae: Genus/species (Merritt and Cummins, 1996 / Morihara and McCafferty, 1979 TAES V.105 / Burks, 1953)
Heptageniidae: Genus/species (Merritt and Cummins, 1996 / Burks, 1953 / Lewis, 1974) / Bednarik and McCafferty, 1979)
Ephemerellidae: Genus (Merritt and Cummins, 1996)
Tricorythidae: Genus (Merritt and Cummins, 1996)
Caenidae: Genus (Merritt and Cummins, 1996)
Baetiscidae: Genus (Merritt and Cummins, 1996)
Leptophlebiidae: Genus (Merritt and Cummins, 1996)
Potamanthidae: Genus (Merritt and Cummins, 1996)
Emphemeridae: Genus (Merritt and Cummins, 1996)
Palingeniidae: Genus (Merritt and Cummins, 1996)
Polymitarcyidae: Genus (Merritt and Cummins, 1996)
ODONATA
ANISOPTERA
Cordulegasteridae: Genus (Merritt and Cummins, 1996 / Needham and Westfall, 1995 / Needham, Westfall and May, 2000).
Gomphidae: Genus (Merritt and Cummins, 1996 / Needham and Westfall, 1955 / Needham Westfall and May, 2000)
Aeshnidae (or Aeschnidae): Genus/species (Merritt and Cummins, 1996 / Needham and Westfall, 1955 / Needham, Westfall and May, 2000)
Corduliidae: Genus (Merritt and Cummins, 1996 / Needham and Westfall, 1955 / Needham, Westfall and May, 2000)
Libellulidae: Genus (Merritt and Cummins, 1996 / Needham and Westfall, 2000)

ZYGOPTERA

Calopterygidae: Genus (Merritt and Cummins, 1996 / Westfall and May, 1996)
Coenagrionidae: Genus (Merritt and Cummins, 1996 / Westfall and May, 1996)
Lestidae: Genus (Merritt and Cummins, 1996 / Westfall and May, 1996)

PLECOPTERA
Pteronarcyidae: Genus (Merritt and Cummins, 1996)
Taeniopterygidae: Genus (Merritt and Cummins, 1996)
Nemouridae: Genus (Merritt and Cummins, 1996)
Leuctridae: Genus (Merritt and Cummins, 1996)
Capniidae: Genus (Merritt and Cummins, 1996)
Perlidae: Genus (Merritt and Cummins, 1996)
Perlodidae: Genus (Merritt and Cummins, 1996)
Chloroperlidae: Genus (Merritt and Cummins, 1996)
HEMIPTERA
Gerridae: Genus (Merritt and Cummins, 1996)
Notonectidae: Genus (Merritt and Cummins, 1996)
Pleidae: Genus (Merritt and Cummins, 1996)
Belostomatidae: Genus (Merritt and Cummins, 1996)
Corixidae: Genus (Merritt and Cummins, 1996)
MEGALOPTERA
Sialidae: Genus (Merritt and Cummins, 1996)
Corydalidae: Genus/species (Merritt and Cummins, 1996)
NEUROPTERA
Sisyridae: Genus (Merritt and Cummins, 1996)
TRICHOPTERA
Hydropsychidae: Genus/species (Merritt and Cummins,1996 / Schmude and Hilsenhoff,1986)
Philopotamidae: Genus (Merritt and Cummins, 1996)
Polycentropodidae: Genus (Merritt and Cummins, 1996)
Psychomyiidae: Genus (Merritt and Cummins, 1996)
Glossosomatidae: Genus (Merritt and Cummins, 1996)
Hydroptilidae: Genus (Merritt and Cummins, 1996)
Rhyacophilidae: Genus (Merritt and Cummins, 1996)
Brachycentridae: Genus (Merritt and Cummins, 1996)
Lepidostomatidae: Genus (Merritt and Cummins, 1996)
Molannidae: Genus (Merritt and Cummins, 1996)
Phryganeidae: Genus (Merritt and Cummins, 1996)
Helicopsychidae: species (Merritt and Cummins, 1996)
Leptoceridae: Genus/species (Merritt and Cummins, 1996 / Glover and Floyd, 2004 / Floyd, 1995)
LEPIDOPTERA
Pyralidae: Genus (Merritt and Cummins, 1996)
COLEOPTERA
Gyrinidae: Genus (Merritt and Cummins, 1996)
Psephenidae (larvae only): Genus (Merritt and Cummins, 1996)
Scirtidae: Genus (Merritt and Cummins, 1996)
Haliplidae: Genus (Merritt and Cummins, 1996)
Hydrophilidae: Genus (Merritt and Cummins, 1996)

Dytiscidae: Genus (Merritt and Cummins, 1996)
Dryopidae: Genus (Merritt and Cummins, 1996)
Elmidae: Genus (Merritt and Cummins, 1996)
DIPTERA
Tipulidae: Genus (Merritt and Cummins, 1996)
Chaoboridae: Genus (Merritt and Cummins, 1996)
Culicidae: Genus (Merritt and Cummins, 1996)
Dixidae: Genus (Merritt and Cummins, 1996)
Psychodidae: Genus (Merritt and Cummins, 1996)
Simuliidae: Genus (Merritt and Cummins, 1996)
Chironomidae: Genus/species (Merritt and Cummins, 1996 / Epler, 1992)
Stratiomyidae: Genus (Merritt and Cummins, 1996)
Tabanidae: Genus (Merritt and Cummins, 1996)
Empididae: Genus (Merritt and Cummins, 1996)
Syrphidae: Genus (Merritt and Cummins, 1996)
Ephydridae: Genus (Merritt and Cummins, 1996)
Sciomyzidae: Genus (Merritt and Cummins, 1996)
Muscidae: Genus (Merritt and Cummins, 1996)
Athericidae: Genus (Merritt and Cummins, 1996)

MOLLUSCA
GASTROPODA: Genus (Pennak, 1989)
PELECYPODA
Corbiculidae: species (Pennak, 1989 / Cummings and Mayer, 1992)
Sphaeriidae: Genus (Pennak, 1989 / Cummings and Mayer, 1992)
Unionidae: Genus/species (Pennak, 1989 / Cummings and Mayer, 1992)

Note:
Class = key to the class level
Family = key to the family level
Genus = key to the genus level
Genus/species = contain genera with only one species for our area or genera which add large numbers and diversity to our samples at the species level. Current species level key are indicated for those genera.
species = only one species should occur for this group in our Illinois occurrence list.

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Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 168

Standard Operating Procedure for Method to Collect Aquatic Macroinvertebrates from Wadeable Streams for Biotic Integrity Assessments

Surface Water Section

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1.0 Scope and Application

The instructions outlined in this Standard Operating Procedure (SOP) describe how to collect and preserve aquatic macroinvertebrate samples from perennial streams and rivers using a quantitative multiple-habitat, 20-jab method (see below). Use these samples to calculate an index of biotic integrity (IBI) and assess the physical, chemical and biological condition of the resource. Collect samples between June 1st and October 15th. These protocols apply to perennial streams and wadeable rivers that typically range in size from 2nd through 6th order. In some situations, these protocols may apply to intermittent streams that meet specific base flow criterion (see below in Section 6.1). Use the macroinvertebrate Index of Biotic Integrity (mIBI) to evaluate 20-jab samples.

Aquatic macroinvertebrates are invertebrates that are visible to the unaided eye, retained in a U.S. Standard No. 30 sieve (595-micron mesh size) and live at least a portion of their lives in the water. Refer to Appendix A for the list of IBI-applicable aquatic macroinvertebrates.

The framework outlined in this SOP is a modification of the "*Multihabitat Approach: D-frame dip-net*" collection methodology that was outlined in EPA, (1999). The modifications consist of additional dip allocation criteria, which improve dip allocation consistency when collecting macroinvertebrates. Hereafter, refer to this modified macroinvertebrate collection method as, the "20-jab" approach.

2.0 Summary of Method(s).

- Collect aquatic macroinvertebrate samples from perennial, 2rd through 6th order wadeable streams.
- Collect aquatic macroinvertebrate samples between June 1st and October 15th.
- Collect the sample with 20- dip-net jabs.
- Use a 600-micron mesh, 18x9-inch rectangular-frame, dip-net.
- Clean equipment between each sample.
- Sample an 18x18-inch area of habitat with each dip-net jab.
- Allocate 20 jabs in the sample reach.
- Use the mean water width of the sample reach to determine how to allocate the 20 jabs among the edge-zone and bottom-zone (*e.g., $\bar{x}$ stream width = bank-zone jabs: <10ft=10, 10-29ft=8, 30-59ft=6 and ≥60ft=4*). See Table 1, in Appendix B.
- Establish the relative amounts of each of the pertinent habitat types within the bottom- zone and bank-zone.
- Bottom zone habitats are: *coarse particle substrate, fine particle substrate, vegetation and plant detritus.* See Table 1, in Appendix B.
- Edge zone habitats are: *submerged terrestrial vegetation, submerged tree roots and brush-debris-jams.* See Table 1, in Appendix B.
- Distribute the bank-zone and bottom-zone jab allotments proportionally among the pertinent habitat types within each of the two respective zones (see Equation 1 and Section 6.2.4).
- Summarize the number of jabs allocated to each habitat in the sample reach.
- Perform 20 jabs.
- Use the jab allocation summary and perform the allocated number of jabs in each of the pertinent habitat types.
- Take jabs in the more productive areas of each habitat type. More productive areas typically occur where current velocity is relatively high and the substrate is relatively stable.
- Distribute multiple jabs within more productive areas, of a particular habitat type, equally along the entire length of the sample reach when possible.

- Composite the collected materials into a 600-micron sieve bucket or a suitably sized watertight plastic tub after each jab is completed.
- Sieve the collected materials to remove excess water (as needed). As needed, discard excess debris after removing all clinging macroinvertebrates- retain these macroinvertebrates in the sample.
- Preserve the sample with 95% ethanol and return it to the lab for subsequent sub-sampling and identification.

3.0 Interferences and Corrective Action.

- Macroinvertebrate communities vary seasonally. Collect aquatic macroinvertebrate samples between June 1st and October 15th.
- Specimens left on the equipment between samples can give false results. Remove all specimens from the equipment before subsequent samples are collected.
- Improper sample preservation makes specimen identification more difficult. Take care to use large enough containers and adequate amounts of 95% ethanol during the initial preservation of the sample. Safeguard preserved samples that contain a relatively large amount of organic matter by decanting the original, spent, preservative and refilling the container using fresh 95% ethanol - perform this maintenance within 1-week of sample collection. Repeat as needed to maintain the sample preservative.

4.0 Safety.

Follow the general field-safety guidelines in the Illinois EPA, Bureau of Water's Surface Water Section, Field Safety Manual (Document Control Number 151) (Illinois EPA, 1994).

5.0 Equipment and Supplies.

- Nitex bottom kick net with an 18x9-inch rectangular opening and a 600-micron mesh size.
- Sieve bucket with a 600-micron stainless steel mesh bottom.
- Plastic rinse pan
- Forceps
- Sample bottles
- 95% ethanol

6.0 General Guidelines

Collect Macroinvertebrate Samples with the 20-jab Method.

6.1 Select a sampling reach that:

- has instream and riparian habitat conditions typical of the entire assessment reach,
- has flow conditions that approximate typical summer base flow,
- has perennial flow, or if intermittent, has continuous width and depth that extends throughout the sampling reach, which should be at least 300 feet. The length of the sample reach should not exceed 800 feet.
- has no highly influential tributary streams,
- has at least one riffle/pool sequence or analog (i.e., run/bend meander or alternate point-bar sequence), if present.,

6.1.1 The 20-jab approach is applicable if:

- Conditions allow the sampler to collect macroinvertebrates (i.e., to take jabs with a dip-net) in all pertinent bottom-zone and bank-zone habitat types that occur in the sampling reach.

AND

- Conditions allow the sampler to apply the 11-transect habitat-characterization, as described in Section E of this manual or to estimate the amount of each of several bottom-zone and bank-zone habitat types using the non-transect method (Appendix B).
- In general, the 20-jab approach applies if more than 50% of the stream is accessible and adequately characterized.

- Between each sample, pick equipment clean of macroinvertebrates and debris. Rinse all sampling equipment in ambient water before collecting the sample.

6.2 Jab allotment (sample reach)

6.2.1 Determine jab allotment for the bottom-zone and bank-zone.

- Follow the instructions provided in *Appendix B* to identify bottom-zone and bank-zone jab allotments. Specifically, use Table 1 (Appendix B) and mean water width of the sample reach to determine the number of jabs allocated to the bottom-zone and bank-zone. Essentially, allocate fewer jabs along the edge as stream size increases. Depending on stream size, jab allotment will range from 4-10 jabs in the bank-zone and from 10-16 jabs in the bottom-zone.

6.2.2 Identify the type and amount of bank-zone and bottom-zone habitat.

- Identify the relative amount of pertinent habitat type in each of the respective bank-zone and bottom-zone. Follow the instructions provided in *Appendix B* to identify the presence and relative proportion of pertinent habitat in the sample reach. Specifically, use the appropriate habitat characterization method, 11-transect versus non-transect method, in Appendix B., to determine the relative proportion of pertinent habitat in each collection-zone. Pertinent bottom-zone habitat types are *fine substrate*, *coarse substrate, detritus and vegetation (Table 1, in Appendix B)*. Pertinent bank-zone habitat types are *submerged terrestrial vegetation, submerged tree roots and brush-debris jams (Table 1, in Appendix B)*.

6.2.3 Summarize and record the relative amount of each of the pertinent habitat types in the collection-zones.

6.2.4 Determine jab allotments for each of the various habitat types within the bottom-zone and bank-zone.

- Take the jab allocation assigned to a particular collection-zone (Section 6.2.3) and in turn, redistribute this allotment of jabs proportionally among the various habitat types within the respective zone (Section 6.2.4). Essentially, allocate more jabs to the more frequently encountered habitat types.
- Based on the definition of each bottom-zone habitat type (Table 1 in Appendix B) and Equation 1 (see below), translate the relative percentage of the given habitat type (Section 6.2.3) to the number of jabs allocated to *fine substrate, coarse substrate, detritus and vegetation*. Similarly, translate the relative percentage of the given bank-zone habitat type to the number of jabs allocated to *submerged terrestrial vegetation, submerged tree roots* and *brush-debris jams*.

- If rounding results in more than 20 jabs for the total allocation across all habitat types, *decrease* the number of jabs allocated to the most-abundant habitat type to limit the total to 20.

- If rounding results in less than 20 jabs for the total allocation across all habitat types, *increase* the number of jabs allocated to the most-abundant habitat type to expand the total to 20.

Number of jabs to perform in a particular habitat type within a particular bank-zone or bottom-zone =

Percentage or length of pertinent habitat type (Table 1 in Appendix B)	÷	Sum of percentages or lengths of all pertinent habitat types	x	Number of collection zone jabs allocated (Table 1 in Appendix B)

6.2.5 Summarize jab allocation

- Within each of the two collection-zones (bank-zone and bottom-zone), summarize the jabs allocated among each pertinent habitat type. For each zone, if the relative percentage of a habitat type is less than 5%, do not allocate jabs to that type. When transforming relative amounts of habitat types into numbers of jabs, round to the nearest whole number (Equation 1).
- In certain circumstances, rounding to the 20 jab target is allowable (see section 6.2.4).
- Occasionally a stream reach will lack bank-zone habitat. In this instance, allocate all 20 jabs to the bottom-zone habitats.

6.2.6 Record the number of jabs allocated to each bottom-zone and bank-zone habitat type.

6.3 Perform 20 jabs.

6.3.1 General guidelines for jabs:

- Collect macroinvertebrates with 20, dip-net jabs, according to the computed jab allotment summary obtained in Section 6.2.6.
- Use an 18x9-inch rectangular dip-net with a Standard #30 (600-micron) mesh size.
- One person performs all jabs taken within both collection zones (bank-zone or bottom-zone).
- For each of the given habitat types, take jabs in the more productive, areas. More productive areas generally occur where current velocity is relatively high. To minimize the potential for sampling bias attributable to uneven spatial distribution of macroinvertebrates throughout an entire sampling reach, distribute multiple jabs in more productive, areas of a particular habitat type as evenly as possible throughout the sampling reach-- providing the particular habitat type and multiple productive areas within the particular habitat type occur throughout the sample reach. In each of the two collection-zones, bank versus bottom, if there is not enough sampling area to allow performing all of the jabs allocated to a particular habitat type in the given zone, then, perform the remaining jabs among the remaining habitat

types in the given zone. Allocate these remaining jabs in proportions as close as possible to the original allotments. If the bank-zone lacks bank-zone habitat, re-allocate all 20 jabs proportionally among bottom-zone habitats.

- Between jabs (as needed), composite the dip-net contents into a 600-micron sieve bucket or large plastic tub. These containers facilitate debris removal and provide convenient storage when compositing the materials collected with the 20 jabs prior to sample preservation.

- Debris removal (e.g., coarse-particle substrates) greatly facilitates laboratory sub-sampling and sorting of the preserved sample. To remove large objects from the sample, place the material in a dip net, sieve bucked or watertight tub and vigorously agitate, rinse, brush, or pick the objects (as needed) to remove attached organisms. Discard the debris after all macroinvertebrates are removed. Retain all detached macroinvertebrates and return them to the sieve bucket. To remove ultra-fine particles (e.g., sitl), rinse the sample in a sieve bucked until the unwanted particles are washed away. Be careful to not splash out any organisms. Remove sand by transferring the collected materials from the dip-net into a shallow plastic pan. Add a small amount of water to the pan (e.g., ~1-inch deep) and swirl the contents being careful to not splash out any organisms. Pour the water and dislodged organisms into the sieve bucket while retaining as much of the sand as possible in the rinse pan.
- After rinsing, drain, bottle and preserve the sample.

6.3.2 Specific instructions:

- To perform a jab, place the net immediately downstream from the targeted bottom-zone or bank-zone habitat type and dislodge macroinvertebrates by disturbing an 18x18-inch area of substrate. At higher water velocities, these activities will flush dislodged substrate and macroinvertebrates directly into the stationary dip-net. At lower velocities, the same activity becomes ineffective and an additional step is required. Immediately after the upper layer of an 18x18-inch patch of substrate is collected, direct several net sweeps through the plume of materials that forms and is momentarily-suspended directly above the just-sampled patch of streambed. If possible, sweep in an upstream direction.

- When large, coarse-particle substrates (e.g., cobble and boulders) are sampled, wash, brush, or pick surface-clinging organisms from each object. Retain all of the dislodged organisms and discard the cleaned object. As needed, use a dip-net, sieve bucket or watertight tub to composite the materials collected with each jab(s). Use the sieve bucket or dip-net to remove excess water or fine particle materials from the sample.

- When fine-particle streambed substrates (e.g., silt/mud, sand) are sampled, disturb and collect bottom materials from the upper 1-inch of streambed in an 18x18-inch patch by repeatedly bumping the leading edge of the dip-net along the streambed surface until the entire sample area is disturbed. Immediately after the upper layer of material from the 18x18-inch patch is collected, complete the jab by repeatedly sweeping the net upstream through the plume of materials that becomes dislodged and momentarily-suspended over the disturbed area of streambed. Before preserving the sample, remove excess fines and sediment (see above).

- Large, pieces of wood are sampled if they occupy an 18x18-inch sample area AND if their dimensions allow fitting these objects into the dip-net AND they have microbial conditioning. When sampling large woody-debris, wash, brush, or pick surface-clinging macroinvertebrates from the collected materials and retain these organisms in the dip net. After all macroinvertebrates are retained, discard the woody debris.

7.0 Sample Preservation.

After completing 20-jabs, sieve and transfer the collected materials to an appropriate leak-proof jar(s). Label the container and preserve the sample with 95% ethanol by immersing the sample material. If a sample contains large amounts of organic debris, check for sufficient preservation within five days (or sooner) of initial "fixing". As needed, decant the spent preservative and add more 95% ethanol to ensure continued sample preservation. Thereafter, periodically check and re-preserve the sample as needed.

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Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 169

Standard Operating Procedure for Methods to Collect Aquatic Macroinvertebrates with Multi-plate Artificial Substrate Samplers

Surface Water Section

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1.0 Scope and Application

The Standard Operating Procedure (SOP) describes how to collect aquatic macroinvertebrates with multi-plate, artificial substrate samplers. Use these samples to evaluate aquatic life designated use attainment as well as NPDES permit compliance. The methods outlined in this SOP, generally applies to flowing waters that are non-wadeable.

Aquatic macroinvertebrates are invertebrates that are visible to the unaided eye, retained in a U.S. Standard No. 30 sieve (595-micron mesh size) and live at least a portion of their lives in the water. Refer to Appendix A for the list of SOP-applicable aquatic macroinvertebrates.

2.0 Summary of Method

- Use multi-plate artificial substrate samplers when rivers and streams are non-wadeable.
- Collect macroinvertebrates during June 1st through October 15th.
- Sample in a manner that places three multi-plate artificial substrate samplers per monitoring site.
- Set the samplers in locations that have at least 0.3 feet per second current velocity.
- Place one sampler on the Right Descending Side,
- Place one sampler on the Left Descending Side
- Place one sampler in the Center of Flow.
- The ideal situation for sampler placement by may not always be possible. To ensure colonization of an adequate representation of the macroinvertebrate community, a minimum of three circular multi-plate samplers are installed by IEPA biological staff at each sampling location and left for a period of four to six weeks (Weber, 1976). Samplers should be hung together to be considered as replicates. In larger river systems, such as the Illinois, Mississippi, Ohio, and Wabash, it is recommended that three replicates (one set) be installed near each bank.
- In all cases, suspend samplers well up in the photic zone.
- Retrieve the samplers after 4-6 weeks.
- At a given monitoring site, composite the macroinvertebrates obtained from the three samplers.
- Randomly subsample the composited macroinvertebrates to a fixed count of 300 ± 20 percent individuals.

3.0 Interferences and Corrective Action

Factors affecting colonization of artificial substrates by macroinvertebrates have been discussed in studies by Mason et al. (1973) and Roby (1976) and summarized in methods manuals prepared by the U.S. Geological Survey (Slack et al, 1973) and the U.S. Environmental Protection Agency (Weber, 1973). Variables affecting artificial substrate performance are minimized when possible and consistent between sampling stations. Factors affecting macroinvertebrate colonization which are considered when planning and conducting aquatic investigations are listed below:

- Depth of sample placement
- Light intensity – shading
- Stream velocity
- Season
- Duration of sample placement
- Fluctuating water levels

- Organism selectivity
- Sedimentation of sampler surfaces
- Predation
- Detritus accumulation
- Organism drift
- Vandalism

Advantages and Limitations (Modified from Weber, 1973)

While some of the factors affecting artificial substrate performance are considered as drawbacks to this sampling approach, there are definite advantages to be considered in utilizing artificial substrates. Some of the more important advantages and limitations of artificial substrates are listed below:

Advantages

- The confounding effects of substrate differences are reduced.
- A higher level of precision is obtained as compared to other sampling devices.
- Quantitatively comparable data are obtained from environments which are virtually impossible to sample with other devices.
- Samples usually contain negligible amounts of extraneous material, permitting quick laboratory processing.
- Artificial substrates may be used as a bioassay tool (Burks and Wilhm, 1977).
- Statistical inferences may be made when replicate samples are taken.
- Substrate standardization allows comparison of data from different investigators.
- In addition to the above, use of uniform artificial substrates allows:
  - biomass and/or productivity estimates
  - calculation of diversity indices
  - detection of subtle differences in certain macroinvertebrate communities not readily apparent from qualitative studies

Limitations

- The need for a long exposure period (4-6 weeks) makes the samplers unsuited for short-term studies.
- Samplers and floats are sometimes difficult to anchor in place and may present a navigation hazard.
- Samplers are vulnerable to vandalism and are often lost.
- Samplers provide no measure of the condition of the natural substrate at a station or of the effect of pollution on that substrate, including settled solids.
- Samplers only record the community that develops during the sampling period, thus reducing the value of the collected fauna as indicators of prior conditions.
- Samplers are selective for drift organisms.

4.0 Safety

Follow the general field-safety guidelines in the Illinois EPA, Bureau of Water Surface Water Section, Field Safety Manual (Document Control Number 151) (Illinois EPA, 1994).

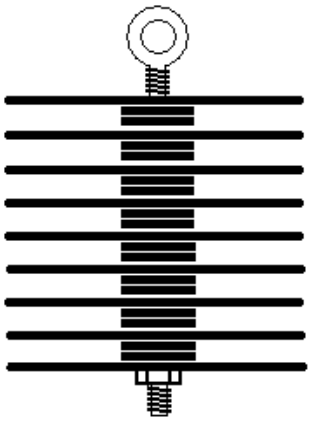
5.0 Equipment and Supplies

-Circular hardboard multi-plate artificial substrate samplers (see schematic)
-2x8x16-inch concrete blocks (1-per sampler)
-An assortment of assorted length threaded rod plus a sufficient number of threaded couplers to join the rods to the cinder blocks and the multi-plate artificial substrate samplers to the rods.
-Sieve bucket with a 600-micron stainless mesh bottom
-Forceps
-Sample bottles
-95% ethanol

6.0 General Instructions

A wide variety of artificial substrates has been developed for the evaluation of benthic macroinvertebrate communities. The first samplers of this type consisted of trays of rocks placed in lakes (Moon, 1935). Other investigators have used barbecue baskets filled with limestone rocks (Mason. et al, 1973) or round spheres (Jacobi, 1971). The hardboard multi-plate type sampler was first used by Hester and Dendy (1962) and numerous variations of this artificial substrate design have subsequently evolved (Arthur and Horning, 1969; Fullner, 1971; Parsons and Tatum, 1974; and McDaniel, 1974). Fullner (1971), concluded in a study comparing the effectiveness of basket and multi-plate samplers that the multi-plate sampler collected a wide variety of invertebrates and its performance compared favorably to that of the basket sampler. Mason et al. (1973) similarly found that multi-plate samplers yield samples comparable to the basket samplers and concluded that acceptable collecting efficiency is attained with three replicates.

Weber (1973) recommended using circular multi-plate samplers consisting of 14 hardboard plates, each 7.5 cm in diameter. A modification of this design was developed for use by Illinois EPA staff biologists in 1974 (see Figure 1).



Sampler design consists of nine circular hardboard plates 0.35 mm thick and 7.25 cm in diameter each separated by two circular hardboard spacers 0.35 mm thick and 2.1 cm in diameter. Plates and spacers are strung on a 3/16" eye bolt and secured with a 3/16" nut. Each sampler has 717.4 cm (0.072 m) available for macroinvertebrate colonization. Note that the above diagram is not to scale.

Figure 1. Circular multi-plate hardboard artificial substrate sampler

Applicable Sampler Use

Use multi-plate artificial substrate samplers for sample reaches that are non-wadeable. In general, the multi-plate sampler approach is applicable if less than 50% of the stream is accessible by wading.

Sampler Placement

Install multi-plate samplers in areas which are representative of overall water quality of the stream in question. Use one cinder block to anchor each sampler. Affix a threaded rod to a 2x8x16-inch cinder block, or equivalent, and attach a multi-plate sampler to the free end of the rod. Use threaded couplers to connect the threaded rod(s), multi-plate samplers and cinder blocks. Use various length rods to account for the various water depths encountered among monitoring sites and to suspend samplers well up in the photic zone. Care is taken when placing samplers in water with fluctuating levels.

- Always install multi-plate samplers in flow with a velocity ≥ 0.3 feet per second (Ohio EPA, 1988).
- Place multi-plate sampler at three locations within a given monitoring site.
- Place one sampler on the Right Descending Side,
- Place one sampler on the Left Descending Side
- Place one sampler in the Center of Flow.
- The ideal situation for sampler placement by may not always be possible. To ensure colonization of an adequate representation of the macroinvertebrate community, a minimum of three circular

multi-plate samplers are installed by IEPA biological staff at each sampling location and left for a period of four to six weeks (Weber, 1976). Samplers should be hung together to be considered as replicates. In larger river systems, such as the Illinois, Mississippi, Ohio, and Wabash, it is recommended that three replicates (one set) be installed near each bank.

-

- In all cases, suspend the samplers well up in the photic zone.

In deeper waters, suspend the samplers by other methods (e.g., floats, docks etc.,). In all cases, suspend the samplers well up in the photic zone.

Sampler Retrieval

Artificial substrate samplers collected after the colonization period of 4-6 weeks and are retrieved individually with the aid of a container with a No. 30 mesh screen bottom. Samplers, once located, are raised very slowly until the collection container can be placed under the sampler and the wire cut. Each sampler along with all organisms attached are then placed in a container of 95% ethanol. If time permits, each circular multi-plate sampler is placed in a bucket of screened water and dismantled. The organisms and debris are then removed from the substrate material. This is accomplished by shaking the material in the bucket of water and by scraping or brushing actions. Care is taken to ensure removal of all the specimens. The water in the bucket is then poured through a U.S. Standard No. 30 sieve to remove the fine particles. All organisms detached in the retrieval process are then placed in sample containers containing 95% ethanol.

Sample processing

- At a given monitoring site, composite the macroinvertebrates obtained from the three samplers.
- Randomly subsample the composited macroinvertebrates to a fixed count of 300 ± 20 percent individuals.

Table 1. Nine plate circular artificial substrate conversions, organisms per sampler to estimated number organisms per square meter.

#/Sampler	#/m2	#/Sampler	#/m2	#Sampler	#/m2	#/Sampler	#/m2
1	14	26	364	51	714	76	1064
2	28	27	378	52	728	77	1078
3	42	28	392	53	742	78	1092
4	56	29	406	54	756	79	1106
5	21	30	1290	55	2365	80	3440
6	258	31	1333	56	2408	81	3483
7	301	32	1376	57	2451	82	3526
8	344	33	1419	58	2494	83	3569
9	387	34	1462	59	2537	84	3612
10	430	35	1505	60	2580	85	3655
11	473	36	1548	61	2623	86	3698
12	516	37	1591	62	2666	87	3741
13	559	38	1634	63	2709	88	3784
14	602	39	1677	64	2752	89	3827
15	645	40	1720	65	2795	90	3870
16	688	41	1763	66	2838	91	3913
17	731	42	1806	67	2881	92	395
18	774	43	1849	68	2924	93	3999
19	817	44	1892	69	2967	94	4042
20	860	45	1935	70	3010	95	4085
21	903	46	1978	71	3053	96	4128
22	946	47	2021	72	3096	97	417
23	989	48	2064	73	3139	98	4214
24	1032	49	2107	74	3182	99	4257
25	1075	50	2150	75	3225	100	4300

Table 1. Nine plate circular artificial substrate conversions, organisms per sampler to estimated number organisms per square meter.

#/Sampler	#/m2	#/Sampler	#/m2	#Sampler	#/m2	#/Sampler	#/m2
1	14	26	364	51	714	76	1064
2	28	27	378	52	728	77	1078
3	42	28	392	53	742	78	1092
4	56	29	406	54	756	79	1106
5	70	30	420	55	770	80	1120
6	84	31	434	56	784	81	1134
7	98	32	48	57	798	83	1148
8	112	33	462	58	812	83	1162
9	126	34	476	59	826	84	1176
10	140	35	490	60	840	85	1190
11	154	36	504	61	854	86	1204
12	168	37	518	62	868	87	1218
13	182	38	532	63	882	88	1232
14	196	39	546	64	896	89	1246
15	210	40	560	65	910	90	1260
16	224	41	574	66	924	91	1274
17	238	42	588	67	938	92	1288
18	252	43	602	68	952	93	1302
19	266	44	616	69	966	94	1316
20	280	45	630	70	980	95	133
21	294	46	644	71	994	96	134
22	308	47	658	72	1008	97	1358
23	322	48	672	73	1022	98	1372
24	336	49	686	74	1036	99	1386
25	350	50	700	75	1050	100	1400

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Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 173

Standard Operating Procedure for Methods to Collect Aquatic Macroinvertebrates with Grab Samplers

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1.0 Scope and Application

The Standard Operating Procedure (SOP) describes how to collect aquatic macroinvertebrates with grab samplers. Occasionally these types of alternative macroinvertebrate collection approaches are required. These alternative approaches are needed when the usual methods of hand-picking, kick sampling or net sweeps are impractical because of the physical setting of the sample reach. Excessive depth in large river systems is one example that may preclude the use of one collection method over another. When conventional sampling devices are impractical, substitute artificial substrate samplers, petite ponar and/or Ekman grabs when appropriate. The time of year, water depth and/or water velocity as well as the study objectives are simultaneously evaluated in order to identify an appropriate sample collection method.

Grabs are devices designed to penetrate the substrate by virtue of their own weight and leverage, and have spring or gravity activated closing mechanisms. In shallow waters, some of these devices may be rigged on poles or rods and physically pushed into the substrate to a predetermined depth. Grabs with spring-activated closing devices include the Ekman, Shipek and Smith-McIntyre; gravity-closing grabs include the Petersen, Ponar and Orange Peel.

Grab samplers currently used by Illinois EPA biological staff include the Petersen, Ponar, Petite Ponar and Ekman. Of the above devices, the Petite Ponar and Ekman are lightweight and designed for hand line operation. The Petersen and Ponar grabs, weighing about 70 lbs. each, are not recommended for hand-line use.

Mechanical grabs are designed for sampling bottom substrates in lakes and slow-to-moderate velocity streams. Because bottom substrates and stream velocity differs widely in the aquatic environment, certain inherent design limitations of grab samplers should be understood before utilizing these samplers in macroinvertebrate investigations.

Aquatic macroinvertebrates are invertebrates that are visible to the unaided eye, retained in a U.S. Standard No. 30 sieve (595-micron mesh size) and live at least a portion of their lives in the water. Refer to Appendix A for the list of SOP-applicable aquatic macroinvertebrates.

2.0 Summary of Method

- Depending on the survey goals, choose an appropriate sample collection location
- Clean the grab sampling equipment
- Set the sampler trigger device
- With a rope, lower the grab sampler to the stream or lake bottom and trigger the jaw closing mechanism
- Retrieve the sampler and empty the grab contents into a sieve bucket
- As needed, rinse the sample in ambient water to remove unwanted debris
- Place the sample in watertight plastic sample bottles
- Label and preserve the sample with 95% Ethanol

3.0 Interferences and Corrective Action

The time of year has an effect on the size, number and general availability of aquatic forms. Investigations for which the objective is to determine the maximum impact of a certain municipality or industry on a stream community are ideally conducted during summer periods of low stream flow and maximum temperatures. Sampling at other times may also be necessary. Dilution for organic and/or toxic wastes will generally be minimal during low flow periods and elevated stream temperatures will produce maximum fluctuations in diurnal oxygen concentrations. Macroinvertebrate samples collected for basin investigations or long term monitoring stations should be collected during the same time frame to preclude seasonal variability in distribution and abundance from meaningful data evaluations. Illinois EPA conducts macroinvertebrate sampling between June 1st and October 15.

Advantages and Limitations (Modified from Weber, 1973)

Advantages

- Quantitatively comparable data are obtained from environments which are virtually impossible to sample with other devices.
- Statistical inferences may be made when replicate samples are taken.
- Allows biomass and/or productivity estimates

Limitations

- Depth of Penetration and Angle of Closure
- Depth of penetration is a very serious problem and depends on the weight of sampler as opposed to the particle size and degree of compaction of the bottom sediments. The Ekman Grab is light in weight and most useful for sampling soft, finely divided substrates composed of varying proportions of fine sand, clay silt, pulpy peat, and muck. For clay hard pan and coarse substrates, such as coarse sand and gravel, the heavier grabs such as the Ponar and Petersen, are most satisfactory. Auxiliary weights may be added to aid penetration of the substrate and to add stability in heavy currents and rough waters.
- Because of differences in the depth of penetration and the angle of "bite" upon closure, data from the different grabs are not comparable. The Ekman essentially encloses a square which is equal in area from the surface to maximum depth of penetration before closure. In soft substrates, for which this grab is best suited, the penetration is quite deep and the angular closure of the spring-loaded jaws has very little effect on the volume of sample collected. In essence, this means that if the depth of penetration is 15 cm. the organisms lying at that depth have the same opportunity to be sampled as those lying near the surface.
- In clam-shell type devices, such as the Petersen and Ponar, the original penetration is often quite shallow. Because of the sharp angle of "bite" upon closure, the area enclosed by the jaws, as they close, decreases at increasing depths of substrate penetration. Therefore, within the enclosed area, organisms found at greater depths do not have an equal opportunity to be sampled.
- Incomplete Jaw Closure:
- Probably one of the most frustrating aspects of sampling macroinvertebrates with various types of grabs relates to the problem of incomplete closure of the jaws resulting in loss of the sample. Any object, such as clumps of vegetation, woody debris and gravel, that cannot be sheared by the closing action of the jaws often prevents complete closure. In the order of their decreasing ability to shear obstructing materials, the common grabs may be ranked: Ponar, Petersen and Ekman. If the Ekman is filled to within more than 5 cm of the top, there may be loss of substrate material on retrieval. An

advantage of the Ekman grab is that the surface of the sediment can be examined upon retrieval, and only those samples in which the sediment surface is undisturbed are retained.

- Bottom Wash-Out:
- All grabs produce a "shock" wave as they descend. This disturbance can affect the efficiency of the sampler by causing an outward wash (blow-out) of flocculent materials near the mud-water interface that may result in inadequate sampling of near-surface organisms such as phantom midge larvae, and some chironomid midges. The shock wave of the Ekman grab is minimized by the sue of hinged, freely-opening top flaps. The Ponar grab is a modified Petersen with side curtains and a screen on the top, allowing water to pass, and thus reducing the shock wave. The shock wave effect is minimized in hand sampling by first obtaining the approximate sampling depth in a trial grab and then marking this depth on the rope with a knot or other means. When approaching this mark on subsequent grabs, the rate of descent is substantially reduced in the final few meters to minimize the bottom wash-out effect.
- In many of the larger rivers, stream velocity presents special problems to grab type sampling devices. In areas of moderate depth and high velocity, it is extremely difficult, if not impossible to get a grab sampler to sit flat on the bottom substrate to achieve a consistent sample "bite". Grab samplers are not used in high velocity situations where a uniform "bite" cannot be ascertained for proper quantification of sample data.

4.0 Safety

Follow the general field-safety guidelines in the Illinois EPA, Bureau of Water Surface Water Section, Field Safety Manual (Document Control Number 151) (Illinois EPA, 1994).

The Petite Ponar dredge is a heavy, center-pivot sampler that presents a pinching hazard when closing. Use caution when handling an open Ponar sampler. Keep the safety pin that locks the jaws open in place at all times other than when collecting a sample. The Eckman dredge is a messenger style spring-loaded jaw sampler that presents a pinching hazard when closing. Use care when handling the sampler while collecting a sample. Transport the sampling device with the jaws in the closed position.

5.0 Equipment and Supplies

- Dredge Sampler Equipment: Petite Ponar dredge or Eckman dredge
- Sieve bucket with Stainless Steel mesh (600 micrometer mesh size)
- Pan (minimum size 12x12x3 inch)
- Ethanol alcohol (90%)
- Non-phosphate Detergent (e.g., Liqui-Nox®)
- Cleaning Brush with non-metallic bristles
- Waterproof Marker and pencil
- Tape and paper for labels
- Sample Bottles:
- Several plastic 1 gallon jugs
- Several 250-500 ml plastic bottles
- Forceps

5.0 General Instructions

The spatial and temporal distribution of aquatic macroinvertebrates necessitates special attention be given to: survey objectives, site selection, sampling season and selection of sampling equipment.

5.1 Determine the Survey objectives
Comparative water quality studies
- physicochemical
- chemical

Faunal studies

5.2 Sampling Season

Macroinvertebrate samples collected for basin investigations or long term monitoring stations should be collected during the same time frame to preclude seasonal variability in distribution and abundance from meaningful data evaluations. Illinois EPA conducts macroinvertebrate sampling between June 1st and October 15.

5.3 Select the appropriate sampling gear

Selection of quantitative sampling equipment is influenced by many factors, some of which include study objectives, manpower considerations and equipment availability. Stream size, depth, velocity and type of bottom substrate are other factors which influence selection of appropriate sampling equipment.

5.4 Site Selection

Select an appropriate sampling location based on the type and goals of the study. In general, the sampling locations should be:
- representative of overall stream conditions
- comparable in bottom substrate, depth, velocity and shading

Walk, wade or float (via boat or canoe) to the desired sampling location. Clean and rinse all sampling equipment before use as follows:
- Between each individual sample, the equipment should be picked clean of any debris
- All equipment can then be rinsed with ambient lake water
- All macrophytes and any other debris must be removed from the boat and trailer to avoid the spread of aquatic invasive species.

5.5 Grab Sample Collection

After verifying that the grab sampler is clean set the sampler trigger device. With a rope, lower the grab sampler to the stream or lake bottom and trigger the jaw closing mechanism. The Petite Ponar dredge is a heavy, center-pivot sampler with jaws that trigger shut when the dredge bumps the bottom. The Eckman dredge is a messenger style spring-loaded jaw unit that snaps shut after the weighted messenger is released from the surface, travels down the anchor line and strikes the samplers trigger mechanism. Use

care when handling the sampler while collecting a sample. Transport the sampling device with the jaws in the closed position. Keep the safety pin that locks the jaws open in place at all times other than when collecting a sample.

Slowly retrieve the dredge from the water and empty the grab contents into a sieve bucket. While retrieving the dredge, take care not to create unnecessary turbulence, water currents or waves that might unintentionally flush macroinvertebrates out of the dredge.

5.6 Process the Grab Sample

Macroinvertebrate samples collected with grab devices are field processed prior to adding preservatives and returning to the office. Samples are processed by placing the bottom materials in a square tub and homogenizing this material with an adequate quantity of water. This mixture of organic matter, sediment and aquatic organisms are then to be poured through a Wildco three gallon wash bucket (No. 30 mesh) to facilitate removal of the finer materials. Aquatic invertebrates and debris retained in the wash bucket are then transferred to a container with appropriate preservative. For samples collected with the Ekman or Petite Ponar, conversion of number of organisms per sample to numbers per square meter is made by using Table 1.

Table 1. Conversion of number of organisms per sample to numbers per square meter for samples collected with the Ekman or Petite Ponar dredges.

#/Sampler	#/m2	#/Sampler	#/m2	#Sampler	#/m2	#/Sampler	#/m2
1	43	26	1118	51	2193	76	3268
2	86	27	1161	52	2236	77	3311
3	129	28	1204	53	2279	78	3354
4	172	29	1247	54	2322	79	3397
5	21	30	1290	55	2365	80	3440
6	258	31	1333	56	2408	81	3483
7	301	32	1376	57	2451	82	3526
8	344	33	1419	58	2494	83	3569
9	387	34	1462	59	2537	84	3612
10	430	35	1505	60	2580	85	3655
11	473	36	1548	61	2623	86	3698
12	516	37	1591	62	2666	87	3741
13	559	38	1634	63	2709	88	3784
14	602	39	1677	64	2752	89	3827
15	645	40	1720	65	2795	90	3870

16	688	41	1763	66	2838	91	3913
17	731	42	1806	67	2881	92	395
18	774	43	1849	68	2924	93	3999
19	817	44	1892	69	2967	94	4042
20	860	45	1935	70	3010	95	4085
21	903	46	1978	71	3053	96	4128
22	946	47	2021	72	3096	97	417
23	989	48	2064	73	3139	98	4214
24	1032	49	2107	74	3182	99	4257
25	1075	50	2150	75	3225	100	4300

6.0 Literature Cited

Illinois EPA 1994. Quality Assurance Plan. Integrated Water Monitoring Program Document. Bureau of Water and Division of Laboratories, Springfield, Illinois.

Weber, C.I. 1973. Biological Field and Laboratory Methods for Measuring the Quality of Surface Waters and Effluents. U.S. EPA Environmental Monitoring Series. EPA-670/4-73-001.

Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 176

Macroinvertebrate Tolerance List and Functional Feeding Group Classification

Surface Water Section

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Appendix A. Illinois Environmental Protection Agency Macroinvertebrate Tolerance List and Functional Feeding Group Classification

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Phylogenetic Order

Major Group	Bios ID	Taxon	Tolerance	Functional Feeding Group
Phylum Platyhelminthes	1	Platyhelminthes	99.9	
Class Turbellaria	2	Turbellaria	6	PR
Order Tricladida	3	Tricladida	6	CG
Family Planariidae	4	Planariidae	6	
	5	Dugesia sp.	6	
	6	Dugesia tigrina	6	PR
	7	Planaria sp.	6	
Phylum Nematomorpha	15	Nematomorpha	99.9	PA
Class Gordioida	18	Gordius sp.	99.9	
Phylum Annelida	30	Annelida	99.9	CG
Class Oligochaeta	31	Oligochaeta	10	CG
Order Branchiobdellida	32	Branchiobdellida	10	PA
Family Branchiobdellidae	33	Branchiobdellidae	10	CG
Order Lumbriculida	34	Lumbriculida	10	
Family Lumbriculidae	35	Lumbriculidae	10	CG
Order Haplotaxida	36	Haplotaxida	10	
Family Aeolosomatidae	37	Aeolosomatidae	10	CF
Family Enchytraeidae	38	Enchytraeidae	10	CG
Family Lumbricidae	39	Lumbricidae	10	CG
Order Tubificida	212	Tubificida	10	
Family Naididae	40	Naididae	10	CG
	41	Allonais sp.	10	CG
	42	Allonais pectinata	10	
	43	Amphichaeta sp.	10	CG
	44	Amphichaeta leydigi	10	
	45	Arcteonais sp.	10	
	46	Arcteonais lomondi	10	CG
	47	Bratislavia sp.	10	CG
	48	Bratislavia unidentata	10	CG
	49	Chaetogaster sp.	10	SH
	50	Chaetogaster diaphanus	10	PR
	51	Chaetogaster diastrophus	10	PR
	52	Chaetogaster limnaei	10	PR
	53	Dero sp.	10	CG
	54	Dero digitata	10	CG
	55	Dero furcata	10	CG
	56	Dero lodeni	10	CG
	57	Dero nivea	10	CG
	109	Dero pectinata	10	CG
	58	Nais sp.	10	CG

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	59	Nais barbata	10	CG
	60	Nais behningi	10	CG
	61	Nais bretscheri	10	CG
	62	Nais communis	10	CG
	63	Nais elinguis	10	CG
	64	Nais pardalis	10	CG
	65	Nais simplex	10	CG
	66	Nais variabilis	10	CG
	110	Paranais sp.	10	
	67	Paranais frici	10	
	68	Ophidonais sp.	10	
	69	Ophidonais serpentina	10	CG
	70	Pristina sp.	10	CG
	71	Pristina aequiseta	10	CG
	72	Pristina leidyi	10	CG
	73	Pristina breviseta	10	CG
	74	Pristina longiseta	10	
	75	Pristina osborni	10	CG
	76	Pristina synclites	10	CG
	77	Slavina sp.	10	CG
	78	Slavina appendiculata	10	CG
	79	Specaria sp.	10	CG
	80	Specaria josinae	10	CG
	81	Stephensoniana sp.	10	
	82	Stephensoniana trivandrana	10	CG
	83	Stylaria sp.	10	
	84	Stylaria fossularis	10	CG
	85	Stylaria lacustris	10	CG
	86	Uncinais sp.	10	
	87	Uncinais uncinata	10	
	88	Vejdovskyella sp.	10	CG
	89	Vejdovskyella intermedia	10	CG
	90	Wapsa sp.	10	
	91	Wapsa mobilis	10	
Family Tubificidae	92	Tubificidae	10	CG
	93	Aulodrilus pigueti	10	CG
	94	Branchiura sp.	10	CG
	95	Branchiura sowerbyi	10	CG
	96	Ilyodrilus sp.	10	
	97	Ilyodrilus templetoni	10	CG
	98	Limnodrilus sp.	10	CG
	99	Limnodrilus cervix	10	CG
	100	Limnodrilus claparedianus	10	CG
	101	Limnodrilus hoffmeisteri	10	CG

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	102	Limnodrilus udekemianus	10	CG
	103	Tubifex sp.	10	CG
	104	Tubifex tubifex	10	CG
	105	Potamothrix vejdovskyi	10	
	108	Quistradrilus multisetosus	10	CG
Class Hirudinea	249	Hirudinea	8	PR
Order Rhynchobdellida	151	Rhynchobdellida	8	
Family Glossiphoniidae	152	Glossiphoniidae	8	PR
	153	Actinobdella sp.	8	
	154	Actinobdella inequiannulata	8	
	155	Alboglossiphonia sp.	8	
	156	Alboglossiphonia heteroclita	8	PR
	157	Batracobdella sp.	8	PR
	158	Batracobdella phalera	8	
	159	Batracobdella picta	8	
Order Pharyngobdellidae	209	Pharyngobdellidae	8	
Family Erpobdellidae	208	Desserobdella phalera	8	
	210	Gloiobdella elongata	8	
Family Glossiphoniidae	160	Glossiphonia sp.	8	PR
	161	Glossiphonia complanata	8	PR
	162	Helobdella sp.	8	PA
	163	Helobdella elongata	8	PR
	164	Helobdella fusca	8	PA
	165	Helobdella papillata	8	PR
	166	Helobdella stagnalis	8	PR
	167	Helobdella triserialis	8	PA
	168	Placobdella montifera	8	PR
	169	Placobdella sp.	8	PR
	170	Placobdella multilineata	8	PR
	171	Placobdella ornata	8	PR
	172	Placobdella papillifera	8	PA
	173	Placobdella parasitica	8	PA
	174	Placobdella pediculata	8	
	175	Theromyzon sp.	8	PR
	176	Theromyzon biannulatum	8	
Family Piscicolidae	177	Piscicolidae	7	
	178	Cystobranchus verrilli	7	
	179	Cystobranchus sp.	7	
	180	Myzobdella sp.	7	
	181	Myzobdella lugubris	7	PR
	182	Piscicola sp.	7	PR
	183	Piscicola milneri	7	PR
	184	Piscicola punctata	7	PR
	185	Piscicolaria sp.	7	

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	186	Piscicolaria reducta	7	
Order Gnathobdellida	211	Gnathobdellida	7	
Family Hirudinidae	188	Hirudinidae	8	PR
	189	Haemopis sp.	7	PR
	190	Haemopis marmorata	7	
	191	Haemopis terrestris	7	
	192	Macrobdella sp.	7	
	193	Macrobdella decora	7	
	194	Philobdella sp.	7	
	195	Philobdella gracilis	7	PR
Family Erpobdellidae	197	Erpobdellidae	8	PR
	198	Dina sp.	8	PR
	199	Dina dubia	8	
	200	Dina parva	8	
	201	Erpobdella sp.	8	
	202	Erpobdella punctata	8	PR
	203	Mooreobdella sp.	8	PR
	204	Mooreobdella fervida	8	
	205	Mooreobdella microstoma	8	PR
	206	Nephelopsis sp.	8	
	207	Nephelopsis obscura	8	PR
Phylum Arthropoda	250	Arthropoda	99.9	
Class Crustacea	251	Crustacea	99.9	CG
Order Isopoda	252	Isopoda	99.9	CG
Family Asellidae	253	Asellidae	6	CG
	254	Caecidotea sp.	6	CG
	256	Caecidotea brevicaudus	6	
	347	Caecidotea communis	6	CG
	257	Caecidotea forbesi	6	
	258	Caecidotea intermedia	6	
	259	Caecidotea kendeighi	6	
	263	Caecidotea tridentata	6	
	268	Caecidotea packardi	6	
	269	Caecidotea spatulata	6	
	270	Caecidotea stygia	6	
	255	Asellus sp.	6	
	272	Lirceus sp.	4	CG
	273	Lirceus fontinalis	4	CG
	274	Lirceus garmani	4	CG
	275	Lirceus lineatus	4	CG
	276	Lirceus louisianae	4	
Order Amphipoda	325	Amphipoda	4	CG
Family Talitridae	326	Hyalellidae	4	
	327	Hyalella sp.	4	CG

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	328	Hyalella azteca	5	CG
Family Gammaridae	329	Gammaridae	4	CG
	341	Gammarus sp.	3	
	344	Gammarus pseudolimnaeus	3	CG
	345	Gammarus troglophilus	3	
	346	Gammarus fasciatus	3	CG
	330	Stygobromus sp.	4	PR
	331	Stygobromus subtilis	4	
	332	Bactrurus sp.	1	
Family Crongonyctidae	348	Crongonyctidae	4	
	335	Crangonyx sp.	4	CG
	336	Crangonyx forbesi	4	CG
	337	Crangonyx gracilis	4	CG
	338	Crangonyx minor	4	
	339	Crangonyx packardi	4	
	340	Crangonyx pseudogracilis	4	
Order Decapoda	400	Decapoda	99.9	SH
Family Cambaridae	401	Cambaridae	5	CG
	402	Cambarellus sp.	5	SH
	403	Cambarellus puer	5	CG
	404	Cambarellus shufeldtii	5	CG
	405	Cambarus sp.	5	CG
	406	Cambarus diogenes	5	
	407	Cambarus rusticiformis	5	
	408	Cambarus tenebrosus	5	
	409	Fallicambarus sp.	5	
	410	Fallicambarus fodiens	5	
	411	Orconectes sp.	5	
	412	Orconectes illinoiensis	5	
	413	Orconectes immunis	5	
	414	Orconectes indianensis	5	
	415	Orconectes kentuckiensis	5	
	416	Orconectes lancifer	5	
	417	Orconectes placidus	5	
	418	Orconectes propinquus	5	
	419	Orconectes rusticus	5	
	420	Orconectes stannardi	5	
	421	Orconectes virilis	5	
	430	Orconectes bisectus	5	
	422	Procambarus sp.	5	SH
	423	Procambarus acutus	5	SH
	424	Procambarus clarki	5	
	425	Procambarus gracilis	5	
	426	Procambarus viaeviridis	5	

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Family Palaemonidae	427	Palaemonidae	4	
	428	Palaemonetes sp.	4	
	429	Palaemonetes kadiakensis	4	
Class Insecta	475	Insecta	99.9	
Order Ephemeroptera	476	Ephemeroptera	3	CG
Family Acanthametropodidae	2605	Acanthametropodidae		
	478	Acanthametropus sp.	3	PR
	479	Acanthametropus pecatonica	3	
Family Ameletidae	2604	Ameletidae		
	480	Ameletus sp.	0	CG
	481	Ameletus lineatus	0	
Family Siphlonuridae	477	Siphlonuridae	3	CG
	482	Siphlonurus sp.	2	CG
	483	Siphlonurus alternatus	2	
	484	Siphlonurus quebecensis	2	
	485	Siphlonurus rapidus	2	
Family Oligoneuriidae	486	Oligoneuriidae	3	CF
Family Isonychiidae	487	Isonychia sp.	3	CF
	488	Isonychia arida	3	
	489	Isonychia bicolor	3	CG
	490	Isonychia rufa	3	
	491	Isonychia sayi	3	
	492	Isonychia sicca	3	
Family Metretopodidae	493	Metretopodidae	3	
	494	Siphloplecton sp.	2	CG
	495	Siphloplecton basale	2	
	496	Siphloplecton interlineatum	2	
Family Baetidae	497	Baetidae	4	CG
	652	Acentrella sp.	4	
	2203	Acerpenna sp.	4	SH
	506	Acerpenna macdunnoughi	4	CG
	498	Baetis sp.	4	CG
	645	Baetis amplus	4	CG
	653	Baetis armillatus	4	
	499	Baetis brunneicolor	4	CG
	500	Baetis ephippiatus	4	
	501	Baetis flavistriga	4	CG
	502	Baetis frondalis	4	
	503	Baetis hageni	4	CG
	504	Baetis intercalaris	7	
	646	Baetis levitans	4	
	505	Baetis longipalpus	6	
	507	Baetis propinquus gr.	4	
	508	Acerpenna pygmaeus	4	

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	509	Baetis quilleri	4	
	510	Baetis tricaudatus	1	CG
	650	Barbaetis cestus	4	
	647	Baetis vagans	4	
	511	Callibaetis sp.	4	CG
	512	Callibaetis ferrugineus	4	
	513	Callibaetis fluctuans	4	
	514	Callibaetis skokianus	4	
	515	Centroptilum sp.	2	CG
	661	Diphetor hageni	4	
	643	Procloeon sp.	4	
	516	Cloeon sp.	3	
	517	Cloeon alamance	3	
	518	Cloeon rubropictum	3	
	519	Heterocloeon sp.	4	SC
	520	Heterocloeon curiosum	4	SC
	651	Plauditus sp.	3	
	660	Plauditus armillatus	4	
	657	Plauditus punctiventris	3	
	521	Pseudocloeon sp.	4	SC
	522	Pseudocloeon carolina	4	
	523	Pseudocloeon dubium	4	SC
	524	Pseudocloeon myrsum	4	
	525	Pseudocloeon parvulum	4	
	656	Pseudocloeon propinquus gr.	4	
	526	Pseudocloeon punctiventris	4	
	527	Paracloeodes sp.	4	SC
	2233	Paracloeodes minutus	5	
Family Arthropleidae	2603	Arthropleidae		
	531	Arthroplea sp.	3	CF
	532	Arthroplea bipunctata	3	
Family Heptageniidae	528	Heptageniidae	3.5	SC
	529	Anepeorus sp.	3.5	PR
	530	Anepeorus simplex	3.5	
	533	Epeorus sp.	1	SC
	534	Epeorus vitreus	0	
	535	Heptagenia diabasia	4	
	644	Nixe sp.	4	SC
	655	Nixe perfida	4	
	536	Heptagenia sp.	3	SC
	537	Heptagenia flavescens	2	
	538	Heptagenia hebe	3	
	539	Heptagenia lucidipennis	3	
	540	Heptagenia maculipennis	3	SC

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	541	Heptagenia marginalis	1	SC
	542	Heptagenia perfida	1	
	543	Heptagenia pulla	0	SC
	544	Rhithrogena sp.	0	SC
	545	Rhithrogena pellucida	0	SC
	550	Stenacron sp.	4	SC
	548	Stenacron interpunctatum	4	
	648	Leucrocuta sp.	3	SC
	654	Leucrocuta hebe	3	
	649	Leucrocuta maculipennis	3	
	551	Maccaffertium sp.	4	SC
	565	Maccaffertium ares	3	
	552	Maccaffertium exiguum	5	
	553	Maccaffertium quinquespinum	5	
	556	Maccaffertium integrum	4	
	557	Maccaffertium luteum	1	SC
	559	Maccaffertium mediopunctatum	2	SC
	558	Maccaffertium nepotellum	5	
	560	Maccaffertium modestum	3	SC
	561	Maccaffertium annexum	4	
	563	Maccaffertium pulchellum	3	SC
	562	Maccaffertium rubromaculatum	2	
	564	Maccaffertium terminatum	4	SC
	566	Maccaffertium vicarium	3	SC
	659	Stenonema sp.	4	SC
	554	Stenonema femoratum	7	SC
Family Ephemerellidae	567	Ephemerellidae	3.5	CG
	568	Attenella sp.	2	CG
	569	Attenella attenuata	2	CG
	570	Dannella sp.	2	
	571	Dannella lita	2	CG
	572	Dannella simplex	2	CG
	573	Drunella sp.	1	PR
	574	Ephemerella cornuta	1	
	575	Drunella cornutella	1	SC
	576	Ephemerella lata	1	
	577	Ephemerella walkeri	1	
	578	Ephemerella sp.	2	CG
	579	Ephemerella aurivillii	2	CG
	580	Ephemerella catawba	2	CG
	581	Ephemerella dorothea	2	CG
	582	Ephemerella excrucians	2	CG
	583	Ephemerella invaria	2	CG
	584	Ephemerella needhami	2	CG

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	585	Ephemerella rotunda	2	
	586	Ephemerella subvaria	2	CG
	587	Eurylophella sp.	4	SC
	588	Eurylophella aestiva	4	
	589	Eurylophella bicolor	4	CG
	590	Ephemerella coxalis	4	
	591	Eurylophella funeralis	4	
	592	Eurylophella lutulenta	4	CG
	593	Eurylophella temporalis	4	CG
	594	Serratella sp.	1	CG
	595	Serratella deficiens	1	CG
	596	Ephemerella frisoni	1	
	597	Serratella sordida	1	CG
Family Leptohyphidae	598	Leptohyphidae	5.5	CG
	662	Leptohyphe sp.	5.5	CG
	599	Tricorythodes sp.	5	CG
Family Caenidae	600	Caenidae	5.5	CG
	601	Brachycercus sp.	3	CG
	602	Caenis sp.	6	CG
Family Baetiscidae	603	Baetiscidae	3	CG
	604	Baetisca bajkovi	3	
	605	Baetisca sp.	3	CG
	606	Baetisca lacustris	3	
	607	Baetisca laurentina	3	
	608	Baetisca obesa	3	
Family Leptophlebiidae	609	Leptophlebiidae	3	CG
	610	Choroterpes sp.	2	CG
	611	Choroterpes basalis	2	CG
	612	Habrophlebiodes sp.	2	SC
	613	Habrophlebiodes americana	2	
	614	Leptophlebia sp.	3	CG
	615	Paraleptophlebia sp.	2	CG
	616	Paraleptophlebia moerens	2	
	617	Paraleptophlebia ontario	2	
	618	Paraleptophlebia praepedita	2	
	619	Paraleptophlebia sticta	2	
Family Potamanthidae	620	Potamanthidae	5	CF
	621	Anthopotamus sp.	4	
	623	Anthopotamus myops	4	
Family Ephemeridae	625	Ephemeridae	5	CG
	626	Ephemera sp.	3	CG
	627	Ephemera simulans	3	CG
	628	Hexagenia sp.	6	CG
	629	Hexagenia atrocaudata	6	CG

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	630	Hexagenia bilineata	6	CG
	631	Hexagenia limbata	5	CG
	632	Hexagenia munda	5	
	633	Hexagenia rigida	6	CG
Family Palingeniidae	635	Pentagenia sp.	4	CF
	636	Pentagenia vittigera	4	CG
Family Polymitarcyidae	637	Polymitarcyidae	3	CG
	638	Ephoron sp.	2	CG
	639	Ephoron album	2	CG
	640	Ephoron leukon	2	CG
	641	Tortopus sp.	4	CG
Order Odonata	700	Odonata	99.9	PR
Family Cordulegastridae	702	Cordulegastridae	4.5	PR
	703	Cordulegaster sp.	2	PR
	704	Cordulegaster maculata	2	PR
	705	Cordulegaster obliqua	2	PR
Family Gomphidae	706	Gomphidae	4.5	PR
	2602	Erpetogomphus sp.	2	
	2229	Erpetogomphus designatus	2	
	713	Dromogomphus sp.	4	PR
	714	Dromogomphus spinosus	4	PR
	707	Arigomphus sp.	7	PR
	722	Gomphus sp.	7	PR
	738	Gomphus amnicola	7	
	716	Gomphus crassus	7	
	723	Gomphus exilis	7	
	717	Gomphus externus	7	PR
	724	Gomphus graslinellus	7	
	710	Gomphus lentulus	7	
	719	Gomphus lineatifrons	7	
	725	Gomphus lividus	7	PR
	739	Gomphus notatus	7	
	740	Gomphus plagiatus	7	
	726	Gomphus quadricolor	7	
	711	Gomphus submedianus	7	
	741	Gomphus spiniceps	7	
	720	Gomphus vastus	7	PR
	712	Gomphus villosipes	7	
	879	Gomphurus sp.	7	
	737	Stylurus sp.	7	PR
	728	Hagenius sp.	3	PR
	729	Hagenius brevistylus	3	PR
	735	Lanthus sp.	6	PR
	730	Ophiogomphus sp.	2	PR

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	731	Ophiogomphus rupinsulensis	2	
	732	Progomphus sp.	5	PR
	733	Progomphus obscurus	5	PR
	734	Stylogomphus sp.	4.5	PR
	736	Stylogomphus albistylus	4.5	PR
Family Aeshnidae	742	Aeshnidae	4.5	PR
	743	Aeshna sp.	4	PR
	744	Aeshna canadensis	4	
	746	Aeshna constricta	4	PR
	747	Aeshna umbrosa	4	
	748	Aeshna verticalis	4	
	749	Anax sp.	5	PR
	750	Anax junius	5	PR
	751	Basiaeschna sp.	2	PR
	752	Basiaeschna janata	2	PR
	753	Boyeria sp.	3	PR
	754	Boyeria vinosa	3	PR
	755	Epiaeschna sp.	1	PR
	756	Epiaeschna heros	1	PR
	758	Nasiaeschna sp.	2	PR
	757	Nasiaeschna pentacantha	2	PR
Family Macromiidae	759	Macromiidae	4.5	PR
	760	Didymops sp.	4	PR
	761	Didymops transversa	4	PR
	762	Macromia sp.	3	PR
	764	Macromia georgina	3	PR
	765	Macromia illinoiensis	3	PR
	766	Macromia pacifica	3	
	767	Macromia taeniolata	3	PR
Family Corduliidae	768	Corduliidae	4.5	PR
	769	Cordulia sp.	2	PR
	770	Cordulia shurtleffi	2	
	771	Epitheca sp.	4	PR
	772	Epicordulia sp.	4.5	PR
	773	Epicordulia princeps	4.5	PR
	774	Helocordulia sp.	2	PR
	775	Neurocordulia sp.	3	PR
	776	Neurocordulia molesta	3	PR
	777	Neurocordulia obsoleta	3	PR
	778	Neurocordulia yamaskanensis	3	
	779	Somatochlora sp.	1	PR
	781	Somatochlora filosa	1	
	782	Somatochlora linearis	1	PR
	783	Somatochlora tenebrosa	1	

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	784	Tetragoneuria sp.	4.5	PR
	785	Tetragoneuria cynosura	4.5	PR
Family Libellulidae	787	Libellulidae	4.5	PR
	788	Celithemis sp.	2	PR
	789	Celithemis elisa	2	
	790	Celithemis eponina	2	
	791	Celithemis monomelaena	2	
	792	Erythemis sp.	5	PR
	793	Erythemis simplicicollis	5	PR
	794	Erythrodiplax sp.	5	PR
	795	Ladona sp.	4.5	PR
	796	Ladona julia	4.5	
	797	Leucorrhinia sp.	4.5	PR
	799	Leucorrhinia intacta	4.5	
	800	Libellula sp.	8	PR
	801	Libellula cyanea	8	
	802	Libellula incesta	8	PR
	803	Libellula luctuosa	8	
	804	Libellula pulchella	8	
	805	Libellula quadrimaculata	8	
	806	Libellula semifasciata	8	PR
	807	Libellula vibrans	8	PR
	809	Pachydiplax sp.	8	PR
	810	Pachydiplax longipennis	8	PR
	811	Pantala sp.	7	PR
	812	Pantala flavescens	7	
	813	Pantala hymenaea	7	
	814	Perithemis sp.	4	PR
	815	Perithemis tenera	4	PR
	816	Plathemis sp.	3	PR
	817	Plathemis lydia	3	PR
	818	Sympetrum sp.	4	PR
	819	Sympetrum ambiguum	4	PR
	820	Sympetrum corruptum	4	
	821	Sympetrum obstrusum	4	
	822	Sympetrum rubicundulum	4	
	823	Sympetrum semicinctum	4	
	824	Sympetrum vicinum	4	
	825	Tramea sp.	4	PR
	826	Tramea carolina	4	PR
	827	Tramea lacerata	4	
	828	Tramea onusta	4	
	829	Zygoptera	99.9	PR
Family Calopterygidae	830	Calopterygidae	3.5	PR

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	831	Calopteryx sp.	4	PR
	832	Calopteryx aequabilis	4	
	833	Calopteryx maculata	4	PR
	834	Hetaerina sp.	3	PR
	835	Hetaerina americana	3	PR
	836	Hetaerina titia	3	PR
Family Lestidae	837	Lestidae	99.9	PR
	838	Archilestes sp.	1	PR
	839	Archilestes grandis	1	
	840	Lestes sp.	6	PR
	841	Lestes disjunctus	6	
	842	Lestes eurinus	6	
	843	Lestes forcipatus	6	
	844	Lestes inaequalis	6	
	845	Lestes rectangularis	6	
	846	Lestes vigilax	6	
Family Coenagrionidae	847	Coenagrionidae	5.5	PR
	848	Amphiagrion sp.	5	PR
	849	Amphiagrion saucium	5	
	850	Anomalagrion sp.	5.5	PR
	851	Anomalagrion hastatum	5.5	PR
	852	Argia sp.	5	PR
	853	Argia apicalis	5	PR
	854	Argia bipunctulata	5	
	997	Argia fumipennis	5	PR
	855	Argia moesta	5	PR
	856	Argia sedula	5	PR
	857	Argia tibialis	5	PR
	858	Argia translata	5	
	859	Argia violacea	5	PR
	860	Chromagrion sp.	5.5	PR
	861	Chromagrion conditum	5.5	
	862	Coenagrion sp.	5.5	PR
	863	Enallagma sp.	6	PR
	864	Enallagma aspersum	6	
	865	Enallagma civile	6	
	866	Enallagma divagans	6	PR
	867	Enallagma exsulans	6	
	868	Enallagma geminatum	6	
	869	Enallagma hageni	6	
	870	Enallagma signatum	6	PR
	871	Enallagma traviatum	6	
	872	Enallagma vesperum	6	PR
	873	Ischnura sp.	6	PR

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	874	Ischnura posita	6	PR
	875	Ischnura verticalis	6	
	876	Nehalennia sp.	7	PR
	877	Nehalennia gracilis	7	
	878	Nehalennia irene	7	
Order Plecoptera	925	Plecoptera	1.5	PR
Family Pteronarcyidae	927	Pteronarcys sp.	2	SH
Family Taeniopterygidae	928	Taeniopterygidae	1.5	SH
	929	Oemopteryx sp.	1.5	SH
	930	Oemopteryx glacialis	1.5	
	931	Strophopteryx sp.	1.5	
	932	Strophopteryx fasciata	1.5	SH
	933	Taeniopteryx sp.	2	SH
	934	Taeniopteryx nivalis	2	SH
	935	Taeniopteryx parvula	2	SH
Family Nemouridae	936	Nemouridae	1.5	SH
	937	Amphinemura sp.	1.5	SH
	938	Nemoura sp.	1	SH
	939	Nemoura venosa	1	
	940	Prostoia sp.	1.5	SH
	941	Soyedina sp.	1.5	SH
Family Leuctridae	942	Leuctridae	1.5	SH
	943	Leuctra sp.	1	SH
Family Capniidae	944	Capniidae	1.5	SH
	945	Allocapnia sp.	2	SH
	946	Allocapnia mystica	1.5	
	947	Allocapnia recta	1.5	
	948	Allocapnia vivipara	1.5	SH
	949	Capnia sp.	1	SH
	950	Capnia vernalis	1	
	951	Paracapnia sp.	1.5	SH
	952	Paracapnia angulata	1.5	
	953	Paracapnia opis	1.5	
Family Perlidae	954	Perlidae	1.5	PR
	955	Acroneuria sp.	1	PR
	956	Acroneuria abnormis	1	PR
	957	Acroneuria arida	1	PR
	958	Acroneuria carolinensis	1	PR
	959	Acroneuria evoluta	1	PR
	960	Acroneuria internata	1	PR
	961	Acroneuria lycorias	1	PR
	962	Attaneuria sp.	1.5	
	963	Attaneuria ruralis	1.5	PR
	964	Neoperla sp.	1	PR

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	965	Neoperla clymene	1	PR
	966	Paragnetina sp.	1.5	PR
	967	Paragnetina media	1.5	PR
	968	Perlesta sp.	4	PR
	971	Atoperla sp.	1	
	969	Perlesta placida	4	
	970	Perlinella sp.	2	PR
	972	Perlinella drymo	2	PR
	973	Perlinella ephyre	2	PR
	974	Phasganophora sp.	1.5	PR
	975	Phasganophora capitata	1.5	
Family Perlodidae	976	Perlodidae	1.5	PR
	977	Hydroperla sp.	1	PR
	978	Hydroperla crosbyi	1	
	979	Isogenoides sp.	1.5	PR
	980	Isoperla sp.	2	PR
	981	Isoperla bilineata	2	
	982	Isoperla clio	2	
	996	Isoperla confusa	2	
	983	Isoperla cotta	2	
	984	Isoperla dicala	2	
	985	Isoperla lata	2	
	986	Isoperla marlynia	2	
	987	Isoperla nana	2	
	988	Isoperla richardsoni	2	
Family Chloroperlidae	989	Chloroperlidae	1.5	PR
	990	Chloroperla sp.	3	
	991	Alloperla sp.	1.5	PR
	992	Hastaperla sp.	1.5	SC
	993	Hastaperla brevis	1.5	
	994	Rasvena sp.	1.5	
	995	Rasvena terna	1.5	CG
Order Hemiptera	1050	Hemiptera	99.9	PR
Family Hebridae	1051	Hebridae	99.9	PR
	1052	Hebrus sp.	99.9	PR
	1053	Merragata sp.	99.9	PR
Family Mesoveliidae	1054	Mesoveliidae	99.9	PR
	1055	Mesovelia sp.	99.9	PR
	1056	Mesovelia mulsanti	99.9	PR
Family Gerridae	1057	Gerridae	99.9	PR
	1058	Gerris sp.	99.9	PR
	1059	Limnogonus sp.	99.9	PR
	1060	Limnogonus hesione	99.9	
	1061	Metrobates sp.	99.9	PR

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	1062	Rheumatobates sp.	99.9	PR
	1063	Trepobates sp.	99.9	PR
Family Veliidae	1064	Veliidae	99.9	PR
	1065	Microvelia sp.	99.9	PR
	1066	Rhagovelia sp.	99.9	PR
Family Notonectidae	1067	Notonectidae	99.9	PR
	1068	Buenoa sp.	99.9	PR
	1069	Notonecta sp.	99.9	PR
Family Pleidae	1070	Pleidae	99.9	PR
	1071	Neoplea sp.	99.9	PR
	1072	Neoplea striola	99.9	
Family Naucoridae	1073	Naucoridae	99.9	PR
	1074	Pelocoris sp.	99.9	PR
	1075	Pelocoris femoratus	99.9	PR
Family Nepidae	1076	Nepidae	99.9	PR
	1077	Nepa sp.	99.9	PR
	1078	Nepa apiculata	99.9	
	1079	Ranatra fusca	99.9	PR
	1080	Ranatra sp.	99.9	PR
	1081	Ranatra kirkaldyi	99.9	PR
	1082	Ranatra nigra	99.9	PR
Family Belostomatidae	1083	Belostomatidae	99.9	PR
	1084	Belostoma sp.	99.9	PR
	1085	Belostoma flumineum	99.9	PR
	1086	Lethocerus sp.	99.9	PR
	1087	Lethocerus americans	99.9	
	1088	Lethocerus griseus	99.9	
	1089	Lethocerus uhleri	99.9	
Family Corixidae	1090	Corixidae	99.9	PR
	1091	Hesperocorixa sp.	99.9	PR
	1092	Hesperocorixa interrupta	99.9	
	1094	Hesperocorixa laevigata	99.9	
	1096	Hesperocorixa lucida	99.9	
	1097	Hesperocorixa nitida	99.9	
	1098	Hesperocorixa obliqua	99.9	
	1100	Hesperocorixa vulgaris	99.9	
	1101	Palmacorixa sp.	99.9	PR
	1102	Palmacorixa buenoi	99.9	
	1103	Palmacorixa gilletteii	99.9	
	1104	Palmacorixa nana	99.9	
	1105	Ramphocorixa sp.	99.9	PR
	1106	Ramphocorixa acuminata	99.9	
	1107	Sigara sp.	99.9	PR
	1108	Sigara alternata	99.9	

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	1109	Sigara compressoidea	99.9	
	1111	Sigara hubbelli	99.9	
	1113	Sigara modesta	99.9	
	1115	Sigara signata	99.9	
	1117	Trichocorixa sp.	99.9	PR
	1118	Trichocorixa calva	99.9	
	1119	Trichocorixa kanza	99.9	
	1120	Trichocorixa macroceps	99.9	
Order Megaloptera	1175	Megaloptera	3.5	
Family Sialidae	1176	Sialidae	3.5	PR
	1177	Sialis sp.	4	PR
	1180	Sialis infumata	4	
	1181	Sialis itasca	4	
	1183	Sialis mohri	4	PR
	1184	Sialis vagans	4	
	1185	Sialis velata	4	
Family Corydalidae	1186	Corydalidae	3	PR
	1187	Chauliodes sp.	4	PR
	1188	Chauliodes pectinicornis	4	PR
	1189	Chauliodes rastricornis	4	PR
	1190	Corydalus sp.	3	PR
	1191	Corydalus cornutus	3	PR
	1192	Nigronia sp.	2	PR
	1193	Nigronia fasciatus	2	PR
	1194	Nigronia serricornis	2	PR
Order Neuroptera	1250	Neuroptera	99.9	PR
Family Sisyridae	1251	Sisyridae	1	PR
	1252	Climacea sp.	1	
	1253	Climacea areolaris	1	
	1254	Sisyra sp.	1	PR
	1255	Sisyra vicaria	1	
Order Trichoptera	1300	Trichoptera	3.5	
Family Hydropsychidae	1301	Hydropsychidae	5.5	CF
	1302	Cheumatopsyche sp.	6	CF
	1304	Diplectrona sp.	2	CF
	1303	Diplectrona metaqui	2	
	1305	Diplectrona modesta	2	CF
	1306	Hydropsyche sp.	5	CF
	1307	Hydropsyche aerata	5	CF
	1308	Hydropsyche arinale	5	
	1309	Hydropsyche betteni	5	CF
	1310	Hydropsyche bidens	5	
	1333	Ceratopsyche cheilonis	4	
	1332	Ceratopsyche bronta	4	CF

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	1311	Hydropsyche cuanis	5	
	1312	Hydropsyche dicantha	5	CF
	1313	Hydropsyche frisoni	5	CF
	1314	Hydropsyche hageni	5	
	1316	Hydropsyche incommoda	5	
	1493	Ceratopsyche morosa	4	
	1317	Hydropsyche orris	4	CF
	1318	Hydropsyche phalerata	2	CF
	1319	Hydropsyche placoda	4	
	1320	Hydropsyche scalaris	5	CF
	1492	Ceratopsyche alternans	5	CF
	1321	Hydropsyche simulans	5	CF
	1322	Hydropsyche valanis	5	CF
	1323	Hydropsyche venularis	5	CF
	1324	Macronema sp.	2	CF
	2217	Macrostemum sp.	2	CF
	1325	Macronema zebratum	2	CF
	1326	Parapsyche sp.	5.5	PR
	1327	Parapsyche apicalis	5.5	
	1328	Potamyia sp.	4	CF
	1329	Potamyia flava	4	CF
	1330	Ceratopsyche sp.	4	CF
	1496	Ceratopsyche alhedra	4	
	1499	Ceratopsyche slossonae	4	
	1337	Ceratopsyche sparna	4	
Family Philopotamidae	1338	Philopotamidae	3.5	CF
	1339	Chimarra sp.	3	CF
	1340	Chimarra aterrima	3	CF
	1341	Chimarra feria	3	CF
	1342	Chimarra obscura	3	CF
	1343	Chimarra socia	3	CF
	1344	Dolophilodes sp.	0	CG
	1345	Dolophilodes distinctus	0	
	1346	Wormaldia sp.	3.5	CF
	1348	Wormaldia shawnee	3.5	
Family Polycentropodidae	1349	Polycentropodidae	3.5	CF
	1350	Cyrnellus sp.	5	CF
	1351	Cyrnellus fraternus	5	CF
	1352	Neureclipsis sp.	3	CF
	1353	Neureclipsis crepuscularis	3	CF
	1354	Neureclipsis bimaculata	3	CF
	1355	Nyctiophylax sp.	1	CF
	1356	Phylocentropus sp.	3.5	CF
	1357	Phylocentropus placidus	3.5	

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	1358	Polycentropus sp.	3	PR
	1359	Polycentropus centralis	3	PR
	1360	Polycentropus cinereus	3	PR
	1361	Polycentropus flavus	3	PR
	1362	Polycentropus glacialis	3	PR
	1363	Polycentropus interruptus	3	PR
	1364	Polycentropus remotus	3	PR
Family Psychomyiidae	1365	Psychomyiidae	3.5	CG
	1366	Lype sp.	3.5	SC
	1367	Lype diversa	3.5	SC
	1368	Psychomyia sp.	2	SC
	1369	Psychomyia flavida	2	CG
Family Glossosomatidae	1370	Glossosomatidae	3.5	SC
	1372	Agapetus sp.	2	SC
	1371	Agapetus illini	2	
	1373	Glossosoma sp.	3.5	SC
	1374	Glossosoma intermedium	3.5	SC
	1375	Protoptila sp.	1	SC
Family Hydroptilidae	1376	Hydroptilidae	3.5	PH
	1377	Agraylea sp.	2	PH
	1378	Agraylea multipunctata	2	
	1379	Hydroptila sp.	2	SC
	1500	Hydroptila waubesiana	2	
	1380	Ithytrichia sp.	1	SC
	1381	Leucotrichia sp.	3	SC
	1382	Leucotrichia pictipes	3	
	1384	Mayatrichia sp.	1	SC
	1383	Mayatrichia ayama	1	SC
	1385	Neotrichia sp.	4	SC
	1386	Ochrotrichia sp.	4	CG
	1387	Orthotrichia sp.	1	SC
	1388	Oxyethira sp.	2	MH
	1389	Stactobiella sp.	3.5	SH
	1390	Stactobiella palmata	3.5	
Family Rhyacophilidae	1391	Rhyacophilidae	3.5	PR
	1392	Rhyacophila sp.	1	PR
	1393	Rhyacophila fenestra	1	
	1394	Rhyacophila fuscula	1	PR
	1395	Rhyacophila lobifera	1	
	1396	Rhyacophila vibox	1	
Family Brachycentridae	1397	Brachycentridae	3.5	CF
	1398	Brachycentrus sp.	1	CF
	1399	Brachycentrus americanus	1	CF
	1400	Brachycentrus lateralis	1	CF

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	1401	Brachycentrus numerosus	1	CF
	1402	Brachycentrus occidentalis	1	CF
	1403	Micrasema sp.	3.5	MH
	1404	Micrasema rusticum	3.5	
Family Lepidostomatidae	1405	Lepidostomatidae	3.5	SH
	1406	Lepidostoma sp.	3	SH
	1407	Lepidostoma liba	3	
Family Limnephilidae	1408	Limnephilidae	3.5	SH
	1409	Anabolia sp.	3.5	SH
	1410	Frenesia sp.	3.5	SH
	1411	Frenesia missa	3.5	
	1412	Goera sp.	3.5	SC
	1413	Hesperophylax sp.	3.5	SH
	1414	Hesperophylax designatus	3.5	SH
	1415	Hydatophylax sp.	2	SH
	1416	Hydatophylax argus	2	SH
	1417	Ironoquia sp.	3.5	SH
	1418	Leptophylax sp.	3.5	SH
	1420	Limnephilus sp.	3	SH
	1421	Neophylax sp.	3	SC
	1422	Neophylax concinnus	3	
	1423	Platycentropus sp.	3	SH
	1424	Platycentropus radiatus	3	
	1425	Pseudostenophylax sp.	3.5	SH
	1426	Pseudostenophylax uniformis	3.5	
	1427	Pycnopsyche sp.	3	SH
	1428	Pycnopsyche guttifer	3	SH
	1429	Pycnopsyche lepida	3	
	1430	Pycnopsyche luculenta	3	
	1431	Pycnopsyche scabripennis	3	SH
	1432	Pycnopsyche subfasciata	3	SH
Family Molannidae	1433	Molannidae	3.5	CG
	1434	Molanna sp.	3.5	SC
	1435	Molanna blenda	3.5	
	1436	Molanna tryphena	3.5	
	1437	Molanna uniophila	3.5	
Family Phryganeidae	1438	Phryganeidae	3.5	SH
	1439	Agrypnia sp.	3	SH
	1441	Agrypnia vestita	3	
	1442	Banksiola sp.	2	SH
	1443	Banksiola crotchi	2	
	1444	Fabria sp.	3.5	SH
	1445	Fabria inornata	3.5	
	1446	Oligostomis sp.	3.5	PR

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	1447	Oligostomis ocelligera	3.5	PR
	1448	Phryganea sp.	3	
	1449	Ptilostomis sp.	3	SH
Family Helicopsychidae	1450	Helicopsychidae	3.5	SC
	1451	Helicopsyche sp.	2	SC
	1452	Helicopsyche borealis	2	SC
Family Leptoceridae	1453	Leptoceridae	3.5	CG
	1454	Ceraclea sp.	3	CG
	1456	Ceraclea ancylus	3	
	1457	Ceraclea cancellata	3	
	1458	Ceraclea diluta	3	
	1459	Ceraclea flava	3	
	1460	Ceraclea maculata	3	
	1462	Ceraclea nepha	3	
	2510	Ceraclea transversa	3	
	1463	Ceraclea resurgens	3	
	1466	Leptocerus sp.	3	SH
	1467	Leptocerus americanus	3	
	1468	Mystacides sp.	2	CG
	1470	Mystacides sepulchralis	2	
	1471	Nectopsyche sp.	3	SH
	1472	Nectopsyche albida	3	
	1473	Nectopsyche candida	3	
	1474	Nectopsyche diarina	3	
	1475	Nectopsyche exquisita	3	
	1476	Nectopsyche pavida	3	
	1477	Oecetis sp.	5	PR
	1478	Oecetis avara	5	PR
	1479	Oecetis cinerascens	5	PR
	1480	Oecetis eddlestoni	5	PR
	1481	Oecetis inconspicua	5	PR
	2521	Oecetis nocturna	5	
	1482	Oecetis ochracea	5	
	1483	Setodes sp.	3.5	
	1484	Triaenodes sp.	3	MH
	1486	Triaenodes injustus	3	
	1487	Triaenodes marginatus	3	sh
	1488	Triaenodes tardus	3	SH
Family Sericostomatidae	1489	Sericostomatidae	3.5	SH
	1490	Agarodes distincta	3.5	
Order Lepidoptera	1550	Lepidoptera	99.9	SH
	1501	Crambidae	99.9	SH
Family Pyralidae	1551	Pyralidae	99.9	SH
	1552	Munroessa sp.	99.9	SH

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	1554	Nymphula sp.	99.9	SH
	1555	Paraponyx sp.	99.9	SH
	1556	Petrophila sp.	5	SC
	1557	Synclita sp.	99.9	SH
Order Coleoptera	1600	Coleoptera	99.9	PR
Family Gyrinidae	1601	Gyrinidae	99.9	PR
	1602	Dineutus sp. (larvae only)	4	PR
	1603	Dineutus assimilis (larvae only)	4	PR
	1604	Dineutus discolor (larvae only)	4	PR
	1607	Gyrinus sp. (larvae only)	4	PR
	1608	Gyrinus aeneolus (larvae only)	4	PR
	1609	Gyrinus analis (larvae only)	4	
Family Psephenidae	1614	Psephenidae	4	SC
	1616	Psephenus sp.	4	SC
	1615	Psephenus herricki	4	SC
Family Scirtidae	1617	Helodidae	7	SC
	1618	Cyphon sp.	7	SC
	1619	Cyphon americanus	7	
	1620	Cyphon collaris	7	
	1621	Cyphon modestus	7	
	1622	Cyphon nebulosus	7	
	1623	Cyphon obscurus	7	
	1624	Cyphon punctatus	7	
	1625	Cyphon perplexus	7	
	1626	Scirtes sp.	7	SH
	1627	Scirtes orbiculatus	7	
	1628	Scirtes tibialis	7	
Family Haliplidae	1629	Haliplidae	99.9	SH
	1630	Haliplus sp.	99.9	MH
	1631	Haliplus fasciatus	99.9	SH
	1632	Haliplus immaculicollis	99.9	
	1633	Haliplus leopardus	99.9	
	1634	Haliplus pantherinus	99.9	
	1635	Haliplus triopsis	99.9	
	1636	Peltodytes sp.	99.9	SH
	1637	Peltodytes duodecimpunctatus	99.9	
	1638	Peltodytes dunavani	99.9	
	1639	Peltodytes lengi	99.9	
	1640	Peltodytes sexmaculatus	99.9	
Family Hydrophilidae	1641	Hydrophilidae	99.9	PR
	1642	Anacaena sp.	99.9	
	1643	Anacaena limbata	99.9	
	1644	Berosus sp.	99.9	PR
	1646	Berosus fraternus	99.9	

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	1647	Berosus infuscatus	99.9	
	1649	Berosus peregrinus	99.9	
	1650	Berosus pugnax	99.9	
	1651	Berosus striatus	99.9	CG
	1652	Crenitis sp.	99.9	PR
	1653	Cymbiodyta sp.	99.9	CG
	1655	Cymbiodyta chamberlaini	99.9	
	1656	Cymbiodyta semistriata	99.9	
	1657	Cymbiodyta vindicata	99.9	
	1658	Enochrus sp.	99.9	CG
	1660	Enochrus cinctus	99.9	
	1662	Enochrus consortus	99.9	
	1663	Enochrus hamiltoni	99.9	
	1664	Enochrus ochraceus	99.9	
	1665	Enochrus perplexus	99.9	
	1666	Enochrus pygmaeus	99.9	
	1667	Enochrus sayi	99.9	
	1803	Helocombus sp.	99.9	
	1668	Helophorus sp.	99.9	SH
	1669	Hydrobius sp.	99.9	PR
	1670	Hydrobius fuscipes	99.9	
	1673	Hydrochara sp.	99.9	CG
	1674	Hydrochus sp.	99.9	SH
	1675	Hydrophilus sp.	99.9	PR
	1676	Laccobius sp.	99.9	PR
	1677	Laccobius agilis	99.9	
	1678	Laccobius minutoides	99.9	
	1679	Paracymus sp.	99.9	PR
	1680	Paracymus subcupreus	99.9	
	1681	Tropisternus sp.	99.9	PR
	1682	Tropisternus blatchleyi	99.9	
	1684	Tropisternus lateralis	99.9	CG
	1685	Tropisternus mixtus	99.9	
	1686	Tropisternus natator	99.9	
Family Dytiscidae	1687	Dytiscidae	99.9	PR
	1688	Acilius sp.	99.9	PR
	1689	Acilius fraternus	99.9	
	1691	Acilius semisulcatus	99.9	
	1692	Agabetes sp.	99.9	PR
	1694	Agabus sp.	99.9	PR
	1696	Agabus ambiguus	99.9	
	1698	Agabus disintegratus	99.9	
	1703	Bidessonotus sp.	99.9	PR
	1704	Bidessonotus inconspicuus	99.9	

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	1705	Celina sp.	99.9	PR
	1706	Celina hubbelli	99.9	
	1707	Copelatus sp.	99.9	PR
	1708	Copelatus chevrolati	99.9	
	1709	Copelatus glyphicus	99.9	
	1710	Coptotomus sp.	99.9	PR
	1713	Cybister sp.	99.9	PR
	1714	Cybister fimbriolatus	99.9	
	1715	Deronectes sp.	99.9	PR
	1717	Dytiscus sp.	99.9	PR
	1719	Dytiscus hybridus	99.9	
	1720	Hydaticus sp.	99.9	PR
	1721	Hydaticus modestus	99.9	
	1722	Hydroporus sp.	99.9	PR
	1725	Hydroporus clypealis	99.9	
	1726	Hydroporus consimilus	99.9	
	1728	Hydroporus niger	99.9	
	1729	Hydroporus rufilabris	99.9	
	1733	Hydroporus vittatipennis	99.9	
	1734	Hydrovatus sp.	99.9	PR
	1735	Hydrovatus pustalatus	99.9	PR
	1736	Hygrotus sp.	99.9	PR
	1737	Ilybius sp.	99.9	PR
	1738	Ilybius biguttulus	99.9	
	1739	Laccophilus sp.	99.9	PR
	1740	Laccophilus fasciatus	99.9	PR
	1741	Laccophilus maculosus	99.9	
	1742	Laccophilus proximus	99.9	PR
	1744	Laccornis sp.	99.9	PR
	1745	Liodessus sp.	99.9	PR
	1748	Matus sp.	99.9	PR
	1749	Matus bicarinatus	99.9	
	1751	Rhantus sp.	99.9	PR
	1752	Rhantus binotatus	99.9	
	1753	Thermonectus sp.	99.9	PR
	1754	Thermonectus basillaris	99.9	PR
	1755	Thermonectus ornaticollis	99.9	
	1756	Uvarus sp.	99.9	PR
Family Psephenidae	1760	Dicranopselaphus	4	SC
	1761	Ectopria sp.	4	SC
	1762	Ectopria nervosa	4	SC
	1763	Ectopria thoracica	4	
Family Dryopidae	1764	Dryopidae	4	SH
	1765	Helichus sp.	4	SH

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	1766	Helichus fastigiatus	4	
	1767	Helichus lithophilus	4	
	1768	Helichus striatus	4	
	1769	Pelonomus sp.	4	CG
	1770	Pelonomus obscurus	4	
Family Elmidae	1771	Elmidae	5	CG
	1772	Ancyronyx sp.	2	
	1773	Ancyronyx variegatus	2	CG
	1774	Dubiraphia sp.	5	CG
	1775	Dubiraphia bivittata	2	
	1776	Dubiraphia minima	5	
	1777	Dubiraphia quadrinotata	7	
	1778	Dubiraphia vittata	7	
	1779	Macronychus sp.	2	
	1780	Macronychus glabratus	2	
	1781	Microcylloepus sp.	2	CG
	1782	Microcylloepus pusillus	2	CG
	1783	Optioservus sp.	4	SC
	1784	Optioservus fastiditus	4	SC
	1785	Optioservus ovalis	4	SC
	1786	Optioservus trivittatus	4	SC
	1787	Promoresia sp.	5	SC
	1788	Stenelmis sp.	7	SC
	1789	Stenelmis bicarinata	7	SC
	1790	Stenelmis crenata	7	
	1791	Stenelmis decorata	7	SC
	1792	Stenelmis lateralis	7	SC
	1793	Stenelmis markeli	7	SC
	1794	Stenelmis mera	7	SC
	1795	Stenelmis musgravei	7	SC
	1796	Stenelmis sexlineata	7	SC
	1797	Stenelmis vittipennis	6	
Family Curculionidae	1799	Curculionidae	99.9	SH
	1800	Listronotus sp.	99.9	CF
Family Scirtidae	1801	Elodes sp.	7	
	1802	Prionocyphon sp.	7	SC
Order Diptera	1850	Diptera	10	
Family Blephariceridae	1851	Blephariceridae	0	SC
	1852	Blepharicera sp.	0	SC
Family Tipulidae	1853	Tipulidae	4	SH
	1854	Tipula sp.	4	SH
	1855	Antocha sp.	5	CG
	1856	Dicranota sp.	4	PR
	1857	Eriocera sp.	7	PR

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	1858	Erioptera sp.	4	CG
	2221	Gonomyia sp.	4	CG
	1859	Helius sp.	5	CG
	1860	Hesperoconopa sp.	2	CG
	1861	Hexatoma sp.	4	PR
	1862	Limnophila sp.	4	PR
	1863	Limonia sp.	3	SH
	1864	Ormosia sp.	4	CG
	1865	Pedicia sp.	4	PR
	1866	Pilaria sp.	4	PR
	1867	Prionocera sp.	4	SH
	1868	Pseudolimnophila sp.	2	PR
Family Chaoboridae	1869	Chaoboridae	8	PR
	1870	Chaoborus sp.	8	PR
	1871	Corethrella sp.	8	PR
Family Culicidae	1873	Culicidae	8	CG
	1875	Aedes sp.	8	CF
	1876	Aedes atropalpus	8	
	1879	Aedes canadensis	8	
	1880	Aedes cinereus	8	
	1881	Aedes communis	8	
	1882	Aedes dorsalis	8	
	1885	Aedes flavescens	8	
	1888	Aedes sollicitans	8	
	1889	Aedes sticticus	8	
	1890	Aedes stimulans	8	
	1894	Aedes triseriatus	8	
	1895	Aedes trivittatus	8	
	1896	Aedes vexans	8	
	1897	Anopheles sp.	6	CF
	1898	Anopheles barberi	6	
	1899	Anopheles crucians	6	
	1900	Anopheles earlei	6	
	1901	Anopheles punctipennis	6	
	1902	Anopheles quadrimaculatus	6	
	1903	Anopheles walkeri	6	
	1904	Culex sp.	8	CF
	1905	Culex erraticus	8	
	1906	Culex peccator	8	
	1907	Culex pipiens	8	CF
	1908	Culex quinquefasciatus	8	
	1909	Culex restuans	8	
	1910	Culex salinarius	8	
	1911	Culex tarsalis	8	

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	1912	Culiseta sp.	8	CG
	1913	Culiseta inornata	8	
	1914	Culiseta melanura	8	
	1917	Mansonia sp.	8	CG
	1918	Mansonia perturbans	8	
	1919	Psorophora sp.	8	PR
	1920	Psorophora ciliata	8	
	1921	Psorophora confinnis	8	PR
	1922	Psorophora cyanescens	8	
	1923	Psorophora discolor	8	
	1924	Psorophora ferox	8	
	1925	Psorophora horrida	8	
	1926	Psorophora howardi	8	
	1927	Psorophora varipes	8	
	1928	Uranotaenia sp.	8	CF
	1929	Uranotaenia sapphirina	8	
Family Dixidae	1930	Dixidae	10	CG
	1931	Dixa sp.	10	CG
	2238	Dixella sp.	10	
Family Psychodidae	1933	Psychodidae	11	CG
	2165	Telmatoscopus sp.	11	CG
	1934	Pericoma sp.	11	CG
	1935	Psychoda sp.	11	CG
Family Ceratopogonidae	1936	Ceratopogonidae	5	PR
	1937	Atrichopogon sp.	2	PR
	1938	Bezzia sp.	5	CG
	2166	Ceratopogon sp.	5	PR
	1939	Culicoides sp.	5	PR
	1940	Dasyhelea sp.	5	CG
	1941	Forcipomyia sp.	5	SC
	2167	Monohelea sp.	5	PR
	2223	Nilobezzia sp.	5	PR
	1942	Palpomyia sp.	6	PR
	2596	Serromyia sp.	5	
	1943	Probezzia sp.	5	PR
	2224	Sphaeromias sp.	5	
	1945	Stilobezzia sp.	5	
Family Simuliidae	1946	Simuliidae	6	CF
	1947	Cnephia sp.	4	CF
	1948	Cnephia pecuarum	4	
	1949	Prosimulium sp.	2	CF
	1950	Prosimulium magnum	2	
	1951	Prosimulium mixtum	2	
	1952	Simulium sp.	6	CF

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	1953	Simulium clarkei	4	
	1954	Simulium corbis	0	
	1955	Simulium decorum	4	CF
	1956	Simulium jenningsi	4	CF
	1957	Simulium luggeri	2	
	1958	Simulium meridionale	1	CF
	1959	Simulium tuberosum	4	CF
	1960	Simulium venustum	6	CF
	1961	Simulium verecundum	6	
	1962	Simulium vittatum	8	CF
Family Chironomidae	1963	Chironomidae	6	CG
Tribe Tanypodinae	2206	Tanypodinae	6	PR
	1965	Ablabesmyia sp.	6	CG
	2168	Ablabesmyia annulata	6	
	2169	Ablabesmyia hauberi	6	
	2501	Ablabesmyia janta	6	
	2502	Ablabesmyia janta var II	6	
	1966	Ablabesmyia mallochi	6	
	1967	Ablabesmyia monilis	6	PR
	1968	Ablabesmyia parajanta	6	
	1969	Ablabesmyia peleensis	6	
	2170	Ablabesmyia tarella	6	
	1970	Clinotanypus sp.	6	PR
	1971	Clinotanypus pinguis	6	PR
	1973	Coelotanypus sp.	4	PR
	1972	Coelotanypus concinnus	6	PR
	2171	Conchapelopia sp.	6	PR
	1974	Djalmabatista sp.	6	PR
	2239	Djalmabatista pulchra	6	
	1975	Guttipelopia sp.	6	PR
	2214	Hayesomyia sp.	5	
	2215	Helopelopia sp.	4	PR
	2509	Hudsonimyia sp.	6	
	1976	Labrundinia sp.	4	PR
	2512	Labrundinia neopilosella	4	
	2241	Labrundinia pilosella	4	
	2172	Labrundinia virescens	6	PR
	1977	Larsia sp.	6	PR
	1978	Macropelopia sp.	7	PR
	2211	Meropelopia sp.	3	
	1979	Natarsia sp.	6	PR
	1980	Nilotanypus sp.	6	PR
	2174	Nilotanypus fimbriatus	6	PR
	2173	Paramerina sp.	6	PR

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	1981	Pentaneura sp.	3	PR
	1982	Procladius sp.	8	PR
	1983	Psectrotanypus sp.	8	PR
	2225	Rheopelopia sp.	3	PR
	1984	Tanypus sp.	8	PR
	2516	Tanypus carinatus	8	
	2175	Tanypus neopunctipennis	8	
	2508	Tanypus punctipennis	8	
	2515	Tanypus stellatus	8	
	1985	Thienemannimyia sp.	6	PR
	2176	Thienemannimyia senata	6	
	1986	Zavrelimyia sp.	8	
	2235	Zavrelimyia sinuosa com	8	
Tribe Diamesinae	2210	Diamesinae	6	
	1988	Diamesa sp.	4	CG
	1989	Odontomesa sp.	6	CG
	1990	Potthastia sp.	6	
	1991	Prodiamesa sp.	3	CG
	1992	Pseudodiamesa sp.	1	CG
	1993	Sympotthastia sp.	6	CG
	1994	Syndiamesa sp.	6	CG
Tribe Orthocladiinae	1995	Orthocladiinae	6	CG
	2202	Orthocladius sp./Cricotopus sp.	6	
	1996	Brillia sp.	6	SH
	1997	Cardiocladius sp.	6	PR
	1998	Chaetocladius sp.	6	CG
	1999	Corynoneura sp.	2	CG
	2177	Corynoneura taris	2	CG
	2000	Cricotopus sp.	8	SH
	2001	Cricotopus bicinctus	10	
	2002	Cricotopus intersectus	8	SH
	2003	Cricotopus sylvestris	8	
	2004	Cricotopus trifascia	6	
	2209	Epoicocladius sp.	6	CG
	2005	Eukiefferiella sp.	4	CG
	2006	Heterotrissocladius sp.	6	CG
	2007	Hydrobaenus sp.	2	SC
	2250	Gymnometriocnemus sp.	6	
	2578	Lopescladius sp.	4	
	2008	Metriocnemus sp.	6	CG
	2009	Nanocladius sp.	3	CG
	2178	Nanocladius distinctus	3	CG
	2010	Orthocladius sp.	4	CG
	2216	Parakiefferiella sp.	5	

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	2011	Parametriocnemus sp.	4	CG
	2179	Paraphaenocladius sp.	6	CG
	2012	Psectrocladius sp.	5	CG
	2597	Psilometriocnemus sp.	6	
	2013	Pseudorthocladius sp.	6	CG
	2014	Rheocricotopus sp.	6	CG
	2180	Rheocricotopus fuscipes	6	
	2246	Rheocricotopus robacki	6	
	2015	Smittia sp.	6	CG
	2220	Stilocladius sp.	6	
	2213	Tvetenia sp.	5	
	2016	Thienemanniella sp.	2	CG
	2017	Thienemanniella xena	2	CG
	2200	Zalutschia sp.	6	SH
Tribe Chironomini	2205	Chironomini	6	CG
	2201	Axarus sp.	6	CG
	2019	Chironomus sp.	11	CG
	2020	Chironomus anthracinus	11	
	2021	Chironomus attenuatus	10	
	2022	Chironomus decorus	11	
	2023	Chironomus plumosus	11	
	2024	Chironomus riparius	11	
	2186	Chironomus staegeri	11	
	2025	Cladopelma sp.	6	CG
	2026	Cryptochironomus sp.	8	PR
	2027	Cryptochironomus digitatus	8	
	2028	Cryptochironomus fulvus	8	PR
	2029	Cryptotendipes sp.	6	CG
	2030	Demicryptochironomus sp.	6	CG
	2031	Dicrotendipes sp.	6	CG
	2523	Dicrotendipes lucifer	6	
	2032	Dicrotendipes modestus	6	CG
	2033	Dicrotendipes neomodestus	6	CG
	2034	Dicrotendipes nervosus	6	
	2517	Dicrotendipes simpsoni	6	
	2524	Dicrotendipes tritomus	6	
	2035	Einfeldia sp.	10	CG
	2189	Einfeldia austeni	10	
	2240	Einfeldia pagana	10	
	2036	Endochironomus sp.	6	SH
	2037	Endochironomus nigricans	6	SH
	2038	Endochironomus subtendens	6	SH
	2039	Glyptotendipes sp.	10	CF
	2533	Glyptotendipes amplus	10	

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2040	Glyptotendipes lobiferus	10	SH
2041	Glyptotendipes paripes	10	
2042	Harnischia sp.	6	CG
2255	Hyporhygma quadripunctatum	4	
2043	Kiefferulus sp.	7	CG
2518	Lipiniella sp.	6	
2044	Microchironomus sp.	6	CG
2045	Microtendipes sp.	6	CF
2046	Microtendipes caducus	6	
2047	Microtendipes pedellus	6	CF
2208	Nilothauma sp.	3	
2048	Parachironomus sp.	8	PR
2194	Parachironomus carinatus	8	PR
2195	Parachironomus directus	8	
2242	Parachironomus frequens	8	
2049	Parachironomus monochromus	8	
2525	Parachironomus pectinatella	4	
2520	Parachironomus tenuicaudatus	8	
2050	Paracladopelma sp.	4	CG
2537	Paracladopelma nereis	4	CG
2052	Paralauterborniella sp.	6	CG
2243	Paralauterborniella nigrohalteralis	6	
2053	Paratendipes sp.	3	CG
2251	Pagastiella sp.	6	
2244	Paratendipes albimanus	3	
2054	Phaenopsectra sp.	4	SC
2055	Phaenopsectra flavipes	4	SC
2507	Phaenopsectra obediens gr.	4	
2245	Phaenopsectra punctipes gr.	4	
2057	Polypedilum sp.	6	SH
2540	Polypedilum aviceps	6	
2058	Polypedilum convictum gr.	6	SH
2065	Polypedilum digitifer	6	SH
2060	Polypedilum fallax	6	SH
2541	Polypedilum flavum	6	
2061	Polypedilum halterale	4	SH
2062	Polypedilum illinoense	5	SH
2059	Polypedilum obtusum	6	SH
2063	Polypedilum scalaenum	6	SH
2064	Polypedilum simulans	6	SH
2066	Pseudochironomus sp.	5	CG
2198	Pseudochironomus fulviventris	5	
2199	Pseudochironomus prasinatus	5	
2218	Robackia sp.	3	CG

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	2067	Saetheria sp.	6	CG
	2514	Seatheria tylus	4	
	2068	Stenochironomus sp.	3	SH
	2069	Stenochironomus hilaris	3	CG
	2219	Stelechomyia	6	CG
	2247	Stelechomyia pulpulchra	99.9	CG
	2070	Stictochironomus sp.	5	
	2519	Stictochironomus caffrarius	5	
	2248	Stictochironomus devinctus	5	
	2071	Tribelos sp.	5	CG
	2197	Tribelos fuscicorne	4	CG
	2232	Tribelos jucundus	5	
	2072	Xenochironomus sp.	4	PR
	2513	Xenochironomus xenolabis	6	
	2227	Xestochironomus sp.	6	
	2249	Xylotopus par.	6	
	3433	Zavreliella sp.	2	PR
	2254	Zavreliella marmorata	2	
Tribe Tanytarsini	2207	Tanytarsini	6	CF
	2074	Cladotanytarsus sp.	7	CG
	2503	Cladotanytarsus daviese	7	
	2504	Cladotanytarsus species a	7	
	2236	Cladotanytarsus species b	7	
	2505	Cladotanytarsus species c	7	
	2237	Cladotanytarsus species f	7	
	2506	Cladotanytarsus species h	7	
	2075	Micropsectra sp.	4	CG
	2076	Paratanytarsus sp.	6	CG
	2077	Rheotanytarsus sp.	6	CF
	2226	Stempellina sp.	2	CG
	2212	Stempellinella sp.	2	CG
	2228	Sublettea sp.	6	CF
	2078	Tanytarsus sp.	7	CF
	2230	Tanytarsus guerlus	7	
	2231	Tanytarsus glabrescan	7	
Family Ptychopteridae	2079	Ptychopteridae	8	CG
	2080	Bittacomorpha sp.	8	CG
	2081	Ptychoptera sp.	8	CG
Family Stratiomyidae	2082	Stratiomyidae	10	CG
	2092	Allognosta sp.	10	CG
	2083	Odostomia sp.	10	CG
	2084	Odostomia cincta	10	
	2085	Oxycera sp.	10	SC
	2086	Stratiomys sp.	10	CF

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	2087	Stratiomys discalis	10	
	2088	Stratiomys meigeni	10	
Family Tabanidae	2089	Tabanidae	7	PR
	2090	Atylotus sp.	7	PR
	2091	Atylotus bicolor	7	
	2143	Chlorotabanus sp.	7	
	2093	Chrysops sp.	7	CG
	2094	Chrysops aberrans	7	
	2097	Chrysops brunneus	7	
	2098	Chrysops callidus	7	
	2100	Chrysops cincticornis	7	
	2101	Chrysops dimmocki	7	
	2103	Chrysops flavidus	7	
	2105	Chrysops geminatus	7	
	2106	Chrysops macquarti	7	
	2108	Chrysops moechus	7	
	2109	Chrysops montanus	7	
	2110	Chrysops niger	7	
	2115	Chrysops striatus	7	
	2116	Chrysops univittatus	7	
	2118	Chrysops vittatus	7	
	2119	Hybomitra sp.	7	PR
	2125	Tabanus sp.	7	PR
	2126	Tabanus atratus	7	
	2127	Tabanus cymatophorus	7	
	2128	Tabanus fairchildi	7	
	2130	Tabanus lineola	7	
	2131	Tabanus marginalis	7	
	2132	Tabanus nigrescens	7	
	2133	Tabanus pumilus	7	
	2134	Tabanus quinquevittatus	7	
	2135	Tabanus reinwardtii	7	
	2136	Tabanus sparus	7	
	2137	Tabanus stygius	7	
	2139	Tabanus subsimilis	7	
	2140	Tabanus sulcifrons	7	
	2141	Tabanus superjumentarius	7	
	2142	Tabanus trimaculatus	7	
Family Dolichopodidae	2144	Dolichopodidae	5	PR
	2145	Hydrophorus sp.	99.9	PR
	2594	Raphium sp.	99.9	
Family Empididae	2146	Empididae	6	PR
	2147	Hemerodromia sp.	6	PR
	2595	Rhamphomyia sp.	1.0	

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Family Syrphidae	2148	Syrphidae	11	CG
	2149	Chrysogaster sp.	11	CG
	2150	Eristalis sp.	11	CG
Family Ephydridae	2151	Ephydridae	8	CG
	2152	Ephydra sp.	8	CG
Family Sciomyzidae	2153	Sciomyzidae	10	PR
	2154	Dictya sp.	10	PR
	2155	Dictya pictipes	10	
Family Muscidae	2156	Muscidae	8	PR
	2157	Limnophora sp.	8	PR
Family Athericidae	2158	Athericidae	10	
	2159	Atherix sp.	10	PR
	2160	Atherix variegata	4	PR
Phylum Mollusca	2300	Mollusca	99.9	
Class Gastropoda	2301	Gastropoda	99.9	SC
Order Mesogastropoda	2599	Mesogastropoda	99.9	
Family Viviparidae	2302	Viviparidae	6	SC
	2303	Campeloma sp.	7	SC
	2304	Lioplax sp.	7	SC
	2305	Viviparus sp.	1	SC
	2306	Valvatidae	6	SC
	2307	Valvata sp.	2	SC
Family Bithyniidae	2308	Bithyniidae	6	
	2309	Bithynia sp.	6	
Family Hydrobiidae	2310	Hydrobiidae	6	SC
	2312	Amnicola sp.	4	SC
	2313	Amnicola walkeri	4	
	2314	Cincinnatia sp.	6	SC
	2315	Marstonia sp.	6	
	2316	Probythinella sp.	6	
	2317	Pyrgulopsis sp.	6	SC
	2318	Somatogyrus sp.	6	
Family Pleuroceridae	2319	Pleuroceridae	6	
	2320	Elimia sp.	6	SC
	2321	Goniobasis sp.	5	SC
	2322	Leptoxis sp.	6	
	2323	Lithasia sp.	6	
	2324	Pleurocera sp.	7	SC
	2325	Pleurocera acuta	7	SC
Family Pomatiopsidae	2326	Pomatiopsidae	6	
	2327	Pomatiopsis sp.	6	
Family Physidae	2328	Physidae	9	SC
	2329	Aplexa sp.	7	
	2330	Physa sp.	9	SC

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	2351	Physa acuta	9	
	2361	Physa integra	9	CG
	2331	Physella sp.	9	SC
	2352	Physella sayi	9	
Family Lymnaeidae	2332	Lymnaeidae	7	SC
	2333	Acella sp.	7	
Order Basommatophora	2598	Basommatophora	7	
	2334	Fossaria sp.	7	SC
	2362	Fossaria obrussa	7	
	2335	Lymnaea sp.	7	SC
	2336	Pseudosuccinea sp.	7	SC
	2355	Pseudosuccinea columella	7	SC
	2337	Stagnicola sp.	7	SC
	2338	Stagnicola emarginatus	7	
Family Planorbidae	2339	Planorbidae	6.5	SC
	2340	Gyraulus sp.	6	SC
	2341	Helisoma sp.	7	SC
	2342	Menetus sp.	6.5	SC
	2343	Planorbella sp.	6.5	SC
	2357	Planorbella truncata	6.5	
	2344	Planorbula sp.	7	SC
	2345	Promenetus sp.	6.5	CG
Family Ancylidae	2346	Ancylidae	7	SC
	2347	Ferrissia sp.	7	SC
	2358	Ferrissia rivularis	7	SC
	2348	Laevapex sp.	6	SC
	2359	Laevapex fuscus	6	SC
	2360	Laevapex diaphanus	6	SC
Class Pelecypoda	2400	Pelecypoda	99.9	CF
Order Unionoida	2600	Unionoida	99.9	
Family Unionidae	2401	Unionidae	1.5	CF
	2402	Actinonaias sp.	1.5	
	2403	Actinonaias carinata	1	
	2404	Actinonaias ellipsiformis	1.5	
	2405	Actinonaias ligamentina	1.5	
	2406	Alasmidonta sp.	1.5	CF
	2407	Alasmidonta calceolus	1.5	
	2408	Alasmidonta marginata	1	
	2409	Alasmidonta triangulata	1.5	
	2410	Amblema sp.	1.5	
	2411	Amblema plicata	1.5	
	2412	Anodonta sp.	3	CF
	2413	Anodonta grandis	3	
	2415	Anodonta imbecilis	3	

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2416	Anodonta suborbiculata	3	
2417	Anodontoides sp.	1.5	CF
2418	Anodontoides ferussacianus	1.5	
2419	Arcidens sp.	1.5	CF
2420	Arcidens confragosus	1.5	
2421	Carunculina sp.	7	CF
2500	Toxolasma paruvus	7	
2423	Carunculina parva	7	
2424	Toxolasma texasensis	7	
2425	Cyclonaias sp.	1.5	
2426	Cyclonaias tuberculata	1.5	
2427	Cyprogenia sp.	1.5	
2428	Cyprogenia irrorata	1.5	
2429	Dysnomia sp.	1.5	
2431	Dysnomia triquetra	1.5	
2432	Elliptio sp.	2	CF
2433	Elliptio crassidens	2	
2434	Elliptio dilatata	2	
2435	Fusconaia sp.	1	
2436	Fusconaia ebena	1	
2437	Fusconaia flava	1	
2438	Lampsilis sp.	1	CF
2439	Lampsilis teres	1	CF
2498	Lampsilis fasciola	1	
2441	Lampsilis radiata	1	
2442	Lampsilis ventricosa	1	
2443	Lampsilis orbiculata	1	
2444	Lampsilis higginsi	1	
2445	Lasmigona sp.	1.5	
2446	Lasmigona complanata	1.5	
2447	Lasmigona compressa	1.5	
2448	Lasmigona costata	1.5	
2449	Leptodea sp.	1.5	CF
2450	Leptodea fragilis	1.5	
2451	Ligumia sp.	1	CF
2452	Ligumia recta	1	
2453	Ligumia subrostrata	1	
2454	Megalonaias sp.	1.5	
2455	Megalonaias nervosa	1.5	
2456	Obliquaria sp.	1	
2457	Obliquaria reflexa	1	
2458	Obovaria sp.	1.5	
2459	Obovaria olivaria	1.5	
2460	Obovaria subrotunda	1.5	

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	2462	Plagiola lineolata	1.5	
	2463	Plethobasus sp.	1.5	
	2464	Plethobasus cyphyus	1.5	
	2465	Pleurobema sp.	1.5	
	2466	Pleurobema cordatum	1.5	
	2467	Proptera sp.	1	
	2468	Proptera alata	1	
	2469	Proptera capax	1	
	2470	Proptera laevissimus	1	
	2471	Ptychobranchus sp.	1.5	
	2472	Ptychobranchus fasciolaris	1.5	
	2473	Quadrula sp.	1.5	
	2474	Quadrula cyclindrica	1.5	
	2475	Quadrula metanerva	1.5	
	2476	Quadrula nodulata	1.5	
	2477	Quadrula pustulosa	1.5	
	2478	Quadrula quadrula	1.5	
	2479	Strophitus sp.	4	
	2480	Strophitus undulatus	4	
	2481	Tritogonia sp.	1	
	2482	Tritogonia verrucosa	1	
	2483	Truncilla sp.	1	
	2484	Truncilla donaciformis	1	
	2485	Truncilla truncata	1	
	2486	Uniomerus sp.	1.5	
	2487	Uniomerus tetralasmus	1.5	
	2488	Villosa sp.	1	
	2489	Villosa iris	1	
	2490	Villosa lienosa	1	
Order Venerioda	2601	Veneroida	5	
Family Pisidiidae	2499	Pisidiidae	5	
Family Sphaeriidae	2491	Sphaeriidae	5	
	2492	Musculium sp.	5	CF
	2493	Musculium transversum	5	CF
	2494	Pisidium sp.	5	CF
	2495	Sphaerium sp.	5	CG
Family Corbiculidae	2497	Corbicula sp.	4	CF
Family Dreissenidae	2234	Dreissena polymorpha	99.9	

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Alphabetic Order

Bios ID	Taxon	Tolerance	Functional Feeding Group
2168	Ablabesmyia annulata	6	
2169	Ablabesmyia hauberi	6	
2501	Ablabesmyia janta	6	
2502	Ablabesmyia janta var II	6	
1966	Ablabesmyia mallochi	6	
1967	Ablabesmyia monilis	6	PR
1968	Ablabesmyia parajanta	6	
1969	Ablabesmyia peleensis	6	
1965	Ablabesmyia sp.	6	CG
2170	Ablabesmyia tarella	6	
2605	Acanthametropodidae		
479	Acanthametropus pecatonica	3	
478	Acanthametropus sp.	3	PR
2333	Acella sp.	7	
652	Acentrella sp.	4	
506	Acerpenna macdunnoughi	4	CG
508	Acerpenna pygmaeus	4	
2203	Acerpenna sp.	4	SH
1689	Acilius fraternus	99.9	
1691	Acilius semisulcatus	99.9	
1688	Acilius sp.	99.9	PR
956	Acroneuria abnormis	1	PR
957	Acroneuria arida	1	PR
958	Acroneuria carolinensis	1	PR
959	Acroneuria evoluta	1	PR
960	Acroneuria internata	1	PR
961	Acroneuria lycorias	1	PR
955	Acroneuria sp.	1	PR
154	Actinobdella inequiannulata	8	
153	Actinobdella sp.	8	
2403	Actinonaias carinata	1	
2404	Actinonaias ellipsiformis	1.5	
2405	Actinonaias ligamentina	1.5	
2402	Actinonaias sp.	1.5	
1876	Aedes atropalpus	8	
1879	Aedes canadensis	8	
1880	Aedes cinereus	8	
1881	Aedes communis	8	
1882	Aedes dorsalis	8	
1885	Aedes flavescens	8	
1888	Aedes sollicitans	8	
1875	Aedes sp.	8	CF

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1889	Aedes sticticus	8	
1890	Aedes stimulans	8	
1894	Aedes triseriatus	8	
1895	Aedes trivittatus	8	
1896	Aedes vexans	8	
37	Aeolosomatidae	10	CF
744	Aeshna canadensis	4	
746	Aeshna constricta	4	PR
743	Aeshna sp.	4	PR
747	Aeshna umbrosa	4	
748	Aeshna verticalis	4	
742	Aeshnidae	4.5	PR
1692	Agabetes sp.	99.9	PR
1696	Agabus ambiguus	99.9	
1698	Agabus disintegratus	99.9	
1694	Agabus sp.	99.9	PR
1371	Agapetus illini	2	
1372	Agapetus sp.	2	SC
1490	Agarodes distincta	3.5	
1378	Agraylea multipunctata	2	
1377	Agraylea sp.	2	PH
1439	Agrypnia sp.	3	SH
1441	Agrypnia vestita	3	
2407	Alasmidonta calceolus	1.5	
2408	Alasmidonta marginata	1	
2406	Alasmidonta sp.	1.5	CF
2409	Alasmidonta triangulata	1.5	
156	Alboglossiphonia heteroclita	8	PR
155	Alboglossiphonia sp.	8	
946	Allocapnia mystica	1.5	
947	Allocapnia recta	1.5	
945	Allocapnia sp.	2	SH
948	Allocapnia vivipara	1.5	SH
2092	Allognosta sp.	10	CG
42	Allonais pectinata	10	
41	Allonais sp.	10	CG
991	Alloperla sp.	1.5	PR
2411	Amblema plicata	1.5	
2410	Amblema sp.	1.5	
2604	Ameletidae		
481	Ameletus lineatus	0	
480	Ameletus sp.	0	CG
2312	Amnicola sp.	4	SC
2313	Amnicola walkeri	4	

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849	Amphiagrion saucium	5	
848	Amphiagrion sp.	5	PR
44	Amphichaeta leydigi	10	
43	Amphichaeta sp.	10	CG
937	Amphinemura sp.	1.5	SH
325	Amphipoda	4	CG
1409	Anabolia sp.	3.5	SH
1643	Anacaena limbata	99.9	
1642	Anacaena sp.	99.9	
750	Anax junius	5	PR
749	Anax sp.	5	PR
2346	Ancylidae	7	SC
1772	Ancyronyx sp.	2	
1773	Ancyronyx variegatus	2	CG
530	Anepeorus simplex	3.5	
529	Anepeorus sp.	3.5	PR
30	Annelida	99.9	CG
2413	Anodonta grandis	3	
2415	Anodonta imbecilis	3	
2412	Anodonta sp.	3	CF
2416	Anodonta suborbiculata	3	
2418	Anodontoides ferussacianus	1.5	
2417	Anodontoides sp.	1.5	CF
851	Anomalagrion hastatum	5.5	PR
850	Anomalagrion sp.	5.5	PR
1898	Anopheles barberi	6	
1899	Anopheles crucians	6	
1900	Anopheles earlei	6	
1901	Anopheles punctipennis	6	
1902	Anopheles quadrimaculatus	6	
1897	Anopheles sp.	6	CF
1903	Anopheles walkeri	6	
623	Anthopotamus myops	4	
621	Anthopotamus sp.	4	
1855	Antocha sp.	5	CG
2329	Aplexa sp.	7	
839	Archilestes grandis	1	
838	Archilestes sp.	1	PR
2420	Arcidens confragosus	1.5	
2419	Arcidens sp.	1.5	CF
46	Arcteonais lomondi	10	CG
45	Arcteonais sp.	10	
853	Argia apicalis	5	PR
854	Argia bipunctulata	5	

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997	Argia fumipennis	5	PR
855	Argia moesta	5	PR
856	Argia sedula	5	PR
852	Argia sp.	5	PR
857	Argia tibialis	5	PR
858	Argia translata	5	
859	Argia violacea	5	PR
707	Arigomphus sp.	7	PR
532	Arthroplea bipunctata	3	
531	Arthroplea sp.	3	CF
2603	Arthropleidae		
250	Arthropoda	99.9	
253	Asellidae	6	CG
255	Asellus sp.	6	
2158	Athericidae	10	
2159	Atherix sp.	10	PR
2160	Atherix variegata	4	PR
971	Atoperla sp.	1	
1937	Atrichopogon sp.	2	PR
963	Attaneuria ruralis	1.5	PR
962	Attaneuria sp.	1.5	
569	Attenella attenuata	2	CG
568	Attenella sp.	2	CG
2091	Atylotus bicolor	7	
2090	Atylotus sp.	7	PR
93	Aulodrilus pigueti	10	CG
2201	Axarus sp.	6	CG
332	Bactrurus sp.	1	
497	Baetidae	4	CG
645	Baetis amplus	4	CG
653	Baetis armillatus	4	
499	Baetis brunneicolor	4	CG
500	Baetis ephippiatus	4	
501	Baetis flavistriga	4	CG
502	Baetis frondalis	4	
503	Baetis hageni	4	CG
504	Baetis intercalaris	7	
646	Baetis levitans	4	
505	Baetis longipalpus	6	
507	Baetis propinquus gr.	4	
509	Baetis quilleri	4	
498	Baetis sp.	4	CG
510	Baetis tricaudatus	1	CG
647	Baetis vagans	4	

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604	Baetisca bajkovi	3	
606	Baetisca lacustris	3	
607	Baetisca laurentina	3	
608	Baetisca obesa	3	
605	Baetisca sp.	3	CG
603	Baetiscidae	3	CG
1443	Banksiola crotchi	2	
1442	Banksiola sp.	2	SH
650	Barbaetis cestus	4	
752	Basiaeschna janata	2	PR
751	Basiaeschna sp.	2	PR
2598	Basommatophora	7	
158	Batracobdella phalera	8	
159	Batracobdella picta	8	
157	Batracobdella sp.	8	PR
1085	Belostoma flumineum	99.9	PR
1084	Belostoma sp.	99.9	PR
1083	Belostomatidae	99.9	PR
1646	Berosus fraternus	99.9	
1647	Berosus infuscatus	99.9	
1649	Berosus peregrinus	99.9	
1650	Berosus pugnax	99.9	
1644	Berosus sp.	99.9	PR
1651	Berosus striatus	99.9	CG
1938	Bezzia sp.	5	CG
1704	Bidessonotus inconspicuus	99.9	
1703	Bidessonotus sp.	99.9	PR
2309	Bithynia sp.	6	
2308	Bithyniidae	6	
2080	Bittacomorpha sp.	8	CG
1852	Blepharicera sp.	0	SC
1851	Blephariceridae	0	SC
753	Boyeria sp.	3	PR
754	Boyeria vinosa	3	PR
1397	Brachycentridae	3.5	CF
1399	Brachycentrus americanus	1	CF
1400	Brachycentrus lateralis	1	CF
1401	Brachycentrus numerosus	1	CF
1402	Brachycentrus occidentalis	1	CF
1398	Brachycentrus sp.	1	CF
601	Brachycercus sp.	3	CG
32	Branchiobdellida	10	PA
33	Branchiobdellidae	10	CG
95	Branchiura sowerbyi	10	CG

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94	Branchiura sp.	10	CG
47	Bratislavia sp.	10	CG
48	Bratislavia unidentata	10	CG
1996	Brillia sp.	6	SH
1068	Buenoa sp.	99.9	PR
256	Caecidotea brevicaudus	6	
347	Caecidotea communis	6	CG
257	Caecidotea forbesi	6	
258	Caecidotea intermedia	6	
259	Caecidotea kendeighi	6	
268	Caecidotea packardi	6	
254	Caecidotea sp.	6	CG
269	Caecidotea spatulata	6	
270	Caecidotea stygia	6	
263	Caecidotea tridentata	6	
600	Caenidae	5.5	CG
602	Caenis sp.	6	CG
512	Callibaetis ferrugineus	4	
513	Callibaetis fluctuans	4	
514	Callibaetis skokianus	4	
511	Callibaetis sp.	4	CG
830	Calopterygidae	3.5	PR
832	Calopteryx aequabilis	4	
833	Calopteryx maculata	4	PR
831	Calopteryx sp.	4	PR
403	Cambarellus puer	5	CG
404	Cambarellus shufeldtii	5	CG
402	Cambarellus sp.	5	SH
401	Cambaridae	5	CG
406	Cambarus diogenes	5	
407	Cambarus rusticiformis	5	
405	Cambarus sp.	5	CG
408	Cambarus tenebrosus	5	
2303	Campeloma sp.	7	SC
949	Capnia sp.	1	SH
950	Capnia vernalis	1	
944	Capniidae	1.5	SH
1997	Cardiocladius sp.	6	PR
2423	Carunculina parva	7	
2421	Carunculina sp.	7	CF
1706	Celina hubbelli	99.9	
1705	Celina sp.	99.9	PR
789	Celithemis elisa	2	
790	Celithemis eponina	2	

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791	Celithemis monomelaena	2	
788	Celithemis sp.	2	PR
515	Centroptilum sp.	2	CG
1456	Ceraclea ancylus	3	
1457	Ceraclea cancellata	3	
1458	Ceraclea diluta	3	
1459	Ceraclea flava	3	
1460	Ceraclea maculata	3	
1462	Ceraclea nepha	3	
1463	Ceraclea resurgens	3	
1454	Ceraclea sp.	3	CG
2510	Ceraclea transversa	3	
2166	Ceratopogon sp.	5	PR
1936	Ceratopogonidae	5	PR
1496	Ceratopsyche alhedra	4	
1492	Ceratopsyche alternans	5	CF
1332	Ceratopsyche bronta	4	CF
1333	Ceratopsyche cheilonis	4	
1493	Ceratopsyche morosa	4	
1499	Ceratopsyche slossonae	4	
1330	Ceratopsyche sp.	4	CF
1337	Ceratopsyche sparna	4	
1998	Chaetocladius sp.	6	CG
50	Chaetogaster diaphanus	10	PR
51	Chaetogaster diastrophus	10	PR
52	Chaetogaster limnaei	10	PR
49	Chaetogaster sp.	10	SH
1869	Chaoboridae	8	PR
1870	Chaoborus sp.	8	PR
1188	Chauliodes pectinicornis	4	PR
1189	Chauliodes rastricornis	4	PR
1187	Chauliodes sp.	4	PR
1302	Cheumatopsyche sp.	6	CF
1340	Chimarra aterrima	3	CF
1341	Chimarra feria	3	CF
1342	Chimarra obscura	3	CF
1343	Chimarra socia	3	CF
1339	Chimarra sp.	3	CF
1963	Chironomidae	6	CG
2205	Chironomini	6	CG
2020	Chironomus anthracinus	11	
2021	Chironomus attenuatus	10	
2022	Chironomus decorus	11	
2023	Chironomus plumosus	11	

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2024	Chironomus riparius	11	
2019	Chironomus sp.	11	CG
2186	Chironomus staegeri	11	
990	Chloroperla sp.	3	
989	Chloroperlidae	1.5	PR
2143	Chlorotabanus sp.	7	
611	Choroterpes basalis	2	CG
610	Choroterpes sp.	2	CG
861	Chromagrion conditum	5.5	
860	Chromagrion sp.	5.5	PR
2149	Chrysogaster sp.	11	CG
2094	Chrysops aberrans	7	
2097	Chrysops brunneus	7	
2098	Chrysops callidus	7	
2100	Chrysops cincticornis	7	
2101	Chrysops dimmocki	7	
2103	Chrysops flavidus	7	
2105	Chrysops geminatus	7	
2106	Chrysops macquarti	7	
2108	Chrysops moechus	7	
2109	Chrysops montanus	7	
2110	Chrysops niger	7	
2093	Chrysops sp.	7	CG
2115	Chrysops striatus	7	
2116	Chrysops univittatus	7	
2118	Chrysops vittatus	7	
2314	Cincinnatia sp.	6	SC
2025	Cladopelma sp.	6	CG
2503	Cladotanytarsus daviese	7	
2074	Cladotanytarsus sp.	7	CG
2504	Cladotanytarsus species a	7	
2236	Cladotanytarsus species b	7	
2505	Cladotanytarsus species c	7	
2237	Cladotanytarsus species f	7	
2506	Cladotanytarsus species h	7	
1253	Climacea areolaris	1	
1252	Climacea sp.	1	
1971	Clinotanypus pinguis	6	PR
1970	Clinotanypus sp.	6	PR
517	Cloeon alamance	3	
518	Cloeon rubropictum	3	
516	Cloeon sp.	3	
1948	Cnephia pecuarum	4	
1947	Cnephia sp.	4	CF

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1972	Coelotanypus concinnus	6	PR
1973	Coelotanypus sp.	4	PR
862	Coenagrion sp.	5.5	PR
847	Coenagrionidae	5.5	PR
1600	Coleoptera	99.9	PR
2171	Conchapelopia sp.	6	PR
1708	Copelatus chevrolati	99.9	
1709	Copelatus glyphicus	99.9	
1707	Copelatus sp.	99.9	PR
1710	Coptotomus sp.	99.9	PR
2497	Corbicula sp.	4	CF
704	Cordulegaster maculata	2	PR
705	Cordulegaster obliqua	2	PR
703	Cordulegaster sp.	2	PR
702	Cordulegastridae	4.5	PR
770	Cordulia shurtleffi	2	
769	Cordulia sp.	2	PR
768	Corduliidae	4.5	PR
1871	Corethrella sp.	8	PR
1090	Corixidae	99.9	PR
1186	Corydalidae	3	PR
1191	Corydalus cornutus	3	PR
1190	Corydalus sp.	3	PR
1999	Corynoneura sp.	2	CG
2177	Corynoneura taris	2	CG
1501	Crambidae	99.9	SH
336	Crangonyx forbesi	4	CG
337	Crangonyx gracilis	4	CG
338	Crangonyx minor	4	
339	Crangonyx packardi	4	
340	Crangonyx pseudogracilis	4	
335	Crangonyx sp.	4	CG
1652	Crenitis sp.	99.9	PR
2001	Cricotopus bicinctus	10	
2002	Cricotopus intersectus	8	SH
2000	Cricotopus sp.	8	SH
2003	Cricotopus sylvestris	8	
2004	Cricotopus trifascia	6	
348	Crongonyctidae	4	
251	Crustacea	99.9	CG
2027	Cryptochironomus digitatus	8	
2028	Cryptochironomus fulvus	8	PR
2026	Cryptochironomus sp.	8	PR
2029	Cryptotendipes sp.	6	CG

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1905	Culex erraticus	8	
1906	Culex peccator	8	
1907	Culex pipiens	8	CF
1908	Culex quinquefasciatus	8	
1909	Culex restuans	8	
1910	Culex salinarius	8	
1904	Culex sp.	8	CF
1911	Culex tarsalis	8	
1873	Culicidae	8	CG
1939	Culicoides sp.	5	PR
1913	Culiseta inornata	8	
1914	Culiseta melanura	8	
1912	Culiseta sp.	8	CG
1799	Curculionidae	99.9	SH
1714	Cybister fimbriolatus	99.9	
1713	Cybister sp.	99.9	PR
2425	Cyclonaias sp.	1.5	
2426	Cyclonaias tuberculata	1.5	
1655	Cymbiodyta chamberlaini	99.9	
1656	Cymbiodyta semistriata	99.9	
1653	Cymbiodyta sp.	99.9	CG
1657	Cymbiodyta vindicata	99.9	
1619	Cyphon americanus	7	
1620	Cyphon collaris	7	
1621	Cyphon modestus	7	
1622	Cyphon nebulosus	7	
1623	Cyphon obscurus	7	
1625	Cyphon perplexus	7	
1624	Cyphon punctatus	7	
1618	Cyphon sp.	7	SC
2428	Cyprogenia irrorata	1.5	
2427	Cyprogenia sp.	1.5	
1351	Cyrnellus fraternus	5	CF
1350	Cyrnellus sp.	5	CF
179	Cystobranchus sp.	7	
178	Cystobranchus verrilli	7	
571	Dannella lita	2	CG
572	Dannella simplex	2	CG
570	Dannella sp.	2	
1940	Dasyhelea sp.	5	CG
400	Decapoda	99.9	SH
2030	Demicryptochironomus sp.	6	CG
54	Dero digitata	10	CG
55	Dero furcata	10	CG

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56	Dero lodeni	10	CG
57	Dero nivea	10	CG
109	Dero pectinata	10	CG
53	Dero sp.	10	CG
1715	Deronectes sp.	99.9	PR
208	Desserobdella phalera	8	
1988	Diamesa sp.	4	CG
2210	Diamesinae	6	
1760	Dicranopselaphus	4	SC
1856	Dicranota sp.	4	PR
2523	Dicrotendipes lucifer	6	
2032	Dicrotendipes modestus	6	CG
2033	Dicrotendipes neomodestus	6	CG
2034	Dicrotendipes nervosus	6	
2517	Dicrotendipes simpsoni	6	
2031	Dicrotendipes sp.	6	CG
2524	Dicrotendipes tritomus	6	
2155	Dictya pictipes	10	
2154	Dictya sp.	10	PR
760	Didymops sp.	4	PR
761	Didymops transversa	4	PR
199	Dina dubia	8	
200	Dina parva	8	
198	Dina sp.	8	PR
1603	Dineutus assimilis (larvae only)	4	PR
1604	Dineutus discolor (larvae only)	4	PR
1602	Dineutus sp. (larvae only)	4	PR
661	Diphetor hageni	4	
1303	Diplectrona metaqui	2	
1305	Diplectrona modesta	2	CF
1304	Diplectrona sp.	2	CF
1850	Diptera	10	
1931	Dixa sp.	10	CG
2238	Dixella sp.	10	
1930	Dixidae	10	CG
2239	Djalmabatista pulchra	6	
1974	Djalmabatista sp.	6	PR
2144	Dolichopodidae	5	PR
1345	Dolophilodes distinctus	0	
1344	Dolophilodes sp.	0	CG
2234	Dreissena polymorpha	99.9	
713	Dromogomphus sp.	4	PR
714	Dromogomphus spinosus	4	PR
575	Drunella cornutella	1	SC

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573	Drunella sp.	1	PR
1764	Dryopidae	4	SH
1775	Dubiraphia bivittata	2	
1776	Dubiraphia minima	5	
1777	Dubiraphia quadrinotata	7	
1774	Dubiraphia sp.	5	CG
1778	Dubiraphia vittata	7	
5	Dugesia sp.	6	
6	Dugesia tigrina	6	PR
2429	Dysnomia sp.	1.5	
2431	Dysnomia triquetra	1.5	
1687	Dytiscidae	99.9	PR
1719	Dytiscus hybridus	99.9	
1717	Dytiscus sp.	99.9	PR
1762	Ectopria nervosa	4	SC
1761	Ectopria sp.	4	SC
1763	Ectopria thoracica	4	
2189	Einfeldia austeni	10	
2240	Einfeldia pagana	10	
2035	Einfeldia sp.	10	CG
2320	Elimia sp.	6	SC
2433	Elliptio crassidens	2	
2434	Elliptio dilatata	2	
2432	Elliptio sp.	2	CF
1771	Elmidae	5	CG
1801	Elodes sp.	7	
2146	Empididae	6	PR
864	Enallagma aspersum	6	
865	Enallagma civile	6	
866	Enallagma divagans	6	PR
867	Enallagma exsulans	6	
868	Enallagma geminatum	6	
869	Enallagma hageni	6	
870	Enallagma signatum	6	PR
863	Enallagma sp.	6	PR
871	Enallagma traviatum	6	
872	Enallagma vesperum	6	PR
38	Enchytraeidae	10	CG
2037	Endochironomus nigricans	6	SH
2036	Endochironomus sp.	6	SH
2038	Endochironomus subtendens	6	SH
1660	Enochrus cinctus	99.9	
1662	Enochrus consortus	99.9	
1663	Enochrus hamiltoni	99.9	

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1664	Enochrus ochraceus	99.9	
1665	Enochrus perplexus	99.9	
1666	Enochrus pygmaeus	99.9	
1667	Enochrus sayi	99.9	
1658	Enochrus sp.	99.9	CG
533	Epeorus sp.	1	SC
534	Epeorus vitreus	0	
627	Ephemera simulans	3	CG
626	Ephemera sp.	3	CG
579	Ephemerella aurivillii	2	CG
580	Ephemerella catawba	2	CG
574	Ephemerella cornuta	1	
590	Ephemerella coxalis	4	
581	Ephemerella dorothea	2	CG
582	Ephemerella excrucians	2	CG
596	Ephemerella frisoni	1	
583	Ephemerella invaria	2	CG
576	Ephemerella lata	1	
584	Ephemerella needhami	2	CG
585	Ephemerella rotunda	2	
578	Ephemerella sp.	2	CG
586	Ephemerella subvaria	2	CG
577	Ephemerella walkeri	1	
567	Ephemerellidae	3.5	CG
625	Ephemeridae	5	CG
476	Ephemeroptera	3	CG
639	Ephoron album	2	CG
640	Ephoron leukon	2	CG
638	Ephoron sp.	2	CG
2152	Ephydra sp.	8	CG
2151	Ephydridae	8	CG
756	Epiaeschna heros	1	PR
755	Epiaeschna sp.	1	PR
773	Epicordulia princeps	4.5	PR
772	Epicordulia sp.	4.5	PR
771	Epitheca sp.	4	PR
2209	Epoicocladius sp.	6	CG
1857	Eriocera sp.	7	PR
1858	Erioptera sp.	4	CG
2150	Eristalis sp.	11	CG
2229	Erpetogomphus designatus	2	
2602	Erpetogomphus sp.	2	
202	Erpobdella punctata	8	PR
201	Erpobdella sp.	8	

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197	Erpobdellidae	8	PR
793	Erythemis simplicicollis	5	PR
792	Erythemis sp.	5	PR
794	Erythrodiplax sp.	5	PR
2005	Eukiefferiella sp.	4	CG
588	Eurylophella aestiva	4	
589	Eurylophella bicolor	4	CG
591	Eurylophella funeralis	4	
592	Eurylophella lutulenta	4	CG
587	Eurylophella sp.	4	SC
593	Eurylophella temporalis	4	CG
1445	Fabria inornata	3.5	
1444	Fabria sp.	3.5	SH
410	Fallicambarus fodiens	5	
409	Fallicambarus sp.	5	
2358	Ferrissia rivularis	7	SC
2347	Ferrissia sp.	7	SC
1941	Forcipomyia sp.	5	SC
2362	Fossaria obrussa	7	
2334	Fossaria sp.	7	SC
1411	Frenesia missa	3.5	
1410	Frenesia sp.	3.5	SH
2436	Fusconaia ebena	1	
2437	Fusconaia flava	1	
2435	Fusconaia sp.	1	
329	Gammaridae	4	CG
346	Gammarus fasciatus	3	CG
344	Gammarus pseudolimnaeus	3	CG
341	Gammarus sp.	3	
345	Gammarus troglophilus	3	
2301	Gastropoda	99.9	SC
1057	Gerridae	99.9	PR
1058	Gerris sp.	99.9	PR
210	Gloiobdella elongata	8	
161	Glossiphonia complanata	8	PR
160	Glossiphonia sp.	8	PR
152	Glossiphoniidae	8	PR
1374	Glossosoma intermedium	3.5	SC
1373	Glossosoma sp.	3.5	SC
1370	Glossosomatidae	3.5	SC
2533	Glyptotendipes amplus	10	
2040	Glyptotendipes lobiferus	10	SH
2041	Glyptotendipes paripes	10	
2039	Glyptotendipes sp.	10	CF

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211	Gnathobdellida	7	
1412	Goera sp.	3.5	SC
706	Gomphidae	4.5	PR
738	Gomphus amnicola	7	
716	Gomphus crassus	7	
723	Gomphus exilis	7	
717	Gomphus externus	7	PR
724	Gomphus graslinellus	7	
710	Gomphus lentulus	7	
719	Gomphus lineatifrons	7	
725	Gomphus lividus	7	PR
739	Gomphus notatus	7	
740	Gomphus plagiatus	7	
726	Gomphus quadricolor	7	
722	Gomphus sp.	7	PR
741	Gomphus spiniceps	7	
711	Gomphus submedianus	7	
720	Gomphus vastus	7	PR
879	Gomphurus sp.	7	
712	Gomphus villosipes	7	
2321	Goniobasis sp.	5	SC
2221	Gonomyia sp.	4	CG
18	Gordius sp.	99.9	
1975	Guttipelopia sp.	6	PR
2250	Gymnometriocnemus sp.	6	
2340	Gyraulus sp.	6	SC
1601	Gyrinidae	99.9	PR
1608	Gyrinus aeneolus (larvae only)	4	PR
1609	Gyrinus analis (larvae only)	4	
1607	Gyrinus sp. (larvae only)	4	PR
613	Habrophlebiodes americana	2	
612	Habrophlebiodes sp.	2	SC
190	Haemopis marmorata	7	
189	Haemopis sp.	7	PR
191	Haemopis terrestris	7	
729	Hagenius brevistylus	3	PR
728	Hagenius sp.	3	PR
1629	Haliplidae	99.9	SH
1631	Haliplus fasciatus	99.9	SH
1632	Haliplus immaculicollis	99.9	
1633	Haliplus leopardus	99.9	
1634	Haliplus pantherinus	99.9	
1630	Haliplus sp.	99.9	MH
1635	Haliplus triopsis	99.9	

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36	Haplotaxida	10	
2042	Harnischia sp.	6	CG
993	Hastaperla brevis	1.5	
992	Hastaperla sp.	1.5	SC
2214	Hayesomyia sp.	5	
1051	Hebridae	99.9	PR
1052	Hebrus sp.	99.9	PR
1766	Helichus fastigiatus	4	
1767	Helichus lithophilus	4	
1765	Helichus sp.	4	SH
1768	Helichus striatus	4	
1452	Helicopsyche borealis	2	SC
1451	Helicopsyche sp.	2	SC
1450	Helicopsychidae	3.5	SC
2341	Helisoma sp.	7	SC
1859	Helius sp.	5	CG
163	Helobdella elongata	8	PR
164	Helobdella fusca	8	PA
165	Helobdella papillata	8	PR
162	Helobdella sp.	8	PA
166	Helobdella stagnalis	8	PR
167	Helobdella triserialis	8	PA
1803	Helocombus sp.	99.9	
774	Helocordulia sp.	2	PR
1617	Helodidae	7	SC
2215	Helopelopia sp.	4	PR
1668	Helophorus sp.	99.9	SH
2147	Hemerodromia sp.	6	PR
1050	Hemiptera	99.9	PR
535	Heptagenia diabasia	4	
537	Heptagenia flavescens	2	
538	Heptagenia hebe	3	
539	Heptagenia lucidipennis	3	
540	Heptagenia maculipennis	3	SC
541	Heptagenia marginalis	1	SC
542	Heptagenia perfida	1	
543	Heptagenia pulla	0	SC
536	Heptagenia sp.	3	SC
528	Heptageniidae	3.5	SC
1860	Hesperoconopa sp.	2	CG
1092	Hesperocorixa interrupta	99.9	
1094	Hesperocorixa laevigata	99.9	
1096	Hesperocorixa lucida	99.9	
1097	Hesperocorixa nitida	99.9	

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1098	Hesperocorixa obliqua	99.9	
1091	Hesperocorixa sp.	99.9	PR
1100	Hesperocorixa vulgaris	99.9	
1414	Hesperophylax designatus	3.5	SH
1413	Hesperophylax sp.	3.5	SH
835	Hetaerina americana	3	PR
834	Hetaerina sp.	3	PR
836	Hetaerina titia	3	PR
520	Heterocloeon curiosum	4	SC
519	Heterocloeon sp.	4	SC
2006	Heterotrissocladius sp.	6	CG
629	Hexagenia atrocaudata	6	CG
630	Hexagenia bilineata	6	CG
631	Hexagenia limbata	5	CG
632	Hexagenia munda	5	
633	Hexagenia rigida	6	CG
628	Hexagenia sp.	6	CG
1861	Hexatoma sp.	4	PR
249	Hirudinea	8	PR
188	Hirudinidae	8	PR
2509	Hudsonimyia sp.	6	
328	Hyalella azteca	5	CG
327	Hyalella sp.	4	CG
326	Hyalellidae	4	
2119	Hybomitra sp.	7	PR
1721	Hydaticus modestus	99.9	
1720	Hydaticus sp.	99.9	PR
1416	Hydatophylax argus	2	SH
1415	Hydatophylax sp.	2	SH
2007	Hydrobaenus sp.	2	SC
2310	Hydrobiidae	6	SC
1670	Hydrobius fuscipes	99.9	
1669	Hydrobius sp.	99.9	PR
1673	Hydrochara sp.	99.9	CG
1674	Hydrochus sp.	99.9	SH
978	Hydroperla crosbyi	1	
977	Hydroperla sp.	1	PR
1641	Hydrophilidae	99.9	PR
1675	Hydrophilus sp.	99.9	PR
2145	Hydrophorus sp.	99.9	PR
1725	Hydroporus clypealis	99.9	
1726	Hydroporus consimilus	99.9	
1728	Hydroporus niger	99.9	
1729	Hydroporus rufilabris	99.9	

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1722	Hydroporus sp.	99.9	PR
1733	Hydroporus vittatipennis	99.9	
1307	Hydropsyche aerata	5	CF
1308	Hydropsyche arinale	5	
1309	Hydropsyche betteni	5	CF
1310	Hydropsyche bidens	5	
1311	Hydropsyche cuanis	5	
1312	Hydropsyche dicantha	5	CF
1313	Hydropsyche frisoni	5	CF
1314	Hydropsyche hageni	5	
1316	Hydropsyche incommoda	5	
1317	Hydropsyche orris	4	CF
1318	Hydropsyche phalerata	2	CF
1319	Hydropsyche placoda	4	
1320	Hydropsyche scalaris	5	CF
1321	Hydropsyche simulans	5	CF
1306	Hydropsyche sp.	5	CF
1322	Hydropsyche valanis	5	CF
1323	Hydropsyche venularis	5	CF
1301	Hydropsychidae	5.5	CF
1379	Hydroptila sp.	2	SC
1500	Hydroptila waubesiana	2	
1376	Hydroptilidae	3.5	PH
1735	Hydrovatus pustalatus	99.9	PR
1734	Hydrovatus sp.	99.9	PR
1736	Hygrotus sp.	99.9	PR
2255	Hyporhygma quadripunctatum	4	
1738	Ilybius biguttulus	99.9	
1737	Ilybius sp.	99.9	PR
96	Ilyodrilus sp.	10	
97	Ilyodrilus templetoni	10	CG
475	Insecta	99.9	
1417	Ironoquia sp.	3.5	SH
874	Ischnura posita	6	PR
873	Ischnura sp.	6	PR
875	Ischnura verticalis	6	
979	Isogenoides sp.	1.5	PR
488	Isonychia arida	3	
489	Isonychia bicolor	3	CG
490	Isonychia rufa	3	
491	Isonychia sayi	3	
492	Isonychia sicca	3	
487	Isonychia sp.	3	CF
981	Isoperla bilineata	2	

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982	Isoperla clio	2	
996	Isoperla confusa	2	
983	Isoperla cotta	2	
984	Isoperla dicala	2	
985	Isoperla lata	2	
986	Isoperla marlynia	2	
987	Isoperla nana	2	
988	Isoperla richardsoni	2	
980	Isoperla sp.	2	PR
252	Isopoda	99.9	CG
1380	Ithytrichia sp.	1	SC
2043	Kiefferulus sp.	7	CG
2512	Labrundinia neopilosella	4	
2241	Labrundinia pilosella	4	
1976	Labrundinia sp.	4	PR
2172	Labrundinia virescens	6	PR
1677	Laccobius agilis	99.9	
1678	Laccobius minutoides	99.9	
1676	Laccobius sp.	99.9	PR
1740	Laccophilus fasciatus	99.9	PR
1741	Laccophilus maculosus	99.9	
1742	Laccophilus proximus	99.9	PR
1739	Laccophilus sp.	99.9	PR
1744	Laccornis sp.	99.9	PR
796	Ladona julia	4.5	
795	Ladona sp.	4.5	PR
2360	Laevapex diaphanus	6	SC
2359	Laevapex fuscus	6	SC
2348	Laevapex sp.	6	SC
2498	Lampsilis fasciola	1	
2444	Lampsilis higginsi	1	
2443	Lampsilis orbiculata	1	
2441	Lampsilis radiata	1	
2438	Lampsilis sp.	1	CF
2439	Lampsilis teres	1	CF
2442	Lampsilis ventricosa	1	
735	Lanthus sp.	6	PR
1977	Larsia sp.	6	PR
2446	Lasmigona complanata	1.5	
2447	Lasmigona compressa	1.5	
2448	Lasmigona costata	1.5	
2445	Lasmigona sp.	1.5	
1550	Lepidoptera	99.9	SH
1407	Lepidostoma liba	3	

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1406	Lepidostoma sp.	3	SH
1405	Lepidostomatidae	3.5	SH
1453	Leptoceridae	3.5	CG
1467	Leptocerus americanus	3	
1466	Leptocerus sp.	3	SH
2450	Leptodea fragilis	1.5	
2449	Leptodea sp.	1.5	CF
662	Leptohyphe sp.	5.5	CG
598	Leptohyphidae	5.5	CG
614	Leptophlebia sp.	3	CG
609	Leptophlebiidae	3	CG
1418	Leptophylax sp.	3.5	SH
2322	Leptoxis sp.	6	
841	Lestes disjunctus	6	
842	Lestes eurinus	6	
843	Lestes forcipatus	6	
844	Lestes inaequalis	6	
845	Lestes rectangularis	6	
840	Lestes sp.	6	PR
846	Lestes vigilax	6	
837	Lestidae	99.9	PR
1087	Lethocerus americans	99.9	
1088	Lethocerus griseus	99.9	
1086	Lethocerus sp.	99.9	PR
1089	Lethocerus uhleri	99.9	
799	Leucorrhinia intacta	4.5	
797	Leucorrhinia sp.	4.5	PR
1382	Leucotrichia pictipes	3	
1381	Leucotrichia sp.	3	SC
654	Leucrocuta hebe	3	
649	Leucrocuta maculipennis	3	
648	Leucrocuta sp.	3	SC
943	Leuctra sp.	1	SH
942	Leuctridae	1.5	SH
801	Libellula cyanea	8	
802	Libellula incesta	8	PR
803	Libellula luctuosa	8	
804	Libellula pulchella	8	
805	Libellula quadrimaculata	8	
806	Libellula semifasciata	8	PR
800	Libellula sp.	8	PR
807	Libellula vibrans	8	PR
787	Libellulidae	4.5	PR
2452	Ligumia recta	1	

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2451	Ligumia sp.	1	CF
2453	Ligumia subrostrata	1	
1408	Limnephilidae	3.5	SH
1420	Limnephilus sp.	3	SH
99	Limnodrilus cervix	10	CG
100	Limnodrilus claparedianus	10	CG
101	Limnodrilus hoffmeisteri	10	CG
98	Limnodrilus sp.	10	CG
102	Limnodrilus udekemianus	10	CG
1060	Limnogonus hesione	99.9	
1059	Limnogonus sp.	99.9	PR
1862	Limnophila sp.	4	PR
2157	Limnophora sp.	8	PR
1863	Limonia sp.	3	SH
1745	Liodessus sp.	99.9	PR
2304	Lioplax sp.	7	SC
2518	Lipiniella sp.	6	
273	Lirceus fontinalis	4	CG
274	Lirceus garmani	4	CG
275	Lirceus lineatus	4	CG
276	Lirceus louisianae	4	
272	Lirceus sp.	4	CG
1800	Listronotus sp.	99.9	CF
2323	Lithasia sp.	6	
2578	Lopescladius sp.	4	
39	Lumbricidae	10	CG
34	Lumbriculida	10	
35	Lumbriculidae	10	CG
2335	Lymnaea sp.	7	SC
2332	Lymnaeidae	7	SC
1367	Lype diversa	3.5	SC
1366	Lype sp.	3.5	SC
561	Maccaffertium annexum	4	
565	Maccaffertium ares	3	
552	Maccaffertium exiguum	5	
556	Maccaffertium integrum	4	
557	Maccaffertium luteum	1	SC
559	Maccaffertium mediopunctatum	2	SC
560	Maccaffertium modestum	3	SC
558	Maccaffertium nepotellum	5	
563	Maccaffertium pulchellum	3	SC
553	Maccaffertium quinquespinum	5	
562	Maccaffertium rubromaculatum	2	
551	Maccaffertium sp.	4	SC

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564	Maccaffertium terminatum	4	SC
566	Maccaffertium vicarium	3	SC
193	Macrobdella decora	7	
192	Macrobdella sp.	7	
764	Macromia georgina	3	PR
765	Macromia illinoiensis	3	PR
766	Macromia pacifica	3	
762	Macromia sp.	3	PR
767	Macromia taeniolata	3	PR
759	Macromiidae	4.5	PR
1324	Macronema sp.	2	CF
1325	Macronema zebratum	2	CF
1780	Macronychus glabratus	2	
1779	Macronychus sp.	2	
1978	Macropelopia sp.	7	PR
2217	Macrostemum sp.	2	CF
1918	Mansonia perturbans	8	
1917	Mansonia sp.	8	CG
2315	Marstonia sp.	6	
1749	Matus bicarinatus	99.9	
1748	Matus sp.	99.9	PR
1383	Mayatrichia ayama	1	SC
1384	Mayatrichia sp.	1	SC
2455	Megalonaias nervosa	1.5	
2454	Megalonaias sp.	1.5	
1175	Megaloptera	3.5	
2342	Menetus sp.	6.5	SC
2211	Meropelopia sp.	3	
1053	Merragata sp.	99.9	PR
2599	Mesogastropoda	99.9	
1056	Mesovelia mulsanti	99.9	PR
1055	Mesovelia sp.	99.9	PR
1054	Mesoveliidae	99.9	PR
493	Metretopodidae	3	
2008	Metriocnemus sp.	6	CG
1061	Metrobates sp.	99.9	PR
1404	Micrasema rusticum	3.5	
1403	Micrasema sp.	3.5	MH
2044	Microchironomus sp.	6	CG
1782	Microcylloepus pusillus	2	CG
1781	Microcylloepus sp.	2	CG
2075	Micropsectra sp.	4	CG
2046	Microtendipes caducus	6	
2047	Microtendipes pedellus	6	CF

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2045	Microtendipes sp.	6	CF
1065	Microvelia sp.	99.9	PR
1435	Molanna blenda	3.5	
1434	Molanna sp.	3.5	SC
1436	Molanna tryphena	3.5	
1437	Molanna uniophila	3.5	
1433	Molannidae	3.5	CG
2300	Mollusca	99.9	
2167	Monohelea sp.	5	PR
204	Mooreobdella fervida	8	
205	Mooreobdella microstoma	8	PR
203	Mooreobdella sp.	8	PR
1552	Munroessa sp.	99.9	SH
2156	Muscidae	8	PR
2492	Musculium sp.	5	CF
2493	Musculium transversum	5	CF
1470	Mystacides sepulchralis	2	
1468	Mystacides sp.	2	CG
181	Myzobdella lugubris	7	PR
180	Myzobdella sp.	7	
40	Naididae	10	CG
59	Nais barbata	10	CG
60	Nais behningi	10	CG
61	Nais bretscheri	10	CG
62	Nais communis	10	CG
63	Nais elinguis	10	CG
64	Nais pardalis	10	CG
65	Nais simplex	10	CG
58	Nais sp.	10	CG
66	Nais variabilis	10	CG
2178	Nanocladius distinctus	3	CG
2009	Nanocladius sp.	3	CG
757	Nasiaeschna pentacantha	2	PR
758	Nasiaeschna sp.	2	PR
1979	Natarsia sp.	6	PR
1073	Naucoridae	99.9	PR
1472	Nectopsyche albida	3	
1473	Nectopsyche candida	3	
1474	Nectopsyche diarina	3	
1475	Nectopsyche exquisita	3	
1476	Nectopsyche pavida	3	
1471	Nectopsyche sp.	3	SH
877	Nehalennia gracilis	7	
878	Nehalennia irene	7	

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876	Nehalennia sp.	7	PR
15	Nematomorpha	99.9	PA
938	Nemoura sp.	1	SH
939	Nemoura venosa	1	
936	Nemouridae	1.5	SH
965	Neoperla clymene	1	PR
964	Neoperla sp.	1	PR
1422	Neophylax concinnus	3	
1421	Neophylax sp.	3	SC
1071	Neoplea sp.	99.9	PR
1072	Neoplea striola	99.9	
1385	Neotrichia sp.	4	SC
1078	Nepa apiculata	99.9	
1077	Nepa sp.	99.9	PR
207	Nephelopsis obscura	8	PR
206	Nephelopsis sp.	8	
1076	Nepidae	99.9	PR
1354	Neureclipsis bimaculata	3	CF
1353	Neureclipsis crepuscularis	3	CF
1352	Neureclipsis sp.	3	CF
776	Neurocordulia molesta	3	PR
777	Neurocordulia obsoleta	3	PR
775	Neurocordulia sp.	3	PR
778	Neurocordulia yamaskanensis	3	
1250	Neuroptera	99.9	PR
1193	Nigronia fasciatus	2	PR
1194	Nigronia serricornis	2	PR
1192	Nigronia sp.	2	PR
2223	Nilobezzia sp.	5	PR
2174	Nilotanypus fimbriatus	6	PR
1980	Nilotanypus sp.	6	PR
2208	Nilothauma sp.	3	
655	Nixe perfida	4	
644	Nixe sp.	4	SC
1069	Notonecta sp.	99.9	PR
1067	Notonectidae	99.9	PR
1355	Nyctiophylax sp.	1	CF
1554	Nymphula sp.	99.9	SH
2457	Obliquaria reflexa	1	
2456	Obliquaria sp.	1	
2459	Obovaria olivaria	1.5	
2458	Obovaria sp.	1.5	
2460	Obovaria subrotunda	1.5	
1386	Ochrotrichia sp.	4	CG

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700	Odonata	99.9	PR
1989	Odontomesa sp.	6	CG
2084	Odostomia cincta	10	
2083	Odostomia sp.	10	CG
1478	Oecetis avara	5	PR
1479	Oecetis cinerascens	5	PR
1480	Oecetis eddlestoni	5	PR
1481	Oecetis inconspicua	5	PR
2521	Oecetis nocturna	5	
1482	Oecetis ochracea	5	
1477	Oecetis sp.	5	PR
930	Oemopteryx glacialis	1.5	
929	Oemopteryx sp.	1.5	SH
31	Oligochaeta	10	CG
486	Oligoneuriidae	3	CF
1447	Oligostomis ocelligera	3.5	PR
1446	Oligostomis sp.	3.5	PR
69	Ophidonais serpentina	10	CG
68	Ophidonais sp.	10	
731	Ophiogomphus rupinsulensis	2	
730	Ophiogomphus sp.	2	PR
1784	Optioservus fastiditus	4	SC
1785	Optioservus ovalis	4	SC
1783	Optioservus sp.	4	SC
1786	Optioservus trivittatus	4	SC
430	Orconectes bisectus	5	
412	Orconectes illinoiensis	5	
413	Orconectes immunis	5	
414	Orconectes indianensis	5	
415	Orconectes kentuckiensis	5	
416	Orconectes lancifer	5	
417	Orconectes placidus	5	
418	Orconectes propinquus	5	
419	Orconectes rusticus	5	
411	Orconectes sp.	5	
420	Orconectes stannardi	5	
421	Orconectes virilis	5	
1864	Ormosia sp.	4	CG
1995	Orthocladiinae	6	CG
2010	Orthocladius sp.	4	CG
2202	Orthocladius sp./Cricotopus sp.	6	
1387	Orthotrichia sp.	1	SC
2085	Oxycera sp.	10	SC
1388	Oxyethira sp.	2	MH

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810	Pachydiplax longipennis	8	PR
809	Pachydiplax sp.	8	PR
2251	Pagastiella sp.	6	
429	Palaemonetes kadiakensis	4	
428	Palaemonetes sp.	4	
427	Palaemonidae	4	
1102	Palmacorixa buenoi	99.9	
1103	Palmacorixa gilletteii	99.9	
1104	Palmacorixa nana	99.9	
1101	Palmacorixa sp.	99.9	PR
1942	Palpomyia sp.	6	PR
812	Pantala flavescens	7	
813	Pantala hymenaea	7	
811	Pantala sp.	7	PR
952	Paracapnia angulata	1.5	
953	Paracapnia opis	1.5	
951	Paracapnia sp.	1.5	SH
2194	Parachironomus carinatus	8	PR
2195	Parachironomus directus	8	
2242	Parachironomus frequens	8	
2049	Parachironomus monochromus	8	
2525	Parachironomus pectinatella	4	
2048	Parachironomus sp.	8	PR
2520	Parachironomus tenuicaudatus	8	
2537	Paracladopelma nereis	4	CG
2050	Paracladopelma sp.	4	CG
2233	Paracloeodes minutus	5	
527	Paracloeodes sp.	4	SC
1679	Paracymus sp.	99.9	PR
1680	Paracymus subcupreus	99.9	
967	Paragnetina media	1.5	PR
966	Paragnetina sp.	1.5	PR
2216	Parakiefferiella sp.	5	
2243	Paralauterborniella nigrohalteralis	6	
2052	Paralauterborniella sp.	6	CG
616	Paraleptophlebia moerens	2	
617	Paraleptophlebia ontario	2	
618	Paraleptophlebia praepedita	2	
615	Paraleptophlebia sp.	2	CG
619	Paraleptophlebia sticta	2	
2173	Paramerina sp.	6	PR
2011	Parametriocnemus sp.	4	CG
67	Paranais frici	10	
110	Paranais sp.	10	

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2179	Paraphaenocladius sp.	6	CG
1555	Paraponyx sp.	99.9	SH
1327	Parapsyche apicalis	5.5	
1326	Parapsyche sp.	5.5	PR
2076	Paratanytarsus sp.	6	CG
2244	Paratendipes albimanus	3	
2053	Paratendipes sp.	3	CG
1865	Pedicia sp.	4	PR
2400	Pelecypoda	99.9	CF
1075	Pelocoris femoratus	99.9	PR
1074	Pelocoris sp.	99.9	PR
1770	Pelonomus obscurus	4	
1769	Pelonomus sp.	4	CG
1638	Peltodytes dunavani	99.9	
1637	Peltodytes duodecimpunctatus	99.9	
1639	Peltodytes lengi	99.9	
1640	Peltodytes sexmaculatus	99.9	
1636	Peltodytes sp.	99.9	SH
635	Pentagenia sp.	4	CF
636	Pentagenia vittigera	4	CG
1981	Pentaneura sp.	3	PR
1934	Pericoma sp.	11	CG
814	Perithemis sp.	4	PR
815	Perithemis tenera	4	PR
969	Perlesta placida	4	
968	Perlesta sp.	4	PR
954	Perlidae	1.5	PR
972	Perlinella drymo	2	PR
973	Perlinella ephyre	2	PR
970	Perlinella sp.	2	PR
976	Perlodidae	1.5	PR
1556	Petrophila sp.	5	SC
2055	Phaenopsectra flavipes	4	SC
2507	Phaenopsectra obediens gr.	4	
2245	Phaenopsectra punctipes gr.	4	
2054	Phaenopsectra sp.	4	SC
209	Pharyngobdellidae	8	
975	Phasganophora capitata	1.5	
974	Phasganophora sp.	1.5	PR
195	Philobdella gracilis	7	PR
194	Philobdella sp.	7	
1338	Philopotamidae	3.5	CF
1448	Phryganea sp.	3	
1438	Phryganeidae	3.5	SH

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1357	Phylocentropus placidus	3.5	
1356	Phylocentropus sp.	3.5	CF
2351	Physa acuta	9	
2361	Physa integra	9	CG
2330	Physa sp.	9	SC
2352	Physella sayi	9	
2331	Physella sp.	9	SC
2328	Physidae	9	SC
1866	Pilaria sp.	4	PR
183	Piscicola milneri	7	PR
184	Piscicola punctata	7	PR
182	Piscicola sp.	7	PR
186	Piscicolaria reducta	7	
185	Piscicolaria sp.	7	
177	Piscicolidae	7	
2499	Pisidiidae	5	
2494	Pisidium sp.	5	CF
168	Placobdella montifera	8	PR
170	Placobdella multilineata	8	PR
171	Placobdella ornata	8	PR
172	Placobdella papillifera	8	PA
173	Placobdella parasitica	8	PA
174	Placobdella pediculata	8	
169	Placobdella sp.	8	PR
2462	Plagiola lineolata	1.5	
7	Planaria sp.	6	
4	Planariidae	6	
2343	Planorbella sp.	6.5	SC
2357	Planorbella truncata	6.5	
2339	Planorbidae	6.5	SC
2344	Planorbula sp.	7	SC
817	Plathemis lydia	3	PR
816	Plathemis sp.	3	PR
1424	Platycentropus radiatus	3	
1423	Platycentropus sp.	3	SH
1	Platyhelminthes	99.9	
660	Plauditus armillatus	4	
657	Plauditus punctiventris	3	
651	Plauditus sp.	3	
925	Plecoptera	1.5	PR
1070	Pleidae	99.9	PR
2464	Plethobasus cyphyus	1.5	
2463	Plethobasus sp.	1.5	
2466	Pleurobema cordatum	1.5	

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2465	Pleurobema sp.	1.5	
2325	Pleurocera acuta	7	SC
2324	Pleurocera sp.	7	SC
2319	Pleuroceridae	6	
1349	Polycentropodidae	3.5	CF
1359	Polycentropus centralis	3	PR
1360	Polycentropus cinereus	3	PR
1361	Polycentropus flavus	3	PR
1362	Polycentropus glacialis	3	PR
1363	Polycentropus interruptus	3	PR
1364	Polycentropus remotus	3	PR
1358	Polycentropus sp.	3	PR
637	Polymitarcyidae	3	CG
2540	Polypedilum aviceps	6	
2058	Polypedilum convictum gr.	6	SH
2065	Polypedilum digitifer	6	SH
2060	Polypedilum fallax	6	SH
2541	Polypedilum flavum	6	
2061	Polypedilum halterale	4	SH
2062	Polypedilum illinoense	5	SH
2059	Polypedilum obtusum	6	SH
2063	Polypedilum scalaenum	6	SH
2064	Polypedilum simulans	6	SH
2057	Polypedilum sp.	6	SH
2326	Pomatiopsidae	6	
2327	Pomatiopsis sp.	6	
620	Potamanthidae	5	CF
105	Potamothrix vejdovskyi	10	
1329	Potamyia flava	4	CF
1328	Potamyia sp.	4	CF
1990	Potthastia sp.	6	
1867	Prionocera sp.	4	SH
1802	Prionocyphon sp.	7	SC
71	Pristina aequiseta	10	CG
73	Pristina breviseta	10	CG
72	Pristina leidyi	10	CG
74	Pristina longiseta	10	
75	Pristina osborni	10	CG
70	Pristina sp.	10	CG
76	Pristina synclites	10	CG
1943	Probezzia sp.	5	PR
2316	Probythinella sp.	6	
423	Procambarus acutus	5	SH
424	Procambarus clarki	5	

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425	Procambarus gracilis	5	
422	Procambarus sp.	5	SH
426	Procambarus viaeviridis	5	
1982	Procladius sp.	8	PR
643	Procloeon sp.	4	
1991	Prodiamesa sp.	3	CG
733	Progomphus obscurus	5	PR
732	Progomphus sp.	5	PR
2345	Promenetus sp.	6.5	CG
1787	Promoresia sp.	5	SC
2468	Proptera alata	1	
2469	Proptera capax	1	
2470	Proptera laevissimus	1	
2467	Proptera sp.	1	
1950	Prosimulium magnum	2	
1951	Prosimulium mixtum	2	
1949	Prosimulium sp.	2	CF
940	Prostoia sp.	1.5	SH
1375	Protoptila sp.	1	SC
2012	Psectrocladius sp.	5	CG
1983	Psectrotanypus sp.	8	PR
1614	Psephenidae	4	SC
1615	Psephenus herricki	4	SC
1616	Psephenus sp.	4	SC
2198	Pseudochironomus fulviventris	5	
2199	Pseudochironomus prasinatus	5	
2066	Pseudochironomus sp.	5	CG
522	Pseudocloeon carolina	4	
523	Pseudocloeon dubium	4	SC
524	Pseudocloeon myrsum	4	
525	Pseudocloeon parvulum	4	
656	Pseudocloeon propinquus gr.	4	
526	Pseudocloeon punctiventris	4	
521	Pseudocloeon sp.	4	SC
1992	Pseudodiamesa sp.	1	CG
1868	Pseudolimnophila sp.	2	PR
2013	Pseudorthocladius sp.	6	CG
1425	Pseudostenophylax sp.	3.5	SH
1426	Pseudostenophylax uniformis	3.5	
2355	Pseudosuccinea columella	7	SC
2336	Pseudosuccinea sp.	7	SC
2597	Psilometriocnemus sp.	6	
1920	Psorophora ciliata	8	
1921	Psorophora confinnis	8	PR

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1922	Psorophora cyanescens	8	
1923	Psorophora discolor	8	
1924	Psorophora ferox	8	
1925	Psorophora horrida	8	
1926	Psorophora howardi	8	
1919	Psorophora sp.	8	PR
1927	Psorophora varipes	8	
1935	Psychoda sp.	11	CG
1933	Psychodidae	11	CG
1369	Psychomyia flavida	2	CG
1368	Psychomyia sp.	2	SC
1365	Psychomyiidae	3.5	CG
927	Pteronarcys sp.	2	SH
1449	Ptilostomis sp.	3	SH
2472	Ptychobranchus fasciolaris	1.5	
2471	Ptychobranchus sp.	1.5	
2081	Ptychoptera sp.	8	CG
2079	Ptychopteridae	8	CG
1428	Pycnopsyche guttifer	3	SH
1429	Pycnopsyche lepida	3	
1430	Pycnopsyche luculenta	3	
1431	Pycnopsyche scabripennis	3	SH
1427	Pycnopsyche sp.	3	SH
1432	Pycnopsyche subfasciata	3	SH
1551	Pyralidae	99.9	SH
2317	Pyrgulopsis sp.	6	SC
2474	Quadrula cyclindrica	1.5	
2475	Quadrula metanerva	1.5	
2476	Quadrula nodulata	1.5	
2477	Quadrula pustulosa	1.5	
2478	Quadrula quadrula	1.5	
2473	Quadrula sp.	1.5	
108	Quistradrilus multisetosus	10	CG
1106	Ramphocorixa acuminata	99.9	
1105	Ramphocorixa sp.	99.9	PR
1079	Ranatra fusca	99.9	PR
1081	Ranatra kirkaldyi	99.9	PR
1082	Ranatra nigra	99.9	PR
1080	Ranatra sp.	99.9	PR
2594	Raphium sp.	99.9	
994	Rasvena sp.	1.5	
995	Rasvena terna	1.5	CG
1066	Rhagovelia sp.	99.9	PR
2595	Rhamphomyia sp.	1	

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1752	Rhantus binotatus	99.9	
1751	Rhantus sp.	99.9	PR
2180	Rheocricotopus fuscipes	6	
2246	Rheocricotopus robacki	6	
2014	Rheocricotopus sp.	6	CG
2225	Rheopelopia sp.	3	PR
2077	Rheotanytarsus sp.	6	CF
1062	Rheumatobates sp.	99.9	PR
545	Rhithrogena pellucida	0	SC
544	Rhithrogena sp.	0	SC
1393	Rhyacophila fenestra	1	
1394	Rhyacophila fuscula	1	PR
1395	Rhyacophila lobifera	1	
1392	Rhyacophila sp.	1	PR
1396	Rhyacophila vibox	1	
1391	Rhyacophilidae	3.5	PR
151	Rhynchobdellida	8	
2218	Robackia sp.	3	CG
2067	Saetheria sp.	6	CG
2153	Sciomyzidae	10	PR
1627	Scirtes orbiculatus	7	
1626	Scirtes sp.	7	SH
1628	Scirtes tibialis	7	
2514	Seatheria tylus	4	
1489	Sericostomatidae	3.5	SH
595	Serratella deficiens	1	CG
597	Serratella sordida	1	CG
594	Serratella sp.	1	CG
2596	Serromyia sp.	5	
1483	Setodes sp.	3.5	
1176	Sialidae	3.5	PR
1180	Sialis infumata	4	
1181	Sialis itasca	4	
1183	Sialis mohri	4	PR
1177	Sialis sp.	4	PR
1184	Sialis vagans	4	
1185	Sialis velata	4	
1108	Sigara alternata	99.9	
1109	Sigara compressoidea	99.9	
1111	Sigara hubbelli	99.9	
1113	Sigara modesta	99.9	
1115	Sigara signata	99.9	
1107	Sigara sp.	99.9	PR
1946	Simuliidae	6	CF

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1953	Simulium clarkei	4	
1954	Simulium corbis	0	
1955	Simulium decorum	4	CF
1956	Simulium jenningsi	4	CF
1957	Simulium luggeri	2	
1958	Simulium meridionale	1	CF
1952	Simulium sp.	6	CF
1959	Simulium tuberosum	4	CF
1960	Simulium venustum	6	CF
1961	Simulium verecundum	6	
1962	Simulium vittatum	8	CF
477	Siphlonuridae	3	CG
483	Siphlonurus alternatus	2	
484	Siphlonurus quebecensis	2	
485	Siphlonurus rapidus	2	
482	Siphlonurus sp.	2	CG
495	Siphloplecton basale	2	
496	Siphloplecton interlineatum	2	
494	Siphloplecton sp.	2	CG
1254	Sisyra sp.	1	PR
1255	Sisyra vicaria	1	
1251	Sisyridae	1	PR
78	Slavina appendiculata	10	CG
77	Slavina sp.	10	CG
2015	Smittia sp.	6	CG
781	Somatochlora filosa	1	
782	Somatochlora linearis	1	PR
779	Somatochlora sp.	1	PR
783	Somatochlora tenebrosa	1	
2318	Somatogyrus sp.	6	
941	Soyedina sp.	1.5	SH
80	Specaria josinae	10	CG
79	Specaria sp.	10	CG
2491	Sphaeriidae	5	
2495	Sphaerium sp.	5	CG
2224	Sphaeromias sp.	5	
1390	Stactobiella palmata	3.5	
1389	Stactobiella sp.	3.5	SH
2338	Stagnicola emarginatus	7	
2337	Stagnicola sp.	7	SC
2219	Stelechomyia	6	CG
2247	Stelechomyia pulpulchra	99.9	CG
2226	Stempellina sp.	2	CG
2212	Stempellinella sp.	2	CG

Functional Feeding Group (ffg)
SC=scraper, PA=parasite, PR=predator, OM=omnivore, GC=gatherer/collector, FC=filter/collector, SH=shredder, PI=piercer
99.9=Taxon excluded from mIBI computation

Appendix A. Illinois Environmental Protection Agency Macroinvertebrate Tolerance List and Functional Feeding Group Classification

January, 2011

548	Stenacron interpunctatum	4	
550	Stenacron sp.	4	SC
1789	Stenelmis bicarinata	7	SC
1790	Stenelmis crenata	7	
1791	Stenelmis decorata	7	SC
1792	Stenelmis lateralis	7	SC
1793	Stenelmis markeli	7	SC
1794	Stenelmis mera	7	SC
1795	Stenelmis musgravei	7	SC
1796	Stenelmis sexlineata	7	SC
1788	Stenelmis sp.	7	SC
1797	Stenelmis vittipennis	6	
2069	Stenochironomus hilaris	3	CG
2068	Stenochironomus sp.	3	SH
554	Stenonema femoratum	7	SC
659	Stenonema sp.	4	SC
81	Stephensoniana sp.	10	
82	Stephensoniana trivandrana	10	CG
2519	Stictochironomus caffrarius	5	
2248	Stictochironomus devinctus	5	
2070	Stictochironomus sp.	5	
1945	Stilobezzia sp.	5	
2220	Stilocladius sp.	6	
2082	Stratiomyidae	10	CG
2087	Stratiomys discalis	10	
2088	Stratiomys meigeni	10	
2086	Stratiomys sp.	10	CF
2479	Strophitus sp.	4	
2480	Strophitus undulatus	4	
932	Strophopteryx fasciata	1.5	SH
931	Strophopteryx sp.	1.5	
330	Stygobromus sp.	4	PR
331	Stygobromus subtilis	4	
84	Stylaria fossularis	10	CG
85	Stylaria lacustris	10	CG
83	Stylaria sp.	10	
736	Stylogomphus albistylus	4.5	PR
734	Stylogomphus sp.	4.5	PR
737	Stylurus sp.	7	PR
2228	Sublettea sp.	6	CF
819	Sympetrum ambiguum	4	PR
820	Sympetrum corruptum	4	
821	Sympetrum obstrusum	4	
822	Sympetrum rubicundulum	4	

Functional Feeding Group (ffg)
C=scraper, PA=parasite, PR=predator, OM=omnivore, GC=gatherer/collector, FC=filter/collector, SH=shredder, PI=piercer
9.9=Taxon excluded from mIBI computation

Appendix A. Illinois Environmental Protection Agency Macroinvertebrate Tolerance List and Functional Feeding Group Classification

January, 2011

823	Sympetrum semicinctum	4	
818	Sympetrum sp.	4	PR
824	Sympetrum vicinum	4	
1993	Sympotthastia sp.	6	CG
1557	Synclita sp.	99.9	SH
1994	Syndiamesa sp.	6	CG
2148	Syrphidae	11	CG
2089	Tabanidae	7	PR
2126	Tabanus atratus	7	
2127	Tabanus cymatophorus	7	
2128	Tabanus fairchildi	7	
2130	Tabanus lineola	7	
2131	Tabanus marginalis	7	
2132	Tabanus nigrescens	7	
2133	Tabanus pumilus	7	
2134	Tabanus quinquevittatus	7	
2135	Tabanus reinwardtii	7	
2125	Tabanus sp.	7	PR
2136	Tabanus sparus	7	
2137	Tabanus stygius	7	
2139	Tabanus subsimilis	7	
2140	Tabanus sulcifrons	7	
2141	Tabanus superjumentarius	7	
2142	Tabanus trimaculatus	7	
928	Taeniopterygidae	1.5	SH
934	Taeniopteryx nivalis	2	SH
935	Taeniopteryx parvula	2	SH
933	Taeniopteryx sp.	2	SH
2206	Tanypodinae	6	PR
2516	Tanypus carinatus	8	
2175	Tanypus neopunctipennis	8	
2508	Tanypus punctipennis	8	
1984	Tanypus sp.	8	PR
2515	Tanypus stellatus	8	
2207	Tanytarsini	6	CF
2231	Tanytarsus glabrescan	7	
2230	Tanytarsus guerlus	7	
2078	Tanytarsus sp.	7	CF
2165	Telmatoscopus sp.	11	CG
785	Tetragoneuria cynosura	4.5	PR
784	Tetragoneuria sp.	4.5	PR
1754	Thermonectus basillaris	99.9	PR
1755	Thermonectus ornaticollis	99.9	
1753	Thermonectus sp.	99.9	PR

Functional Feeding Group (ffg)
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Appendix A. Illinois Environmental Protection Agency Macroinvertebrate Tolerance List and Functional Feeding Group Classification

January, 2011

176	Theromyzon biannulatum	8	
175	Theromyzon sp.	8	PR
2016	Thienemanniella sp.	2	CG
2017	Thienemanniella xena	2	CG
2176	Thienemannimyia senata	6	
1985	Thienemannimyia sp.	6	PR
1854	Tipula sp.	4	SH
1853	Tipulidae	4	SH
641	Tortopus sp.	4	CG
2500	Toxolasma paruvus	7	
2424	Toxolasma texasensis	7	
826	Tramea carolina	4	PR
827	Tramea lacerata	4	
828	Tramea onusta	4	
825	Tramea sp.	4	PR
1063	Trepobates sp.	99.9	PR
1486	Triaenodes injustus	3	
1487	Triaenodes marginatus	3	sh
1484	Triaenodes sp.	3	MH
1488	Triaenodes tardus	3	SH
2197	Tribelos fuscicorne	4	CG
2232	Tribelos jucundus	5	
2071	Tribelos sp.	5	CG
1118	Trichocorixa calva	99.9	
1119	Trichocorixa kanza	99.9	
1120	Trichocorixa macroceps	99.9	
1117	Trichocorixa sp.	99.9	PR
1300	Trichoptera	3.5	
3	Tricladida	6	CG
599	Tricorythodes sp.	5	CG
2481	Tritogonia sp.	1	
2482	Tritogonia verrucosa	1	
1682	Tropisternus blatchleyi	99.9	
1684	Tropisternus lateralis	99.9	CG
1685	Tropisternus mixtus	99.9	
1686	Tropisternus natator	99.9	
1681	Tropisternus sp.	99.9	PR
2484	Truncilla donaciformis	1	
2483	Truncilla sp.	1	
2485	Truncilla truncata	1	
103	Tubifex sp.	10	CG
104	Tubifex tubifex	10	CG
212	Tubificida	10	
92	Tubificidae	10	CG

Functional Feeding Group (ffg)
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99.9=Taxon excluded from mIBI computation

Appendix A. Illinois Environmental Protection Agency Macroinvertebrate Tolerance List and Functional Feeding Group Classification

January, 2011

2	Turbellaria	6	PR
2213	Tvetenia sp.	5	
86	Uncinais sp.	10	
87	Uncinais uncinata	10	
2486	Uniomerus sp.	1.5	
2487	Uniomerus tetralasmus	1.5	
2401	Unionidae	1.5	CF
2600	Unionoida	99.9	
1929	Uranotaenia sapphirina	8	
1928	Uranotaenia sp.	8	CF
1756	Uvarus sp.	99.9	PR
2307	Valvata sp.	2	SC
2306	Valvatidae	6	SC
89	Vejdovskyella intermedia	10	CG
88	Vejdovskyella sp.	10	CG
1064	Veliidae	99.9	PR
2601	Veneroida	5	
2489	Villosa iris	1	
2490	Villosa lienosa	1	
2488	Villosa sp.	1	
2302	Viviparidae	6	SC
2305	Viviparus sp.	1	SC
91	Wapsa mobilis	10	
90	Wapsa sp.	10	
1348	Wormaldia shawnee	3.5	
1346	Wormaldia sp.	3.5	CF
2072	Xenochironomus sp.	4	PR
2513	Xenochironomus xenolabis	6	
2227	Xestochironomus sp.	6	
2249	Xylotopus par.	6	
2200	Zalutschia sp.	6	SH
3433	Zavreliella sp.	2	
2254	Zavreliella marmorata	2	
2235	Zavrelimyia sinuosa com	8	
1986	Zavrelimyia sp.	8	PR
829	Zygoptera	99.9	PR

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Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 177

Methods Utilized to Determine the Types and Amounts of Pertinent Macroinvertebrate Habitats in Perennial Wadeable Streams for 20-Jab Allocation

Surface Water Section

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Appendix B. Methods utilized to determine the types and amounts of pertinent macroinvertebrate habitats in perennial wadeable streams for 20-jab allocation.

Use the average stream width in the sampling reach to allocate jabs between bank-zone and bottom-zone habitats. Use Table 1 (see below) to identify all pertinent, macroinvertebrate habitats. Note that Table 1 definitions were developed exclusively for 20-jab collections and may not fulfill the requirements of other sampling methods.

Table 1 (Appendix B). 20-jab macroinvertebrate collection-zone criteria and categories plus habitat types.

Mean water width (to nearest foot)	Assumed width of bank-zone	Bank-zone jabs	Bottom-zone jabs
< 10 ft	25% of water width per bank	10	10
10-29 ft	20% of water width per bank	8	12
30-59 ft	15% of water width per bank	6	14
≥60	10% of water width per bank	4	16

Bottom-zone habitat type	Definition
- *Fine substrate*:	Streambed surface predominantly comprising silt/mud to fine gravel (i.e., particles < 8mm in diameter of intermediate dimension).
- *Coarse substrate*:	Streambed surface predominantly comprising medium gravel to boulder (i.e., particles ≥ 8 mm in diameter of intermediate dimension).
- *Plant detritus*:	Streambed surface predominantly comprising nonliving plant material (e.g., leaves, twigs).
- *Vegetation*:	Streambed surface predominantly comprising living plant material (e.g., aquatic macrophytes, filamentous algae, submerged terrestrial plants).

Bank-zone habitat type	Definition
- *Submerged terrestrial vegetation:*	Living, terrestrial plants (along stream banks) of which submerged portions provide cover or attachment sites for macroinvertebrates.
- *Submerged tree roots*:	Living tree roots (along stream banks) of which submerge portions provide cover or attachment sites for macroinvertebrates.
- *Brush-debris jams*:	Non-living, submerged, woody material (e.g., branches, twigs, smaller logs) that occurs above the streambed surface and that appears to have microbial conditioning. Exclude recent deadfall that lacks microbial conditioning.

When using the definitions in Table 1, (see above) base all classifications on conditions that exist within the sample reach at the time of the macroinvertebrate collection. If water turbidity or excessive depth prevents seeing the entire wetted stream channel throughout the sampling reach, the sampler may use tactile cues to obtain a reasonably accurate estimate of the amount of each habitat type. However, in most cases, if more than half of the wetted stream channel cannot be seen, touched, or otherwise appropriately-characterized, it is unknown that the 20-jab method will apply.

Habitat characterization methods

Two approved habitat characterization methods exist for use with the 20-jab sample approach. Review the criteria (see below) included with the 11-transect and the non-transect methods and decide which of the two habitat characterizations apply. Based on this decision, use the pre-defined habitat types (Table 1 above) and the chosen characterization method to determine the amount of pertinent macroinvertebrate habitat present in the sample reach. It is important to note the difference between 11-transect and non-transect methods. The 11-transect method establishes 11- equally spaced transects, which results in a relatively consistent number of observation points based on stream width. It should also be pointed out that observations taken along the transect do not always represent the predominant substrate at a given point. In many situations there is an intermixture of substrates types along a transect (e.g. silt, sand and fine gravel). In these circumstances an equal number of points in a transect are assigned to each of the substrate types in the intermixture (e.g. 3-silt, 3-sand, 3-fine gravel). More importantly, it is unknown how well the substrate types observed at the established points along a transect represent the relative amounts of substrate types that occur between the transects. These unknowns may be significant since the area between the transects is the majority of the sample reach. Conversely, the non-transect approach provides an estimate of the relative amounts of each substrate type throughout the sample reach. Both the 11-transect and non-transect methods rely on visual and tactile cues and both methods provide estimates that account for the intermixture of substrates as described above.

1.0 11-transect habitat characterization method

Use the 11-transect method if: the type or amount of habitat encountered during the last appraisal differs obviously from the present appraisal OR; the 11-transect habitat method has never been applied in the sample reach OR; the habitat appraisal would be made by qualified, trained personnel with fewer than two years of experience in measuring and characterizing instream physical habitat (including stream-bottom composition) for purposes of natural-resource management.

Table 2 (Appendix B). 11-transect "point-increment" habitat categories.

Substrates Types:

Name	Particle-Size Range
[1]*Silt/mud*	*< 0.062 mm*
[1]*Sand*	*0.062 – 2 mm*
[1]*Fine gravel*	*2 – 8 mm*
[2]*Medium gravel*	*8 – 16 mm*
[2]*Coarse gravel*	*16 – 64 mm*
[2]*Small cobble*	*64 – 128 mm*
[2]*Large cobble*	*128 – 256 mm*
[2]*Boulder*	*256 – 4000 mm*
Bedrock	*> 4000 mm*

Bottom Types:
Claypan/Compacted soil
[3]*Plant detritus*
[4]*Vegetation*

Instream Cover Types:
[6]*Submerged terrestrial vegetation*
[7]*Submerged tree roots*
[5]*Brush-debris jam*
Boulder (not embedded)
Undercut bank
Rock/clay ledge
[5]*Log*
[4]*Aquatic vegetation*
Other (please specify)

Point-increment observation to collection-zone translation
[1]*Fine substrate* (Bottom-zone habitat type)
[2]*Coarse substrate* (Bottom-zone habitat type)
[3]*Plant detritus* (Bottom-zone habitat type)
[4]Vegetation (Bottom-zone OR Bank-zone) (macrophytes, filamentous algae and submerged terrestrial vegetation)
[5]*Brush-debris jams* (Bank-zone habitat type)
[6]*Submerged terrestrial vegetation* (Bank-zone habitat type)
[7]*Submerged tree roots* (Bank-zone habitat type)

- When applicable, coarsely measure and estimate habitat conditions by applying the appropriate parts of the 11-transect method as described in *Section E:* of this manual. Specifically, use the point-transect approach to identify the predominant "substrate type" or "bottom type" (Table 2 in Appendix B) at each of many points distributed regularly on the wetted stream bed throughout the entire sampling reach. Also, per each of ten segments in the sampling reach, coarsely measure the area occupied by each of the nine "instream cover type"(s) (Table 2 in Appendix B). Often there is an intermixture of substrate types. In these situations an equal number of points in a transect are assigned to each of the substrate types in the intermixture (e.g. 3-silt, 3-sand, 3-fine gravel). Instream cover is measured in square feet for the 11-transect method.

Bottom-zone habitat classification based on 11-transect methodology

Refer to the assumed width of bank-zone criteria provided in Table 1 to determine the width of the bottom-zone.

Based on 11-transect habitat information, translate each of the pertinent point-increment transect observations of "*substrate*" and "*bottom type*" (Table 2) into an observation point that is classified as one of the four corresponding 20-jab "*bottom-zone"* habitat types (Table 1). Note that claypan and bedrock are not considered as a bottom-zone habitat type for applying the 20-jab method and the area of wetted stream bottom that consists of claypan should be ignored (in the denominator in Equation 1) when

considering the relative percentage of relevant bottom-zone habitat types. Explicit translation instructions are included in the Table 2 footnotes.

Per each of the four 20-jab bottom-zone habitat types, calculate the number of times a specified types occur among all of the 11-transect point-increment observations.

Use Equation 1 (see below) to calculate the relative percentage of each of the four, 20-jab, bottom-zone habitat types in the sampling reach as:

Equation 1. *Relative percentage of each bottom-zone habitat type* =

$$\frac{\textit{Sum of the points (from all transects) at which the bottom-zone habitat type occurred}}{\textit{Sum of the points (from all transects) at which any of the four bottom-zone habitat types occurred}} \times 100$$

Record the relative percentage result for each, 20-jab, bottom-zone habitat type. Note: claypan and bedrock are not included in bottom zone allocations (Table 2).

Bank-zone habitat classification based on 11-transect methodology

Refer to the assumed width of bank-zone criteria provided in Table 1 to determine the width of the bank-zone collection area.

Submerged terrestrial vegetation

- With the 11-transect habitat characterization method, the spatial coverage of submerged terrestrial vegetation is determined for both banks within each of the ten stream segments delineated by the eleven transects. In each of the ten segments, use a known distance such as the length of a wading rod or the distance between transects, to coarsely measure the total length (ft) of *submerged terrestrial vegetation* present (Table 2). Sum the ten segment totals and record this grand total as the amount of *submerged terrestrial vegetation* that occurs along the banks of the sample reach. The linear amount (ft) of submerged terrestrial vegetation that occurs along the banks of the sample reach- as determined by the 11-transect method- is considered equivalent to the amount of submerged terrestrial vegetation that occurs in the assumed bank-zone of the 20-jab collection area.

Submerged tree roots

- With the 11-transect habitat characterization method, the spatial coverage of submerged tree roots is determined for both banks within each of the ten stream segments delineated by the eleven transects. In each of the ten segments, use known distance such as the length of a wading rod or the distance between transects, to coarsely measure the total length (ft) of *submerged tree roots* present (Table 2). Sum the ten segment totals and record this grand total as the amount of *submerged tree roots* that occurs along the banks of the sample reach. The linear amount (ft) of submerged tree roots that occurs along the banks of the sample reach- as determined by the 11-transect method- is considered equivalent to the amount of submerged tree roots that occurs in the assumed bank-zone of the 20-jab collection area.

Brush-debris jams

- The amount of *log* plus *brush-debris jam* habitat is calculated in a slightly different manner than the previous two bank-zone habitats. Here, consider each *log* plus *brush-debris jams* as bank-zone habitat, regardless of where the woody materials are located in the sample reach—

provided that the woody material occurs at a depth and water velocity that allow safe and sufficient sampling of macroinvertebrates with a dip net. Coarsely measure the single longest dimension covered by each log and brush-debris jam. Sum these lengths to yield the total length of *logs* plus *brush-debris jams* within each of the ten stream segments. In turn, sum the ten segment totals and record the grand total length of *log* plus *brush-debris jams* that occurs in the sample reach.

Use Equation 2 (see below) to calculate the relative percentage of each bank-zone habitat type in the sampling reach as

Equation 2. Relative percentage of each bank-zone habitat type =

Total length that a given bank-zone habitat type occurred in the sample reach	÷	*Sum of total length for each of the three bank-zone habitat types that occur in the sample reach*	*x 100*

Record the relative percentage of each bank-zone habitat type.

2.0 Non-Transect habitat characterization method

For qualified, trained personnel having two or more years of experience in measuring and characterizing instream physical habitat (including stream-bottom composition) for purposes of natural-resource management, use either the 11-transect (see above) or the non-transect approach to characterize macroinvertebrate habitat within the sample reach.

Bottom-zone habitat classification based on non-transect estimations

- Estimate and record the percent surface area of the relevant portion of wetted stream bottom that consists of each of the four bottom-zone habitat types (Table 1). Note that claypan and bedrock are not considered as a bottom-zone habitat type for applying the 20-jab method and the area of wetted stream bottom that consists of claypan should be ignored (in the denominator in Equation 1) when considering the relative percentage of relevant bottom-zone habitat types.

Bank-zone habitat classification based on non-transect estimations

- Estimate and record the percentage of bank length occupied by each of the three bank-zone habitat types. Estimate and record *logs* plus *submerged terrestrial vegetation* (Table 1 in Appendix B) and *submerged tree roots* (Table 1 in Appendix B) as the length of bank-zone covered by each habitat type in the sampling reach. When estimating the amount of *logs* plus *brush-debris jams (*Table 1*)* in the sampling reach, consider all logs and brush-debris jams as bank-zone habitat, regardless of where the snag occurs in the sample reach—provided that the log and brush-debris jam occurs at a depth and water velocity that allow safe and sufficient sampling of macroinvertebrates with a dip-net. Estimate the single longest dimension covered by each log and brush-debris jam and then sum these lengths to yield the total length of *logs* plus *Brush-debris jams* in the sample reach. The percentage of bank length is from IEPA STREAM ASSESSMENT FORM-Non-Transect Habitat Information.

Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 178

Genus-List: Macroinvertebrate-Index of Biotic Integrity (m-IBI) Tolerance List and Functional Feeding Group Classification

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Appendix C. Genus-List: Illinois Environmental Protection Agency Macroinvertebrate-Index of Biotic Integrity (m-IBI) Tolerance List and Functional Feeding Group Classification

January, 2011

Phylogenetic Order

Major Group	Bios ID	Taxon	Tolerance	Functional Feeding Group
Phylum Platyhelminthes	1	Platyhelminthes	99.9	
Class Turbellaria	2	Turbellaria	6	PR
Phylum Nematomorpha	15	Nematomorpha	99.9	PA
Phylum Annelida	30	Annelida	99.9	CG
Class Oligochaeta	31	Oligochaeta	10	CG
Class Hirudinea	249	Hirudinea	8	PR
Phylum Arthropoda	250	Arthropoda	99.9	
Class Crustacea	251	Crustacea	99.9	CG
Order Isopoda	252	Isopoda	99.9	CG
Family Asellidae	253	Asellidae	6	CG
	254	Caecidotea sp.	6	CG
	255	Asellus sp.	6	CG
	272	Lirceus sp.	4	CG
Order Amphipoda	325	Amphipoda	4	CG
Family Talitridae	326	Hyalellidae	4	
	327	Hyalella sp.	4	CG
Family Gammaridae	329	Gammaridae	4	CG
	341	Gammarus sp.	3	CG
	330	Stygobromus sp.	4	PR
	332	Bactrurus sp.	1	
Family Crongonyctidae	348	Crongonyctidae	4	
	335	Crangonyx sp.	4	CG
Order Decapoda	400	Decapoda	99.9	SH
Family Cambaridae	401	Cambaridae	5	CG
Family Palaemonidae	427	Palaemonidae	4	
	428	Palaemonetes sp.	4	
Class Insecta	475	Insecta	99.9	
Order Ephemeroptera	476	Ephemeroptera	3	CG
Family Acanthametropodidae	2605	Acanthametropodidae		
	478	Acanthametropus sp.	3	PR
Family Ameletidae	2604	Ameletidae		
	480	Ameletus sp.	0	CG
Family Siphlonuridae	477	Siphlonuridae	3	CG
	482	Siphlonurus sp.	2	CG
Family Oligoneuriidae	486	Oligoneuriidae	3	CF
Family Isonychiidae	487	Isonychia sp.	3	CF
Family Metretopodidae	493	Metretopodidae	3	
	494	Siphloplecton sp.	2	CG

Functional Feeding Group (ffg)
SC=scraper, PA=parasite, PR=predator, OM=omnivore, GC=gatherer/collector, FC=filter/collector, SH=shredder, PI=piercer
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Appendix C. Genus-List: Illinois Environmental Protection Agency Macroinvertebrate-Index of Biotic Integrity (m-IBI) Tolerance List and Functional Feeding Group Classification

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Family Baetidae	497	Baetidae	4	CG
	652	Acentrella sp.	4	
	2203	Acerpenna sp.	4	SH
	498	Baetis sp.	4	CG
	511	Callibaetis sp.	4	CG
	515	Centroptilum sp.	2	CG
	663	Diphetor sp.	4	
	643	Procloeon sp.	4	
	519	Heterocloeon sp.	4	SC
	651	Plauditus sp.	3	
	521	Pseudocloeon sp.	4	SC
	527	Paracloeodes sp.	4	SC
Family Arthropleidae	2603	Arthropleidae	3	CF
	531	Arthroplea sp.	3	CF
Family Heptageniidae	528	Heptageniidae	3.5	SC
	529	Anepeorus sp.	3.5	PR
	533	Epeorus sp.	1	SC
	644	Nixe sp.	4	SC
	536	Heptagenia sp.	3	SC
	544	Rhithrogena sp.	0	SC
	550	Stenacron sp.	4	SC
	648	Leucrocuta sp.	3	SC
	551	Maccaffertium sp.	4	SC
	659	Stenonema sp.	4	SC
Family Ephemerellidae	567	Ephemerellidae	3.5	CG
	568	Attenella sp.	2	CG
	570	Dannella sp.	2	CG
	573	Drunella sp.	1	PR
	578	Ephemerella sp.	2	CG
	587	Eurylophella sp.	4	SC
	594	Serratella sp.	1	CG
Family Leptohyphidae	598	Leptohyphidae	5.5	CG
	662	Leptohyphe sp.	5.5	CG
	599	Tricorythodes sp.	5	CG
Family Caenidae	600	Caenidae	5.5	CG
	601	Brachycercus sp.	3	CG
	602	Caenis sp.	6	CG
Family Baetiscidae	603	Baetiscidae	3	CG
	605	Baetisca sp.	3	CG
Family Leptophlebiidae	609	Leptophlebiidae	3	CG
	610	Choroterpes sp.	2	CG
	612	Habrophlebiodes sp.	2	SC

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	614	Leptophlebia sp.	3	CG
	615	Paraleptophlebia sp.	2	CG
Family Potamanthidae	620	Potamanthidae	5	CF
	621	Anthopotamus sp.	4	
Family Ephemeridae	625	Ephemeridae	5	CG
	626	Ephemera sp.	3	CG
	628	Hexagenia sp.	6	CG
Family Palingeniidae	635	Pentagenia sp.	4	CF
Family Polymitarcyidae	637	Polymitarcyidae	3	CG
	638	Ephoron sp.	2	CG
	641	Tortopus sp.	4	CG
Order Odonata	700	Odonata	99.9	PR
Family Cordulegastridae	702	Cordulegastridae	4.5	PR
	703	Cordulegaster sp.	2	PR
Family Gomphidae	706	Gomphidae	4.5	PR
	2602	Erpetogomphus sp.	2	
	713	Dromogomphus sp.	4	PR
	707	Arigomphus sp.	7	PR
	722	Gomphus sp.	7	PR
	737	Stylurus sp.	7	PR
	728	Hagenius sp.	3	PR
	735	Lanthus sp.	6	PR
	730	Ophiogomphus sp.	2	PR
	732	Progomphus sp.	5	PR
	734	Stylogomphus sp.	4.5	PR
Family Aeshnidae	742	Aeshnidae	4.5	PR
	743	Aeshna sp.	4	PR
	749	Anax sp.	5	PR
	751	Basiaeschna sp.	2	PR
	753	Boyeria sp.	3	PR
	755	Epiaeschna sp.	1	PR
	758	Nasiaeschna sp.	2	PR
Family Macromiidae	759	Macromiidae	4.5	PR
	760	Didymops sp.	4	PR
	762	Macromia sp.	3	PR
Family Corduliidae	768	Corduliidae	4.5	PR
	769	Cordulia sp.	2	PR
	771	Epitheca sp.	4	PR
	772	Epicordulia sp.	4.5	PR
	774	Helocordulia sp.	2	PR
	775	Neurocordulia sp.	3	PR
	779	Somatochlora sp.	1	PR

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	784	Tetragoneuria sp.	4.5	PR
Family Libellulidae	787	Libellulidae	4.5	PR
	788	Celithemis sp.	2	PR
	792	Erythemis sp.	5	PR
	794	Erythrodiplax sp.	5	PR
	795	Ladona sp.	4.5	PR
	797	Leucorrhinia sp.	4.5	PR
	800	Libellula sp.	8	PR
	809	Pachydiplax sp.	8	PR
	811	Pantala sp.	7	PR
	814	Perithemis sp.	4	PR
	816	Plathemis sp.	3	PR
	818	Sympetrum sp.	4	PR
	825	Tramea sp.	4	PR
	829	Zygoptera	99.9	PR
Family Calopterygidae	830	Calopterygidae	3.5	PR
	831	Calopteryx sp.	4	PR
	834	Hetaerina sp.	3	PR
Family Lestidae	837	Lestidae	99.9	PR
	838	Archilestes sp.	1	PR
	840	Lestes sp.	6	PR
Family Coenagrionidae	847	Coenagrionidae	5.5	PR
	848	Amphiagrion sp.	5	PR
	852	Argia sp.	5	PR
	860	Chromagrion sp.	5.5	PR
	862	Coenagrion sp.	5.5	PR
	863	Enallagma sp.	6	PR
	873	Ischnura sp.	6	PR
	876	Nehalennia sp.	7	PR
Order Plecoptera	925	Plecoptera	1.5	PR
Family Pteronarcyidae	927	Pteronarcys sp.	2	SH
Family Taeniopterygidae	928	Taeniopterygidae	1.5	SH
	929	Oemopteryx sp.	1.5	SH
	931	Strophopteryx sp.	1.5	SH
	933	Taeniopteryx sp.	2	SH
Family Nemouridae	936	Nemouridae	1.5	SH
	937	Amphinemura sp.	1.5	SH
	938	Nemoura sp.	1	SH
	940	Prostoia sp.	1.5	SH
	941	Soyedina sp.	1.5	SH
Family Leuctridae	942	Leuctridae	1.5	SH
	943	Leuctra sp.	1	SH

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Family Capniidae	944	Capniidae	1.5	SH
	945	Allocapnia sp.	2	SH
	949	Capnia sp.	1	SH
	951	Paracapnia sp.	1.5	SH
Family Perlidae	954	Perlidae	1.5	PR
	955	Acroneuria sp.	1	PR
	962	Attaneuria sp.	1.5	PR
	964	Neoperla sp.	1	PR
	966	Paragnetina sp.	1.5	PR
	968	Perlesta sp.	4	PR
	971	Atoperla sp.	1	
	970	Perlinella sp.	2	PR
	974	Phasganophora sp.	1.5	PR
Family Perlodidae	976	Perlodidae	1.5	PR
	977	Hydroperla sp.	1	PR
	979	Isogenoides sp.	1.5	PR
	980	Isoperla sp.	2	PR
Family Chloroperlidae	989	Chloroperlidae	1.5	PR
	990	Chloroperla sp.	3	
	991	Alloperla sp.	1.5	PR
	992	Hastaperla sp.	1.5	SC
	994	Rasvena sp.	1.5	CG
Order Hemiptera	1050	Hemiptera	99.9	PR
Order Megaloptera	1175	Megaloptera	3.5	
Family Sialidae	1176	Sialidae	3.5	PR
	1177	Sialis sp.	4	PR
Family Corydalidae	1186	Corydalidae	3	PR
	1187	Chauliodes sp.	4	PR
	1190	Corydalus sp.	3	PR
	1192	Nigronia sp.	2	PR
Order Neuroptera	1250	Neuroptera	99.9	PR
Family Sisyridae	1251	Sisyridae	1	PR
	1252	Climacea sp.	1	
	1254	Sisyra sp.	1	PR
Order Trichoptera	1300	Trichoptera	3.5	
Family Hydropsychidae	1301	Hydropsychidae	5.5	CF
	1330	Ceratopsyche sp.	4	CF
	1302	Cheumatopsyche sp.	6	CF
	1304	Diplectrona sp.	2	CF
	1306	Hydropsyche sp.	5	CF
	1324	Macronema sp.	2	CF
	2217	Macrostemum sp.	2	CF

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	1326	Parapsyche sp.	5.5	PR
	1328	Potamyia sp.	4	CF
Family Philopotamidae	1338	Philopotamidae	3.5	CF
	1339	Chimarra sp.	3	CF
	1344	Dolophilodes sp.	0	CG
	1346	Wormaldia sp.	3.5	CF
Family Polycentropodidae	1349	Polycentropodidae	3.5	CF
	1350	Cyrnellus sp.	5	CF
	1352	Neureclipsis sp.	3	CF
	1355	Nyctiophylax sp.	1	CF
	1356	Phylocentropus sp.	3.5	CF
	1358	Polycentropus sp.	3	PR
Family Psychomyiidae	1365	Psychomyiidae	3.5	CG
	1366	Lype sp.	3.5	SC
	1368	Psychomyia sp.	2	SC
Family Glossosomatidae	1370	Glossosomatidae	3.5	SC
	1372	Agapetus sp.	2	SC
	1373	Glossosoma sp.	3.5	SC
	1375	Protoptila sp.	1	SC
Family Hydroptilidae	1376	Hydroptilidae	3.5	PH
	1377	Agraylea sp.	2	PH
	1379	Hydroptila sp.	2	SC
	1380	Ithytrichia sp.	1	SC
	1381	Leucotrichia sp.	3	SC
	1384	Mayatrichia sp.	1	SC
	1385	Neotrichia sp.	4	SC
	1386	Ochrotrichia sp.	4	CG
	1387	Orthotrichia sp.	1	SC
	1388	Oxyethira sp.	2	MH
	1389	Stactobiella sp.	3.5	SH
Family Rhyacophilidae	1391	Rhyacophilidae	3.5	PR
	1392	Rhyacophila sp.	1	PR
Family Brachycentridae	1397	Brachycentridae	3.5	CF
	1398	Brachycentrus sp.	1	CF
	1403	Micrasema sp.	3.5	MH
Family Lepidostomatidae	1405	Lepidostomatidae	3.5	SH
	1406	Lepidostoma sp.	3	SH
Family Limnephilidae	1408	Limnephilidae	3.5	SH
	1409	Anabolia sp.	3.5	SH
	1410	Frenesia sp.	3.5	SH
	1412	Goera sp.	3.5	SC
	1413	Hesperophylax sp.	3.5	SH

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	1415	Hydatophylax sp.	2	SH
	1417	Ironoquia sp.	3.5	SH
	1418	Leptophylax sp.	3.5	SH
	1420	Limnephilus sp.	3	SH
	1421	Neophylax sp.	3	SC
	1423	Platycentropus sp.	3	SH
	1425	Pseudostenophylax sp.	3.5	SH
	1427	Pycnopsyche sp.	3	SH
Family Molannidae	1433	Molannidae	3.5	CG
	1434	Molanna sp.	3.5	SC
Family Phryganeidae	1438	Phryganeidae	3.5	SH
	1439	Agrypnia sp.	3	SH
	1442	Banksiola sp.	2	SH
	1444	Fabria sp.	3.5	SH
	1446	Oligostomis sp.	3.5	PR
	1448	Phryganea sp.	3	
	1449	Ptilostomis sp.	3	SH
Family Helicopsychidae	1450	Helicopsychidae	3.5	SC
	1451	Helicopsyche sp.	2	SC
Family Leptoceridae	1453	Leptoceridae	3.5	CG
	1454	Ceraclea sp.	3	CG
	1466	Leptocerus sp.	3	SH
	1468	Mystacides sp.	2	CG
	1471	Nectopsyche sp.	3	SH
	1477	Oecetis sp.	5	PR
	1483	Setodes sp.	3.5	
	1484	Triaenodes sp.	3	SH
Family Sericostomatidae	1489	Sericostomatidae	3.5	SH
	1502	Agarodes sp.	3.5	
Order Lepidoptera	1550	Lepidoptera	99.9	SH
Family Pyralidae	1551	Pyralidae	99.9	SH
	1556	Petrophila sp.	5	SC
Order Coleoptera	1600	Coleoptera	99.9	PR
Family Gyrinidae	1601	Gyrinidae	99.9	PR
	1602	Dineutus sp. (larvae only)	4	PR
	1607	Gyrinus sp. (larvae only)	4	PR
Family Psephenidae	1614	Psephenidae	4	SC
	1760	Dicranopselaphus sp.	4	SC
	1761	Ectopria sp.	4	SC
	1616	Psephenus sp.	4	SC
Family Scirtidae	1617	Helodidae	7	SC
	1618	Cyphon sp.	7	SC

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	1626	Scirtes sp.	7	SH
Family Dryopidae	1764	Dryopidae	4	SH
	1765	Helichus sp.	4	SH
	1769	Pelonomus sp.	4	CG
Family Elmidae	1771	Elmidae	5	CG
	1772	Ancyronyx sp.	2	CG
	1774	Dubiraphia sp.	5	CG
	1779	Macronychus sp.	2	
	1781	Microcylloepus sp.	2	CG
	1783	Optioservus sp.	4	SC
	1787	Promoresia sp.	5	SC
	1788	Stenelmis sp.	7	SC
Family Scirtidae	1801	Elodes sp.	7	
	1802	Prionocyphon sp.	7	SC
Order Diptera	1850	Diptera	10	
Family Blephariceridae	1851	Blephariceridae	0	SC
	1852	Blepharicera sp.	0	SC
Family Tipulidae	1853	Tipulidae	4	SH
	1854	Tipula sp.	4	SH
	1855	Antocha sp.	5	CG
	1856	Dicranota sp.	4	PR
	1857	Eriocera sp.	7	PR
	1858	Erioptera sp.	4	CG
	2221	Gonomyia sp.	4	CG
	1859	Helius sp.	5	CG
	1860	Hesperoconopa sp.	2	CG
	1861	Hexatoma sp.	4	PR
	1862	Limnophila sp.	4	PR
	1863	Limonia sp.	3	SH
	1864	Ormosia sp.	4	CG
	1865	Pedicia sp.	4	PR
	1866	Pilaria sp.	4	PR
	1867	Prionocera sp.	4	SH
	1868	Pseudolimnophila sp.	2	PR
Family Chaoboridae	1869	Chaoboridae	8	PR
	1870	Chaoborus sp.	8	PR
	1871	Corethrella sp.	8	PR
Family Culicidae	1873	Culicidae	8	CG
	1875	Aedes sp.	8	CF
	1897	Anopheles sp.	6	CF
	1904	Culex sp.	8	CF
	1912	Culiseta sp.	8	CG

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	1917	Mansonia sp.	8	CG
	1919	Psorophora sp.	8	PR
	1928	Uranotaenia sp.	8	CF
Family Dixidae	1930	Dixidae	10	CG
	1931	Dixa sp.	10	CG
	2238	Dixella sp.	10	
Family Psychodidae	1933	Psychodidae	11	CG
	2165	Telmatoscopus sp.	11	CG
	1934	Pericoma sp.	11	CG
	1935	Psychoda sp.	11	CG
Family Ceratopogonidae	1936	Ceratopogonidae	5	PR
	1937	Atrichopogon sp.	2	PR
	1938	Bezzia sp.	5	CG
	2166	Ceratopogon sp.	5	PR
	1939	Culicoides sp.	5	PR
	1940	Dasyhelea sp.	5	CG
	1941	Forcipomyia sp.	5	SC
	2167	Monohelea sp.	5	PR
	2223	Nilobezzia sp.	5	PR
	1942	Palpomyia sp.	6	PR
	2596	Serromyia sp.	5	
	1943	Probezzia sp.	5	PR
	2224	Sphaeromias sp.	5	
	1945	Stilobezzia sp.	5	
Family Simuliidae	1946	Simuliidae	6	CF
	1947	Cnephia sp.	4	CF
	1949	Prosimulium sp.	2	CF
	1952	Simulium sp.	6	CF
Family Chironomidae	1963	Chironomidae	6	CG
Tribe Tanypodinae	2206	Tanypodinae	6	PR
	1965	Ablabesmyia sp.	6	CG
	1970	Clinotanypus sp.	6	PR
	1973	Coelotanypus sp.	4	PR
	2171	Conchapelopia sp.	6	PR
	1974	Djalmabatista sp.	6	PR
	1975	Guttipelopia sp.	6	PR
	2214	Hayesomyia sp.	5	
	2215	Helopelopia sp.	4	PR
	2509	Hudsonimyia sp.	6	
	1976	Labrundinia sp.	4	PR
	1977	Larsia sp.	6	PR
	1978	Macropelopia sp.	7	PR

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	2211	Meropelopia sp.	3	
	1979	Natarsia sp.	6	PR
	1980	Nilotanypus sp.	6	PR
	2173	Paramerina sp.	6	PR
	1981	Pentaneura sp.	3	PR
	1982	Procladius sp.	8	PR
	1983	Psectrotanypus sp.	8	PR
	2225	Rheopelopia sp.	3	PR
	1984	Tanypus sp.	8	PR
	1985	Thienemannimyia sp.	6	PR
Tribe Diamesinae	2210	Diamesinae	6	
	1988	Diamesa sp.	4	CG
	1989	Odontomesa sp.	6	CG
	1990	Potthastia sp.	6	
	1991	Prodiamesa sp.	3	CG
	1992	Pseudodiamesa sp.	1	CG
	1993	Sympotthastia sp.	6	CG
	1994	Syndiamesa sp.	6	CG
Tribe Orthocladiinae	1995	Orthocladiinae	6	CG
	2202	Orthocladius sp./Cricotopus sp.	6	
	1996	Brillia sp.	6	SH
	1997	Cardiocladius sp.	6	PR
	1998	Chaetocladius sp.	6	CG
	1999	Corynoneura sp.	2	CG
	2209	Epoicocladius sp.	6	CG
	2005	Eukiefferiella sp.	4	CG
	2006	Heterotrissocladius sp.	6	CG
	2007	Hydrobaenus sp.	2	SC
	2250	Gymnometriocnemus sp.	6	
	2578	Lopescladius sp.	4	
	2008	Metriocnemus sp.	6	CG
	2009	Nanocladius sp.	3	CG
	2216	Parakiefferiella sp.	5	
	2011	Parametriocnemus sp.	4	CG
	2179	Paraphaenocladius sp.	6	CG
	2012	Psectrocladius sp.	5	CG
	2597	Psilometriocnemus sp.	6	
	2013	Pseudorthocladius sp.	6	CG
	2014	Rheocricotopus sp.	6	CG
	2015	Smittia sp.	6	CG
	2220	Stilocladius sp.	6	
	2213	Tvetenia sp.	5	

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	2016	Thienemanniella sp.	2	CG
	2200	Zalutschia sp.	6	SH
Tribe Chironomini	2205	Chironomini	6	CG
	2201	Axarus sp.	6	CG
	2019	Chironomus sp.	11	CG
	2025	Cladopelma sp.	6	CG
	2026	Cryptochironomus sp.	8	PR
	2029	Cryptotendipes sp.	6	CG
	2030	Demicryptochironomus sp.	6	CG
	2031	Dicrotendipes sp.	6	CG
	2035	Einfeldia sp.	10	CG
	2036	Endochironomus sp.	6	SH
	2039	Glyptotendipes sp.	10	CF
	2042	Harnischia sp.	6	CG
	2255	Hyporhygma sp.	4	
	2043	Kiefferulus sp.	7	CG
	2518	Lipiniella sp.	6	
	2044	Microchironomus sp.	6	CG
	2045	Microtendipes sp.	6	CF
	2208	Nilothauma sp.	3	
	2048	Parachironomus sp.	8	PR
	2050	Paracladopelma sp.	4	CG
	2053	Paratendipes sp.	3	CG
	2251	Pagastiella sp.	6	
	2054	Phaenopsectra sp.	4	SC
	2057	Polypedilum sp.	6	SH
	2066	Pseudochironomus sp.	5	CG
	2218	Robackia sp.	3	CG
	2067	Saetheria sp.	6	CG
	2068	Stenochironomus sp.	3	SH
	2219	Stelechomyia sp.	6	CG
	2070	Stictochironomus sp.	5	
	2071	Tribelos sp.	5	CG
	2072	Xenochironomus sp.	4	PR
	2227	Xestochironomus sp.	6	
	2249	Xylotopus par.	6	
	1986	Zavrelimyia sp.	8	PR
Tribe Tanytarsini	2207	Tanytarsini	6	CF
	2074	Cladotanytarsus sp.	7	CG
	2075	Micropsectra sp.	4	CG
	2076	Paratanytarsus sp.	6	CG
	2077	Rheotanytarsus sp.	6	CF

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	2226	Stempellina sp.	2	CG
	2212	Stempellinella sp.	2	CG
	2228	Sublettea sp.	6	CF
	2078	Tanytarsus sp.	7	CF
Family Ptychopteridae	2079	Ptychopteridae	8	CG
	2080	Bittacomorpha sp.	8	CG
	2081	Ptychoptera sp.	8	CG
Family Stratiomyidae	2082	Stratiomyidae	10	CG
	2092	Allognosta sp.	10	CG
	2083	Odostomia sp.	10	CG
	2085	Oxycera sp.	10	SC
	2086	Stratiomys sp.	10	CF
Family Tabanidae	2089	Tabanidae	7	PR
	2090	Atylotus sp.	7	PR
	2143	Chlorotabanus sp.	7	
	2093	Chrysops sp.	7	CG
	2119	Hybomitra sp.	7	PR
	2125	Tabanus sp.	7	PR
Family Dolichopodidae	2144	Dolichopodidae	5	PR
Family Empididae	2146	Empididae	6	PR
	2147	Hemerodromia sp.	6	PR
	2595	Rhamphomyia sp.	1.0	
Family Syrphidae	2148	Syrphidae	11	CG
	2149	Chrysogaster sp.	11	CG
	2150	Eristalis sp.	11	CG
Family Ephydridae	2151	Ephydridae	8	CG
	2152	Ephydra sp.	8	CG
Family Sciomyzidae	2153	Sciomyzidae	10	PR
	2154	Dictya sp.	10	PR
Family Muscidae	2156	Muscidae	8	PR
	2157	Limnophora sp.	8	PR
Family Athericidae	2158	Athericidae	10	
	2159	Atherix sp.	10	PR
Phylum Mollusca	2300	Mollusca	99.9	
Class Gastropoda	2301	Gastropoda	99.9	SC
Order Mesogastropoda	2599	Mesogastropoda	99.9	
Family Viviparidae	2302	Viviparidae	6	SC
	2303	Campeloma sp.	7	SC
	2304	Lioplax sp.	7	SC
	2305	Viviparus sp.	1	SC
Family Valvatidae	2306	Valvatidae	6	SC
	2307	Valvata sp.	2	SC

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Family Bithyniidae	2308	Bithyniidae	6	
	2309	Bithynia sp.	6	
Family Hydrobiidae	2310	Hydrobiidae	6	SC
	2312	Amnicola sp.	4	SC
	2314	Cincinnatia sp.	6	SC
	2315	Marstonia sp.	6	
	2316	Probythinella sp.	6	
	2317	Pyrgulopsis sp.	6	SC
	2318	Somatogyrus sp.	6	
Family Pleuroceridae	2319	Pleuroceridae	6	
	2320	Elimia sp.	6	SC
	2321	Goniobasis sp.	5	SC
	2322	Leptoxis sp.	6	
	2323	Lithasia sp.	6	
	2324	Pleurocera sp.	7	SC
Family Pomatiopsidae	2326	Pomatiopsidae	6	
	2327	Pomatiopsis sp.	6	
Family Physidae	2328	Physidae	9	SC
	2329	Aplexa sp.	7	
	2330	Physa sp.	9	SC
	2331	Physella sp.	9	SC
Family Lymnaeidae	2332	Lymnaeidae	7	SC
	2333	Acella sp.	7	
Order Basommatophora	2598	Basommatophora	7	
	2334	Fossaria sp.	7	SC
	2335	Lymnaea sp.	7	SC
	2336	Pseudosuccinea sp.	7	SC
	2337	Stagnicola sp.	7	SC
Family Planorbidae	2339	Planorbidae	6.5	SC
	2340	Gyraulus sp.	6	SC
	2341	Helisoma sp.	7	SC
	2342	Menetus sp.	6.5	SC
	2343	Planorbella sp.	6.5	SC
	2344	Planorbula sp.	7	SC
	2345	Promenetus sp.	6.5	CG
Family Ancylidae	2346	Ancylidae	7	SC
	2347	Ferrissia sp.	7	SC
	2348	Laevapex sp.	6	SC
Class Pelecypoda	2400	Pelecypoda	99.9	CF
Order Unionoida	2600	Unionoida	99.9	
Family Unionidae	2401	Unionidae	1.5	CF
Order Venerioda	2601	Veneroida	5	

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Family Sphaeriidae	2491	Sphaeriidae/Pisidiidae	5	
Family Corbiculidae	2497	Corbicula sp.	4	CF
Family Dreissenidae	2234	Dreissena sp.	99.9	

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Alphabetic Order

Major Group	Bios ID	Taxon	Tolerance	Functional Feeding Group
	1965	Ablabesmyia sp.	6	CG
Family Acanthametropodidae	2605	Acanthametropodidae		
	478	Acanthametropus sp.	3	PR
	2333	Acella sp.	7	
	652	Acentrella sp.	4	
	2203	Acerpenna sp.	4	SH
	955	Acroneuria sp.	1	PR
	1875	Aedes sp.	8	CF
	743	Aeshna sp.	4	PR
Family Aeshnidae	742	Aeshnidae	4.5	PR
	1372	Agapetus sp.	2	SC
	1502	Agarodes sp.	3.5	
	1377	Agraylea sp.	2	PH
	1439	Agrypnia sp.	3	SH
	945	Allocapnia sp.	2	SH
	2092	Allognosta sp.	10	CG
	991	Alloperla sp.	1.5	PR
Family Ameletidae	2604	Ameletidae		
	480	Ameletus sp.	0	CG
	2312	Amnicola sp.	4	SC
	848	Amphiagrion sp.	5	PR
	937	Amphinemura sp.	1.5	SH
Order Amphipoda	325	Amphipoda	4	CG
	1409	Anabolia sp.	3.5	SH
	749	Anax sp.	5	PR
Family Ancylidae	2346	Ancylidae	7	SC
	1772	Ancyronyx sp.	2	CG
	529	Anepeorus sp.	3.5	PR
Phylum Annelida	30	Annelida	99.9	CG
	1897	Anopheles sp.	6	CF
	621	Anthopotamus sp.	4	

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	1855	Antocha sp.	5	CG
	2329	Aplexa sp.	7	
	838	Archilestes sp.	1	PR
	852	Argia sp.	5	PR
	707	Arigomphus sp.	7	PR
	531	Arthroplea sp.	3	CF
Family Arthropleidae	2603	Arthropleidae	3	CF
Phylum Arthropoda	250	Arthropoda	99.9	
Family Asellidae	253	Asellidae	6	CG
	255	Asellus sp.	6	CG
Family Athericidae	2158	Athericidae	10	
	2159	Atherix sp.	10	PR
	971	Atoperla sp.	1	
	1937	Atrichopogon sp.	2	PR
	962	Attaneuria sp.	1.5	PR
	568	Attenella sp.	2	CG
	2090	Atylotus sp.	7	PR
	2201	Axarus sp.	6	CG
	332	Bactrurus sp.	1	
Family Baetidae	497	Baetidae	4	CG
	498	Baetis sp.	4	CG
	605	Baetisca sp.	3	CG
Family Baetiscidae	603	Baetiscidae	3	CG
	1442	Banksiola sp.	2	SH
	751	Basiaeschna sp.	2	PR
Order Basommatophora	2598	Basommatophora	7	
	1938	Bezzia sp.	5	CG
	2309	Bithynia sp.	6	
Family Bithyniidae	2308	Bithyniidae	6	
	2080	Bittacomorpha sp.	8	CG
	1852	Blepharicera sp.	0	SC
Family Blephariceridae	1851	Blephariceridae	0	SC
	753	Boyeria sp.	3	PR
Family Brachycentridae	1397	Brachycentridae	3.5	CF
	1398	Brachycentrus sp.	1	CF

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	601	Brachycercus sp.	3	CG
	1996	Brillia sp.	6	SH
	254	Caecidotea sp.	6	CG
Family Caenidae	600	Caenidae	5.5	CG
	602	Caenis sp.	6	CG
	511	Callibaetis sp.	4	CG
Family Calopterygidae	830	Calopterygidae	3.5	PR
	831	Calopteryx sp.	4	PR
Family Cambaridae	401	Cambaridae	5	CG
	2303	Campeloma sp.	7	SC
	949	Capnia sp.	1	SH
Family Capniidae	944	Capniidae	1.5	SH
	1997	Cardiocladius sp.	6	PR
	788	Celithemis sp.	2	PR
	515	Centroptilum sp.	2	CG
	1454	Ceraclea sp.	3	CG
	2166	Ceratopogon sp.	5	PR
Family Ceratopogonidae	1936	Ceratopogonidae	5	PR
	1330	Ceratopsyche sp.	4	CF
	1998	Chaetocladius sp.	6	CG
Family Chaoboridae	1869	Chaoboridae	8	PR
	1870	Chaoborus sp.	8	PR
	1187	Chauliodes sp.	4	PR
	1302	Cheumatopsyche sp.	6	CF
	1339	Chimarra sp.	3	CF
Family Chironomidae	1963	Chironomidae	6	CG
Tribe Chironomini	2205	Chironomini	6	CG
	2019	Chironomus sp.	11	CG
	990	Chloroperla sp.	3	
Family Chloroperlidae	989	Chloroperlidae	1.5	PR
	2143	Chlorotabanus sp.	7	
	610	Choroterpes sp.	2	CG
	860	Chromagrion sp.	5.5	PR
	2149	Chrysogaster sp.	11	CG
	2093	Chrysops sp.	7	CG

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	2314	Cincinnatia sp.	6	SC
	2025	Cladopelma sp.	6	CG
	2074	Cladotanytarsus sp.	7	CG
	1252	Climacea sp.	1	
	1970	Clinotanypus sp.	6	PR
	1947	Cnephia sp.	4	CF
	1973	Coelotanypus sp.	4	PR
	862	Coenagrion sp.	5.5	PR
Family Coenagrionidae	847	Coenagrionidae	5.5	PR
Order Coleoptera	1600	Coleoptera	99.9	PR
	2171	Conchapelopia sp.	6	PR
Family Corbiculidae	2497	Corbicula sp.	4	CF
	703	Cordulegaster sp.	2	PR
Family Cordulegastridae	702	Cordulegastridae	4.5	PR
	769	Cordulia sp.	2	PR
Family Corduliidae	768	Corduliidae	4.5	PR
	1871	Corethrella sp.	8	PR
Family Corydalidae	1186	Corydalidae	3	PR
	1190	Corydalus sp.	3	PR
	1999	Corynoneura sp.	2	CG
	335	Crangonyx sp.	4	CG
Family Crongonyctidae	348	Crongonyctidae	4	
Class Crustacea	251	Crustacea	99.9	CG
	2026	Cryptochironomus sp.	8	PR
	2029	Cryptotendipes sp.	6	CG
	1904	Culex sp.	8	CF
Family Culicidae	1873	Culicidae	8	CG
	1939	Culicoides sp.	5	PR
	1912	Culiseta sp.	8	CG
	1618	Cyphon sp.	7	SC
	1350	Cyrnellus sp.	5	CF
	570	Dannella sp.	2	CG
	1940	Dasyhelea sp.	5	CG
Order Decapoda	400	Decapoda	99.9	SH
	2030	Demicryptochironomus sp.	6	CG

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	1988	Diamesa sp.	4	CG
Tribe Diamesinae	2210	Diamesinae	6	
	1760	Dicranopselaphus sp.	4	SC
	1856	Dicranota sp.	4	PR
	2031	Dicrotendipes sp.	6	CG
	2154	Dictya sp.	10	PR
	760	Didymops sp.	4	PR
	1602	Dineutus sp. (larvae only)	4	PR
	663	Diphetor sp.	4	
	1304	Diplectrona sp.	2	CF
Order Diptera	1850	Diptera	10	
	1931	Dixa sp.	10	CG
	2238	Dixella sp.	10	
Family Dixidae	1930	Dixidae	10	CG
	1974	Djalmabatista sp.	6	PR
Family Dolichopodidae	2144	Dolichopodidae	5	PR
	1344	Dolophilodes sp.	0	CG
Family Dreissenidae	2234	Dreissena sp.	99.9	
	713	Dromogomphus sp.	4	PR
	573	Drunella sp.	1	PR
Family Dryopidae	1764	Dryopidae	4	SH
	1774	Dubiraphia sp.	5	CG
	1761	Ectopria sp.	4	SC
	2035	Einfeldia sp.	10	CG
	2320	Elimia sp.	6	SC
Family Elmidae	1771	Elmidae	5	CG
Family Scirtidae	1801	Elodes sp.	7	
Family Empididae	2146	Empididae	6	PR
	863	Enallagma sp.	6	PR
	2036	Endochironomus sp.	6	SH
	533	Epeorus sp.	1	SC
	626	Ephemera sp.	3	CG
	578	Ephemerella sp.	2	CG
Family Ephemerellidae	567	Ephemerellidae	3.5	CG
Family Ephemeridae	625	Ephemeridae	5	CG

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Order Ephemeroptera	476	Ephemeroptera	3	CG
	638	Ephoron sp.	2	CG
	2152	Ephydra sp.	8	CG
Family Ephydridae	2151	Ephydridae	8	CG
	755	Epiaeschna sp.	1	PR
	772	Epicordulia sp.	4.5	PR
	771	Epitheca sp.	4	PR
	2209	Epoicocladius sp.	6	CG
	1857	Eriocera sp.	7	PR
	1858	Erioptera sp.	4	CG
	2150	Eristalis sp.	11	CG
	2602	Erpetogomphus sp.	2	
	792	Erythemis sp.	5	PR
	794	Erythrodiplax sp.	5	PR
	2005	Eukiefferiella sp.	4	CG
	587	Eurylophella sp.	4	SC
	1444	Fabria sp.	3.5	SH
	2347	Ferrissia sp.	7	SC
	1941	Forcipomyia sp.	5	SC
	2334	Fossaria sp.	7	SC
	1410	Frenesia sp.	3.5	SH
Family Gammaridae	329	Gammaridae	4	CG
	341	Gammarus sp.	3	CG
Class Gastropoda	2301	Gastropoda	99.9	SC
	1373	Glossosoma sp.	3.5	SC
Family Glossosomatidae	1370	Glossosomatidae	3.5	SC
	2039	Glyptotendipes sp.	10	CF
	1412	Goera sp.	3.5	SC
Family Gomphidae	706	Gomphidae	4.5	PR
	722	Gomphus sp.	7	PR
	2321	Goniobasis sp.	5	SC
	2221	Gonomyia sp.	4	CG
	1975	Guttipelopia sp.	6	PR
	2250	Gymnometriocnemus sp.	6	
	2340	Gyraulus sp.	6	SC

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Family Gyrinidae	1601	Gyrinidae	99.9	PR
	1607	Gyrinus sp. (larvae only)	4	PR
	612	Habrophlebiodes sp.	2	SC
	728	Hagenius sp.	3	PR
	2042	Harnischia sp.	6	CG
	992	Hastaperla sp.	1.5	SC
	2214	Hayesomyia sp.	5	
	1765	Helichus sp.	4	SH
	1451	Helicopsyche sp.	2	SC
Family Helicopsychidae	1450	Helicopsychidae	3.5	SC
	2341	Helisoma sp.	7	SC
	1859	Helius sp.	5	CG
	774	Helocordulia sp.	2	PR
Family Scirtidae	1617	Helodidae	7	SC
	2215	Helopelopia sp.	4	PR
	2147	Hemerodromia sp.	6	PR
Order Hemiptera	1050	Hemiptera	99.9	PR
	536	Heptagenia sp.	3	SC
Family Heptageniidae	528	Heptageniidae	3.5	SC
	1860	Hesperoconopa sp.	2	CG
	1413	Hesperophylax sp.	3.5	SH
	834	Hetaerina sp.	3	PR
	519	Heterocloeon sp.	4	SC
	2006	Heterotrissocladius sp.	6	CG
	628	Hexagenia sp.	6	CG
	1861	Hexatoma sp.	4	PR
Class Hirudinea	249	Hirudinea	8	PR
	2509	Hudsonimyia sp.	6	
	327	Hyalella sp.	4	CG
Family Talitridae	326	Hyalellidae	4	
	2119	Hybomitra sp.	7	PR
	1415	Hydatophylax sp.	2	SH
	2007	Hydrobaenus sp.	2	SC
Family Hydrobiidae	2310	Hydrobiidae	6	SC
	977	Hydroperla sp.	1	PR

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	1306	Hydropsyche sp.	5	CF
Family Hydropsychidae	1301	Hydropsychidae	5.5	CF
	1379	Hydroptila sp.	2	SC
Family Hydroptilidae	1376	Hydroptilidae	3.5	PH
	2255	Hyporhygma sp.	4	
Class Insecta	475	Insecta	99.9	
	1417	Ironoquia sp.	3.5	SH
	873	Ischnura sp.	6	PR
	979	Isogenoides sp.	1.5	PR
Family Isonychiidae	487	Isonychia sp.	3	CF
	980	Isoperla sp.	2	PR
Order Isopoda	252	Isopoda	99.9	CG
	1380	Ithytrichia sp.	1	SC
	2043	Kiefferulus sp.	7	CG
	1976	Labrundinia sp.	4	PR
	795	Ladona sp.	4.5	PR
	2348	Laevapex sp.	6	SC
	735	Lanthus sp.	6	PR
	1977	Larsia sp.	6	PR
Order Lepidoptera	1550	Lepidoptera	99.9	SH
	1406	Lepidostoma sp.	3	SH
Family Lepidostomatidae	1405	Lepidostomatidae	3.5	SH
Family Leptoceridae	1453	Leptoceridae	3.5	CG
	1466	Leptocerus sp.	3	SH
	662	Leptohyphe sp.	5.5	CG
Family Leptohyphidae	598	Leptohyphidae	5.5	CG
	614	Leptophlebia sp.	3	CG
Family Leptophlebiidae	609	Leptophlebiidae	3	CG
	1418	Leptophylax sp.	3.5	SH
	2322	Leptoxis sp.	6	
	840	Lestes sp.	6	PR
Family Lestidae	837	Lestidae	99.9	PR
	797	Leucorrhinia sp.	4.5	PR
	1381	Leucotrichia sp.	3	SC
	648	Leucrocuta sp.	3	SC

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	943	Leuctra sp.	1	SH
Family Leuctridae	942	Leuctridae	1.5	SH
	800	Libellula sp.	8	PR
Family Libellulidae	787	Libellulidae	4.5	PR
Family Limnephilidae	1408	Limnephilidae	3.5	SH
	1420	Limnephilus sp.	3	SH
	1862	Limnophila sp.	4	PR
	2157	Limnophora sp.	8	PR
	1863	Limonia sp.	3	SH
	2304	Lioplax sp.	7	SC
	2518	Lipiniella sp.	6	
	272	Lirceus sp.	4	CG
	2323	Lithasia sp.	6	
	2578	Lopescladius sp.	4	
	2335	Lymnaea sp.	7	SC
Family Lymnaeidae	2332	Lymnaeidae	7	SC
	1366	Lype sp.	3.5	SC
	551	Maccaffertium sp.	4	SC
	762	Macromia sp.	3	PR
Family Macromiidae	759	Macromiidae	4.5	PR
	1324	Macronema sp.	2	CF
	1779	Macronychus sp.	2	
	1978	Macropelopia sp.	7	PR
	2217	Macrostemum sp.	2	CF
	1917	Mansonia sp.	8	CG
	2315	Marstonia sp.	6	
	1384	Mayatrichia sp.	1	SC
Order Megaloptera	1175	Megaloptera	3.5	
	2342	Menetus sp.	6.5	SC
	2211	Meropelopia sp.	3	
Order Mesogastropoda	2599	Mesogastropoda	99.9	
Family Metretopodidae	493	Metretopodidae	3	
	2008	Metriocnemus sp.	6	CG
	1403	Micrasema sp.	3.5	MH
	2044	Microchironomus sp.	6	CG

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	1781	Microcylloepus sp.	2	CG
	2075	Micropsectra sp.	4	CG
	2045	Microtendipes sp.	6	CF
	1434	Molanna sp.	3.5	SC
Family Molannidae	1433	Molannidae	3.5	CG
Phylum Mollusca	2300	Mollusca	99.9	
	2167	Monohelea sp.	5	PR
Family Muscidae	2156	Muscidae	8	PR
	1468	Mystacides sp.	2	CG
	2009	Nanocladius sp.	3	CG
	758	Nasiaeschna sp.	2	PR
	1979	Natarsia sp.	6	PR
	1471	Nectopsyche sp.	3	SH
	876	Nehalennia sp.	7	PR
Phylum Nematomorpha	15	Nematomorpha	99.9	PA
	938	Nemoura sp.	1	SH
Family Nemouridae	936	Nemouridae	1.5	SH
	964	Neoperla sp.	1	PR
	1421	Neophylax sp.	3	SC
	1385	Neotrichia sp.	4	SC
	1352	Neureclipsis sp.	3	CF
	775	Neurocordulia sp.	3	PR
Order Neuroptera	1250	Neuroptera	99.9	PR
	1192	Nigronia sp.	2	PR
	2223	Nilobezzia sp.	5	PR
	1980	Nilotanypus sp.	6	PR
	2208	Nilothauma sp.	3	
	644	Nixe sp.	4	SC
	1355	Nyctiophylax sp.	1	CF
	1386	Ochrotrichia sp.	4	CG
Order Odonata	700	Odonata	99.9	PR
	1989	Odontomesa sp.	6	CG
	2083	Odostomia sp.	10	CG
	1477	Oecetis sp.	5	PR
	929	Oemopteryx sp.	1.5	SH

Functional Feeding Group (ffg)
SC=scraper, PA=parasite, PR=predator, OM=omnivore, GC=gatherer/collector, FC=filter/collector, SH=shredder, PI=piercer
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Appendix C. Genus-List: Illinois Environmental Protection Agency Macroinvertebrate-Index of Biotic Integrity (m-IBI) Tolerance List and Functional Feeding Group Classification

January, 2011

Class Oligochaeta	31	Oligochaeta	10	CG
Family Oligoneuriidae	486	Oligoneuriidae	3	CF
	1446	Oligostomis sp.	3.5	PR
	730	Ophiogomphus sp.	2	PR
	1783	Optioservus sp.	4	SC
	1864	Ormosia sp.	4	CG
Tribe Orthocladiinae	1995	Orthocladiinae	6	CG
	2202	Orthocladius sp./Cricotopus sp.	6	
	1387	Orthotrichia sp.	1	SC
	2085	Oxycera sp.	10	SC
	1388	Oxyethira sp.	2	MH
	809	Pachydiplax sp.	8	PR
	2251	Pagastiella sp.	6	
	428	Palaemonetes sp.	4	
Family Palaemonidae	427	Palaemonidae	4	
	1942	Palpomyia sp.	6	PR
	811	Pantala sp.	7	PR
	951	Paracapnia sp.	1.5	SH
	2048	Parachironomus sp.	8	PR
	2050	Paracladopelma sp.	4	CG
	527	Paracloeodes sp.	4	SC
	966	Paragnetina sp.	1.5	PR
	2216	Parakiefferiella sp.	5	
	615	Paraleptophlebia sp.	2	CG
	2173	Paramerina sp.	6	PR
	2011	Parametriocnemus sp.	4	CG
	2179	Paraphaenocladius sp.	6	CG
	1326	Parapsyche sp.	5.5	PR
	2076	Paratanytarsus sp.	6	CG
	2053	Paratendipes sp.	3	CG
	1865	Pedicia sp.	4	PR
Class Pelecypoda	2400	Pelecypoda	99.9	CF
	1769	Pelonomus sp.	4	CG
Family Palingeniidae	635	Pentagenia sp.	4	CF
	1981	Pentaneura sp.	3	PR

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January, 2011

	1934	Pericoma sp.	11	CG
	814	Perithemis sp.	4	PR
	968	Perlesta sp.	4	PR
Family Perlidae	954	Perlidae	1.5	PR
	970	Perlinella sp.	2	PR
Family Perlodidae	976	Perlodidae	1.5	PR
	1556	Petrophila sp.	5	SC
	2054	Phaenopsectra sp.	4	SC
	974	Phasganophora sp.	1.5	PR
Family Philopotamidae	1338	Philopotamidae	3.5	CF
	1448	Phryganea sp.	3	
Family Phryganeidae	1438	Phryganeidae	3.5	SH
	1356	Phylocentropus sp.	3.5	CF
	2330	Physa sp.	9	SC
	2331	Physella sp.	9	SC
Family Physidae	2328	Physidae	9	SC
	1866	Pilaria sp.	4	PR
	2343	Planorbella sp.	6.5	SC
Family Planorbidae	2339	Planorbidae	6.5	SC
	2344	Planorbula sp.	7	SC
	816	Plathemis sp.	3	PR
	1423	Platycentropus sp.	3	SH
Phylum Platyhelminthes	1	Platyhelminthes	99.9	
	651	Plauditus sp.	3	
Order Plecoptera	925	Plecoptera	1.5	PR
	2324	Pleurocera sp.	7	SC
Family Pleuroceridae	2319	Pleuroceridae	6	
Family Polycentropodidae	1349	Polycentropodidae	3.5	CF
	1358	Polycentropus sp.	3	PR
Family Polymitarcyidae	637	Polymitarcyidae	3	CG
	2057	Polypedilum sp.	6	SH
Family Pomatiopsidae	2326	Pomatiopsidae	6	
	2327	Pomatiopsis sp.	6	
Family Potamanthidae	620	Potamanthidae	5	CF
	1328	Potamyia sp.	4	CF

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January, 2011

	1990	Potthastia sp.	6	
	1867	Prionocera sp.	4	SH
	1802	Prionocyphon sp.	7	SC
	1943	Probezzia sp.	5	PR
	2316	Probythinella sp.	6	
	1982	Procladius sp.	8	PR
	643	Procloeon sp.	4	
	1991	Prodiamesa sp.	3	CG
	732	Progomphus sp.	5	PR
	2345	Promenetus sp.	6.5	CG
	1787	Promoresia sp.	5	SC
	1949	Prosimulium sp.	2	CF
	940	Prostoia sp.	1.5	SH
	1375	Protoptila sp.	1	SC
	2012	Psectrocladius sp.	5	CG
	1983	Psectrotanypus sp.	8	PR
Family Psephenidae	1614	Psephenidae	4	SC
	1616	Psephenus sp.	4	SC
	2066	Pseudochironomus sp.	5	CG
	521	Pseudocloeon sp.	4	SC
	1992	Pseudodiamesa sp.	1	CG
	1868	Pseudolimnophila sp.	2	PR
	2013	Pseudorthocladius sp.	6	CG
	1425	Pseudostenophylax sp.	3.5	SH
	2336	Pseudosuccinea sp.	7	SC
	2597	Psilometriocnemus sp.	6	
	1919	Psorophora sp.	8	PR
	1935	Psychoda sp.	11	CG
Family Psychodidae	1933	Psychodidae	11	CG
	1368	Psychomyia sp.	2	SC
Family Psychomyiidae	1365	Psychomyiidae	3.5	CG
Family Pteronarcyidae	927	Pteronarcys sp.	2	SH
	1449	Ptilostomis sp.	3	SH
	2081	Ptychoptera sp.	8	CG
Family Ptychopteridae	2079	Ptychopteridae	8	CG

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Appendix C. Genus-List: Illinois Environmental Protection Agency Macroinvertebrate-Index of Biotic Integrity (m-IBI) Tolerance List and Functional Feeding Group Classification

January, 2011

	1427	Pycnopsyche sp.	3	SH
Family Pyralidae	1551	Pyralidae	99.9	SH
	2317	Pyrgulopsis sp.	6	SC
	994	Rasvena sp.	1.5	CG
	2595	Rhamphomyia sp.	1	
	2014	Rheocricotopus sp.	6	CG
	2225	Rheopelopia sp.	3	PR
	2077	Rheotanytarsus sp.	6	CF
	544	Rhithrogena sp.	0	SC
	1392	Rhyacophila sp.	1	PR
Family Rhyacophilidae	1391	Rhyacophilidae	3.5	PR
	2218	Robackia sp.	3	CG
	2067	Saetheria sp.	6	CG
Family Sciomyzidae	2153	Sciomyzidae	10	PR
	1626	Scirtes sp.	7	SH
Family Sericostomatidae	1489	Sericostomatidae	3.5	SH
	594	Serratella sp.	1	CG
	2596	Serromyia sp.	5	
	1483	Setodes sp.	3.5	
Family Sialidae	1176	Sialidae	3.5	PR
	1177	Sialis sp.	4	PR
Family Simuliidae	1946	Simuliidae	6	CF
	1952	Simulium sp.	6	CF
Family Siphlonuridae	477	Siphlonuridae	3	CG
	482	Siphlonurus sp.	2	CG
	494	Siphloplecton sp.	2	CG
	1254	Sisyra sp.	1	PR
Family Sisyridae	1251	Sisyridae	1	PR
	2015	Smittia sp.	6	CG
	779	Somatochlora sp.	1	PR
	2318	Somatogyrus sp.	6	
	941	Soyedina sp.	1.5	SH
Family Sphaeriidae	2491	Sphaeriidae/Pisidiidae	5	
	2224	Sphaeromias sp.	5	
	1389	Stactobiella sp.	3.5	SH

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January, 2011

	2337	Stagnicola sp.	7	SC
	2219	Stelechomyia sp.	6	CG
	2226	Stempellina sp.	2	CG
	2212	Stempellinella sp.	2	CG
	550	Stenacron sp.	4	SC
	1788	Stenelmis sp.	7	SC
	2068	Stenochironomus sp.	3	SH
	659	Stenonema sp.	4	SC
	2070	Stictochironomus sp.	5	
	1945	Stilobezzia sp.	5	
	2220	Stilocladius sp.	6	
Family Stratiomyidae	2082	Stratiomyidae	10	CG
	2086	Stratiomys sp.	10	CF
	931	Strophopteryx sp.	1.5	SH
	330	Stygobromus sp.	4	PR
	734	Stylogomphus sp.	4.5	PR
	737	Stylurus sp.	7	PR
	2228	Sublettea sp.	6	CF
	818	Sympetrum sp.	4	PR
	1993	Sympotthastia sp.	6	CG
	1994	Syndiamesa sp.	6	CG
Family Syrphidae	2148	Syrphidae	11	CG
Family Tabanidae	2089	Tabanidae	7	PR
	2125	Tabanus sp.	7	PR
Family Taeniopterygidae	928	Taeniopterygidae	1.5	SH
	933	Taeniopteryx sp.	2	SH
Tribe Tanypodinae	2206	Tanypodinae	6	PR
	1984	Tanypus sp.	8	PR
Tribe Tanytarsini	2207	Tanytarsini	6	CF
	2078	Tanytarsus sp.	7	CF
	2165	Telmatoscopus sp.	11	CG
	784	Tetragoneuria sp.	4.5	PR
	2016	Thienemanniella sp.	2	CG
	1985	Thienemannimyia sp.	6	PR
	1854	Tipula sp.	4	SH

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January, 2011

Family Tipulidae	1853	Tipulidae	4	SH
	641	Tortopus sp.	4	CG
	825	Tramea sp.	4	PR
	1484	Triaenodes sp.	3	SH
	2071	Tribelos sp.	5	CG
Order Trichoptera	1300	Trichoptera	3.5	
	599	Tricorythodes sp.	5	CG
Class Turbellaria	2	Turbellaria	6	PR
	2213	Tvetenia sp.	5	
Family Unionidae	2401	Unionidae	1.5	CF
Order Unionoida	2600	Unionoida	99.9	
	1928	Uranotaenia sp.	8	CF
	2307	Valvata sp.	2	SC
Family Valvatidae	2306	Valvatidae	6	SC
Order Venerioda	2601	Veneroida	5	
Family Viviparidae	2302	Viviparidae	6	SC
	2305	Viviparus sp.	1	SC
	1346	Wormaldia sp.	3.5	CF
	2072	Xenochironomus sp.	4	PR
	2227	Xestochironomus sp.	6	
	2249	Xylotopus par.	6	
	2200	Zalutschia sp.	6	SH
	1986	Zavrelimyia sp.	8	PR
	829	Zygoptera	99.9	PR

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Appendix E:

Biological Criteria for the Protection of Aquatic Life: Volume III. Standardized Biological Field and Laboratory Methods for Assessing Fish and Macroinvertebrate Communities

State of Ohio
Environmental Protection Agency

Ecological Assessment Section
Division of Water Quality
Planning & Assessment

Biological Criteria for the Protection of Aquatic Life: Volume III: Standardized Biological Field Sampling and Laboratory Methods for Assessing Fish and Macroinvertebrate Communities

First Update September 30, 1989)

P.O. Box 1049, 1800 WaterMark Dr., Columbus, Ohio 43266-0149

Volume III, pp. V-1-7 to V-1-9. Replaces Tables V-1-1 and V-1-2 with Table V-1.

Table V-1. Current taxonomic keys and the level of taxonomy routinely used by the Ohio EPA for various macroinvertebrate taxonomic classifications.

- Porifera: Species (Pennak 1989)
- Coelenterata: Genus (Pennak 1989)
- Platyhelminthes: Class (Pennak 1989)
- Nemertea: Phylum (Pennak 1989)
- Nematomorpha: Phylum/genus (Pennak 1989)
- Ectoprocta: Genus/species (Thorp and Covich 1991)
- Entoprocta: Species (Thorp and Covich 1991)
- Annelida
  - Oligochaeta: Class (Pennak 1989)
  - Hirudinea: Species (Klemm 1982)
- Arthropoda
  - Crustacea
    - Isopoda: Genus (Pennak 1989)
    - Amphipoda: Genus (Pennak 1989)
      - Gammarus: Species (Holsinger 1972)
    - Decapoda
      - Cambarus and Fallicambarus: Species (Jezerinac and Thoma 1984, Jezerinac 1993)
      - Palaemonetes: Species (Pennak 1989)
  - Arachnoidea: Class (Pennak 1989)
  - Insecta
    - Ephemeroptera: Genus (Edmunds *et al.* 1976, Merritt and Cummins 1996)
      - Baetidae: Genus/species (Morihara and McCafferty 1979, McCafferty and Waltz 1990, Lugo-Ortiz and McCafferty 1998)
        - Pseudocloeon: Species (McCafferty and Waltz 1995)
      - Heptageniidae
        - Stenonema: Species (Bednarik and McCafferty 1979)
      - Ephemerellidae
        - Dannella: Species (Allen and Edmunds 1962)
        - Ephemerella: Species (Allen and Edmunds 1965)
        - Eurylophella: Species (Funk and Sweeney 1994)
        - Serratella: Species (Allen and Edmunds 1963b)
      - Baetiscidae
        - Baetisca: Species (Burks 1953)
    - Ephemeroidea: Species (McCafferty 1975)
    - Odonata: Family/genus (Merritt and Cummins 1996)
      - Anisoptera: Genus/species (Needham and Westfall 1955, Walker 1958, Walker and Corbett 1975)
    - Plecoptera: Genus (Stewart and Stark 1988)
      - Perlidae
        - Acroneuria: Species (Hitchcock 1974)
        - Paragnetina: Species (Hitchcock 1974)
        - Perlinella: Species (Kondratieff *et al.* 1988)
      - Perlodidae: Species (Hitchcock 1974)
    - Hemiptera: Genus (Hilsenhoff 1995, Merritt and Cummins 1996)
    - Megaloptera: Genus (Merritt and Cummins 1996)
      - Nigronia: Species (Neunzig 1966)
    - Neuroptera: Genus (Merritt and Cummins 1996)
    - Trichoptera: Genus (Wiggins 1996, Merritt and Cummins 1996)
      - Philopotamidae: Species (Ross 1944)
      - Hydropsychidae
        - Hydropsyche and Ceratopsyche: Species (Schuster and Etnier 1978)
      - Rhyacophilidae
        - Rhyacophila: Species (Flint 1962, Weaver and Sykora 1979)
      - Leptoceridae
        - Ceraclea: Species (Resh 1976)
        - Mystacides: Species (Yamamoto and Wiggins 1964)
        - Nectopsyche: Species (Haddock 1977)
        - Oecetis: Species (Floyd 1995)
        - Triaenodes/Ylodes: Species (Glover 1996)
    - Lepidoptera: Genus (Merritt and Cummins 1996)
    - Coleoptera: Genus (Hilsenhoff 1995, Merritt and Cummins 1996)
      - Dryopoidea: Genus/species (Brown 1972)
    - Diptera: Family/genus (Merritt and Cummins 1996)
      - Ceratopogonidae
        - Atrichopogon: Species (Johannsen 1935)
      - Chironomidae: Genus/species groups (Wiederholm 1983)
        - Ablabesmyia: Species (Roback 1985)
        - Labrundinia: Species (Roback 1987)
        - Tanypus: Species (Roback 1977)
        - Corynoneura: Species (Simpson and Bode 1980, Bolton In Prep.)
        - Eukiefferiella and Tvetenia: Species groups (Bode 1983)
        - Nanocladius: Species (Saether 1977, Simpson and Bode 1980, Bolton In Prep.)
        - Parakiefferiella: Species (Bolton In Prep.)
        - Rheocricotopus: Species (Saether 1985)
        - Thienemanniella: Species (Hestenes and Saether 2000)
        - Chironomus: Species groups (Oliver and Roussel 1983)
        - Dicrotendipes: Species (Epler 1987)
        - Endochironomus and Tribelos: Species (Grodhaus 1987)
        - Parachironomus: Species (Simpson and Bode 1980, Bolton In Prep.)
        - Polypedilum: Species groups/species (Maschwitz 2000, Bolton In Prep.)
        - Tanytarsini: Genus/species groups/species (Simpson and Bode 1980, Bolton In Prep.)
      - Muscidae: Species (Johannsen 1935)
- Mollusca
  - Gastropoda: Genus/species (Burch 1982)
  - Pelecypoda
    - Sphaeriidae: Genus (Burch 1972)
    - Unionidae: Species (Waters 1995)

Volume III, pp. V-1-11 to V-1-15. Add the following new citations to the References section.

Floyd, M.A. 1995. Larvae of the caddisfly genus Oecetis (Trichoptera: Leptoceridae) in North America. Bulletin of the Ohio Biological Survey Vol. 10, No. 3. 85 pp.

Funk, D.H. and B.W. Sweeney. 1994. The larvae of eastern North American Eurylophella Tiensuu (Ephemeroptera: Ephemerellidae). Transactions of the American Entomological Society 120(3):209-286.

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Hestenes, T.C. and O.A. Saether. 2000. Three new Nearctic Thienemanniella Kieffer species with a review of the Nearctic species. Late 20th Century Research on Chironomidae. An Anthology from the 13th International Symposium on Chironomidae: pp. 103-127. Shaker Verlag, Aachen.

Hilsenhoff, W.L. 1995. Aquatic insects of Wisconsin. Keys to Wisconsin genera and notes on biology, habitat, distribution and species. Publication Number 3 of the Natural History Museums Council. University of Wisconsin - Madison.

Jezerinac, R.F. 1993. A new subgenus and species of crayfish (Decapoda:Cambaridae) of the genus Cambarus, with an amended description of the subgenus Lacunicambarus. Proc. Biol. Soc. Wash. 106(3): pp. 532-544.

Kondratieff, B.C., R.F. Kirchner, and K.W. Stewart. 1988. A review of Perlinella Banks (Plecoptera: Perlidae). Annals of the Entomological Society of America 81(1):19-27.

Lugo-Ortiz, C.R. and W.P. McCafferty. 1998. A new North American genus of Baetidae (Ephemeroptera) and key to Baetis complex genera. Ent. News 109(5): 345-353.

Maschwitz, D.E. and E.F. Cook. 2000. Revision of the Nearctic species of the genus Polypedilum Kieffer (Diptera: Chironomidae) in the subgenera P. (Polypedilum) Kieffer and P. (Uresipedilum) Oyewo and Saether. Bulletin of the Ohio Biological Survey. New Series 12(3): 1-135.

McCafferty, W.P. and R.D. Waltz. 1995. Labiobaetis (Ephemeroptera: Baetidae): new status, new North American species, and related new genus. Ent. News 106(1): 19-28.

McCafferty, W.P. and R.D. Waltz. 1990. Revisionary synopsis of the Baetidae (Ephemeroptera) of North and Middle America. Transactions of the American Entomological Society 116(4):769-799.

Merritt, R.W. and K.W. Cummins (editors). 1996. An introduction to the aquatic insects of North America. 3rd edition. Kendall/Hunt Publishing Company, Dubuque, Iowa.

Pennak, R.W. 1989. Fresh-water invertebrates of the United States. 3rd edition. John Wiley & Sons, New York, New York.

Saether, O.A. 1985. A review of the genus Rheocricotopus Thienemann & Harnisch, 1932, with the description of three new species (Diptera: Chironomidae). Spixiana Supplement 11:59-108.

Thorp, J.H. and A.P. Covich (editors). 1991. Ecology and classification of North American freshwater invertebrates. Academic Press, San Diego, California.

Waters, G.T. 1995. A guide to the freshwater mussels of Ohio. 3rd edition. The Ohio Department of Natural Resources, Division of Wildlife, Columbus, Ohio.

March, 2001

New Citations (cont)

Weaver, J.S., III and J.L. Sykora. 1979. The Rhyacophila of Pennsylvania with larval descriptions of R. banksi and R. carpenteri (Trichoptera: Rhyacophilidae). Annals of Carnegie Museum. Carnegie Museum of Natural History 48(22): 403-423.

Wiggins, G.B. 1996. Larvae of the North American caddisfly genera (Trichoptera). 2nd edition. University of Toronto Press, Toronto, Canada.

Yamamoto, T. and G.B. Wiggins. 1964. A comparative study of the North American species in the caddisfly genus Mystacides (Trichoptera: Leptoceridae). Can. J. Zool. 42: 1105-1126.

NOTICE TO USERS

All methods and procedures for the use of biological criteria contained and/or referred to in these volumes supercede those described in any previous Ohio EPA manuals, reports, policies, and publications dealing with biological evaluation, designation of aquatic life uses, or the determination and evaluation of aquatic life use attainment. Users of these criteria and the supporting field methods, data analyses, and study design should conform to that presented or referenced in these volumes (and subsequent revisions) in order to be applicable under the Ohio Water Quality Standards (WQS; OAC 3745-1).

Three volumes comprise the supporting documentation for setting and using biological criteria in Ohio. All three volumes are needed to use the biological criteria, implement the field and laboratory procedures, and understand the principles behind their development, use, and application. These volumes are:

Ohio Environmental Protection Agency. 1987. *Biological criteria for the protection of aquatic life: Volume I. The role of biological data in water quality assessment.* Division of Water Quality Monitoring and Assessment, Surface Water Section, Columbus, Ohio.

Ohio Environmental Protection Agency. 1987. *Biological criteria for the protection of aquatic life: Volume II. Users manual for biological field assessment of Ohio surface waters.* Division of Water Quality Monitoring and Assessment, Surface Water Section, Columbus, Ohio.

Ohio Environmental Protection Agency. 1989. *Biological criteria for the protection of aquatic life: Volume III. Standardized biological field sampling and laboratory methods for assessing fish and macroinvertebrate communities.* Division of Water Quality Monitoring and Assessment, Columbus, Ohio.

In addition, one other publication from the Stream Regionalization Project is recommended to all users:

Whittier, T.R., D.P. Larsen, R.M. Hughes, C.M. Rohm, A.L. Gallant, and J.M. Omernik. 1987. *The Ohio stream regionalization project: a compendium of results.* U.S. EPA - Environmental Res. Lab, Corvallis, OR. EPA/600/3-87/025. 66 pp.

These documents can be obtained by writing:

Ohio Environmental Protection Agency
Division of Water Quality Monitoring and Assessment
1800 WaterMark Drive, P.O. Box 1049
Columbus, Ohio 43266-0149

Other recommended and helpful literature is listed in the references of each volume.

FOREWARD

This volume is excerpted from the Ohio EPA Manual of Surveillance Methods and Quality Assurance Practices (6th Update). The macroinvertebrate methods are from section V, subsection 1 and the fish methods are from section V, subsection 4 of this manual. They are produced here to accompany the supporting technical documentation for the establishment and use of biological criteria in Ohio.

Acknowledgements

Jeff DeShon, Jack Freda, and Mike Bolton provided the primary input to the macroinvertebrate section. Chris Yoder, Marc Smith, Roger Thoma, Randy Sanders, and Ed Rankin were responsible for the fish section. Ed Rankin was the primary originator of the Qualitative Habitat Evaluation Index (QHEI) which is described in the fish section. Pam Jacques provided typing support.

Subsection 1. Macroinvertebrates

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Part A) Field Methods - Quantitative Sampling

Part B) Field Methods - Qualitative Sampling

Part C) Laboratory Methods - Quantitative Sampling

1) Macroinvertebrate Counts and Identifications

2) Macroinvertebrate Data Analysis

a) Invertebrate Community Index

b) Community Similarity Index

c) Rank Correlation Coefficient

d) Coefficient of Variation

Part D) Laboratory and Data Analysis Methods - Qualitative Sampling

Part A
Field Methods -
Quantitative Sampling

The primary sampling equipment used for the collection of benthic macroinvertebrates is the modified Hester-Dendy multiple-plate artificial substrate sampler. The sampler is constructed of 1/8 inch tempered hardboard cut into three inch square plates and one inch square spacers. A total of eight plates and twelve spacers are used for each sampler. The plates and spacers are placed on a 1/4 inch eyebolt so that there are three single spaces, three double spaces, and one triple space between the plates. The total surface area of the sampler, excluding the eyebolt, is 145.6 square inches.

Samplers placed in streams are tied to a concrete construction block which anchors them in place and prevents the multiple- plates from coming into contact with the natural substrates. In water deeper than four feet, a float (1 qt. cubitainer) is attached to the samplers to keep them within four feet of the surface. Whenever possible, the samplers are placed in runs rather than pools or riffles and an attempt is made to establish stations in as similar an ecological situation as possible. All samplers are exposed for a six week period. A set of samplers consists of three multiple-plate samplers (three square feet) at National Ambient Water Quality Monitoring Network (NAWQMN) stations and five multiple-plate samplers at all other sampling locations. All NAWQMN stations and most routine monitoring stations are sampled during the time period of June 15 to September 30.

Retrieval of the samplers is accomplished by cutting them from the block and placing them in one quart plastic containers while still submersed. Care is taken to avoid disturbing the samplers and thereby dislodging any organisms. Enough formalin is added to each container to equal an approximate 10% solution. Qualitative samples of macroinvertebrates inhabiting the natural substrates are also collected at the time of sampler retrieval. In shallow water, samples are taken in a stream segment covering all available habitats in the near vicinity where the samplers were placed. Samples are collected using triangular ring frame 30-mesh dip nets and hand picking with forceps. Grab samplers (i.e., Ekman, Peterson, or Ponar) can also be used in deep water. The qualitative sampling continues until, by gross examination, no new taxa are being taken. A station description sheet (Figure V-1-1) is filled out by the collector at the time of sampler retrieval. The substrate is described using the categories for substrate characterization indicated in the USEPA biological field manual (Weber, 1973).

In those situations where quantitative biological samples are collected from the natural substrates using a Surber square foot sampler (30-mesh netting), the collector stands on the downstream side of the sampler and works the substrate using a hand cultivator with two inch tines. Large rocks are gently scrubbed with a brush. The material collected is placed in sealed containers, preserved in 10% formalin, and transported to the laboratory. Three to five Surber samples are taken at each site.

Figure V-1-1. Station description sheet used by macroinvertebrate field crews (Front).

Ohio EPA Surface Water Section
Macroinvertebrate Field Sheet

Stream_____ Stream Code_____ RM____ Date Collected_____

Location_____ Date Set_____

_____ Collected By_____

Sampling Method: HD(No.____) - DN/HP - Surber - Grab (Type_____) - Other_____

HD Sampler Site: Depth_____ Canopy_____ Current (Set)_____ Current (Ret)_____

HD Condition: Disturbed Yes/No Comment:_____
Debris Yes/No Comment:_____
Silt/Solids None - Slight - Moderate - Heavy

DN/HP Sampling: Total Time____ Habitats: Pool - Riffle - Run - Margin - Backwater

---

Physical Characteristics

Flow Condition: High - Moderate - Low - Interstitial - Intermittent - Dry
Current Velocity: Fast - Moderate - Slow - ND
Channel Morphology: Natural - Channelized - Channelized (Recovered) - Impounded
Bank Erosion: Extensive - Moderate - Slight - None
Riffle Development: Extensive - Moderate - Sparse - Absent
Riffle Quality: Good - Fair - Poor Embedded: Yes/No
Clarity: Clear - Murky - Turbid
Color: None - Green - Brown - Grey - Other( )
Canopy: Open - 75% - 50% - 25% - Closed

Substrate Characteristics

Percent of:	Pool	Riffle	Run
Bedrock( )			
Boulder( )			
Rubble( )			
Coarse Gravel			
Fine Gravel			
Sand			
Silt			
Clay/Hardpan			
Detritus			
Peat			
Muck			
Other( )			
Macrophytes( )			
Algae( )			
Artifacts( )			
Compaction(F,M,S)			
Depth (Average)			
Width (Average)			

Predominant Land Use (L,R,B)

Forest Open Pasture Wetland
Shrub Closed Pasture Other
Old Field Urban ( )
Rowcrop Residential/Park
Industrial Mining/Construction

Riparian Vegetation

Left	Width	Right	Width	Type
				Large trees
				Small trees
				Shrubs
				Grass/Weeds
				None

Margin Habitat

Undercut Banks Root Mats
Grass Water Willow
Shallows Clay/Hardpan
Rip Rap Bulkhead
Other( )

Margin Quality: Good - Fair - Poor

Figure V-1-1 (Continued). Station description sheet used by macroinvertebrate field crews (Back).

Biological Characteristics

Riffle

Predominant Organisms: _____

Other Common Organisms: _____

Density: High - Moderate - Low

Diversity: High - Moderate - Low

Run

Predominant Organisms: _____

Other Common Organisms: _____

Density: High - Moderate - Low

Diversity: High - Moderate - Low

Pool

Predominant Organisms: _____

Other Common Organisms: _____

Density: High - Moderate - Low

Diversity: High - Moderate - Low

Margin

Predominant Organisms: _____

Other Common Organisms: _____

Density: High - Moderate - Low

Diversity: High - Moderate - Low

Other Notable Collections: _____

Potential Pollution Sources: _____

Evidence of Pollution: _____

Photo Numbers: _____

Comments: _____

_____

_____

_____

_____

_____

_____

In those situations where Ekman, Peterson, or Ponar grab samples are used for quantitative purposes, three to five samples are collected and then treated in essentially the same manner as the Surber samples. The material collected with the grab is washed through a bucket with a 30-mesh screen bottom, placed in sealed containers, preserved in 10% formalin, and returned to the laboratory.

Part B
Field Methods - Qualitative Sampling

When only qualitative samples are collected the methods are similar to those employed when collecting qualitative samples in conjunction with artificial substrate samples except that:

a) A more intensive sampling effort is required.

b) The sampling area is more rigidly defined.

c) More extensive field notes concerning the biological and physical condition of each station are required.

d) A preliminary biological community assessment is made on site.

Each station is sampled at least once between June 15 and September 30. Organisms are collected from the natural substrates using triangular ring frame 30-mesh dip nets and forceps, and are preserved in 70% alcohol. Collections are made for a minimum of 30 minutes, then continue until no new taxa are evident in gross examinations. Whenever possible, a riffle, run, margin, and pool habitat are sampled at each station and an attempt is made to sample areas which are physically similar from site to site. Stations should be sampled in order, moving from upstream to downstream, to detect any changes between sites.

As in quantitative sampling, the station description sheet (Figure V-1-1) is filled out at each station at the time of collection. In addition, the length of sampling time and the presence of riffle, run, margin, and pool habitats are noted. Predominant populations and estimates of community density and diversity in each habitat type are noted on the sheet. A preliminary biological community assessment is made after each station is sampled.

Part C
Laboratory Methods - Quantitative Sampling

Samples are coded and sample numbers are immediately entered into a log book upon arrival at the laboratory. Samples are given a log number derived from the date, e.g., 871108-10, where 87 represents the year, 11 represents the month, and 08 the day. The number following this six digit date, i.e., the number 10 in the previous example, indicates that this was the 10th sampled logged that day. Other information in the log book includes the name(s) of field personnel that collected the sample, date, stream or lake name, basin name, entity (where applicable), general location, sample type, sampling method(s) used, the person who conducted the analyses, and any other comments considered pertinent to the collection and analysis of the sample.

1) *Macroinvertebrate Counts and Identifications*

Composite samples consisting of five multiple-plate samplers are used in station evaluations for routine monitoring. However, replicate samples (three multiple-plate samplers) are reported to the USEPA for NAWQMN stations. Replicate sets of five multiple-plate samplers can be used if deemed necessary in those cases where sampling is for litigation purposes. In all cases, the multiple-

plate(s) is (are) disassembled in a bucket of water, cleaned of organisms and debris, and discarded. The organism/debris mixture is then passed through U.S. Standard Testing Sieves number 30 (0.589 mm openings) and number 40 (0.425 mm openings). The material retained in each sieve is preserved in properly labeled and coded jars of 70% alcohol.

The following procedures are used during the course of analyzing an artificial substrate, Surber, or grab sample:

a) Sorting of the sample is done in a white enamel pan followed by scanning under the dissecting microscope (10x magnification). Subsamples are produced using the following guidelines:

1) The Folsom sample splitter is used for all subsampling. (In an effort to determine the accuracy of the Folsom sample splitter, a sample composed of 200 individuals of five frequently collected organisms was prepared and repeatedly split. Statistical analysis of the data yielded a chi- square value of 2.56, df=4, which was not significant at the 95% probability level.)

2) After an entire sample has been sorted, subsampling within families containing unmanageable numbers is acceptable.

3) Very large samples may be subsampled prior to sorting - but only after examination in a white enamel pan to remove obvious rare taxa, e.g., crayfish, hellgramites, non-hydropsychid caddisflies.

4) A minimum of 250 organisms is identified, with at least 50-100 midges, 70 caddisflies, 70 mayflies.

b) Dipterans of the family Chironomidae are prepared for identification by clearing the larvae in hot 10% KOH for 30 minutes and then mounting in water on microscope slides. Permanent slides for the voucher collection are mounted in Euparol mounting medium.

c) Material retained in the # 40 screen is counted and identified or counted and extrapolated when identification is impossible or impractical. (Artificial substrate sample only.)

d) Organisms determined to be dead before the time of collection are discarded.

e) When only one sex or life stage can be identified it is assumed that the other sex or stage is the same species.

f) Sections of bryozoan colonies are removed from the plates and saved for identification. Only colonies, not individuals, are counted. (Artificial substrate sample only.)

g) Early instars that cannot be identified are extrapolated where possible.

h) Species level identifications are made where possible and practical. Generic or higher level classifications are made if specimens are damaged beyond identification, in those cases where taxonomy is incomplete or laborious and time-consuming, or where the specimen is an unidentifiable early instar.

i) Organisms are listed in tables following the laboratory table format (Table V-1-1).

j) Two end fragments of an oligochaete are counted as one individual. Fragments without ends are not counted.

k) Any taxonomic key in the laboratory may be used as an aid in the identification of an organism. However, the final identification and name used are taken from the asterisked references in Table V-1-2. Also indicated is the level of taxonomy attainable with the keys listed.

Table V-1-1. Phylogenetic order for macroinvertebrate listing including level of taxonomy generally used.

Porifera:	Species
Coelenterata:	Genus
Platyhelminthes:	Class
Nematomorpha:	Genus
Bryozoa:	Species
Entoprocta:	Species
Annelida	
Oligochaeta:	Class
Hirudinea:	Species
Arthropoda	
Crustacea	
Isopoda:	Genus
Amphipoda:	Genus/Species
Decapoda:	Species
Arachnoidea	
Hydracarina:	Class
Insecta	
Ephemeroptera	
Siphlonuridae:	Genus
Baetidae:	Genus
Oligoneuriidae:	Genus
Heptageniidae:	Genus/Species
Leptophlebiidae	Genus
Ephemerellidae:	Species
Tricorythidae:	Genus
Caenidae:	Genus
Baetiscidae:	Species
Potamanthidae:	Genus
Ephemeridae:	Genus
Polymitarcyidae:	Species
Odonata	
Zygoptera	
Calopterygidae:	Genus
Lestidae:	Species
Coenagrionidae:	Family/Genus
Anisoptera	
Aeshnidae:	Species
Gomphidae:	Species
Cordulegastridae:	Species
Macromiidae:	Species
Corduliidae:	Species
Libellulidae:	Species
Plecoptera	
Pteronarcyidae:	Genus
Peltoperlidae:	Genus
Taeniopterygidae:	Genus
Nemouridae:	Species
Leuctridae:	Genus
Capniidae:	Genus
Perlidae:	Species
Perlodidae:	Species
Chloroperlidae:	Genus
Hemiptera	
Belostomatidae:	Genus
Nepidae:	Genus
Pleidae:	Genus
Naucoridae:	Genus
Corixidae:	Genus
Notonectidae:	Genus
Megaloptera	
Sialidae:	Genus
Corydalidae:	Species
Neuroptera:	Genus
Trichoptera	
Philopotamidae:	Genus/Species
Psychomyiidae:	Species
Polycentropodidae:	Genus
Hydropsychidae:	Genus/Species
Rhyacophilidae:	Genus/Species
Glossosomatidae:	Genus
Hydroptilidae:	Genus/Species
Phryganeidae:	Genus
Brachycentridae:	Genus
Limnephilidae:	Genus
Lepidostomatidae:	Genus
Beraeidae:	Genus
Sericostomatidae:	Genus
Odontoceridae:	Genus
Molannidae:	Genus
Helicopsychidae:	Species
Calamoceratidae:	Genus
Leptoceridae:	Genus/Species
Lepidoptera:	Genus

Coleoptera	
Gyrinidae:	Genus
Haliplidae:	Genus
Dytiscidae:	Genus
Noteridae:	Genus
Hydrophilidae:	Genus
Hydraenidae:	Genus
Psephenidae:	Species
Dryopidae:	Genus
Scirtidae:	Family
Elmidae:	Genus/Species
Limnichidae:	Genus
Heteroceridae:	Family
Ptilodactylidae:	Family
Chrysomelidae:	Family
Curculionidae:	Family
Lampyridae:	Family
Diptera	
Tipulidae:	Genus
Psychodidae:	Genus
Ptychopteridae:	Genus
Dixidae:	Genus
Chaoboridae:	Genus
Culicidae:	Genus
Thaumaleidae:	Genus
Simuliidae:	Genus
Certopogonidae:	Family/Genus/Species
Chironomidae	
Tanypodinae:	Genus/Species
Diamesinae:	Genus/Species
Prodiamesinae:	Genus/Species
Orthocladinae:	Genus/Species
Chironominae	
Chironomini:	Genus/Species
Pseudochironomini:	Genus/Species
Tanytarsini:	Genus/Species
Tabanidae:	Genus/Species
Athericidae:	Species
Stratiomyidae:	Genus
Empididae:	Family
Dolichopodidae:	Family
Syrphidae:	Family/Genus
Sciomyzidae:	Family/Genus
Ephydridae:	Family/Genus
Muscidae:	Species
Mollusca	
Gastropoda:	Family/Genus/Species
Pelecypoda:	Family/Genus/Species

Table V-1-2 Level of macroinvertebrate taxonomy attainable using keys (Asterisked references are used for final identifications)

Porifera: Pennak* (1978)/Species
Coelenterata: Pennak* (1978)/Species
Platyhelminthes: Pennak* (1978)/Species
Nematomorpha: Pennak* (1978)/Genus
Bryozoa: Pennak* (1978)/Species
Annelida
- Hirudinea: Klemm* (1982)/Species

Isopoda
- *Asellus*: Williams* (1972)/Species

Amphipoda
- Specific Keys: Pennak* (1978)/Species
- Gammaridae: Holsinger* (1972)/Species

Decapoda
- *Cambarus* and *Fallicambarus*: Jezerinac and Thoma* (1982)/ Species
- *Procambarus* and *Orconectes*: Jezerinac* (1978)/ Species

Ephemeroptera
- Generic Keys: Edmunds et al. (1976), Merritt and Cummins* (1984)/Genus
- *Baetis*: Morihara and McCafferty* (1979)/Species
- *Stenonema*: Bednarik and McCafferty* (1979)/Species
- *Attenella*: Allen and Edmunds* (1961)/Species
- *Dannella*: Allen and Edmunds* (1962)/Species
- *Drunella*: Allen and Edmunds* (1962)/Species
- *Ephemerella*: Allen and Edmunds* (1965)/Species
- *Eurylophella*: Allen and Edmunds* (1965)/Species
- *Serratella*: Allen and Edmunds* (1963)/Species
- *Ephemeroidea*: McCafferty* (1975)/Species
- Other Species Keys: Burks* (1953)/Species

Odonata
- Generic Keys: Merritt and Cummins* (1984)/Genus
- Zygoptera: Walker* (1953)/Species
- Anisoptera: Needham and Westfall* (1955), Walker (1958), Walker and Corbett (1975)/Species

Plecoptera
- Generic Keys: Stewart and Stark* (1988)/Genus
- Species Keys: Hitchcock* (1974)/Species
- *Agnetina*: Stark* (1986)/Species

Hemiptera Generic Keys: Hilsenhoff (1982), Merritt and Cummins* (1984)/Genus

Megaloptera
- Generic Keys: Merritt and Cummins* (1984)/Genus
- *Chauliodes*: Cuyler* (1958)/Species
- *Nigronia*: Neunzig* (1966)/Species

Neuroptera
- Generic Keys: Merritt and Cummins* (1984)/Genus

Trichoptera
- Generic keys: Wiggins* (1977)/Genus
- *Hydropsyche*: Schefter et al.* (1986)/Genus Schuster and Etnier (1978)/Species
- *Rhyacophila*: Flint* (1962)/Species
- *Nectopsyche*: Haddock* (1977)/Species

Lepidoptera
- Generic Keys: Merritt and Cummins* (1984)/Genus

Coleoptera
- Generic Keys: Hilsenhoff (1982), Merritt and Cummins* (1984)/Genus
- Dryopoidea: Brown* (1972)/Species

Diptera
- Generic Keys: McAlpine et al.* (1981) (exc. Chironomidae)/Genus
- Simuliidae: Stone* (1964)/Species
- Chironomidae
  - Generic Keys: Wiederholm* (1983)/Genus
  - *Ablabesmyia*: Roback* (1985)/Species
  - *Clinotanypus*: Roback* (1976)/Species
  - *Coelotanypus*: Roback* (1974)/Species
  - *Labrundinia*: Roback* (1987)/Species
  - *Natarsia* and *Psectrotanypus*: Roback* (1978)/Species
  - *Nilotanypus*: Roback* (1986)/Species
  - *Tanypus*: Roback* (1977)/Species
  - *Pagastia*: Oliver and Roussel* (1982)/Species
  - *Monodiamesa*: Saether* (1973)/Species
  - *Brillia*: Oliver and Roussel* (1983)/Species
  - *Eukiefferiella* and *Tvetenia*: Bode* (1983)/Species group
  - *Nanocladius*: Saether* (1977)/Species
  - *Orthocladius* (*Orthocladius*): Soponis* (1977)/Species
  - *Axarus*: Roback* (1963)/Species
  - *Dicrotendipes*: Epler* (1987)/Species
  - *Endochironomus*, Tribelos, and Endotribelos: Grodhaus*(1987)/Species
  - *Paracladopelma* and *Saetheria*: Jackson* (1977)/Species
  - *Polypedium* (*Polypedilum*): Maschwitz* (1976)/Species
  - Other Species keys: Simpson and Bode* (1980)/Species
- Tabanidae: Pechuman et al.* (1983)/Species
- Athericidae: Webb (1977)*/Species
- Muscidae: Johannsen* (1935)/Species

Mollusca
- Gastropoda: Burch* (1982)/Species
- Pelecypoda: Burch* (1972)/Species

2) Macroinvertebrate Data Analysis

a) *Invertebrate Community Index*

The principle measure of overall macroinvertebrate community condition used by the Biological Field Evaluations Group is the Invertebrate Community Index (ICI), a measurement derived in-house from information collected over many years. The ICI is a modification of the Index of Biotic Integrity (IBI) for fish developed by Karr (1981). The ICI consists of ten structural community metrics, each with four scoring categories of 6, 4, 2, and 0 points (Table V-1-3). The point system evaluates a sample against a database of 247 relatively undisturbed reference sites throughout Ohio. Six points will be scored if a given metric has a value comparable to those of exceptional stream communities, 4 points for those metric values characteristic of more typical good communities, 2 points for metric values slightly deviating from the expected range of good values, and 0 points for metric values strongly deviating from the expected range of good values. The summation of the individual metric scores (determined by the relevant attributes of an invertebrate sample with some consideration given to stream drainage area) results in the ICI value. Metrics 1-9 are all generated from the artificial substrate sample data while Metric 10 is based solely on the qualitative sample data. More discussion of the derivation of the ICI including descriptions of each metric and the data plots and other information used to score each metric can be found in Ohio EPA (1987).

Table V-1-3. Invertebrate Community Index (ICI) Metrics and Scoring Criteria Based on Macroinvertebrate Community Data From 247 Reference Sites Throughout Ohio.

Metric	Scoring Criteria 0	2	4	6
1. Total Number of Taxa	Scoring of each metric varies with drainage area; see Ohio EPA (1987).			
2. Total Number of Mayfly Taxa				
3. Total Number of Caddisfly Taxa				
4. Total Number of Dipteran Taxa				
5. Percent Mayflies				
6. Percent Caddisflies				
7. Percent Tribe Tanytarsini Midges				
8. Percent Other Dipterans and Non-Insects				
9. Percent Tolerant Organisms				
10. Total Number of Qualitative Ephemroptera, Plecoptera, and Trichoptera (EPT) Taxa				

b) *Community Similarity Index*

A coefficient of similarity (c) between two stations can be calculated using Van Horn's (1950) equation modified from the general formula described by Gleason (1920):

$$c = \frac{2w}{a + b}$$

The variables in this expression can be based either on the number of taxa present or absent at each station or on actual numerical data collected at each site. If the presence/absence method is being used:

a = the number of taxa collected at one station,
b = the number of taxa collected at the other station, and
w = the number of taxa common to both stations.

When actual numerical data are being used, each taxon is assigned a prominence value calculated by multiplying the density of the taxon by the square root of its frequency of occurrence (Beals, 1961; Burlington, 1962). In this case:

a = the sum of the prominence values of all of the taxa at one station,
b = the sum of the prominence values of all of the taxa at the other station, and
w = the sum of the prominence values of all of the taxa of one station which it has in common with the other station. The lower of the two resulting values of w is used in the equation.

c) Rank Correlation Coefficient

A rank correlation coefficient between measured biological, chemical, or other physical data can be calculated using the formula defined by Spearman (1904):

$$r_s = 1 - \frac{6 \sum_{i=1}^{n} D_i^2}{n(n^2 - 1)}$$

where n = the number of paired observations ($x_i y_i$) and
D_i = the rank of x_i minus the rank of y_i.

d) Coefficient of Variation

In cases where replicate analyses are conducted (e.g., litigation purposes or NAWQMN stations), a coefficient of variation between replicates is determined following the procedures outlined by Li (1964) using the formula:

$$CV = \frac{s}{\bar{x}} \times 100$$

where s = the sample standard deviation and:
x = the sample mean.

Part D
Laboratory Methods and Data Analysis - Qualitative Sampling

Samples are entered and logged as outlined in Subsection 1, part c. Samples are examined using a dissecting microscope and a tabulated listing of the organisms identified is compiled. Dipterans of the family Chironomidae are prepared as outlined in Subsection 1, Part c. Taxonomic guides used for final identifications are the same as listed in Subsection 1, Part c. Assessment of the macroinvertebrate community condition is finalized using the preliminary assessment made in the field tempered with information on taxa richness and composition from the laboratory identified sample.

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Procedure No.	WQPA-SWS-3	Date Issue	9-30-89
Revision No.	6	Date Effective	9-30-89

Subsection 4. Fish

Part A) Training

1) *Sampling Methods*

All new full-time field personnel in the Fish Evaluation Group receive in-house training in the following procedures prior to the start of the field season. A senior staff member also accompanies the new field crew leader for at least the first two weeks of the field sampling season (and thereafter if necessary) instructing in all aspects of the field sampling. Individuals are then permitted to proceed on their own with periodic conferences with the Fish Evaluation Group supervisor to assure the sampling effort is being conducted in accordance with the procedures described herein.

New part-time summer field personnel receive copies of the fish section of the Quality Assurance Manual (Subsection 4) and are given pre-field season training on the procedures involved in the fish sampling program for a one week period prior to the field season.

2) Species Identification

All new field personnel, summer or full-time, are given a test consisting of a collection of different Ohio fish species to identify and count to determine their familiarity with Ohio fish taxonomy and their ability to accurately count large numbers of fish. Full-time field crew leaders perform or supervise *all* of the actual field identifications and counts with the summer personnel assisting.

Part B) Field Methods

1) *Sampling Site Selection*

The selection of fish sampling sites is based upon several factors including, but not limited to, the following:

1) location of point source dischargers;
2) stream use designation evaluation issues;

3) location of physical habitat features (e.g. dams, changes in geology, changes in stream order, presence of a stream confluence, etc.);
4) location of nonpoint sources of pollution; and,
5) variations in macrohabitat.
6) proximity to ecoregion boundaries.

Each study area has a set number of biological sampling sites allocated based on the number and complexity of the priority issues requiring field evaluation. Optimum placement of sampling sites is determined recognizing practical access and resource constraints. The principal objectives of each survey determine where sampling sites will be located. Generally, sites are located upstream from all pollution sources to determine the background condition for the study area. Should the upstream portion of the stream be impacted, an alternate site may be chosen on an adjacent stream with similar watershed characteristics. Reference sites within the same ecoregion may also be used in this role (these are listed in Ohio EPA 1987). The role of upstream sites is not necessarily to provide a biological performance level against which downstream sites are compared since the ecoregion biocriteria fill this niche for the respective aquatic life use designations. Upstream sites are, however, important in defining any site or watershed specific background conditions that might temporarily or permanently influence eventual aquatic life use attainment in the downstream reaches. Selection of sampling sites within a segment is accomplished by selecting the most *typical* habitat available in an effort to represent the current potential of that segment. An attempt should be made to sample typically similar macrohabitats at all sampling sites established within the study area.

To address point source discharge concerns, at least one site is situated upstream from the primary process wastewater outfall(s), one within the mixing zone, and sites located at intervals downstream from the mixing zone (i.e. dependent on stream size and mixing characteristics) to determine the near and far field impacts, the longitudinal extent and severity of any impact, and to determine if and where recovery occurs. Spacing of the downstream sampling sites is based on physical macrohabitat characteristics, access to the segment, other adjacent point and nonpoint sources, stream size, and other factors. An attempt is made to place sampling sites between point sources where sufficient distance between each exists. Sampling sites may also be situated in the mouths of major tributaries to determine any potential effects on the mainstem. Localized areas of macrohabitat modification such as instream impoundments or channelized sections alter macrohabitat available for fish and can affect community structure and function. Generally, these areas are not *typical* of the macrohabitat in a free-flowing river or stream. However, these areas are often times impacted by the principal sources targeted for evaluation in certain study areas (particularly in urban areas), therefore, sampling sites are located within these modified areas as needed. These areas should be sampled in order to understand the underlying influence that they exert on biological performance and eventual aquatic life use attainment.

2) Fish Sampling Procedures

a) Introduction

The principal method used by Ohio EPA to obtain fish relative abundance and distribution data is pulsed direct current electrofishing. As with any single method there exists inherent sampling selectivity and sampling bias. Pulsed D.C. electrofishing is, however, widely viewed as the single most effective method for sampling fish communities in lotic habitats. Twelve different fish sampling techniques have been assigned sampler type codes. Six codes are currently recognized as valid for generating fish relative abundance data for the purpose of calculating Index of Biotic Integrity (IBI) and Modified Index of Well-

Being (Iwb) scores from which aquatic life use attainment is partially judged (Table V-4-1). The remaining codes are assigned to seldom used or currently experimental methods. This system of letter codes superceded a system of numerical codes used prior to 1984. The use of any one of these sampling methods is dependent on the type of information required and the type of aquatic habitat being sampled. Since 1979 certain methods have been modified or abandoned (e.g. seining). The boat mounted and wading electrofishing methods are the most commonly used fish sampling techniques by Ohio EPA in lotic habitats. The boat electrofishing methods (sampler type A) are used to sample the largest streams and rivers (Table V-4-1). Wading methods (sampler types D, E, and F) are used in wadable streams. These are the most frequently used sampler types and are regarded as suitable for calculating IBI and modified Iwb scores (Ohio EPA 1987). Sampler type B (18' boat, circular electrode array) is used in the deeper rivers (e.g. Ohio River) and embayments (e.g. Lake Erie tributary river mouths). This is also considered to be an acceptable method. Sampler type C is used in free-flowing rivers to sample riffle habitats. This method is used only to supplement the boat methods and the data is not used to calculate the IBI or modified Iwb. Sampler types G and H are seining methods and are no longer in routine use. The fyke net and hoop net methods (types I and J) may be necessary in lentic, wetland, or large river habitats. The experimental gill net method (type K) may be necessary to sample for mid-channel and pelagic species. These *passive* methods (types I through K) are seldom used and only in special situations to supplement routine electrofishing sampling.

Fish sampling is preferably conducted between mid-June and early October, when stream and river flows are generally low, pollution stresses are potentially the greatest, and the fish community is most stable and sedentary. Sampling may be conducted outside of this time period, but the results may not be applicable for Ohio EPA biocriteria purposes. The use and applicability of this data will be evaluated on a case-by-case basis. Special studies are conducted by the Fish Evaluation Group on a periodic basis to determine the effectiveness of each sampling method, comparability of methods, necessary sampling frequency, evaluate new and emerging techniques, and to better understand gear selectivity and effectiveness.

Table V-4-1. Designation of sampler types and description of fish sampling methods used by Ohio EPA (revised June 1, 1984).

Sampling Method Description	Sampler Type	Relative Abundance Based On	Data Collected #	Data Collected Wt[a]
Boat-mounted electro-fishing - *straight electrode array*	A	Per 1.0 km	X	X
Boat-mounted electro-fishing - *circular electrode array*	B	Per 1.0 km	X	X
Boat longline - riffle method)[b]	C	Per 0.3 km	X	X
Sportyak-generator unit	D	Per 0.3 km	X	X
Longline generator unit	E	Per 0.3 km	X	X
Back-pack electro-fishing - battery unit	F	Per 0.3 km	X	X
Backpack electrofishing-seine combination[c]	G	Per 0.3 km	X	
Seines[d]	H	Per 0.3 km	X	
Fyke net[d]	I	Per 24 hours	X	X
Hoop net[d]	J	Per 24 hours	X	X
Gill net[d]	K	Per 24 hours	X	X
Boat-mounted electro-fishing - *straight electrode array* NIGHT	N	Per 1.0 km	X	X
Boat mounted electro-fishing - *circular electrode array* NIGHT	M	Per 1.0 km	X	X
Reserved	L-Z[e]			

[a]Weight data is taken if modified Iwb is needed.

[b]Experimental method in conjunction with sampler type A.

[c]Discontinued method .

[d]Method is not suitable for calculating IBI or modified Iwb scores.

[e]These codes are available for methods developed in the future.

b) Pulsed D.C. Electrofishing Methods and Equipment:

Selection of the Appropriate Sampler Type

Selection of the appropriate sampler type is dependent upon the type of data needed, the type of macrohabitat being sampled, and the size and *depth* of the water body being sampled. This is a critical part of the sampling process since data quality essentially determines data applicability for the purposes of evaluating attainment of aquatic life uses. Thus it is important that the appropriate sampler type be used.

Boat electrofishing methods (sampler type A) are used in moderate to large sized streams and rivers where the use of wading methods are both impractical and less efficient. These include streams and rivers that have pools deep enough to accommodate the 12', 14', or 16' boats and equipment. Sites sampled with the boat methods are referred to as *boat sites*. The usual drainage area range of boat sites is 150 to more than 6000 sq. mi. although the 12' boat method has been used for sites as small as 75 sq. mi. where pool depths exceed 1.5 - 2.0 m and greater. This situation is the most frequently encountered in the Western Allegheny Plateau ecoregion (southeastern Ohio). The 12' electrofishing boat is the smallest of the boat-mounted devices and is used in moderate sized streams that generally cannot be navigated by the larger boats, usually 150 - 400 sq. mi. drainage area. The 14' and 16' electrofishing boats are used in larger rivers where near continuous navigation is possible (usually greater than 400 - 500 sq. mi.). The 18' boat electrofishing method is designed for use in the largest and deepest rivers, impoundments, and embayments. This boat employs either a straight (sampler type A) or circular (sampler type B) electrode array. Night electrofishing may be appropriate for the largest rivers (e.g., Ohio River, impounded sections of the Muskingum R.) where the drainage area exceeds 6000 - 7000 sq. mi. Depending on the electrode array used this method is termed sampler type N (straight array) or sampler type M (circular array).

Wading methods are used in smaller, wadable streams that cannot accommodate the boat methods due to the physical limitations of the stream channel. These are referred to as *wading sites* and range from the smallest headwater areas (<20 sq. mi. drainage area) to sites of 400 - 500 sq. mi. The Sportyak-generator method (sampler type D) is used in streams that range in size from 5-20 m in width and 0.5 - 1.0m in depth (average). There is a great deal of overlap in terms of drainage area between the sites where either the wading or boat sampler types may be most appropriate. The key factors in making the choice between these two methods is pool width and depth *and* access for the sampling equipment. The longline-generator method (sampler type E) is used in areas where the pools are separated by shallow riffles which make the use of the Sportyak method impractical. Both methods will sample the same site with equal efficiency. The backpack electrofishing method (sampler type F) is used in very shallow, small headwaters streams where the longline method is not necessary to secure an adequate sample. Streams that are more than five times the width of the anode net ring and more than twice the depth of the same should not be sampled with the backpack method (sampler type F). The seining methods (sampler types G and H) were used in the past, but have been discontinued by Ohio EPA. These sampler types are retained only to accommodate data generated by non-Ohio EPA entities and to make possible the use of historical data. Results generated by these latter methods (sampler types G and H) may not be suitable for determining aquatic life use attainment using the IBI and modified Iwb.

Selection of any of the previously described methods is based on the best professional judgement of the field crew leader and information gathered in a pre-survey

reconnaissance of the stream. Reconnaissance should take place during low-flow conditions if at all possible. Drainage area, stream length, and stream order are good physical indicators which aid in the selection of the appropriate sampling gear. Information to be collected during the reconnaissance includes the general width and depth of the stream, presence of riffles, dams, log jams and other impediments to navigation, access sites, and location of pollution sources and tributaries. All of these factors are used in choosing the appropriate sampler type(s).

General Cautions Concerning Field Conditions

Electrofishing should be conducted *only* during "normal" water flow and clarity conditions. What constitutes "normal" can vary from stream to stream. Generally "normal" water conditions in Ohio occur during *below* annual average river discharge levels. Under these conditions the surface of the water generally will have a "placid" appearance. Abnormally turbid conditions are to be avoided as are elevated flow and current. All of these adversely affect sampling efficiency and may rule out data applicability for calculating the modified Iwb and IBI. Since the ability of the netter to see stunned fish is critical, sampling should take place only during periods of "normal" water clarity and flow. Most Ohio surface waters have some background turbidity due to planktonic algae and suspended sediment and very few, if any, are entirely clear. Rainfall and subsequent runoff can cause increased turbidity due to the increased presence of suspended sediment (clays and silt). In most areas this imparts a light to medium brown coloration in the water. Floating debris such as sticks and other trash are usually obvious on the surface. Visibility under such conditions is seldom more than a few inches. Such conditions should be avoided and sampling should be delayed until the water returns to its "normal" clarity. High flow should be avoided for the obvious safety reasons, but this also reduces sampling efficiency. The boat methods are particularly affected as it becomes more difficult for the driver to maneuver the boat into areas of cover and current heterogeneity. These cautions apply to all of the electrofishing methods.

Boat Electrofishing Methods and Equipment

The boat methods (sampler types A and B) include the use of 12', 14', 16', and 18' john boats rigged for electrofishing. Equipment type, electrode design, and sampling methods follow the rationale and procedures outlined in Gammon (1973, 1976) and Novotny and Priegel (1974). Figure V-4-1 provides a diagrammatic description of the boat apparatus. A Smith-Root Type VI-A[1] or 3.5 GPP electrofishing unit[2] is used in the 12', 14', 16' and 18' boats. The Type VI-A unit rectifies 60HZ 240VAC (which is supplied by a 3500 or 4500 watt gasoline powered alternator) to pulsed DC. The pulse configuration consists of a triangular wave that can be adjusted to 60 or 120 pulses/second. Six voltage settings from 166 to 996 VDC in 166 volt increments are available. The voltage setting used in a particular situation is determined on a trial and error basis by increasing the voltage setting until a pulse width of 4-5 milliseconds produces an amperage reading of 8 amperes. In Ohio waters during June through October, relative conductivity values normally range from 300-600 umhos/cm. This generally results in a voltage selection of 336, 504, or 672 VDC. Conductivity values below this range may require higher voltage settings, whereas higher conductivity values may require lower voltage settings. The Smith-Root Model 3.5 GPP gas powered alternator and pulsator also delivers pulsed DC current. The pulse configuration consists of a fast rise, slow decay pulse which can be interrupted into 30, 60 or 120 pulses/second. The voltage range is continuously variable between 0-1000 volts and is adjusted by a percent-of-range rheostat to maintain the output amperage between 4 and 11 amps.

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[1] Use of product or company name does not signify endorsement.

[2] Smith-Root, Inc. 14014 N.E. Salmon Creek Ave., Vancouver, Washington 98665.

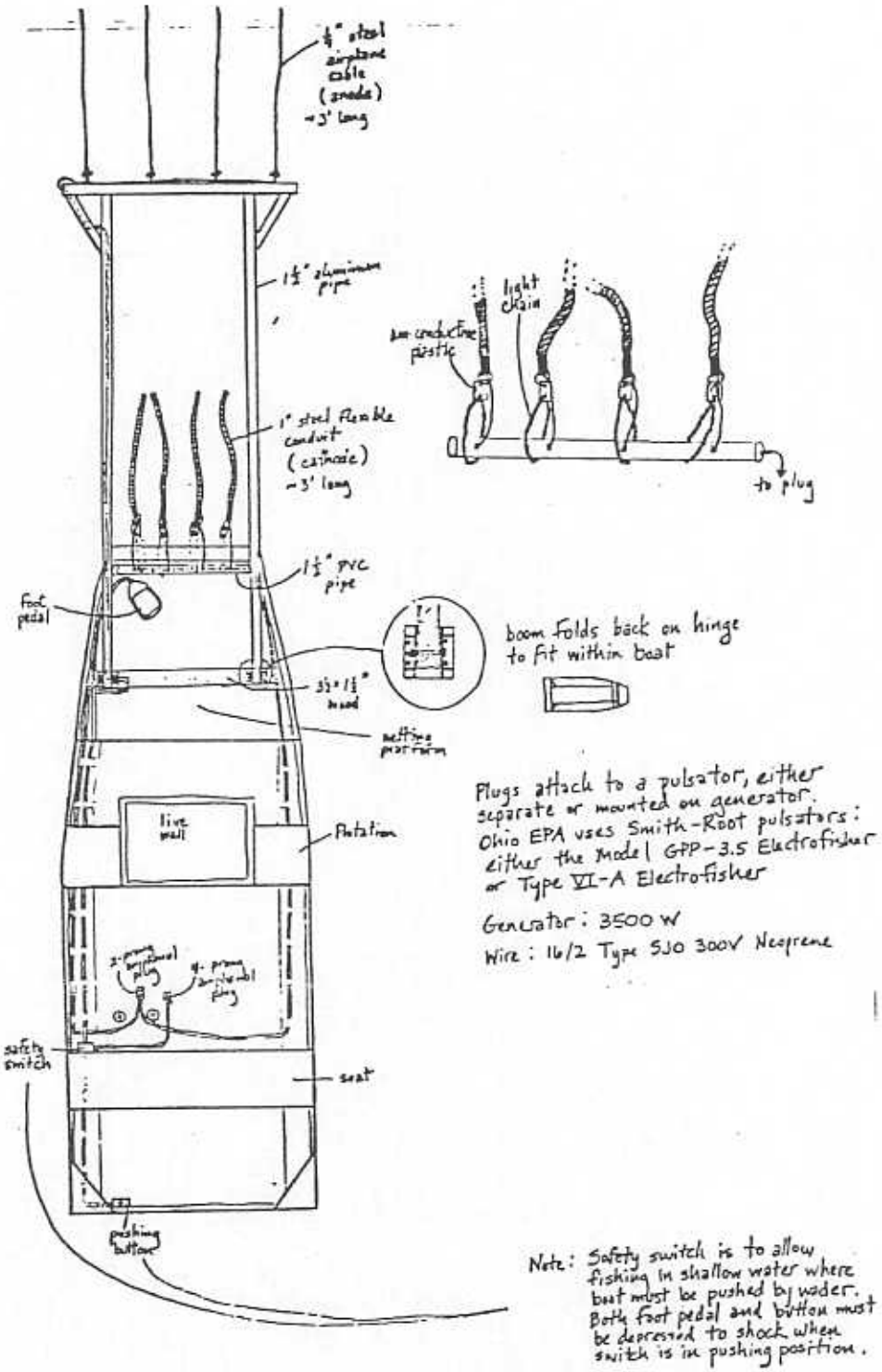


Figure V-4-1. Diagram of the boat electrofishing apparatus used by Ohio EPA to sample large river and stream fish communities.

The optimum range is selected on a trial and error basis by increasing the range until the indicator light flickers. Other comparable pulsed D.C. electrofishing units are acceptable for use as long as their performance is comparable to the aforementioned designs.

Pulsed DC current is transmitted through the water by an arrangement of anodes and cathodes suspended in the water from the boat. On the 12', 14' and 16' boats, four 32" long 1/4" diameter stainless steel aircraft cable anodes are hung from a retractable aluminum boom which extends in front of the boat. Boom length varies according to boat size and is approximately 3.05m on the 18' boat, 2.75m on the 16' boat, 2.15m on the 14' boat, and 2.0m on the 12' boat. Boom width varies from approximately 1.55 to 1.65m being wider on the larger boats. Four anodes are positioned on the front of the boom in a line perpendicular to the length of the boat. Four 64" lengths of 1" O.D. flexible galvanized steel conduit serve as cathodes, and are suspended directly from the bow in a line perpendicular to the length of the boat. The width of this array ranges from 0.75m on the 12' boat to 0.90m on the larger boats. Anodes and cathodes are replaced when damaged or worn. Safety equipment includes a positive pressure cut-off foot-pedal switch located on the bow deck and an emergency toggle cut-off switch adjacent to the stern seat. There is a magnetic-hydraulic circuit breaker on the Type VI-A electrofishing units.

For night electrofishing the equipment includes four 75 watt floodlamps attached to a guardrail which is mounted on the bow. These floodlamps are powered by 120 VAC produced by a separate gasoline powered generator.

A boat sampling crew consists of a *netter* and a *driver*. It is the netter's primary responsibility to capture all fish sighted; the driver's responsibility is to maneuver the boat as effectively as possible giving the netter the *best* opportunity to capture stunned fish (the driver may assist in netting stunned fish that appear at the rear or behind the boat). Both tasks are skill dependent with the boat maneuvering task requiring the most experience to gain adequate proficiency. Each sampling zone is fished in a downstream direction by slowly and steadily maneuvering the electrofishing boat as close to shore and submerged objects as possible by rowing or motoring. This may require *frequent* turning, backing, shifting (forward, reverse), changing speed, etc. in areas of moderate to extensive cover. The electrofishing boat is pushed on the transom by the driver when the water is too shallow to motor or row. A hand actuated positive pressure cut-off switch located on the inside of the transom is used during this procedure in addition to the bow foot-pedal switch. Both the netter and driver are clad in chest waders and rubber gloves. The netter also wears a jacket type personal flotation device. Safety equipment includes a positive pressure cut-off switch located on the bow deck and inside the transom.

Boat Sampling Site Selection

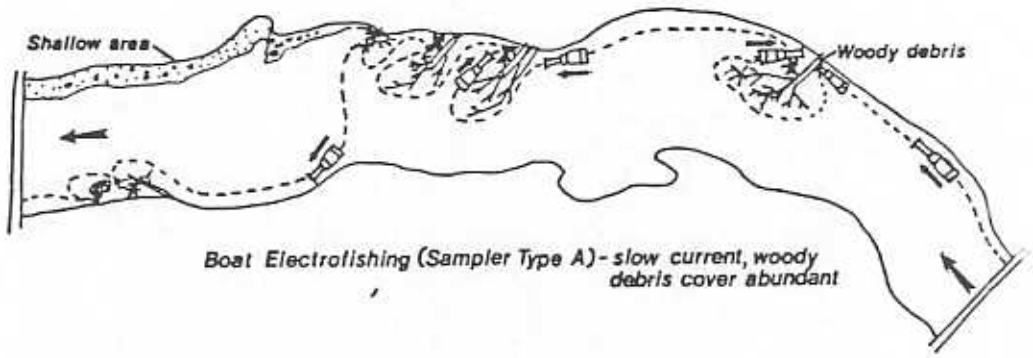
Sampling sites are selected along the shoreline with the most diverse macrohabitat features. This is generally along the gradual outside bends of the larger rivers but is not invariable. In free-flowing habitats part of each zone should include a run-type of habitat *if at all practical*. This of course is determined by the availability of such areas. Boat electrofishing zones generally measure 0.5 kilometers (km) in length, although shorter distances may be necessary in given instances. Distance is measured with a Topometric Products Limited (R) Hip Chain (preferred method) or a Ranging 620 optical rangefinder. Sampling sites are measured by securing the hip chain thread to a stationary object and then wading or motoring the length of the sampling zone. The length of the zone is then measured by the hip-chain counter. When using the optical rangefinder each zone is measured in increments approximating 50 m and accumulated to a distance of 0.5

km. This method is used only with boat methods where the use of the hip chain is impractical. Sampling site locations are verified on 7 1/2 minute USGS topographical maps. Hip chains and rangefinders are calibrated prior to being used in the field on a marked course and adjusted as necessary. The calibration results are recorded in a log book. Water depth in centimeters (cm) is determined to the nearest 10 cm at a minimum of ten locations in each zone with a marked dip net. The average depth is then recorded on the fish data sheet. The boundaries of each electrofishing zone are clearly marked on stationary objects (e.g. trees, bridge piers, etc.) with fluorescent orange paint. The starting point is marked with an arrow pointing in a downstream direction and the ending point is marked with a visible capital "E". This enables accurate relocation of the site on subsequent sampling dates. If the sampling zone is disjunct additional marks are necessary. If the zone stops and then resumes on the same bank then X marks where sampling stops and an arrow indicates where sampling resumes. If the zone switches banks then an arrow pointing skyward indicates the point to switch banks and an arrow pointing down on the opposite shore indicates where the zone resumes. The location of each sampling zone is indexed by river mile (using the river mile index contained in the Ohio EPA PEMSO RMI system) and marked on 7 1/2 minute USGS topographical maps for permanent reference.

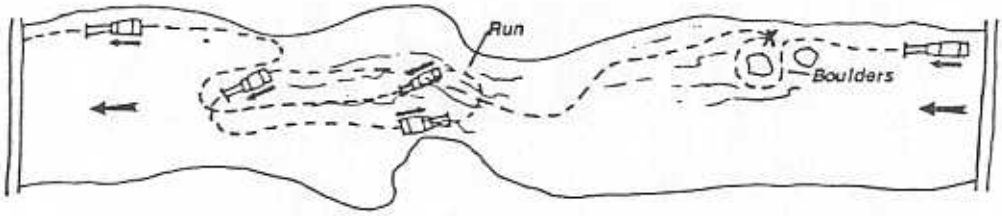
Boat Electrofishing Techniques

Each boat sampling zone is electrofished two or three times during the sampling season starting (whenever possible) at the farthest upstream zone and sampling sequentially downstream until one pass is completed. The remaining one or two sequential passes occur later in the sampling season. Sampling passes should take place at least three to four weeks apart for a three pass effort. If only two passes are planned, five to six weeks should elapse between individual sampling passes. Individual sampling zones are electrofished from upstream to downstream by slowly and steadily maneuvering the electrofishing boat as close to the shore and submerged objects as possible. It is absolutely critical to sample *carefully*, particularly at difficult sites where there is extensive woody debris *or* moderately fast to swift current. Figure V-4-2 provides a diagrammatic portrayal of how two different boat electrofishing zones should be sampled. In zones with extensive woody debris and slow current it is necessary to maneuver the boat in and out of the "pockets" of habitat formed by the debris. If the water depth approaches 100-200 cm it is usually necessary to "wait" for the fish to appear. In moderately fast or swift current it is necessary to conduct fast turns and maneuvers in order to put the netter in a good position to capture stunned fish. The efficiency is enhanced if the electrofishing boat and electric field can be kept moving downstream at a pace just slightly greater than the current velocity. Fish are usually oriented into the current and must either swim into the approaching electrical field or turn sideways to escape downstream. This latter movement presents an increased voltage gradient making the fish more susceptible to the electric current. It is often necessary to pass over the fast water sections of these zones twice. Also, portions of zones with continuous fast current can be effectively sampled by "backing" the boat downstream and occasionally pausing to allow the netter to capture stunned fish. The driver may need to assist with netting when large numbers of fish are stunned. Attempting to electrofish such fast water areas in an upstream direction *only* will greatly diminish sampling efficiency.

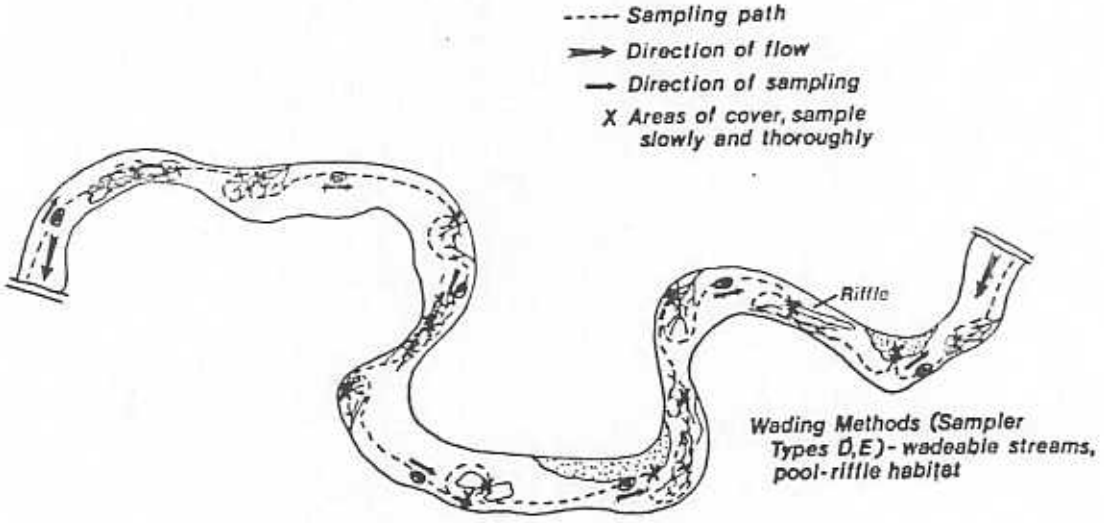
Although sampling is done according to zone length, the amount of time spent electrofishing each zone is an equally important consideration. Time fished can legitimately vary depending on the current, number of fish being collected, and amount and type of cover within a zone. However, there is a general *minimum* amount of time that should be spent sampling each boat zone.



Boat Electrofishing (Sampler Type A) - slow current, woody debris cover abundant



Boat Electrofishing (Sampler Type A) - moderate to fast current, swift runs, boulders



Wading Methods (Sampler Types D,E) - wadeable streams, pool-riffle habitat

Figure V-4-2. Diagramatic portrayal of proper boat electrofishing technique at two different river sampling locations and wading methods technique in a typical pool-run-riffle stream habitat.

Based on an analysis of 1187 electrofishing samples where time fished was compared to various catch results (lwb, numbers, weight, species) that are sensitive to the relative level of effort expended. Inspection of the results show that *at least* 1300 to 1600 seconds should be spent sampling any 0.5 km boat electrofishing zone. This time will likely increase to more than 2000 seconds in slower flowing zones that have numerous downed trees, logs, and other submerged structure. Moderately fast to swift flowing zones may also take longer to sample since the boat must be maneuvered back upstream to cover such areas thoroughly.

Netters are required to wear a pair of polarized sunglasses to facilitate seeing stunned fish in the water during each electrofishing run. An exception to this is with night sampling where sunglasses are not worn. A boat net with an 2.5m long handle and 7.62mm Atlas mesh knotless netting is used to capture stunned fish as they are attracted to the anode array and/or stunned. An effort is made to capture every fish sighted by both the netter and driver.

Captured fish are immediately placed in an on-board livewell for later processing. Water is replaced regularly in warm weather to maintain adequate dissolved oxygen levels in the water and to minimize mortality.

A field crew consists of a minimum of three persons (whenever possible), a boat driver, a netter, and a support vehicle driver. Limited access to most rivers and streams requires the electrofishing boat to be launched at an upstream point with a two person crew. The third crew member is responsible for maintaining contact with the electrofishing boat and meeting the boat at points downstream. Smaller rivers that are not continuously navigable are sampled by locating put-in-and-take-out access points at each sampling location.

The distance of stream or river covered per day is dependent upon the number of sampling zones and ease of navigation. Generally, four to seven zones can be sampled in a 10 to 20 mile segment each day. Relative abundance data collected with this method is expressed as numbers/km and weight (kg)/1.0 km.

The 18' electrofishing boat can be used with either a standard straight electrode array (sampler type A) or with a circular electrode array (sampler type B). The circular array is outfitted according to the specifications listed in Novotny and Priegel (1974). Anode configuration is circular and can be altered by adding or removing electrodes or changing the surface area exposure of each electrode depending on the conductivity of the water. Anodes are added in very low conductivity water less than (100-150 umhos) or removed in extremely high conductivity water greater than (900 umhos). These sampling methods are being tested in rivers where average sampling zone depth is consistently deeper than 150-200 cm (e.g. Lake Erie river mouths, lower Muskingum River, Ohio River, etc.) and in lakes, reservoirs, and impoundments. In these larger and deeper water bodies sampling is also conducted at night. Otherwise, sampling is conducted essentially the same as the methods just described for smaller rivers and streams.

Wading Electrofishing Methods and Equipment and Sampling Techniques

The Sportyak-generator wading method (Sampler type D) is used to sample smaller, wadable streams where access by a 12' john boat is not possible. The longline-generator (sampler type E) method is used in streams that are too shallow to sample with the Sportyak-generator method. The backpack electrofishing method (sampler type F) may be used in lieu of the longline-generator method in *only* the smallest headwaters streams following the restrictions that were previously stated. The Sportyak-generator

method (Sampler type D) employs a light, plastic boat with the capacity to carry a small portable generator/pulsator, and livewell. The Ohio EPA presently uses a 2.1 m Sportyak to carry a model 1736 DCV T&J combination generator/pulsator pulsed DC electrofishing unit and a 30 gallon plastic holding tank. The T&J electrofishing unit has the capability to supply 125 or 250 volts pulsed DC at a maximum of 1750 watts. At sites which have pool width and depth characteristics that suggest the need for the 12' boat method, but which is not accessible may require the use of the more powerful Smith-Root 3.5 GPP unit rigged for use with the Sportyak. This arrangement provides the additional power needed to efficiently sample pools that are consistently more than 1m deep and wider than 30-40m. A 15.2cm wide by approximately 45.7cm long stainless steel strip attached from the bow of the Sportyak acts as a cathode. Spring cord attached directly to the T&J unit supplies pulsed direct current to the anode. The anode is the net ring attached to a 1.8m long tubular fiberglass net pole. A positive pressure switch mounted on the net pole must be depressed to complete the switch circuit and allow electrical current to the electrodes (see Figure V-4-3 for a diagrammatic description).

Procedures for sampling require a two or three person crew, all wearing chest waders and rubber gloves. The primary netter operates the anode net ring while one crew member guides the Sportyak and the third crew member assists in capturing fish. This method is also diagrammed in Figure V-4-2. All habitat types are thoroughly sampled in an *upstream* direction for a distance of 150-200 m. The primary netter works the net ring beneath undercut banks, in and around brush piles, log jams, large boulders and other submerged structure. An effective technique for capturing fish under such objects is to thrust the anode ring into and under the structure with the current on and then *quickly* withdraw the anode ring in one swift motion. This has the effect of drawing fish out from under such structure making their capture possible. Sampling effort is usually concentrated on one side of the stream and some switching from one stream bank to the other may be necessary to sample all habitat types. In riffle and run areas the primary netter *rakes* the anode ring from upstream to downstream,allowing it to drift with the current. At the same time the assist netter blocks off an area downstream of the anode ring. This minimizes escape and avoidance of the electrical field by riffle species. When the holding tank is full of fish or sampling is completed the fish are processed (see Fish Counting and Weighing Procedures).

Sampling procedures for the longline method (Sampler type E) are similar. The longline-generator method uses the same electrofishing unit as the Sportyak method. The longline consists of 100 meters of heavily insulated 4-insulator wire. The anode is the net ring (as in the Sportyak method). The cathode is a floating aluminum plate attached 3m behind the net pole. The backpack electrofishing units (Sampler type F) used are a design supplied by the Michigan Department of Natural Resources[3] that produces 100 or 200 VDC (pulsed) or a Coeffelt Model BP-2 electrofishing unit[4] that produces a similar output. Both units are powered by a 12 VDC power source (motorcycle battery). The net ring serves as the anode and is attached to the end of a 1.8m net pole. A positive pressure switch mounted on the net pole is used to turn the unit on and off and as a safety switch. The cathode configuration on the Michigan DNR unit consists of a piece of copper that approximates 1000 cm^2. A 2.4m long section of 3.8mm plastic jacketed stainless steel cable with a 0.3m section exposed at the tip serves as the cathode for the Coeffelt unit. Both are trailed behind the backpack unit which is worn by the primary netter. Batteries are recharged daily and one charge is usually adequate for sampling one location, or 2-3 hours, whichever occurs first.

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[3]E. Schultz, P.O. Box 225, Grayling, Michigan 49738

[4] .Coeffelt Electronics Co. Inc., 2019 W. Union Ave., Englewood, Colo. 80110.

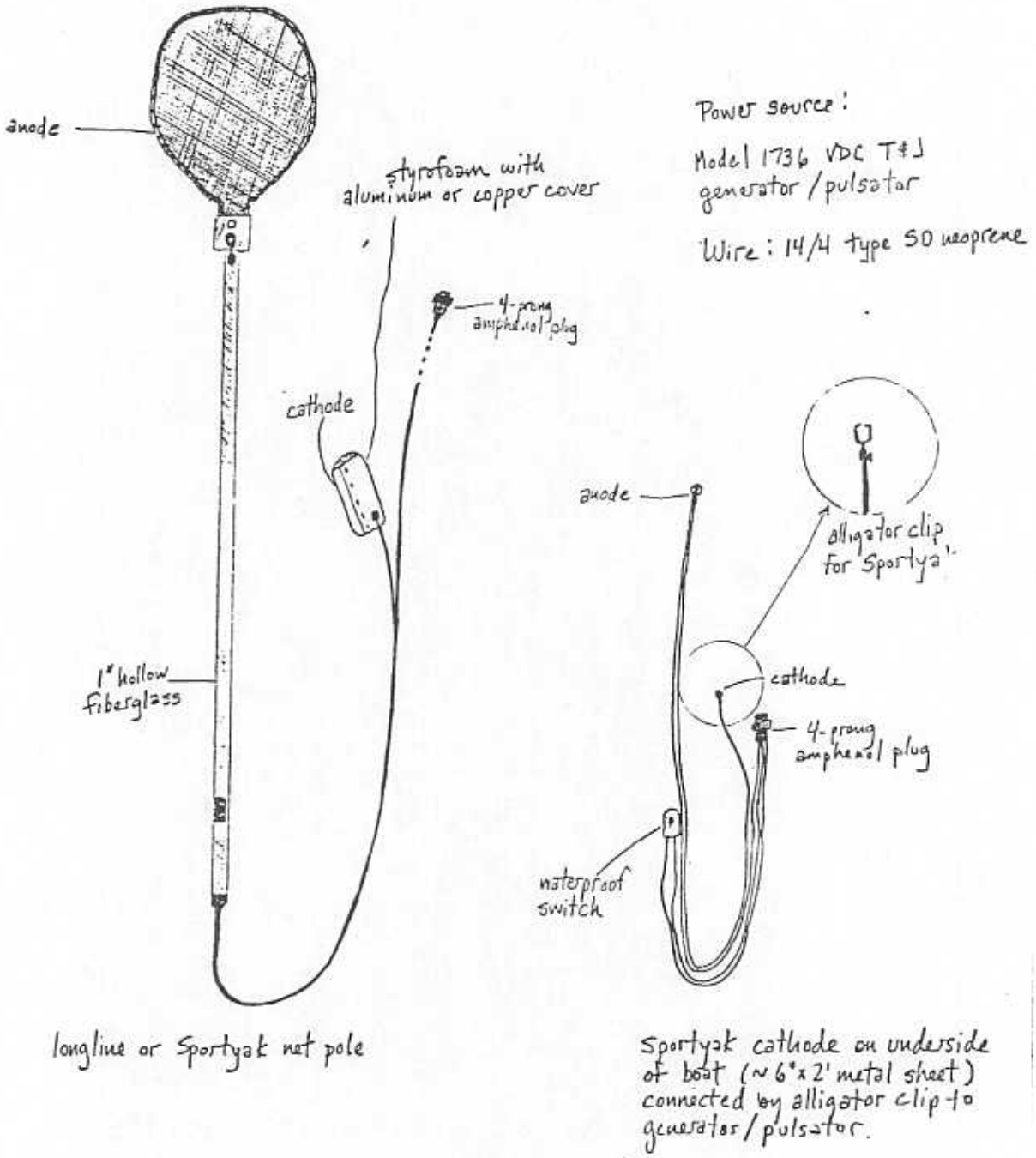

Figure V-4-3. Diagram of the net pole/electrode apparatus used with the Sportyak-generator and long-line electrofishing methods by Ohio EPA to stream fish communities.

Two or three individual sampling passes are preferred with the wading methods although one pass may be sufficient in small streams or certain non-complex situations. The number of passes affects how the catch data and biological indices are used to make environmental evaluations (Ohio EPA 1987). Relative abundance data is expressed in terms of numbers and weight (kg)/0.3km.

Seine Sampling Methods and Equipment and Sampling Techniques

The procedures and equipment used with the backpack electrofishing/seine methods (sampler type G) are generally the same as the backpack electrofishing method (sampler type F), except that seines are used in conjunction with the backpack electrofishing unit. This method was used to generate relative abundance data suitable for calculating the IBI in the years 1977-1981. The use of seines was discontinued in 1982 due to the relatively high degree of variability in the data caused by differing levels of skill between field crews. A detailed description of the methods can be found in earlier versions of this manual. While this method and seines alone may be used by non-Ohio EPA entities to generate fish relative abundance data it may not be acceptable to generate IBI or modified Iwb scores for aquatic life use attainment purposes. This will be evaluated on a case-by-case basis.

c) Passive Gear Methods and Equipment

Passive gear methods are those in which the sampling device is stationary and the capture of fish is dependent on their movements into the sampling device. These methods are not used on a routine basis by Ohio EPA and are considered experimental.

Four types of passive gear (fyke nets, trap nets, modified hoop nets, and gill nets) may be used to supplement boat electrofishing data in large rivers, estuaries, marshes, wetlands, lakes or impoundments. Fyke nets and trap nets are used in shallow water while modified hoop nets and gill nets are used in deep or open water.

Fyke nets (Sampler type I) are used in areas where a side channel can be completely blocked off by the two side leads which "funnel" fish into the net. Locations such as tributaries, marsh channels, or other channels off of the main channel are potential sampling sites. Fyke nets are set by anchoring the cod end just upstream of the channel confluence with the river, with the open end facing the main channel. The two side wings are angled toward the shoreline which blocks as much of the channel as possible. A center lead extends into the main channel helping to guide fish into the net. The Maine fyke net consists of a 4.5m body (11.4mm stretched mesh) supported by five square spring steel frames with three internal throats on the first three frames. Two 9m x 1.2 m wings and one 22.5m center lead are attached to the open end of the net. The cod end and all leads are anchored and floats attached to each anchor.

Trap nets (Sampler type J) are used to sample impoundments and wide river channels with slow velocity conditions. Trap nets are set in structurally complex areas where fish movement and density are anticipated to be highest in order to maximize net catches. One center lead is fastened to shore and the net is set perpendicular to the shore with the cod end anchored and marked with a float. Net dimensions are similar to those of the fyke net except a shorter 15m center lead is used. Modified hoop nets are used when sampling the deeper mid-channel areas. Modified hoop nets have been used to successfully capture fish moving upstream and downstream. By connecting two hoop nets together facing in opposite directions and placing them parallel to the flow, it is possible to discern fish movement in both the upstream and downstream directions. Modified hoop nets are set in

mid-channel parallel to the flow and anchored and marked with floats at both ends.

Gill nets (Sampler type K) are set in open water areas to sample fishes in large rivers, lakes, and impoundments where portions of the fish community are not accessible to shoreline electrofishing. Gill nets can be set at the surface, mid-depth, or on the bottom, depending on the objectives of the sampling and intended target species within the fish community. Gill nets are anchored in open water areas and marked with floats on both ends. Monofilament experimental gill nets are 37.5 m long with 7.5 m panels of 15.2mm, 22.9mm, 25.4mm, 40.6mm, and 50.8mm bar mesh.

All passive gear is checked and emptied 12 to 24 hours after setting. Standard procedures are used to process fish captured by passive gear. Data collected by passive gear can used to determine relative abundance which are expressed as numbers/24 hours and weight (kg)/24 hours. These results *have not* been used by Ohio EPA to calculate IBI and modified Iwb scores for aquatic life use attainment purposes.

3) Field Counting and Weighing Procedures

a) Handling Live Specimens

All sampling methods require placing captured fish in a livewell for processing when sampling each site is complete or when the livewell is full. Water in the livewell is changed as needed to minimize mortality of the captured fish. Fish are released immediately after they are identified *to species*, examined for external anomalies, and, if necessary, weighed. Efforts are made to minimize handling and holding times.

b) Field Identification

The majority of captured fish are identified to species in the field; however, *any* uncertainty about the field identification of individual fish *requires* their preservation for later laboratory identification (see **Part C**). Fish are preserved for future identification in borax buffered 10% formalin and labeled by date, river or stream, and river mile. Identification is required to the *species* level at a minimum and may be necessary to the sub-specific level in certain instances (*e.g.* banded killifish).

The collection techniques used may not be consistently effective for fish less than 15-20 mm in length, thus inclusion in the catch is not recommended. Also, Angermier and Karr (1986) and Angermier and Schlosser (1988) recommend that fish of this size (young-of-the-year) not be included in IBI calculations as they may unduly bias its function as a long-term aquatic ecosystem health measure. Ohio EPA supports this recommendation.

c) Weighing Procedures

For samples of species which are comprised entirely of one size class (*e.g.* adults, juveniles, young-of-the-year), two methods may be used. For larger species (*e.g.* carp, redhorse, most sunfish), where the adult fish are of a similar size, the catch may be weighed as separate individuals *or* in aggregate as a species. All results are recorded on the fish data sheet (Figure V-4-4). For catches with more than 15 individuals per species a *subsample* of 15 fish is weighed as individuals or in aggregate. If there is a *noticeable* variation in sizes between individual fish of a species individual weights should be taken using the subsampling technique if necessary. With smaller species (*e.g.* most minnows and darters) mass weighing in aggregate is recommended. If more than 50 individuals of a species comprise the catch a subsample of at least 50 fish is weighed and the remainder are counted. If extremely high numbers of a particular species are collected and the fish are of a relatively uniform size, the number of individuals may be determined by mass weighing all fish collected and extrapolating the numbers from a counted

Figure V-4-4. Ohio EPA fish field data sheet.

Fish Data Sheet

Collector Recorder

Field Crew: ______ Time of Day: ______ Page ___ of ___

River/Stream: ______ Location: ______

Date: ______

River Code: ______ Sampler Type: ____ Time Fished: ' ___ " ___ Total Seconds ______

RM: ______ Depth: ______ Observed Flow: ______

Distance: ______ Data Source: ______ Number of Species: ______

Anomalies: A-anchor worm; B-black spot; C-leeches; D-deformities; E-eroded fins, F-fungus; L-lesions; M-multiple DELT anomalies, N-blind, P-parasites, Y-popeye; S-emaciated; W-swirled scales; T-tumors; Z-other/. [H-Heavy; L-Light are combined with anomalies A, B, and C]

SPECIES	# WEIGHED	TOTAL COUNTED	WEIGHT (GRAMS)					ANOMALIES					

OhioEPA

EPA 4508

Mass Weighing Convention: Total Weight (g) — 536 (12) — Number Weighed

and weighed *subsample*.

Samples that are comprised of two distinct size classes of fish (*e.g.* adults and juveniles) of a species are processed as two, separate size groups. Adults and juveniles are recorded separately on the fish data sheet by adding an "A" to the species code for adults and a "B" for juveniles. For example, if both adult and juvenile white suckers occur in the same sample the adult numbers and weights are recorded as family-species code 40-016A with juvenile numbers and weights recorded as 40-016B. Although each is listed separately on the fish data sheet they are treated as a subsample of the same species in any subsequent data analyses. The FINS (Fish Information System) programs are designed to calculate relative numbers and weight data based on the input of the weighted subsample data.

Individual fish weighing less than 1000 grams (g) are weighed to the nearest 1g on a Homs 1000 spring dial scale (1000g capacity x 2g intervals). Fish weighing more than 1000g are weighed to the nearest 25g on a Universal Accu-weigh spring dial scale M1250 (with air dash pot; 12000 g capacity in 50 g increments). All scales are checked once each week with National Bureau of Standards Class F check weights (up to 2000g in 1g increments) and adjusted as necessary.

d) External Anomalies

All fish that are weighed whether done individually, in aggregate, or subsampled (only the fish that are actually weighed) are examined for the presence of gross external anomalies and their occurrence is recorded on the fish data sheet (Figure V-4-4) and subsequently entered into FINS. In order to standardize the procedure for counting and identifying anomalies the following criteria should be followed.

All fish that are *weighed* are examined for gross external anomalies. These are anomalies that are visible to the naked eye when the fish are captured, identified, sorted, weighed and counted. Table V-4-2 lists the types of anomalies which are recorded on the fish data sheet and subsequently entered into FINS. 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 V-4-2). An external anomaly is defined as the presence of externally visible skin or subcutaneous disorders, and is expressed as percent (weighted) of affected fish among all fish *weighed.* This is computed for each type of anomaly for each species in each sample. It is computed as a weighted number (i.e. based on percent incidence among weighed fish times the total number of that fish species in the sample). Then the total percent anomalies for a specific type of anomaly or group of anomalies can be calculated for one 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 sheet (Fig. V-4-4).

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* sp.), infections, and protozoan parasites (e.g. *Myxosoma cerebalis*; 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 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 D.O. or high temperatures in combination with chronic toxicity (Pippy and Hare 1969; Sniezko 1962).

Table V-4-2. Codes utilized to record external anomalies on fish.

Anomaly Code	Description
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 and tumors, etc.) on the same individual fish.
AL	Anchor worm - Light infestation: fish with five or fewer attached worms and/or previous attachment sites.
AH	Anchor worm - Heavy infestation: fish with six 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 diameter of the eye.
CL	Leeches - Light infestation: fish with five or fewer attached leeches and/or previous attachment sites.
CH	Leeches - Heavy infestation: fish with six or more attached leeches and/or previous attachment sites.
F	Fungus.
I	Ich.
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).
Y	Popeye disease.
W	Swirled scales.
Z	Other, not included above.

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, characteristic sores left by anchor worms and leeches should not be included unless they are enlarged by this infection. Obvious injuries, however, 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 shasta*; Post 1983) may cause tumor like masses, but these *should not* be considered as tumors. Parasite masses can be squeezed and broken between the thumb and forefinger whereas true tumors are firm and not easily broken (P. Baumann, pers. comm.).

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 former 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 on fish in Ohio streams 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, two 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 are 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 two suckers (one on each end) and the ability to contract or elongate their body. They may occur almost anywhere on the external surface of fish, but are most frequently seen on the anterioventral surface of bullheads (*Ictalurus* sp.). 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* - This 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 fishes 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 a 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 urged 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.

4) Fish Sampling Site Habitat Evaluation: Qualitative Habitat Evaluation Index (QHEI)

A general evaluation of macrohabitat is made by the *fish field crew leader* while sampling each location using the Ohio EPA Site Description Sheet - Fish (Figure V-4-5). This form is used to tabulate data and information for calculating the **Qualitative Habitat Evaluation Index (QHEI)**. The following guidance should be used when completing the site evaluation form.

Figure V-4-5. Front side of the Ohio EPA Site Description Sheet for evaluating the geographical and physical characteristics of fish sampling locations. This is used to record information for the calculation of the Qualitative Habitat Evaluation Index (QHEI).

Ohio EPA Site Description Sheet - Fish

QHEI SCORE: ☐

Stream_____ RM_____ Date_____ River Code_____

Location_____ Crew:_____

1] SUBSTRATE (Check *ONLY* Two Substrate *TYPE BOXES; Check all types present*); **SUBSTRATE SCORE:** ☐

TYPE	POOL	RIFFLE	TYPE	POOL	RIFFLE
☐☐-BLDER /SLABS[10]	____	____	☐☐-GRAVEL [7]	____	____
☐☐-BOULDER [9]	____	____	☐☐-SAND [6]	____	____
☐☐-COBBLE [8]	____	____	☐☐-BEDROCK[5]	____	____
☐☐-HARDPAN [4]	____	____	☐☐-DETRITUS[3]	____	____
☐☐-MUCK [2]	____	____	☐☐-ARTIFIC.[0]	____	____

Substrate Origin (Check all): ☐-LIMESTONE [1] ☐-RIP/RAP [0] ☐-TILLS [1] ☐-HARDPAN [0] ☐-SANDSTONE [0] ☐-SHALE [-1]

Silt Cover (Check One): ☐-SILT HEAVY [-2] ☐-SILT MODERATE [-1] ☐-SILT NORMAL [0] ☐-SILT FREE [1]

Extent Of Embeddness (Check One): ☐—EXTENSIVE [-2] ☐—MODERATE[-1] ☐—LOW[0] ☐—NONE[1]

TOTAL NUMBER OF SUBSTRATE TYPES: ☐> 4 [2] ☐— <= 4 [0] ☐-COAL FINES [-2]

NOTE: (Ignore sludge that originates from point-sources; score is based on natural substrates)

COMMENTS_____

2] INSTREAM COVER **COVER SCORE:** ☐

TYPE (Check *All* That Apply)

☐-UNDERCUT BANKS [1] ☐-DEEP POOLS [2] ☐-OXBOWS [1]
☐-OVERHANGING VEGETATION [1] ☐-ROOTWADS [1] ☐-AQUATIC MACROPHYTES [1]
☐-SHALLOWS (IN SLOW WATER) [1] ☐-BOULDERS [1] ☐-LOGS OR WOODY DEBRIS [1]

AMOUNT(Check *ONLY* One or check 2 and *AVERAGE*)

☐ - EXTENSIVE > 75% [11]
☐ - MODERATE 25-75% [7]
☐ - SPARSE 5-25% [3]
☐ - NEARLY ABSENT < 5% [1]

COMMENTS:_____

3] CHANNEL MORPHOLOGY: (Check *ONLY* One PER Category OR check 2 and *AVERAGE*) **CHANNEL:** ☐

SINUOSITY	DEVELOPMENT	CHANNELIZATION	STABILITY	MODIFICATIONS/OTHER	
☐ - HIGH [4]	☐ - EXCELLENT [7]	☐ - NONE [6]	☐ - HIGH [3]	☐ - SNAGGING	☐ - IMPOUND.
☐ - MODERATE [3]	☐ - GOOD [5]	☐ - RECOVERED [4]	☐ - MODERATE [2]	☐ - RELOCATION	☐ - ISLANDS
☐ - LOW [2]	☐ - FAIR [3]	☐ - RECOVERING [3]	☐ - LOW [1]	☐ - CANOPY REMOVAL	☐ - LEVEED
☐ - NONE [1]	☐ - POOR [1]	☐ - RECENT OR NO RECOVERY [1]		☐ - DREDGING	☐ - BANK SHAPING
				☐ - ONE SIDE CHANNEL MODIFICATIONS	

COMMENTS:_____

4] RIPARIAN ZONE AND BANK EROSION - (check ONE box per bank or check 2 and AVERAGE per bank) **RIPARIAN:** ☐

River Right Looking Downstream

RIPARIAN WIDTH L R (Per Bank)	EROSION/RUNOFF - FLOOD PLAIN QUALITY L R (Most Predominant Per Bank)	L R (Per Bank)	BANK EROSION
☐☐-WIDE>50m [4]	☐☐-FOREST, SWAMP [3]	☐☐-URBAN OR INDUSTRIAL[0]	☐ ☐-NONE OR LITTLE [3]
☐☐-MODERATE 10-50 [3]	☐☐-OPEN PASTURE/ ROWCROP[0]	☐☐-SHRUB OR OLD FIELD[2]	☐ ☐-MODERATE.[2]
☐☐-NARROW 5-10m [2]	☐☐- RESID.,PARK,NEW FIELD [1]	☐☐-CONSERV. TILLAGE [1]	☐ ☐-HEAVY OR SEVERE[1]
☐☐-VERY NARROW 1-5m [1]	☐☐-FENCED PASTURE [1]	☐☐-MINING/CONSTRUCTION [0]	
☐☐-NONE[0]			

COMMENTS:_____

POOL/GLIDE AND RIFFLE/RUN QUALITY **POOL:** ☐

MAX. DEPTH (Check 1)	MORPHOLOGY (Check 1)	POOL/RUN/RIFFLE CURRENT VELOCITY (Check *All* That Apply)	
☐- >1m [6]	☐-POOL WIDTH > RIFFLE WIDTH [2]	☐-TORRENTIAL[-1]	☐-EDDIES[1]
☐- 0.7-1m [4]	☐-POOL WIDTH = RIFFLE WIDTH [1]	☐-FAST[1]	☐-INTERSTITIAL[-1]
☐- 0.4-0.7m [2]	☐-POOL WIDTH < RIFFLE W. [0]	☐-MODERATE [1]	☐-INTERMITTENT[-2]
☐- < 0.4m [1]		☐-SLOW [1]	
☐—<0.2m [Pool = 0]			

☐-NO POOL[0]

COMMENTS:_____

RIFFLE: ☐

RIFFLE/RUN DEPTH	RIFFLE/RUN SUBSTRATE	RIFFLE/RUN EMBEDDEDNESS	
☐ - GENERALLY >10 cm,MAX>50 [4]	☐-STABLE (e.g.,Cobble, Boulder) [2]	☐-EXTENSIVE [-1]	☐-MODERATE[0]
☐ - GENERALLY >10 cm,MAX<50 [3]	☐-MOD. STABLE (e.g.,Pea Gravel) [1]	☐-LOW. [1]	☐-NONE[2]
☐ - GENERALLY 5-10 cm [1]	☐-UNSTABLE (Gravel,Sand) [0]		
☐ - GENERALLY < 5 cm [Riffle = 0]			

☐-NO RIFFLE[0]

COMMENTS_____ **GRADIENT:** ☐

6] Gradient (feet/mile): _____ %POOL:_____ %RIFFLE:_____ %RUN:_____

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Geographical Information

1) *Stream, River Mile (RM), Date* – The official stream name may be found in the Gazetteer of Ohio Streams (Ohio DNR, 1960) or on USGS 7.5 minute topographic maps. If the stream is unnamed, a name and stream code is assigned by the *Surface Water Section Database Coordinator*. Usually the name of a nearby landmark is used for the stream name. A basin-river code is also assigned from the FINS river code system. The River Mile (RM) designations used are found on 7.5 minute topo maps stored at the Ohio EPA, Office of Planning, 1800 WaterMark Drive (PEMSO RMI maps), one of five Ohio EPA District offices (maps for that district), and Ohio EPA, Division of Water Quality Monitoring Assessment laboratory at 1030 King Avenue.

2) Specific Location

A brief description of the sampling location should include proximity to a local landmark such as a bridge, road, discharge outfall, railroad crossing, park, tributary, dam, etc.

3) Field Sampling Crew

The field crew involved with the sampling is noted on the sheet with the person who filled out the sheet listed first. QHEI information is to be completed by the crew leader *only*.

*4) Habitat Characteristics: **QHEI** Metrics*

The **Qualitative Habitat Evaluation Index (QHEI)** is a physical habitat index designed to provide an empirical, quantified evaluation of the general lotic *macrohabitat* characteristics that are important to fish communities. A detailed analysis of the development and use of the QHEI is available in Rankin (1989). The **QHEI** is composed of six principal metrics each of which are described below. The maximum possible **QHEI** site score is 100. Each of the metrics are scored individually and then summed to provide the total **QHEI** site score. This is completed at least once for each sampling site during each year of sampling. An exception to this convention would be when substantial changes to the macrohabitat have occurred between sampling passes.Standardized definitions for pool, run, and riffle habitats, for which a variety of existing definitions and perceptions exist, are essential for accurately using the **QHEI**. For consistency the following definitions are taken from Platts *et al.* (1983). It is recommended that this reference also be consulted prior to scoring individual sites.

Riffle and Run Habitats:

Riffle - areas of the stream with fast current velocity and shallow depth; the water surface is visibly broken.

Run - areas of the stream that have a rapid, non-turbulent flow; runs are deeper than riffles with a faster current velocity than pools and are generally located downstream from riffles where the stream narrows; the stream bed is often flat beneath a run and the water surface is not visibly broken.

Pool and Glide Habitats:

Pool[5] - an area of the stream with slow current velocity and a depth greater than riffle and run areas; the stream bed is often concave and stream width frequently is the greatest; the water surface slope is nearly zero.

Glide - this is an area common to most modified stream channels that do not have distinguishable pool, run, and riffle habitats; the current and flow is similar to that of a canal; the water surface gradient is nearly zero.

The following is a description of each of the six **QHEI**

[5]If a pool or glide has a maximum depth of less than 20 cm, it is deemed to have lost its functionality and the metric is scored a 0.

metrics and the individual metric components. Guidelines on how to score each is presented. Generally, metrics are scored by checking boxes. In certain cases the biologist completing the **QHEI** sheet may interpret a habitat characteristic as being intermediate between the possible choices; in cases where this is allowed (denoted by the term **"Double-Checking"**) two boxes may be checked and their scores averaged.

Metric 1: Substrate

This metric includes two components, *substrate type* and *substrate quality*.

Substrate type

Check the two most common substrate types in the stream reach. If one substrate type predominates (greater than approximately 75-80% of the bottom area *OR* what is clearly the most *functionally* predominant substrate) then this substrate type should be checked twice. **DO NOT CHECK MORE THAN TWO BOXES**. *Note the category for artificial substrates.* Spaces are provided to note the presence (by check marks, or estimates of % if time allows) of *all* substrate types present in pools and riffles that each comprise at least 5% of the site (*i.e.*, they occur in sufficient quantity to support species that may commonly be associated with the habitat type). This section must be filled out completely to permit future analyses of this metric. If there are more than four substrate types in the zone that are present in greater than approximately 5% of the sampling area check the appropriate box.

Substrate quality

Substrate *origin* refers to the "parent" material that the stream substrate is derived from. Check **ONE** box under the substrate origin column *unless* the parent material is from multiple sources (*e.g.*, limestone and tills). **Embeddedness** is the degree that cobble, gravel, and boulder substrates are surrounded, impacted in, or covered by fine materials (sand and silt). Substrates should be considered embedded if >50% of surface of the substrates are embedded in fine material. Embedded substrates cannot be easily dislodged. This also includes substrates that are concreted or "armour-plated". Naturally sandy streams are not considered embedded; however, a sand predominated stream that is the result of anthropogenic activities that have buried the natural coarse substrates is considered embedded. Boxes are checked for *extensiveness* (area of sampling zone) of the embedded substrates as follows: **Extensive** — > 75% of site area, **Moderate** — 50-75%, **Sparse** — 25-50%, **Low** — < 25%.

Silt Cover is the extent that substrates are covered by a silt layer (*i.e.*, more than 1 inch thickness). **Silt Heavy** means that nearly all of the stream bottom is layered with a deep covering of silt. **Moderate** includes extensive coverings of silts, but with some areas of cleaner substrate (*e.g.*, riffles). **Normal** silt cover includes areas where silt is deposited in small amounts along the stream margin *or* is present as a "dusting" that appears to have little functional significance. If substrates are exceptionally clean the **Silt Free** box should be checked.

Substrate types are defined as:

a) *Bedrock* - solid rock forming a continuous surface.
b) *Boulder* - rounded stones over 256 mm in diameter(10 in.) or large "slabs" more than 256 mm in length (*Boulder slabs*).
c) *Cobble* - stones from 64-256 mm (2 1/2 - 10in.) in diameter.
d) *Gravel* - mixture of rounded course material from 2-64 mm (1/12 - 2 1/2 in.) in diameter.
e) *Sand* - materials 0.06 - 2.0 mm in diameter, gritty texture when rubbed between fingers.

f) *Silt* - 0.004 - 0.06 mm in diameter, generally this is fine material which feels "greasy" when rubbed between fingers.

g) *Hardpan* - particles less than 0.004 mm in diameter, usually clay, which forms a dense, gummy surface that is difficult to penetrate.

h) *Marl* - calcium carbonate; usually greyish-white; often contains fragments of mollusc shells.

i) *Detritus* - dead, unconsolidated organic material covering the bottom which could include sticks, wood and other partially or undecayed coarse plant material.

j) *Muck* - black, fine, flocculent, completely decomposed organic matter (*does not include* sewage sludge).

k) *Artificial* - substrates such as rock baskets, gabions, bricks, trash, concrete etc., placed in the stream for reasons *OTHER* than habitat mitigation

Sludge is defined as a thick layer of organic matter, that is decidedly of human or animal origin. **NOTE: SLUDGE THAT ORIGINATES FROM POINT SOURCES IS NOT INCLUDED; THE SUBSTRATE SCORE IS BASED ON THE UNDERLYING MATERIAL.**

Substrate Metric Score:

Although the theoretical maximum metric score is > 20 the maximum score allowed for the **QHEI** is limited to **20 points.**

Metric 2: Instream Cover

This metric consists of *instream cover type* and *instream cover amount.* All of the cover types that are present in greater than approximately 5% of the sampling area (i.e., they occur in sufficient quantity to support species that may commonly be associated with the habitat type) should be checked. Cover should not be counted when it is in areas of the stream with insufficient depth (usually < 20 cm) to make it useful. For example a logjam in 5 cm of water contributes very little if any cover, and at low flow may be dry. Other cover types with limited utility in shallow water include *undercut banks* and *overhanging vegetation*, *boulders*, and *rootwads*. Under *amount*, one or two boxes may be checked. *Extensive* cover is that which is present throughout the sampling area, generally greater than about 75% of the stream reach. Cover is *moderate* when it occurs over 25-75% of the sampling area. Cover is *sparse* when it is present in less than 25% of the stream margins (sparse cover usually exists in one or more isolated patches). Cover is *nearly absent* when no large patch of any type of cover exists anywhere in the sampling area. This situation is usually found in recently channelized streams or other highly modified reaches (*e.g.* ship channels). If cover is thought to be intermediate in amount between two categories, *check two boxes and* ***average*** *their scores.* Cover types include: 1) undercut banks, 2) overhanging vegetation, 3) shallows (in slow water), 4) logs or woody debris, 5) deep pools (> 70 cm), 6) oxbows, 7) boulders, 8) aquatic macrophytes, and 9) rootwads (tree roots that extend into stream). Do not check undercut banks AND rootwads unless undercut banks exist *along with* rootwads as a major component.

Cover Metric Score:

Although the theoretical maximum score is > 20 the maximum score assigned for the QHEI for the instream cover metric is limited to **20 points**

Metric 3: Channel Morphology

This metric emphasizes the quality of the stream channel that relates to the creation and stability of macrohabitat. It includes channel sinuosity (*i.e.* the

degree to which the stream meanders), channel development, channelization, and channel stability. One box under each should be checked unless conditions are considered to be intermediate between two categories; in these cases *check two boxes and* ***average*** *their scores.*

a) *Sinuosity* - **No** sinuosity is a straight channel. **Low** sinuosity is a channel with only 1 or 2 poorly defined outside bends in a sampling reach, or perhaps slight meandering within modified banks. **Moderate** sinuosity is more than 2 outside bends, with at least one bend well defined. **High** sinuosity is more than 2 or 3 well defined outside bends with deep areas outside and shallow areas inside. Sinuosity may be more conceptually described by the ratio of the stream distance between two points on the channel of a stream and the straight-line distance between these same two points, taken from a topographic map. Check *only* one box.

b) *Development* - This refers to the development of riffle/pool complexes. **Poor** means *riffles* are absent, or if present, shallow with sand and fine gravel substrates; *pools*, if present are shallow. Glide habitats, if predominant, receive a **Poor** rating. **Fair** means riffles are poorly developed or absent; however, pools are more developed with greater variation in depth. **Good** means better defined riffles present with larger substrates (gravel, rubble or boulder); pools have variation in depth and there is a distinct transition between pools and riffles. **Excellent** means development is similar to the Good category except the following characteristics must be present: pools must have a maximum depth of >1m and deep riffles and runs (>0.5m) must also be present. In streams sampled with wading methods, a sequence of riffles, runs, and pools must occur more than once in a sampling zone. Check *one* box.

c) *Channelization* - This refers to anthropogenic channel modifications. **Recovered** refers to streams that have been channelized in the past, but which have recovered most of their natural channel characteristics. **Recovering** refers to channelized streams which are still in the process of regaining their former, natural characteristics; however, these habitats are still degraded. This category also applies to those streams, especially in the Huron/Erie Lake Plain ecoregion (NW Ohio), that were channelized long ago and have a riparian border of mature trees, but still have **Poor** channel characteristics. **Recent** or **No Recovery** refers to streams that were recently channelized or those that show no significant recovery of habitats (*e.g.* drainage ditches, grass lined or rock rip-rap banks, etc.). The specific type of habitat modification is checked in the last two columns but not scored.

d) *Stability* - This refers to channel stability. Artificially stable (concrete) stream channels receive a **High** score. Even though they are generally a negative influence on fish the negative effects are related to features other than their stability. Channels with **Low** stability are usually characterized by fine substrates in riffles that often change location, have unstable and severely eroding banks, and a high bedload that slowly creeps downstream. Channels with **Moderate** stability are those that appear to maintain stable riffle/pool and channel characteristics, but which exhibit some symptoms of instability, *e.g.* high bedload, eroding or false banks, or show the effects of wide fluctuations in water level. Channels with **High** stability have stable banks and substrates, and little or no erosion and bedload.

e) *Modifications/Other* - Check the appropriate box if

impounded, islands present, or leveed (these are not included in the QHEI scoring) as well as the appropriate source of habitat modifications.

The maximum QHEI metric score for Channel Morphology is **20 points**.

Metric 4: Riparian Zone and Bank Erosion

This metric emphasizes the quality of the riparian buffer zone and quality of the floodplain vegetation. This includes riparian zone width, floodplain quality, and extent of bank erosion. Each of the three components require scoring the left *and* right banks (looking downstream). The *average* of the left and right banks is taken to derive the component value. One box per bank should be checked unless conditions are considered to be intermediate between two categories; in these cases *check two boxes and* ***average*** *their scores.*

a) *Width of the Floodplain* - This is the width of the riparian (stream side) vegetation. Width estimates are only done for forest, shrub, swamp, and old field vegetation. Old field refers to the a fairly mature successional field that has stable, woody plant growth; this generally does not include weedy urban or industrial lots that often still have high runoff potential. Two boxes, one each for the left and right bank (looking downstream), should be checked and then averaged.

b) *Floodplain Quality* - The two most predominant floodplain quality types should be checked, one each for the left and right banks (includes urban, residential, etc.), and then averaged. By floodplain we mean the areas *immediately outside* of the riparian zone *or greater than 100 feet from the stream*, whichever is wider on each side of the stream. These are areas adjacent to the stream that can have direct runoff and erosional effects during normal wet weather. We do not limit it to the riparian zone and it is much less encompassing than the stream basin.

c) *Bank Erosion* - The following Streambank Soil Alteration Ratings from Platts *et al.* (1983) should be used; check one box for each side of the stream and average the scores. False banks are used in the sense of Platts *et al.* (1983) to mean banks that are no longer adjacent to the normal flow of the channel but have been moved back into the floodplain most commonly as a result of livestock trampling.

1) **None** - streambanks are stable and not being altered by water flows or animals (e.g. livestock) - Score **3**.
2) **Little** - streambanks are stable, but are being lightly altered along the transect line; less than 25% of the streambank is receiving any kind of stress, and if stress is being received it is very light; less than 25% of the streambank is false, broken down or eroding - Score **3**.
3) **Moderate** - streambanks are receiving moderate alteration along the transect line; at least 50 percent of the streambank is in a natural stable condition; less than 50% of the streambank is false, broken down or eroding; false banks are rated as altered - Score **2**.
4) **Heavy** - streambanks have received major alterations along the transect line; less than 50% of the streambank is in a stable condition; over 50% of the streambank is false, broken down, or eroding - Score **1**.
5) **Severe** - streambanks along the transect line are severely altered; less than 25% of the streambank is in a stable condition; over 75% of the streambank is false, broken down, or eroding - Score **1**.

The maximum score for Riparian Zone and Erosion metric is **10 points**.

Metric 5: Pool/Glide and Riffle-Run Quality

This metric emphasizes the quality of the pool, glide and/or riffle-run habitats. This includes pool depth, overall diversity of current velocities (in pools *and* riffles), pool morphology, riffle-run depth, riffle-run substrate, and riffle-run substrate quality.

A) **Pool/Glide Quality**

1) *Maximum depth of pool or glide*; check one box only (Score **0** to **6**). Pools or glides with maximum depths of *less than* 20 cm are considered to have lost their function and the *total metric* is scored a **0**. No other characteristics need be scored in this case.

2) *Current Types* - check each current type that is present in the stream (including riffles and runs; score - **2** to **4**), definitions are:

Torrential - extremely turbulent and fast flow with large standing waves; water surface is very broken with no definable, connected surface; usually limited to gorges and dam spillway tailwaters.

Fast - mostly non-turbulent flow with small standing waves in riffle-run areas; water surface may be partially broken, but there is a visibly connected surface.

Moderate - non-turbulent flow that is detectable and visible (i.e. floating objects are readily transported downstream); water surface is visibly connected.

Slow - water flow is perceptible, but very sluggish.

Eddies - small areas of circular current motion usually formed in pools immediately downstream from riffle-run areas.

Interstitial - water flow that is perceptible only in the interstitial spaces between substrate particles in riffle-run areas.

Intermittent - no flow is evident anywhere leaving standing pools that are separated by dry areas.

4) *Morphology* - Check *Wide* if pools are wider than riffles, *Equal* if pools and riffles are the same width, and *Narrow* if the riffles are wider than the pools (Score **0** to **2**). If the morphology varies throughout the site *average* the types. If the entire stream area (including areas outside of the sampling zone) is pool or riffle, then check riffle = pool.

Although the theoretical maximum score is > 12 the maximum score assigned for the QHEI for the Pool Quality metric is limited to **12 points.**

B) **Riffle-Run Quality**

(score **0** for this metric if no riffles are present)

1) *Riffle/Run Depth* - select one box that most closely describes the depth characteristics of the riffle (Score **0 to 4**). If the riffle is generally less than 5 cm in depth riffles are considered to have loss their function and the entire riffle metric is scored a **0**.

2) *Riffle/Run Substrate Stability*—select one box from each that best describes the substrate type and stability of the riffle habitats (Score **0** to **2**).

3) *Riffle/Run Embeddedness*—**Embeddedness** is the degree that cobble, gravel, and boulder substrates are surrounded or covered by fine material (sand, silt). We consider substrates embedded if >50% of surface of the substrates are embedded in fine material—these substrates cannot be easily dislodged. This also includes substrates that are concreted. Boxes are checked for *extensiveness* (riffle area of sampling

zone) with embedded substrates: **Extensive** — > 75% of stream area, **Moderate** — 50-75%, **Sparse** — 25-50%, **Low** — < 25%.

The maximum score assigned for the QHEI for the Riffle/Run Quality metric is **8 points**.

Metric 6: Map Gradient

Local or map gradient is calculated from USGS 7.5 minute topographic maps by measuring the elevation drop through the sampling area.This is done by measuring the stream length between the first contour line upstream and the first contour line downstream of the sampling site and dividing the distance by the contour interval. If the contour lines are closely "packed" a minimum distance of at least one mile should be used. Some judgement may need to be exercised in certain anomalous areas (*e.g.* in the vicinity of waterfalls, impounded areas, etc.) and this can be compared to an in-field, visual estimate which is recorded on the back of the habitat sheet.

Scoring for ranges of stream gradient takes into account the varying influence of gradient with stream size, preferably measured as drainage area in square miles or stream width. Gradient classifications (Table V-4-3) were modified from Trautman (p 139, 1981) and scores were assigned, by stream size category, after examining scatterplots of IBI vs natural log of gradient in feet/mile. Scores are listed in Table V-4-3

The maximum QHEI metric score for Gradient is **10 points**.

*Computing the Total **QHEI** Score:*

To compute the total **QHEI** score, add the components of each metric to obtain the metric scores and then sum the metric scores to obtain the total **QHEI** score. The **QHEI** metric scores cannot exceed the Metric Maximum Score indicated below.

QHEI SCORING (Maximum = 100)

QHEI Metric	Metric Component	Component Scoring Range	Metric Max. Score
1) Substrate	a) Type	0 to 21	20
	b) Quality	-5 to 3	
2) Instream Cover	a) Type	0 to 10	20
	b) Amount	1 to 11	
3) Channel Morphology	a) Sinuosity	1 to 4	20
	b) Development	1 to 7	
	c) Channelization	1 to 6	
	d) Stability	1 to 3	
4) Riparian Zone	a) Width	0 to 4	10
	b) Quality	0 to 3	
	c) Bank Erosion	1 to 3	
5a) Pool Quality	a) Max. Depth	0 to 6	12
	b) Current	-2 to 4	
	c) Morphology	0 to 2	
5b) Riffle Quality	a) Depth	0 to 4	8
	b) Substr Stab.	0 to 2	
	c) Substr Embd.	-1 to 2	
6)Gradient		2 to 15	10
TOTAL	Maximum Score		100

Additional Information

Additional information is recorded on the reverse side of the Site Description Sheet (Fig. V-4-6) and is described as follows:

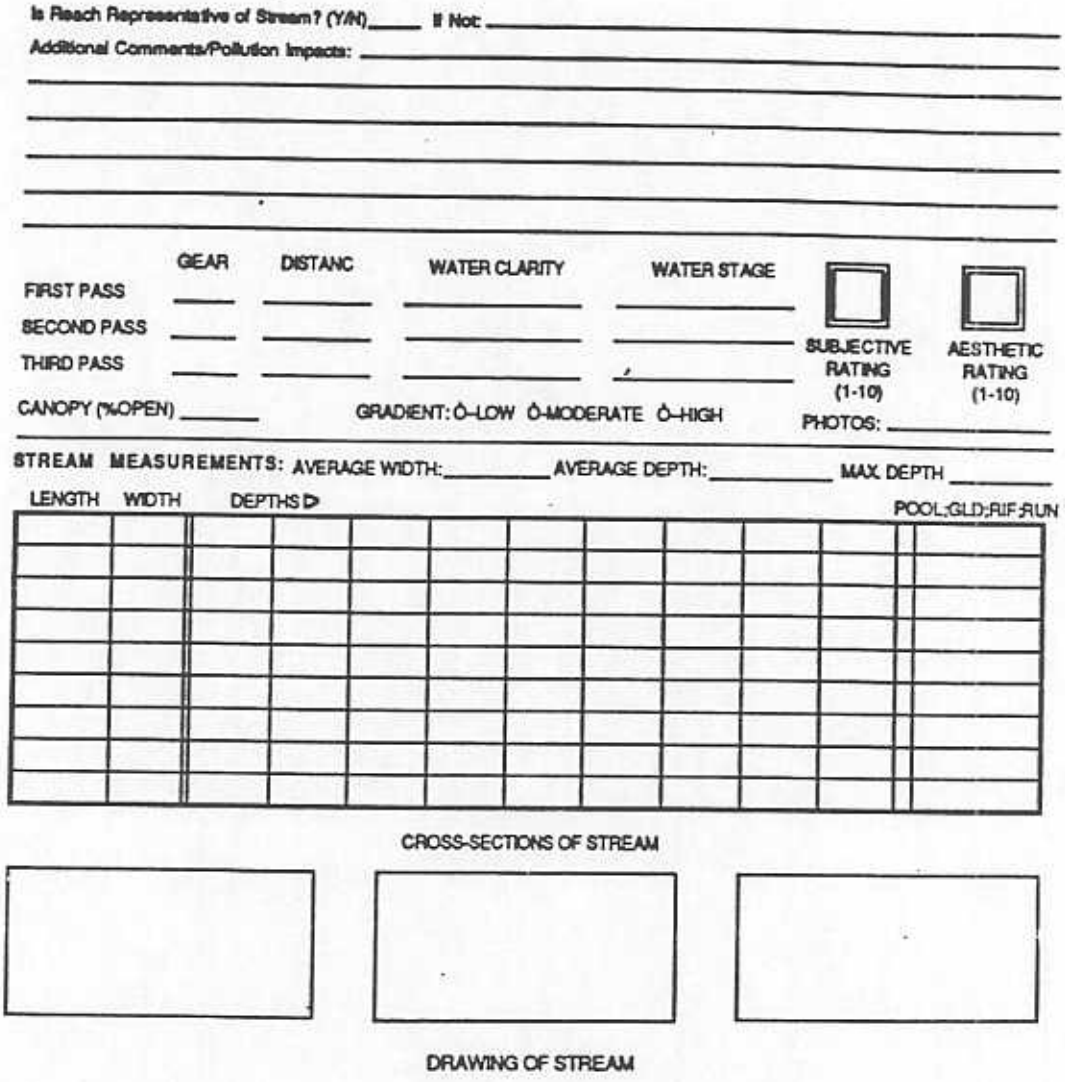
1) *Additional Comments/Pollution Impacts* - Different types of pollution sources (*e.g.* wastewater treatment plant, feedlot, industrial discharge, nonpoint source

Table V-4-3. Classification of stream gradients for Ohio, corrected for stream size. Modified from Trautman (p 139, 1981). Scores were derived from plots of IBI versus the natural log of gradient for each stream size category.

Average Stream Width (m)	Drainage Area (sq mi)	Gradient (ft/mile) Very Low	Low	Low-Moderate	Moderate	Moderate High	High	Very High[1]
0.3-4.7	0-9.2	0-1.0 2	1.1-5.0 4	5.1-10.0 6	10.1-15.0 8	15.1-20 10	20.1-30 10	30.1-40 8
4.8-9.2	9.2-41.6	0-1.0 2	1.1-3.0 4	3.1-6.0 6	6.1-12.0 10	12.1-18.0 10	18.0-30 8	30.1-40 6
9.2-13.8	41.6-103.7	0-1.0 2	1.1-.2.5 4	2.6-5.0 6	5.1-7.5 8	7.6-12.0 10	12.1-20 8	20.1-30 6
13.9-30.6	103.7-622.9	0-1.0 4	1.1-2.0 6	2.1-4.0 8	4.1-6.0 10	6.1-10.0 10	10.1-15 8	15.1-25 6
>30.6	>622.9	-	0-0.5 6	0.6-1.0 8	1.1-2.5 10	2.6-4.0 10	4.1-9.0 10	>9.0 8

[1] Any site with a gradient > than the upper bound of the "very high" gradient classification is assigned a score of 4.

Figure V-4-6. Reverse side of the Ohio EPA Site Description Sheet for evaluating the geographical and physical characteristics of fish sampling locations. This is used to record additional information about the sampling site and adjacent area.



inputs) are noted with their proximity (in 0.1 mile increments) to the sampling site; any evidence of litter either instream or on the stream bank is also noted.

2) *Sampling Gear/Distance Sampled* - The type of fish sampling gear used during each pass is specified (See Table V-4-1) and any variation in sampling procedures is noted (*e.g.*, sampler type A specifies sampling along one shoreline of 0.5 km, but due to local restrictions, sampling may be performed on both shorelines to accumulate 0.5 km); the total sampling distance in kilometers for each sampling site for each pass is recorded.

3) *Water Clarity* - The following descriptions can be used as a guide:

a) Clear - bottom is clearly visible (if shallow enough) and the water contains no apparent color or staining.

b) Stained - usually a brownish (or other) color to the water; the bottom may be visible in shallow areas.

c) Turbid - bottom seldom visible at more than a few inches; caused by suspended sediment particles.

The apparent source of stained (*e.g.* tannic acid, leaf decay, etc.) and turbid (*e.g.* runoff [clay/silt], algae/diatoms, sewage, etc.) may be specified under additional comments.

4) *Water Stage* - This is the general water level of the stream during each pass; suggested descriptors are: a) flood, b) high, c) elevated, d) normal, e) low, and f) interstitial. (Note: sampling should not be conducted during flood or high flows).

5) *Canopy* - This is the percentage of the sampling site that is *not* covered or shaded by woody bank vegetation. In wide streams and rivers this determination should be made along both sides of the river or stream (*i.e.*, the percent of the sampling path that is open).

6) *Gradient* - Check the box that best describes the gradient at the site. This will be used to check the accuracy of gradients taken from topographic maps.

7) *Field Crew* - The names of all individuals involved with the sampling/site description at each site are included.

8) *Photographs* - The number of each photograph taken is recorded; the subject of the photograph is briefly described.

9) *Stream Measurements (optional)* - When measuring the individual sampling sites, length, width, and average and maximum depth information should be recorded; each measurement should be recorded as either a riffle, run, or pool or glide by placing an X in the correct box to the right of where measurements are recorded (Figure V-4-6); see the introduction for definitions of riffles, runs, etc.

The number of width measurements is left to the discretion of the field crew leader. Short riffles may require only one or two width measurements while long pools will probably require more depending on the degree of variation that exists in the stream's width. Depth measurements should be made in association with individual width measurements. Depths should be taken at the stream margins and various points across the stream. Up to nine depth measurements may be taken depending on the variability in the stream bottom. Maximum depth is the deepest spot in the stream section sampled. One purpose of this information is to calculate pool volume.

10) *Stream Diagram - Cross-sections:* Two or three cross-sections of the stream are drawn to provide information on features of the stream bank, stream bottom, stream channel, and floodplain. A series of well defined stream channels (downstream view) are shown in Figure V-4-7. Definitions of these terms follow Platts *et al.* (1983):

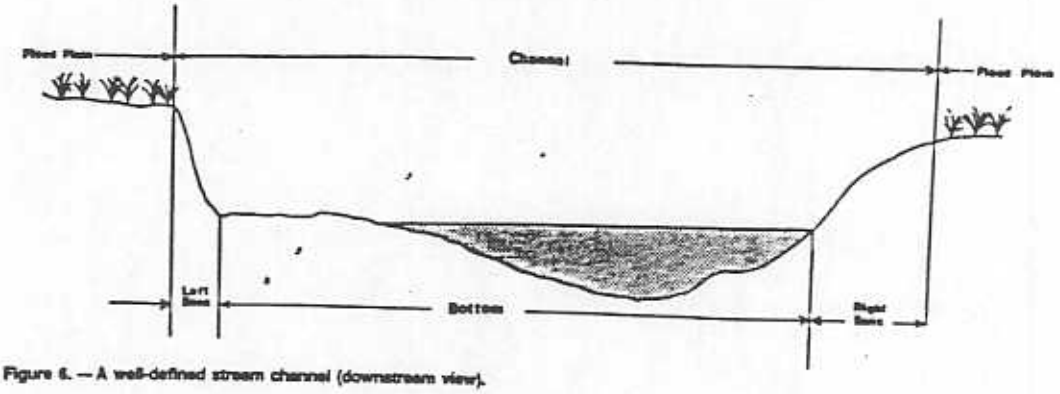


Figure 6. — A well-defined stream channel (downstream view).

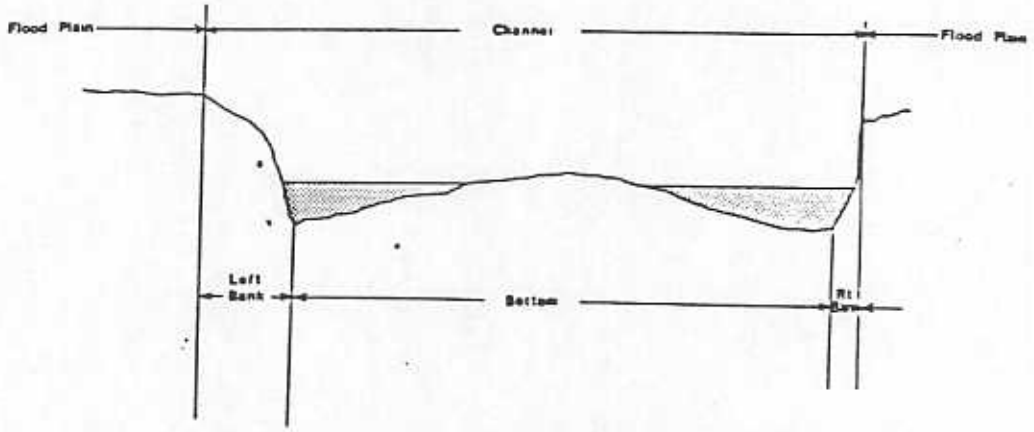


Figure 7. — A well-defined stream channel with concentrated low flows and exposed bottom (downstream view).

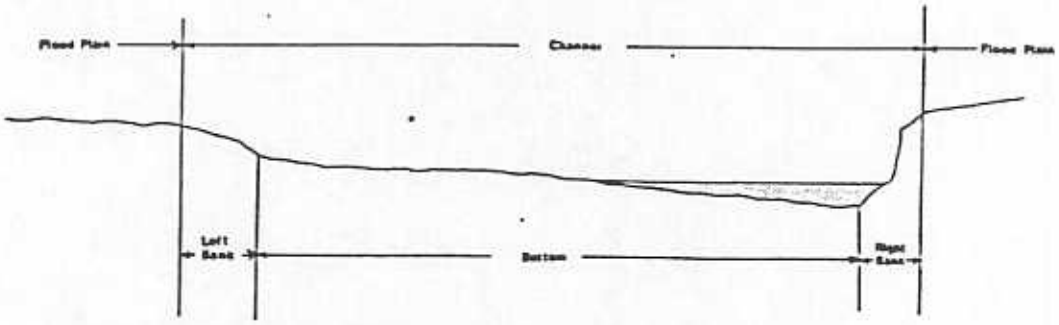


Figure 8. — Stream channel cross channel section on a bend in a stream.

Figure V-4-7. Three variations of a well defined stream channel (downstream view) (from Platts et al. 1983; Figs. 6-8).

Channel - The cross-section containing the stream that is distinct from the surrounding area due to breaks in the general slope of the land, lack of terrestrial vegetation, and changes in the composition of the substrate materials. The channel is made up of streambanks and stream bottoms.

Banks - The portion of the channel cross-section that tends to restrict lateral movement of water. The banks often have a slope steeper than 45° and exhibit a distinct break in slope from the stream bottom.Also, an obvious change in substrate materials is a reliable delineation of the bank.

Stream bottom - The portion of the channel cross-section not classified as bank. The bottom is usually composed of stream sediments or water transported debris and may be covered by rooted or clinging aquatic vegetation. In some geologic formations, the stream bottom may consist of bedrock rather than sediments.

Flood plain - The area adjacent to the channel that is seasonally submerged under water. Usually the flood plain is a low area covered by various types of riparian vegetation.

Stream Map

The entire sampling zone is sketched in the area provided. Important physical features are noted on the map with standard symbols used where possible. The sampling path taken is described along with any other pertinent information

Part C): Laboratory Methods

1) Handling Preserved Materials

a) Preservation Techniques - Fish that are preserved for subsequent identification or for vouchers are immersed in a fixative solution as soon as possible after capture. This helps retain chromatophore patterns which aid in identification. The recommended fixative is a solution of one part commercially prepared formalin and nine parts water with one teaspoon of borax added per 1/2 gallon of fixative. The borax acts as a buffer which neutralizes the acidic effect of the formalin, retarding shrinkage, preventing the hardening of soft body parts, and preventing decalcification of the tissues (Lagler *et al.* 1962). Temperatures greater than 80°F (26.7°C) necessitate a stronger solution of one part formalin to seven or eight parts water. Large fish or containers with closely packed fish also require stronger concentrations of formalin. Strong solutions of formalin can cause gaping or distortion of the mouth and gills, thus care should be taken to obtain correct concentrations when making up the solutions. Specimens more than a few inches long should be slit along the right side of the abdomen prior to preservation; fish heavier than 1 - 2 pounds should also be injected in the muscles on each side of the backbone. Fish normally remain in the formalin solution for at least 2-3 weeks to fix the tissues. Fish are then rinsed in clean water to wash off any excess formalin. The fish are allowed to drain for one-half hour. The fish are then placed in a 35% alcohol wash for 2-3 weeks, switched to a 50% alcohol wash for 2-3 weeks, and placed in a 70% aqueous solution of ethyl alcohol for permanent storage.

Preserving containers are labelled as soon as the fish are collected detailing essential aspects of the sample as completely as possible. Minimum information to be recorded is the stream or river name, location, date, river mile, and principal collector. This information may be written on the initial preserving container with a waterproof marker. If paper is used for making labels it should be 100% rag (which is waterproof) and labeled with India ink or a soft lead pencil.

b) Laboratory Identification and Verification - As discussed previously, fish are field identified by the field crew leader and *when the identification is certain.* However, if there is

any uncertainty the fish are preserved and brought back to the laboratory for verification. In the Ohio EPA laboratory, keys available in Becker (1983), Clay (1975), Pflieger (1975), Scott and Crossman (1973), and Trautman (1957, 1981) are used to identify the preserved fish. Scientific nomenclature follows the recommendations of the American Fisheries Society (Robins *et al.* 1980).

Identifications are verified in-house by at least two trained, full-time Ohio EPA staff. Once taxonomic verification is made, the information is transferred to the fish data sheet for the respective location and either entered into or corrected in FINS. If there remains any question as to the identity of a specimen, it is taken to the Ohio State Museum of Zoology (OSUMZ) for identification by the Curator of Fishes.

c) Disposition -.Ohio EPA maintains an up-to-date reference collection of Ohio and midwestern U.S. region fishes at the Ohio EPA Fish Laboratory. New species or unique specimens are added to the collection as they are encountered. Duplicate specimens are deposited in the OSUMZ where they are permanently catalogued.

2) Data Handling and Analysis

a) Data sheets - Fish data sheets (see example, Figure V-4-4) are completed in the following manner:

1) *Field Crew* - **Sampler** is the individual who actually nets the fish; **Recorder** is the individual who records the data; and **Driver** is the individual who either drives the field vehicle or assists with netting.

2) *Time* - military time sampling started and completed.

3) *River/Stream* - major river or stream being sampled.

4) *Location* - location described as adjacent to, upstream or downstream from a notable landmark.

5)*Date* - month/date/year.

6) *River Code* - assigned number found in FINS RIVLST printout, originally derived from Ohio Gazetteer of Streams (**NOTE:** *contact Central Office Data Coordinator for unnamed or unlisted stream codes*).

7) *River Mile* - river mile (from the *middle* of the electrofishing zone) to the nearest 0.1 mile determined by inspection of PEMSO river mile index maps.

8) *Distance* - electrofishing distance in kilometers to the nearest 0.01 km.

9) *Sampler Type* - sampler type letter code should be noted here (letter codes can be found in Table V-4-1).

10) *Depth* - average depth for the sampling zone to the nearest 10 cm, determined by measuring at 10 random locations with marked depth poles.

11) *Data Source* - two digit code designating the group responsible for data collection, *i.e.* Central Office, Southwest District Office, etc. (**NOTE:** *contact Central Office for Data Source codes*).

12) *Time Fished* - actual time devoted to sampling fish *in seconds.*

13) *Number of Species* - number of species of fish captured during each sampling (hybrids and exotics are *not* included and should be indicated separately).

14) *Species Codes* - each species and any hybrids are recorded by a family-species code following the system presented in Table V-4-5 (located at the end of **Subsection** 4). External anomalies if any, are recorded, for each species according to guidance stated previously.

Additional information that can be entered into FINS includes purpose of the data, latitude and longitude, site drainage area (sq. mi.), local gradient, sample designation, flow, temperature (^{o}C), and dissolved oxygen (mg/l) (Figure V-4-7).

b) Data Storage and Compilation

All completed fish data sheets are logged by the field crew leaders to prevent loss and assure that all sites are sampled according to the plan of study. The data sheets are filed

according to survey, river mile, and date, in that order, at the Ohio EPA Fish Laboratory. The Fish Evaluation Group Leader then logs the data sheets onto master tracking sheets kept at the Ohio EPA Fish Laboratory. Data is then entered into the Fish Information System (FINS) which was developed by Ohio EPA for the purpose of storing and analyzing fish relative abundance data. Data are entered in the format presented in Figure V-4-7. Each data entry is then logged by basin-river code, date of entry, river mile, and date of sampling by the Surface Water Section Data Coordinator. Both the fish data sheet and log book are then initialed by the data entry operator. The data sheets are then assembled in a notebook along with site description sheets, maps of the sampling sites and the preliminary study plans. This is then filed for future reference at the Ohio EPA Fish Laboratory. Any subsequent changes that are made to the fish data sheets are initialed and dated. After all data for a survey have been entered into FINS the entered data are proofread by the field crew leader for accuracy. All corrections or updates are then entered into FINS. Occasionally data from a sampling run may be considered invalid for calculating IBI and modified Iwb scores (*e.g.* due to elevated water levels during sampling, etc.). Although these data are entered into FINS they are designated as invalid samples for calculating community evaluation indices.

c) Analytical Methods

Relative abundance data are analyzed for both community (all species included) and population (single species) parameters. FINS is designed to perform a wide array of analyses. Presently, summarized data from FINS is downloaded to a Sperry PC/IT microcomputer for further detailed analyses. Relative abundance is expressed in terms of numbers/unit distance (or time for passive gear) and weight (kg)/unit distance (or time for passive gear). Community analyses include the number of species per sample, cumulative species per sampling location, Shannon indices (H) based on numbers and weight, the modified Index of Well-Being (Iwb), and the Index of Biotic Integrity modified for application to Ohio waters and Ohio EPA sampling methods. The specific details of how these indices and evaluations are derived is described in the "Users Manual for Biological Field Evaluation of Ohio Surface Waters" (Ohio EPA 1987).

Table V-4-4. Family-species codes used by Ohio EPA fish field crews to code fish data sheets and for data entry into the Fish Information System (FINS).

FISH INFORMATION SYSTEM (FINS) FAMILY CODES	
Family	Code
Petromyzondidae	01
Polyodontidae	04
Acipenseridae	08
Lepisosteidae	10
Amiidae	15
Hiodontidae	18
Clupeidae	20
Salmonidae	25
Osmeridae	30
Umbridae	34
Esocidae	37
Catostomidae	40
Cyprinidae	43
Ictaluridae	47
Anguillidae	50
Cyprinodontidae	54
Poeciliidae	57
Gadidae	60
Percopsidae	63
Aphredoderidae	68
Atherinidae	70
Percichthyidae	74
Centrarchidae	77
Percidae	80
Sciaenidae	85
Cottidae	90
Gasterosteidae	95

Figure V-4-8. Data entry format used to enter raw fish relative abundance into the Fish Information System (FINS). The example below is data collected on September 9, 1984 in the Scioto R. at river mile 125.6. Species 43-001 includes 34 individuals from which a subsample of 15 were individually weighed. Species 43-043 includes 356 individuals of which a subsample of 54 individuals were collectively or "mass" weighed. Anomalies were recorded from the weighed subsample only.

DATE	09/09/84	DEPTH (CM)	120	PURPOSE	01	INVALID SAMPLE	N
RIV CODE	02-001	DATA SOURCE	01	LATITUDE	39 54 03	DESIGNATION	01
RIV MILE	125.6	STREAM ORDER	6	LONGITUDE	83 00 01	FLOW CFS	345
DISTANCE	0.5	TIME (SEC)	1587	DRAIN. AREA	4500	TEMPERATURE	26.8
SAMPLER TYPE	A	OBSERVED FLOW	C	GRADIENT	4.5	DISS. OXYGEN	5.8

NUMBER OF SPECIES 12

ANOMALIES (Y or N?) Y

FAMILY-SPECIES CODE 43-001-C

NUMBER WEIGHED 15 TOTAL COUNT 34

WEIGHT (GRAMS)

ANOMALIES

2350	4550	3450	2000	1225	A	L	P	___	___	___
985	634	5800	1250	1300	2	4	1	___	___	___
2300	3400	230	3250	5700						

FAMILY-SPECIES CODE 43-043-C

NUMBER WEIGHED 54 TOTAL COUNT 356

WEIGHT (GRAMS)

ANOMALIES

2345	BL	E	___	___	___	___
	12	3	___	___	___	___

(additional weight and anomalies spaces would follow for the remaining 10 species)

Table V-4-5. Family-species codes used by Ohio EPA fish field crews to code fish data sheets and for data entry into the Fish Information System (FINS). Designation of Ohio fish species for the purposes of the Index of Biotic Integrity, the Modified Index of Well-Being (Iwb), and the Fish Information System (FINS). Explanation of column headings appears at the end of the table.

FINS Code	Species	Spc Grp	Feed Guild	TOL	IBI Grp	Riv Size	Brd Gld	Hab Pref	Family
01001	Silver lamprey	0	P	-	-	L	N	B	Petromyzontidae
01002	Northern brook lamprey	0	F	R	-	-	N	P	Petromyzontidae
01003	Ohio lamprey	0	P	S	-	-	N	B	Petromyzontidae
01004	Mountain brook lamprey	0	F	S	-	-	N	P	Petromyzontidae
01005	Sea lamprey	0	P	-	E	-	N	B	Petromyzontidae
01006	Least brook lamprey	0	F	-	-	H	N	P	Petromyzontidae
01007	American brook lamprey	0	F	R	-	H	N	P	Petromyzontidae
04001	Paddlefish	0	F	S	-	L	S	B	Polyodontidae
08001	Lake sturgeon	0	V	-	-	L	S	B	Acipenseridae
08002	Shovelnose sturgeon	0	I	-	-	L	S	P	Acipenseridae
10001	Alligator gar	L	P	-	-	L	M	P	Lepisosteidae
10002	Shortnose gar	L	P	-	-	L	M	P	Lepisosteidae
10003	Spotted gar	L	P	-	-	L	M	P	Lepisosteidae
10004	Longnose gar	L	P	-	-	L	M	P	Lepisosteidae
15001	Bowfin	O	P	-	-	-	C	P	Amiidae
18001	Goldeye	W	I	R	-	L	M	B	Hiodontidae
18002	Mooneye	W	I	R	-	L	M	B	Hiodontidae
20001	Skipjack herring	W	P	-	-	L	M	B	Clupeidae
20002	Alewife	O	-	-	E	-	M	P	Clupeidae
20003	Gizzard shad	GS	O	-	-	-	M	P	Clupeidae
20004	Threadfin shad	GS	O	-	-	L	M	P	Clupeidae
25001	Brown trout	SA	-	-	E	-	N	B	Salmonidae
25002	Rainbow trout	SA	-	-	E	-	N	B	Salmonidae
25003	Brook trout	SA	-	-	-	-	N	B	Salmonidae
25004	Lake trout	SA	P	-	F	-	N	P	Salmonidae
25005	Coho salmon	SA	-	-	E	-	N	P	Salmonidae
25006	Chinook salmon	SA	-	-	E	-	N	P	Salmonidae
25007	Cisco or Lake Herring	WF	-	-	-	-	M	P	Salmonidae
25008	Lake whitefish	WF	V	-	-	-	M	P	Salmonidae
30001	Rainbow smelt	0	-	-	-	-	M	P	Osmeridae
34001	Central mudminnow	T	I	T	-	-	C	P	Umbridae
37001	Grass pickerel	P	P	P	-	-	M	P	Esocidae
37002	Chain pickerel	P	P	-	F	-	M	P	Esocidae
37003	Northern pike	P	P	-	F	-	M	P	Esocidae
37004	Muskellunge	P	P	-	F	-	M	P	Esocidae
37005	N. Pike x Muskellunge	P	P	-	E	-	-	-	Esocidae
37006	Grass P. x Chain	P.	P	P-	F	-	-	-	Esocidae
40001	Blue sucker	R	I	R	R	L	S	R	Catostomidae
40002	Bigmouth buffalo	C	I	-	C	L	M	P	Catostomidae
40003	Black buffalo	C	I	-	C	L	M	P	Catostomidae
40004	Smallmouth buffalo	C	I	-	C	L	M	P	Catostomidae
40005	Quillback	C	O	-	C	-	M	P	Catostomidae
40006	River carpsucker	C	O	-	C	L	M	P	Catostomidae

Table V-4-5. Continued

FINS Code	Species	Spc Grp	Feed Guild	TOL	IBI Grp	Riv Size	Brd Gld	Hab Pref	Family
40007	Highfin carpsucker	C	O	-	C	L	M	P	Catostomidae
40008	Silver redhorse	R	I	M	R	-	S	P	Catostomidae
40009	Black redhorse	R	I	I	R	-	S	P	Catostomidae
40010	Golden redhorse	R	I	M	R	-	S	P	Catostomidae
40011	Shorthead redhorse	R	I	M	R	-	S	P	Catostomidae
40012	Greater redhorse	R	I	R	R	-	S	P	Catostomidae
40013	River redhorse	R	I	I	R	-	S	P	Catostomidae
40014	Harelip sucker	R	-	S	R	-	S	P	Catostomidae
40015	Northern hog sucker	R	I	M	R	-	S	R	Catostomidae
40016	White sucker	R	O	T	W	-	S	B	Catostomidae
40017	Longnose sucker	R	I	-	R	-	S	P	Catostomidae
40018	Spotted sucker	R	I	-	R	-	S	P	Catostomidae
40019	Lake chubsucker	R	I	-	R	-	M	P	Catostomidae
40020	Creek chubsucker	R	I	-	R	P	M	P	Catostomidae
43001	Common carp	G	O	T	G	-	M	P	Cyprinidae
43002	Goldfish	G	O	T	G	-	M	P	Cyprinidae
43003	Golden shiner	N	I	T	N	-	M	P	Cyprinidae
43004	Hornyhead chub	M	I	I	N	-	N	B	Cyprinidae
43005	River chub	M	I	I	N	-	N	B	Cyprinidae
43006	Silver chub	M	I	-	N	L	M	P	Cyprinidae
43007	Bigeye chub	M	I	I	N	-	S	R	Cyprinidae
43008	Streamline chub	M	I	R	N	L	S	R	Cyprinidae
43009	Gravel chub	M	I	M	N	L	S	R	Cyprinidae
43010	Speckled chub	M	I	S	N	L	M	R	Cyprinidae
43011	Blacknose dace	M	G	T	N	H	S	R	Cyprinidae
43012	Longnose dace	M	I	R	N	-	S	R	Cyprinidae
43013	Creek chub	M	G	T	N	P	N	B	Cyprinidae
43014	Tonguetied minnow	M	I	S	N	-	N	P	Cyprinidae
43015	Suckermouth minnow	M	I	-	N	-	S	R	Cyprinidae
43016	Southern redbelly dace	M	H	-	N	H	S	B	Cyprinidae
43017	Redside dace	M	I	I	N	H	S	P	Cyprinidae
43018	Rosyside dace	M	I	S	N	H	S	P	Cyprinidae
43019	Pugnose minnow	N	I	R	N	-	M	P	Cyprinidae
43020	Emerald shiner	N	I	-	N	-	S	P	Cyprinidae
43021	Silver shiner	N	I	I	N	-	S	P	Cyprinidae
43022	Rosyface shiner	N	I	I	N	-	S	R	Cyprinidae
43023	Redfin shiner	N	I	-	N	-	N	P	Cyprinidae
43024	Rosefin shiner	N	I	M	N	-	S	P	Cyprinidae
43025	Striped shiner	N	I	-	N	-	S	B	Cyprinidae
43026	Common shiner	N	I	-	N	-	S	P	Cyprinidae
43027	River shiner	N	I	-	N	L	S	P	Cyprinidae
43028	Spottail shiner	N	I	P	N	L	M	P	Cyprinidae
43029	Blackchin shiner	N	I	S	N	-	M	P	Cyprinidae
43030	Bigeye shiner	N	I	R	N	-	S	B	Cyprinidae
43031	Steelcolor shiner	N	I	P	N	-	M	P	Cyprinidae
43032	Spotfin shiner	N	I	-	N	-	M	B	Cyprinidae

Table V-4-5. Continued

FINS Code	Species	Spc Grp	Feed Guild	TOL	IBI Grp	Riv Size	Brd Gld	Hab Pref	Family
43033	Bigmouth shiner	N	I	-	N	-	M	B	Cyprinidae
43034	Sand shiner	N	I	M	N	-	M	B	Cyprinidae
43035	Mimic shiner	N	I	I	N	-	M	B	Cyprinidae
43036	Ghost shiner	N	I	-	N	L	M	P	Cyprinidae
43037	Blacknose shiner	N	I	R	N	-	M	P	Cyprinidae
43038	Pugnose shiner	N	I	S	N	-	M	P	Cyprinidae
43039	Silverjaw minnow	M	I	-	N	P	M	B	Cyprinidae
43040	Mississippi silvery minnow	M	H	-	N	-	M	P	Cyprinidae
43041	Bullhead minnow	N	O	-	N	-	C	P	Cyprinidae
43042	Fathead minnow	M	O	T	N	P	C	B	Cyprinidae
43043	Bluntnose minnow	M	O	T	N	P	C	B	Cyprinidae
43044	Central stoneroller	M	H	-	N	-	N	B	Cyprinidae
43045	Common carp x Goldfish	G	O	T	G	-	-	-	Cyprinidae
43046	Popeye shiner	N	I	S	N	-	S	P	Cyprinidae
43047	Grass carp	G	-	-	E	-	M	B	Cyprinidae
43048	Red shiner	N	I	-	E	-	N	P	Cyprinidae
43049	Common x Rosyface Shiner	N	I	-	-	-	-	-	Cyprinidae
43057	Striped shiner/Stoneroller	M	-	-	-	-	-	-	Cyprinidae
43058	Common shiner/Stoneroller	M	-	-	-	-	-	-	Cyprinidae
43059	Striped shiner/Horny chub	M	I	-	-	-	-	-	Cyprinidae
43999	Hybrid Minnow	M	-	-	-	-	-	-	Cyprinidae
47001	Blue catfish	F	C	-	F	L	C	P	Ictaluridae
47002	Channel catfish	F	-	-	F	-	C	P	Ictaluridae
47003	White catfish	F	I	-	E	-	C	P	Ictaluridae
47004	Yellow bullhead	F	I	T	-	-	C	P	Ictaluridae
47005	Brown bullhead	F	I	T	-	-	C	P	Ictaluridae
47006	Black bullhead	F	I	P	-	-	C	P	Ictaluridae
47007	Flathead catfish	F	P	-	F	L	C	B	Ictaluridae
47008	Stonecat	O	I	I	-	-	C	R	Ictaluridae
47009	Mountain madtom	O	I	R	-	-	C	R	Ictaluridae
47010	Northern madtom	O	I	R	-	-	C	R	Ictaluridae
47011	Scioto madtom	O	I	S	-	-	C	R	Ictaluridae
47012	Brindled madtom	O	I	I	-	-	C	B	Ictaluridae
47013	Tadpole madtom	O	I	-	-	-	C	B	Ictaluridae
50001	American eel	O	C	-	-	-	M	P	Anguillidae
54000	Western Banded killifish	T	I	S	-	-	M	P	Cyprinodontidae
54001	Eastern Banded killifish	T	I	T	E	-	M	P	Cyprinodontidae
54002	Blackstripe topminnow	T	I	-	-	-	M	P	Cyprinodontidae
57001	Mosquitofish	O	I	-	E	-	N	P	Poeciliidae
60001	Burbot	O	-	-	-	-	S	B	Gadidae
63001	Trout-perch	O	I	-	-	-	M	P	Percopsidae
68001	Pirate perch	O	I	-	-	-	M	P	Aphredoderidae
70001	Brook silverside	O	I	M	-	-	M	P	Atherinidae
74001	White bass	W	P	-	F	L	M	P	Percichthyidae
74002	Striped bass	W	P	-	E	-	M	P	Percichthyidae
74003	White perch	W	-	-	E	-	M	P	Percichthyidae

Table V-4-5. Continued

FINS Code	Species	Spc Grp	Feed Guild	TOL	IBI Grp	Riv Size	Brd Gld	Hab Pref	Family
74004	White bass x White perch	W	-	-	-	-	-	-	Percichthyidae
74005	Striped bass x White bass	W	-	-	E	-	-	-	Percichthyidae
77001	White crappie	B	-	-	S	-	C	P	Centrarchidae
77002	Black crappie	B	-	-	S	-	C	P	Centrarchidae
77003	Rock bass	B	C	-	S	-	C	P	Centrarchidae
77004	Smallmouth bass	B	C	M	F	-	C	P	Centrarchidae
77005	Spotted bass	B	C	-	F	-	C	P	Centrarchidae
77006	Largemouth bass	B	C	-	F	-	C	P	Centrarchidae
77007	Warmouth	S	C	-	S	-	C	P	Centrarchidae
77008	Green sunfish	S	I	T	S	P	C	P	Centrarchidae
77009	Bluegill	S	I	P	S	-	C	P	Centrarchidae
77010	Orangespotted sunfish	S	I	-	S	-	C	P	Centrarchidae
77011	Longear sunfish	S	I	M	S	-	C	P	Centrarchidae
77012	Redear sunfish	S	I	-	E	-	C	P	Centrarchidae
77013	Pumpkinseed	S	I	P	S	-	C	P	Centrarchidae
77014	Bluegill x Pumpkinseed	S	-	-	-	-	-	-	Centrarchidae
77015	Green x Bluegill	S	-	-	-	-	-	-	Centrarchidae
77016	Green x Pumpkinseed	S	-	-	-	-	-	-	Centrarchidae
77017	Longear x Bluegill	S	-	-	-	-	-	-	Centrarchidae
77018	Bluegill x Orangespotted	S	-	-	-	-	-	-	Centrarchidae
77019	Green x Orangespotted	S	-	-	-	-	-	-	Centrarchidae
77020	Pumpkinseed x Longear	S	-	-	-	-	-	-	Centrarchidae
77021	Green x Longear	S	-	-	-	-	-	-	Centrarchidae
77022	O'spotted x Pumpkinseed	S	-	-	-	-	-	-	Centrarchidae
77023	Longear x Orangespotted	S	-	-	-	-	-	-	Centrarchidae
77024	Green x Warmouth	S	-	-	-	-	-	-	Centrarchidae
77025	Warmouth x Pumpkinseed	S	-	-	-	-	-	-	Centrarchidae
77998	Green Sunfish Hybrid	S	-	-	-	-	-	-	Centrarchidae
77999	Hybrid Sunfish	S	-	-	-	-	-	-	Centrarchidae
80001	Sauger	V	P	-	F	L	S	P	Percidae
80002	Walleye	V	P	-	F	-	S	P	Percidae
80003	Yellow perch	V	-	-	-	-	M	P	Percidae
80004	Dusky darter	D	I	M	D	-	S	B	Percidae
80005	Blackside darter	D	I	-	D	-	S	B	Percidae
80006	Longhead darter	D	I	S	D	-	S	R	Percidae
80007	Slenderhead darter	D	I	R	D	L	S	R	Percidae
80008	River darter	D	I	-	D	L	S	R	Percidae
80009	Channel darter	D	I	S	D	-	S	P	Percidae
80010	Gilt darter	D	I	S	D	-	S	B	Percidae
80011	Logperch	D	I	M	D	-	S	B	Percidae
80012	Crystal darter	D	I	S	D	-	S	R	Percidae
80013	Eastern sand darter	D	I	R	D	-	S	R	Percidae
80014	Johnny darter	D	I	-	D	P	C	B	Percidae
80015	Greenside darter	D	I	M	D	-	S	R	Percidae
80016	Banded darter	D	I	I	D	-	S	R	Percidae
80017	Variegate darter	D	I	I	D	-	S	R	Percidae

Table V-4-5. Continued

FINS Code	Species	Spc Grp	Feed Guild	TOL	IBI Grp	Riv Size	Brd Gld	Hab Pref	Family
80018	Spotted darter	D	I	R	D	-	S	R	Percidae
80019	Bluebreast darter	D	I	R	D	-	S	R	Percidae
80020	Tippecanoe darter	D	I	R	D	-	S	R	Percidae
80021	Iowa darter	D	I	-	D	-	M	P	Percidae
80022	Rainbow darter	D	I	M	D	-	S	R	Percidae
80023	Orangethroat darter	D	I	-	D	P	S	B	Percidae
80024	Fantail darter	D	I	-	D	H	C	R	Percidae
80025	Least darter	D	I	-	D	-	N	B	Percidae
80026	Sauger x Walleye	V	P	-	E	-	-	-	Percidae
85001	Freshwater drum	F	-	P	-	L	M	P	Sciaenidae
90001	Spoonhead sculpin	SC	-	-	-	-	C	P	Cottidae
90002	Mottled sculpin	SC	I	-	-	H	C	R	Cottidae
90003	Slimy sculpin	SC	-	-	-	-	-	-	Cottidae
90004	Deepwater sculpin	SC	-	-	-	-	-	-	Cottidae
95001	Brookstickleback	O	I	-	-	H	C	P	Gasterosteidae

Table V-4-5, Continued

SPCLST - Legend for Species Designations

The following letter symbol designations are used to classify Ohio fish species according to their taxonomic, functional, structural, pollution tolerance, and ecological characteristics. These designations provide the basis for the Fish Information System (FINS) to calculate metrics for the Index of Biotic Integrity (FINIBI) and the Modified Index of Well-Being (FINLS2) as well as other uses.

SPC GRP (Species Group)[a]
- O - Other
- L - Gars
- W - Large River Species
- GS - Gizzard Shad
- SA - Salmonid
- WF - Whitefish
- T - Tolerant
- P - Pickerels
- R - Round-bodied Suckers
- C - Deep-bodied Suckers
- G - Carp/Goldfish
- N - Shiners
- M - Minnows
- F - Catfish, Drum
- B - Blackbass, Crappie
- S - Sunfish
- V - Non-darter Percidae
- D - Darters
- SC - Sculpins

FEED GUILD (Feeding Guild)[b]
- P - Piscivore
- F - Filter Feeder
- V - Invertivore
- I - Specialist Insectivore
- O - Omnivore
- G - Generalist
- H - Herbivore
- C - Carnivore

TOL (Pollution Tolerance)
- R - Rare Intolerant
- S - Special Intolerant
- I - Common Intolerant
- M - Moderately Intolerant
- T - Highly Tolerant
- P - Moderately Tolerant

BRD GLD (Breeding Guild)[c]
- N - Complex, no parental care
- C - Complex with parental care
- M - simple, miscellaneous
- S - simple lithophils

IBI GRP (IBI Group)[b]
- E - Exotic (non-native)
- F - Sport Species
- R - Round-bodied Sucker
- C - Deep-bodied Sucker
- W - White sucker
- G - Carp/Goldfish
- N - Other Cyprinidae
- S - Sunfish (less Blackbasses)
- D - Darters

RIV SIZ (River Size)
- L - Large River Species
- H - Headwaters Species
- P - Pioneering Species

HAB PRF (Habitat Pref.)[c]
- P - prefers pools
- R - preferes riffles
- B - prefers both

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[a]these designations are not for use in any FINS analytical programs.

[b]designations are patterned after Karr et al. (1986).

[c]designations are patterned after Berkman and Rabeni (1987).

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Appendix F:

Surface Water Collection Procedures and Sediment Collection SOP

Protecting Lake Michigan & Our Waterways.
North Shore Water Reclamation District

Procedure FLD-RIV
Revision No. 0
Org. Date: 04/13/18
Rev. Date

TITLE:

Surface Water Collection Procedures

WRITTEN BY: **APPROVED BY:**

Robert Flood

The use of this SOP is governed by the North Shore Water Reclamation District's Quality Assurance Manual and associated Quality SOPs. Implementation of this SOP must always comply with the requirements of the Quality Assurance Manual and the Quality SOPs.

SCOPE AND APPLICATION:

This SOP is applicable to the collection of representative surface water samples from rivers, streams, lakes or any other surface waters. This procedure is a grab sample method that utilizes a stainless steel bucket or dip sampler to collect a surface water grab sample.

SUMMARY OF METHOD:

Sampling situations can vary widely depending on the location and characteristics of the water body. Generally, a surface water grab sample is accomplished through the use of one of the following techniques:

- Dip sampler
- Stainless steel or polyethylene bucket (polyethylene not for collection of organic samples)
- Direct method

SAFETY PRECAUTIONS:

1. Personal Protection
   Work or disposable gloves are recommended. Hip boots or waders may or may not be required during sample collection.

2. Chemical hazards
   Pre-preserved sample containers may contain hazardous chemicals. Handle all samples carefully to minimize exposure.
3. Biological Hazards
   Water samples may contain potential health hazards. Handle all samples carefully to minimize exposure.

INTERFERENCES:

The two most common interferences in surface water collection include cross contamination and improper collection technique.

1. Cross contamination can be eliminated through the use of dedicated or disposable sampling equipment or proper cleaning/decontamination procedures.
2. Improper sample collection can occur when using contaminated sampling equipment or poor technique. It is important to collect the sample in the most representative area. Care should be taken to minimize bottom substrate disturbance and avoid surface scum or debris.

EQUIPMENT AND SUPPLIES:

1. Stainless steel bucket with rope or dip sampler
2. Deionized rinse water
3. Decontamination equipment and supplies
3. Appropriate sample bottles
4. Cooler with ice packs
5. Field Instrumentation
5. Field Log Book and Sample Chain of Custody

REAGENTS AND STANDARDS:

Reagents may be used for preservation of samples. Preservatives will be specific to the analysis and determined by the laboratory. Cleaning solutions may be used for decontamination of sampling equipment.

SAMPLE PRESERVATION, CONTAINERS, HANDLING AND STORAGE:

Once samples have been collected, the following procedures should be followed:

1. Transfer the sample into a suitable, properly labeled sample container specific for the analysis to be performed.
2. Preserve the sample, if appropriate. Pre-preserved sample containers are preferred for simplicity and convenience. Do not overfill containers if they are pre-preserved.

3. Cap the container securely and cool immediately by placing in a sample cooler with wet ice or reusable ice packs.
4. Record all relevant information in the sample log book and NSWRD Field Collection Sheets.
5. Deliver samples to the laboratory and follow NSWRD chain of custody procedures. See the appropriate section of the NSWRD Laboratory QAP for additional guidance.

QUALITY CONTROL:

All personnel involved in the sample collection process must be properly trained and understand the sampling SOP. Any deviations must be recorded in the field book and/or on the field collection sheet. The laboratory supervisor must be notified of any deviations from the SOP and evaluate appropriately.

All field equipment shall be maintained following manufacturers recommendations. All field equipment shall be inspected, calibrated and tested prior to sampling events and after the equipment returns from the field. Any problems encountered or maintenance required must be noted in the equipment maintenance log book.

CALIBRATION AND STANDARDIZATION:

Field meters must be calibrated daily following manufacturers calibration procedures and documented in the field instrument calibration log book.

PROCEDURE:

Prior to being used for sample collection or holding, all sampling equipment is decontaminated and cleaned following procedures outlined in the NSWRD Laboratory Quality Assurance Project Plan.

1. <u>Preparation</u>
   a. Determine the sample locations by performing a general site survey if possible. Prior knowledge of the locations will aid in determining exact equipment needs and safety considerations. Sample sites may need to be adjusted based on access, property boundaries or obstructions.
   b. Determine the equipment needs and make sure everything is in working order.

2. <u>Sample Collection</u>
   a. Take sample at the specified location. If sampling a river or stream, sample at the middle of the main channel at mid-depth. Collect the sample from a representative site on the stream. Try to locate an area where the water is well mixed and the velocity of flow is great enough that the chance of solids settling is minimal. Depending on the site

characteristics, the sampler may use a bucket, pole sampler or wade in and collect the sample. Lower the sampling device into the stream. When it is properly positioned, activate the bucket to collect a sample by tipping the bucket gently. Avoid top floating debris if possible. It is important not to disturb the bottom substrate during the collection process. If excess dirt, gravel, or other foreign material is collected, discard the sample, and repeat the sampling. Once the sample has been collected, fill each sample bottle to the appropriate mark taking care not to overfill pre-preserved bottles.

b. Field measurements should be performed on site after all of the sample bottles have been filled.
c. Record collection date, time and field measurements in the field book and/or field collection sheet.

REFERENCES:

1. North Shore Water Reclamation District Quality Assurance Plan

2. Standard Methods for the Examination of Water and Wastewater, 22nd ed,2012.

Illinois Environmental Protection Agency

Bureau of Water

Document Control Number 174

Standard Operating Procedure for Surficial Sediment Collection

Surface Water Section
1021 North Grand Avenue East
P.O. Box 19276
Springfield, Illinois 62794-9276
Contact: Bureau of Water, Quality Assurance Officer 217-782-3362

1.0 Scope and Application

This Standard Operating Procedure (SOP) describes how to collect and handle surficial sediment samples from streams and lakes. Surficial sediments are deposited solid materials that have settled from the water column to the stream or lake bottom. The surface layer of sediment is collected and analyzed to identify the presence and amounts of various organic and inorganic compounds. This SOP focuses on collecting surficial sediments within a limited range of particle sizes. To facilitate laboratory analysis and to achieve comparability of results among sediment samples from different streams or different reaches within a stream, all sediment samples submitted for analysis are restricted to comprising particles of 63 microns or smaller in size. For this reason, samplers focus on sites predominated by fine sediments: sand (0.0625 – 2 mm in diameter), silt/mud (0.0039 - 0.0625 mm), clay (< 0.0039 mm), or combinations of these. For streams, this SOP pertains to collecting sediment during low-flow conditions. Both stream and lake sediment samples are typically collected May – October.

2.0 Summary of Method

In Illinois streams and lakes, surficial sediments commonly comprise silt, clay, fine organic material, and larger particles (sand, gravel, etc.). In streams during periods of low flow, the smaller sediment particles settle to the bottom in areas of slowest water velocity such as pools or areas downstream of bars, logs, or other flow obstructions. For streams, the upper ½ inch (1-2 cm) layer of sediment is collected. After retrieval, the sediment is sieved with properly cleaned stainless-steel equipment, placed in quart jars on ice to allow the sediment to settle from the water column. After settling, the supernatant is poured off and the sediment is placed in appropriate bottles and forwarded to the laboratory. For lakes, sediment is collected from the uppermost ½ inch (1-2 cm) layer of dredge-collected material, placed in the appropriate bottles and forwarded to the laboratory.

3.0 Interferences and Corrective Action

3.1 Dirt, oils, or other unintended substances on equipment used to collect a sample can contaminate the sediment sample and bias the laboratory results. Equipment is field cleaned and rinsed prior to use before each station is sampled.

3.2 Use stainless-steel sampling equipment when possible. If stainless-steel equipment is not available, it is acceptable to use other equipment if all non-stainless-steel surfaces that directly contact the sampled sediment are covered with epoxy paint.

3.3 Care should be taken to minimize disturbance of sediment prior to sample collection. When collecting sediment by wading using a hand held spoon, collect the composite portions from downstream to upstream. Avoid areas previously disturbed by earlier sampling (i.e., macroinvertebrate, fish population, etc.). On lakes or large rivers, when deploying an Ekman or

Petite Ponar dredge, lower the sampler slowly to avoid washing out the uppermost sediment layer. Raise the sampler slowly to minimize material loss.

3.4 Larger particles and organic matter may clog the pores of the sieve. Rinse the sieve often in ambient stream water to clear the sieve.

3.5 Minimize windblown material (e.g., plant seeds, dust, etc.) from being deposited in the sample pan and sieve by positioning sieving location in a protected area.

4.0 Safety

4.1 Follow the general field-safety guidelines in Illinois EPA, Bureau of Water's *Surface Water Section Field Safety Manual* (Document Control Number 151).

4.2 Do not use or store isopropyl alcohol near sparks, flames, or any other source of excessive heat. Use caution when storing isopropyl alcohol in a closed vehicle during warm weather.

4.3 Isopropyl alcohol can irritate eyes and skin. Wear protective eyewear, gloves, and an apron (recommended) when using this product. Flush with clean water in the event of eye or dermal contact.

4.4 Collecting a stream or lake sediment sample requires wading into or being on the water. Prior to wading to collect a sample, determine the safety of entering the stream. Be aware that water depth, water velocity, and type of stream bottom influence the ability to safely wade a stream. When collecting a sample by boat, follow all boating-safety guidelines.

4.5 The Petite Ponar dredge is a heavy, center-pivot sampler that presents a pinching hazard when closing. Use caution when handling an open Ponar sampler. Keep the safety pin that locks the jaws open in place at all times other than when collecting a sample. The Eckman dredge is a messenger style spring-loaded jaw sampler that presents a pinching hazard when closing. Use care when handling the sampler while collecting a sample. Transport the sampling device with the jaws in the closed position.

5.0 Equipment and Supplies

Stainless-steel Equipment:

- ASTM-E11 No. 230 sieve (63-micron mesh)
- Pan (minimum size 12x12x3 inch)
- Bowl (minimum size 8 qt. capacity)
- Long-handled “serving” spoon (minimum 12” long)

Grab Sampler:

- Petite Ponar dredge (15 cm x15cm; sample area 232 cm^2; dry weight 11 kg.)
- Eckman dredge (15 cm x15cm; sample area 232 cm^2; dry weight 3 kg.)

Rope to suspend dredge
One-Quart Glass Jars with Lids and Labels: up to four per site
Aluminum Foil
Isopropyl alcohol (pesticide grade)
Deionized Water
Non-phosphate Detergent (e.g., Liqui-Nox)
Cleaning Brush with non-metallic bristles
Waterproof Marker
Laboratory Sample Bottles:
One (1) 250-ml glass bottle for organic analysis
One (1) 250-ml plastic (non-preserved) for metals analysis
Media for Documenting/Describing the Sampling Event and Samples (e.g., hardcopy "Request for Laboratory Analysis" forms, laptop computer)
Coolers with ice for cooling samples

It is the responsibility of the sample technician to select the appropriate sampling equipment and method for the type of media to be collected in order to meet the monitoring objectives.

6.0 Cleaning Sampling Equipment in the Field

Clean and rinse all sampling equipment before using it at each stream reach or lake. Clean equipment as follows:

- Wash all sampling equipment—including quart jars and foil liners (not used for lakes) with a non-phosphate detergent (i.e., Liqui-nox) by using a brush to remove dirt or oil.
- Rinse all equipment with deionized water.
- Rinse all equipment with isopropyl alcohol.
- Rinse again all equipment with deionized water.
- Rinse all equipment with ambient stream or lake water.

7.0 Sediment Sample Collection

7.1 Stream-Sediment Collection – Wadable Streams: Typically, Illinois EPA sediment sampling in streams corresponds closely with biological-assemblage sampling; therefore, the station for sediment sampling is the section of stream (reach) selected for biological sampling. To sufficiently reflect sediment conditions of the stream, the sampler composites sediments from various appropriate sites throughout the sampling reach. Typically, appropriate sediment deposits are in areas of reduced velocity such as pools or areas downstream of gravel (or sand) bars, logs, or other flow obstructions. This section (7.1) refers to sample collection in relatively shallow stream reaches that are accessible by wading.

7.1.1 Clean and rinse all sediment sampling equipment as described in Section 6.0 prior to sampling.

7.1.2 Identify multiple sediment-deposition sites throughout the sampling reach. At identified sites, use the long-handled spoon and bowl to collect and composite sediment from the top (i.e., 1/2 inch) layer. To facilitate sieving, try to minimize the amount of sand and larger particles collected.

7.1.3 Using a one-quart glass jar and the stainless-steel, 63-micron sieve, pour approximately 2 quarts of sieved stream water through the sieve and into the stainless-steel pan. Use only ambient stream water from the immediate vicinity of sediment collections. Use of water from another source may contaminate and/or dilute the sample.

7.1.4 Place a spoonful of sediment into the 63-micron stainless-steel sieve. If there is a considerable amount of aggregated clay/muck in the sediment, add a small amount of water to break down the aggregate to facilitate sieving.

7.1.5 Place the sieve in the stainless-steel pan, with the mesh below the water level. Use short (less than ½ inch), pulsing strokes of the sieve to pulse water up and down through the sieve. After about 2-3 minutes, stop the pulsing action and raise the sieve out of the water. Most of the 63-micron or smaller particles and all of the water will pass through the sieve and be deposited in the pan.

7.1.6 Rinse the sieve in the stream to remove any material remaining in the sieve.

7.1.7 Repeat the prior three steps until approximately a 3-4 mm thick layer of sediment is captured in the pan. Generally, 30-45 minutes of sieving is required to assure an adequate amount of sample.

7.1.8 By gently rocking the pan with the water and sieved sediment, re-suspend the sediment and pour the contents into up to four rinsed one-quart jars filling the jars to the shoulders. Cover each jar with foil and seal it with the lid. If, after filling the jars, there is a significant amount of settled material in the pan, wait several minutes for the sediment in the jars to settle, pour off the supernatant and re-fill the jars to the shoulders.

7.1.9 Properly label the jars and place them in a cooler with sufficient ice (a volume of ice equal to at least the volume occupied by samples, but preferably twice the volume of ice to samples) surrounding the jars to cool them to 6^{o}C or less. Fill out the appropriate sample/sampling documentation (e.g., "Request for Laboratory Analysis" form).

7.1.10 Allow the contents of the jars to settle for a minimum of 12 hours (e.g., overnight). After the sediment (including all fine or organic material) has settled, decant the overlying water (supernatant) from each of the jars. Transfer the sediment from the jars into appropriately labeled 250-ml laboratory sample bottles. Fill each bottle to approximately one-half full (i.e., 125 ml) to ensure enough material for lab analysis.

7.1.11 Place laboratory sample bottles into a cooler with sufficient ice to maintain samples at 6°C or less, or place the sample bottles refrigerator. Allow samples to settle for at least 24 hours. After sediments (including all fine or organic material**)** settle, decant excess water from each bottle (to prevent expansion and cracking) and place it in a standard freezer that maintains a temperature of -10°C or less.

7.2 Stream-Sediment Collection – Non-Wadable Streams: Typically, Illinois EPA sediment sampling in streams corresponds closely with biological-assemblage sampling; therefore, the station for sediment sampling is the section of stream (reach) selected for biological sampling. To sufficiently reflect sediment conditions of the stream, the sampler composites sediments from various appropriate sites throughout the sampling reach. Typically, appropriate sediment deposits are in areas of reduced velocity such as pools or areas downstream of point bars, islands, logs, or other flow obstructions. This section (7.2) refers to sample collection in streams sampled by using a rope-suspended Petite Ponar or Eckman dredge style sampling device. These streams are generally too deep to wade and are accessed by boat.

7.2.1 Clean and rinse all sediment sampling equipment as described in Section 6.0 prior to sampling.

7.2.2 Identify multiple sediment-deposition sites throughout the sampling reach. Set sediment sampler jaws to sampling position and slowly lower the dredge until it contacts the bottom. If using the Petite Ponar dredge, allow enough slack to release the locking pin, thus allowing the jaws to close upon retrieval. Allow a few seconds for the Petite Ponar dredge to sink into the sediment and slowly lift the sample to the surface. If using the Eckman dredge, hold the rope taut after allowing the dredge to sink into the sediment. Release the brass messenger down the rope to trip the dredge mechanism, closing the jaws. Slowly lift the sample to the surface.

7.2.3 Deposit the dredged sediment into a clean stainless-steel pan.

7.2.4 Remove the top ½ inch layer from the sediment sample by using a clean stainless-steel spoon and deposit in a bowl. Repeat collection procedure until a sufficient amount of composited sample is collected.

7.2.5 To process the sample, complete steps described in Sections 7.1.3 – 7.1.11.

7.3 Lake-Sediment Collection - Typical Illinois lake sediments are generally comprised of fine particles. Samples are collected from a boat using a rope-suspended Petite Ponar or Eckman dredge sampling device. Due to the fine particle size of lake sediments, it is not necessary to sieve samples. Occasionally when retrieving a lake-bottom sample with a dredge, the fine sediment will be lost because a stick, shell, or other large debris prevents the jaws from completely closing, allowing the sediment to wash out.

7.3.1 Clean and rinse all sediment sampling equipment as described in Section 6.0 prior to sampling the first station on the lake. Between stations scrub away any mud or other debris with a brush and rinse all collection equipment thoroughly with ambient water.

7.3.2 Set sediment sampler jaws to sampling position and slowly lower the dredge until it contacts the bottom. If using the Petite Ponar dredge, allow enough slack to release the locking pin, thus allowing the jaws to close upon retrieval. Allow a few seconds for the Petite Ponar dredge to sink into the sediment and slowly lift the sample to the surface. If using the Eckman dredge, hold the rope taut after allowing the dredge to sink into the sediment. Release the brass messenger down the rope to trip the dredge mechanism, closing the jaws. Slowly lift the sample to the surface.

7.3.3 Deposit the dredged sediment into a clean stainless-steel pan.

7.3.4 Collect the sample from the top sediment layer (1/2 inch) by using a clean stainless-steel spoon, or collect sediment directly into appropriately labeled 250-ml laboratory sample bottles. Avoid collecting any large organic debris (sticks, leaves, etc.), large particles (sand, gravel etc) and/or sediment material that has come into contact with the pan. Fill each bottle to approximately one-half full (i.e., 125 ml) to ensure enough material for lab analysis. Fill out the appropriate sample/sampling documentation (e.g., "Request for Laboratory Analysis" form).

7.3.5 Place bottles in a cooler with sufficient ice (a volume of ice equal to at least the volume occupied by samples, but preferably twice the volume of ice to samples) to maintain samples at 6^{o}C or less, or place the sample bottles refrigerator. Allow samples to settle for at least 24 hours. After sediments, including all fine or organic material, settle, decant excess water from each bottle (to prevent expansion and cracking) and place it in a standard freezer that maintains a temperature of -10^{o}C or less.

8.0 Sample Preservation, Holding Time and Shipment

Freeze sediment samples until delivery to the laboratory for analysis. Frozen samples may be held for up to six months before laboratory analysis. Take care to adequately pack glass sample bottles in shipping cooler to avoid breakage during shipment to laboratory and to keep samples frozen. Deliver completed RFLA sheets to the laboratory with sample bottles.

9.0 References

American Society for Testing and Materials. 1990. Standard Practice for Decontamination of Field Equipment Used at Nonradioactive Waste Sites. (Designation D 5088 90). Philadelphia, PA.

United States Geological Survey. 1998. Chapter 9. *National Field Manual for the Collection of Water Quality Data*. U.S. Department of the Interior. Reston, Virginia.

M:\Plandocs\Henebry\Documents for Web Site\SedimentCollectionSOP\SOP_CollectingSurficialSediment_DCN013_Rev 12112009.doc

Appendix G:

Continuous DO Monitoring SOP

Standard Operating Procedure
Continuous Recording of Selected Water Quality Parameters

Datasondes will be used in the Upper Desplaines River study area to record continuous water quality data for selected parameters over 3-5 day consecutive periods. The instruments will consist of either YSI 6-Series V2 model or EXO2 model units and used in accordance with the manufacturer specifications (YSI 2017). Each monitoring crew is required to maintain a calibration and maintenance log for each Datasonde Unit. The log will have consecutively numbered pages and include the following information at a minimum: date, Datasonde Model, Datasonde I.D. Number, description of monitoring (survey name), calibration comments, maintenance performed, and crew leader name. Each instrument will be clearly identified (*e.g.,* the make, model, serial and/or I.D. number) to differentiate among multiple units. The appropriate calibration procedure must be followed and if the instrumentation does not have an electronic program that maintains a running calibration log, the results will be recorded in the logbook each time that unit is used, along with the date and name/initials of the person performing the calibration. If any difficulties are encountered during calibration or if the instrument will not hold calibration, this information will also be recorded. Malfunctioning equipment will not be used to collect data and will be scheduled for maintenance and/or repair and recorded in the log indicating what was done to correct the problem, along with the date and initials of the person that identified the problem.

General Unit Operation and Maintenance

The datasondes use batteries as a power source – the EXO2 AA and the 6920V2 uses C batteries. When using alkaline batteries, users can expect approximately 90 days of deployment from a fully loaded datasonde that records once every 15 minutes at 20˚C with a central wiper which rotates once every logging interval. Deployment times can vary depending on water temperature, sampling rate, sensor payload, wiper frequency, and brand of battery. Estimated battery life is approximately 90 days for EXO2. Battery life is dependent on sensor configuration and is given for a typical sensor ensemble.

An EXO datasonde is always in one of three operational states, Off, Awake, or Asleep. These states determine the power usage and logging potential. When Off, the unit is not powered and cannot collect data. Users can apply power internally, using batteries, or externally with an EXO field cable attached from the topside port to an EXO Handheld, DCP, or other approved power source. Once power is applied it is either Awake or Asleep. When Asleep, the unit remains in a very low power setting and waits for a user command or its next scheduled logging interval. When Awake the unit is fully powered and ready to collect data remaining in that state for five minutes after its last communication via Bluetooth or 30 seconds after its last communication via the topside port. The unit also automatically awakens 15 seconds before its next scheduled logging interval. Within the Awake state, the unit has three modes, which are activated via KorEXO software. When “Inactive (Off),” no data is logged. In “Real-Time” mode, the unit continuously records data at a user-specified interval (default is 2 Hz). The

"Sample/Hold" mode allows users to easily synchronize data between the data logger and an external data collection platform.

Proper maintenance is essential for obtaining accurate and quality data. Preventative maintenance includes frequent inspection and cleaning and checking for leaks. A maintenance kit is provided by the manufacturer and includes lubricants and replacement O-rings. Each probe and the wipe require specific steps for cleaning and maintenance. Annual maintenance is also performed and is done in accordance with the manufacturers specifications with needed repairs done by the manufacturer. Storage of the units when they are not being used includes short-term and long-term procedures (YSI 2017).

Parameter Specific Operation

The Datasonde units provide for a baseline of four parameters, dissolved oxygen (D.O.), pH, conductivity (relative and specific), and temperature. The EXO units have the capability to measure additional parameters with the addition of specific probes. However, for this survey the four baseline parameters will be emphasized. The Datasonde units are maintained and operated in accordance with the manufacturer instructions and specifications. Units are set in a representative location at a site and secured to prevent theft and vandalism and are checked as often as necessary to ensure data quality. Each unit is secured in a PVC housing designed to protect the unit from damage while submerged while permitting water to flow over the sensors. Each unit and the protective housing are secured to a stationary object or stakes driven into the bottom. A summary of parameter-specific procedures follows:

Dissolved Oxygen Measurement

Dissolved oxygen (D.O.) is measured in mg/L and with an optical sensor and is derived by the Stern-Volmer equation. Variables that affect D.O. measurements include temperature, salinity, and barometric pressure. Temperature and salinity are compensated for during instrument calibration and field use with instrument software settings. Barometric pressure is adjusted for by sensor calibration to a standard pressure. Calibration is performed at the time of unit deployment and checked daily when deployed over multiple days.

pH Measurement

The unit should be maintained and operated in accordance with the manufacturer instructions and specifications. Calibration is performed at the time of unit deployment using two reference buffers. If the expected reading is alkaline, pH 7 and 10 buffers are used. If the expected reading is acidic, pH 7 and 4 buffers are used. The value of the sample should register within 0.2 S.U. of the selected buffers. Buffer solutions should be clearly marked with the expiration date and only used within the expiration date denoting this information is the calibration log. If a temperature compensating pH probe is not used the instrument should be calibrated under field conditions. The buffer solutions and unknown solutions will need to be at the same temperature (i.e., within ±2˚C) prior to measurement. If this is not the case, the temperature of the buffer solution can be adjusted by submerging the closed bottle of buffer solution in the test water for several minutes prior to use. Since the pH of reference buffer solution varies

slightly with temperature, it will be necessary to use the pH value of the buffer at the adjusted temperature when standardizing the instrument (see Table 1). A table of these values should be available in the field. Using of temperature compensating pH probes will eliminate this step.

***Table 1**. Variation of standard pH buffers with temperature.*

pH electrodes will be stored, cleaned and maintained according to the manufacturer specifications. Storage solutions may include buffers or a solution of saturated KCl.
If the pH electrode becomes coated with deposits during use, it is cleaned using a mild detergent and a soft cloth, or by soaking for short time in a solution of 0.1 N hydrochloric acid, followed by a thorough rinse with distilled water.

Conductivity Measurement
Calibration is performed at the time of unit deployment and logged. Table 2 shows the relationship between conductivity and ambient temperature.

***Table 2**. Variation of a 0.01N KCl conductivity standard with temperature.*

Temperature (˚C)	Conductivity (µS/cm)	Temperature (˚C)	Conductivity (µS/cm)
15	1147	23	1359
16	1173	24	1386
17	1199	25	1413
18	1225	26	1441
19	1251	27	1468
20	1278	28	1496
21	1305	29	1524
22	1332	30	1552

The conductivity sensor uses four internal, pure-nickel electrodes to measure solution conductance. Two of the electrodes are current driven, and two are used to measure the voltage drop. The measured voltage drop is then converted into a conductance value in milliSiemens (mS/cm) and the results are reported in microSiemens (µS/cm). Values are reported as relative conductivity and/or specific conductance at 25°C.

Temperature Measurement
The temperature sensor uses a highly stable and aged thermistor with extremely low-drift characteristics measuring temperature via resistance. The measured resistance is then converted to temperature using an algorithm. The temperature sensor receives a multi-point NIST traceable wet calibration during factory assembly and the accuracy specification of 0.01˚C

is valid for expected life of the probe. No calibration or maintenance of the temperature sensor is required, but accuracy checks can be conducted against a NIST-traceable temperature probe supplied by a user.

Reference

YSI. 2017. EXO User Manual - Advanced Water Quality Monitoring Platform. Item# 603789REF Revision G. YSI Incorporated, Yellow Springs, Ohio. 153 pp.

Appendix H:

Benthic Periphyton Sampling SOP

Standard Operating Procedure
Benthic Periphyton Sampling

Benthic periphyton is collected to provide data on chlorophyll a content in support of the determination of the effect of nutrients as part of a combined nutrient approach that includes the diel D.O. flux as measured by a Datasonde continuous monitors deployed at the same location. The results of the biological assemblage assessment (fish and macroinvertebrates) and concentrations of total phosphorus and nitrogen are also part of the combined assessment. This SOP focuses on collection of benthic periphyton for chlorophyll a analysis.

Sample Collection

Benthic periphyton samples will be collected during a representative low flow period between early July and late August and to coincide with Datasonde deployment. Field data is recorded on a periphyton sample collection form (Figure 1). Field sampling procedures are based on substrate characteristics as follows:

1. At sites where coarse substrate (cobbles, woody materials, etc.) are present that can be removed from the water the procedure is as follows:

    a) Collect a sample of benthic periphyton by scraping a known area from each of ten to twenty (usually fifteen) large gravel to cobble size rocks from a glide-riffle-run complex. Place the substrate in a plastic funnel which drains into a 500-mL plastic bottle. Use an area delimiter to define a known area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter into the funnel by brushing with a stiff-bristled toothbrush for 30 seconds. Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed, and that the entire surface within the delimiter is scrubbed.
    b) Fill a wash bottle with stream water. Wash the dislodged periphyton from each substrate into a funnel into a 500-mL amber bottle. Combine and blend the slurry with a rechargeable stick blender (Cuisinart model CSB-77 or equivalent). Draw three 5 ml aliquots from the blended slurry and filter on Whatman GF/C1.2 micron glass fiber filter in the field, and place on ice or freeze on dry ice for overnight trip or shipping.

2. At sites with large coarse substrates that are too large to remove from the water (bedrock, large woody materials, boulders, etc.) the procedure is as follows:

    a) Use the area delimiter to define a known area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimiter using the tip of the syringe in a scraping motion.
    b) While dislodging periphyton with the syringe tip, simultaneously pull back on the syringe

Figure 1. *Periphyton sample for benthic chlorophyll a analysis field collection form.*

Periphyton Sample Collection Form

Site Description _____ Site Code_____

Sampler name(s) _____

Date (mm/dd/yyyy)_____ Time (military)_____

Present weather conditions_____

Past 24 hrs. weather conditions _____

Past Week Flow Conditions _____

Slurry Volume (ml): _____ Number of rocks sampled: _____

Area scraped per rock: _____cm^2

Volume Filtered:

Filter 1: __5mL__
Filter 2: __5mL__
Filter 3: __5mL__

GPS unit X: _____Y:_____

---

c) plunger to draw the dislodged periphyton into the syringe. Repeat for ten to twenty (usually fifteen) location within sampling area.
d) Empty the syringes into the same 500-mL plastic amber bottle as above.
e) Combine and blend the slurry with a rechargeable stick blender (Cuisinart model CSB-77 or equivalent). Draw three 5 ml aliquots from the blended slurry and filter on Whatman GF/C1.2 micron glass fiber filter in the field, and place on ice or freeze on dry ice for overnight trip or shipping.

3. At sites with no coarse substrates (cobbles and larger) the procedure is as follows:

a) Use the area delimiter to confine a known area of soft sediments.
b) Vacuum the top 1 cm of sediments from within the delimited area into a de-tipped 60-mL syringe.
c) Empty the syringe into the same 500-mL plastic bottle as above.

d) Combine and blend the slurry with a rechargeable stick blender (Cuisinart model CSB-77 or equivalent). Draw three 5 ml aliquots from the blended slurry and filter on Whatman GF/C1.2 micron glass fiber filter in the field, and place on ice or freeze on dry ice for overnight trip or shipping.

Three separate subsamples of the slurry (defined volume) are field filtered thru three separate filters. The filters are then placed in individual zip lock bags wrapped in aluminum foil and placed on ice for shipping. Each sample is labeled by site code, date, time of collection, and sample collector. Samples received at the lab are stored at -20°C for a maximum of 28 days until analysis.

Equipment Maintenance

Care must be taken that all required equipment is properly cleaned prior to the preparation of equipment blanks and collection of samples. Clean all non-metal equipment with dilute HCl acid rinse. Soap (non-phosphate) and tap water are used on all equipment followed by a distilled water rinse. In the field, where such cleaning is not possible, a distilled water rinse is done before collecting a sample at each site.

Appendix I:

Stream Nutrient Assessment Procedure (SNAP)

Proposed Stream Nutrient Assessment Procedure (SNAP)

<table>
<tr><th>STEP 1</th><th>STEP 2</th><th>STEP 3</th><th colspan="2">STEP 4</th></tr>
<tr><th>Biological Criteria</th><th>DO Swing [2]</th><th>Benthic Chlorophyll [3]</th><th colspan="2">Preliminary Assessment:
Trophic Condition Status
of Evaluated ~~Reach~~ Segment or
Waterbody</th></tr>
<tr><td rowspan="3">All indices attaining
or in non-significant
departure[1]</td><td rowspan="2">Normal or low swings
(≤6.5 mg/l)</td><td>Low to moderate
(≤320 mg/m^2)</td><td colspan="2">Attaining use /
Not threatened</td></tr>
<tr><td>High
(>320 mg/m^2)</td><td rowspan="3">Attaining use,
but may be threatened</td><td rowspan="3">See
Flow
Chart A</td></tr>
<tr><td rowspan="2">Wide swings
(>6.5 mg/l)</td><td>Low
(≤182 mg/m^2)</td></tr>
<tr><td>Moderate to high
(>182 mg/m^2)</td></tr>
<tr><td rowspan="3">Non-attaining
(one or more indices
below non-significant
departure)</td><td rowspan="2">Normal or low swings
(≤6.5 mg/l)</td><td>Low to moderate
(≤320 mg/m^2)</td><td>Impaired, but cause(s) other
than nutrients</td><td>See
Flow
Chart B</td></tr>
<tr><td>High
(>320 mg/m^2)</td><td rowspan="2">Impaired; likely nutrients
over-enrichment</td><td rowspan="2">See
Flow
Chart C</td></tr>
<tr><td rowspan="2">Wide swings
(>6.5 mg/l)</td><td>Low
(≤182 mg/m^2)</td></tr>
<tr><td>Moderate to high
(>182 mg/m^2)</td><td>Impaired;
Nutrients over-enrichment</td></tr>
</table>

Proposed Stream Nutrient Assessment Procedure (SNAP) -- continued

_____

Notes:

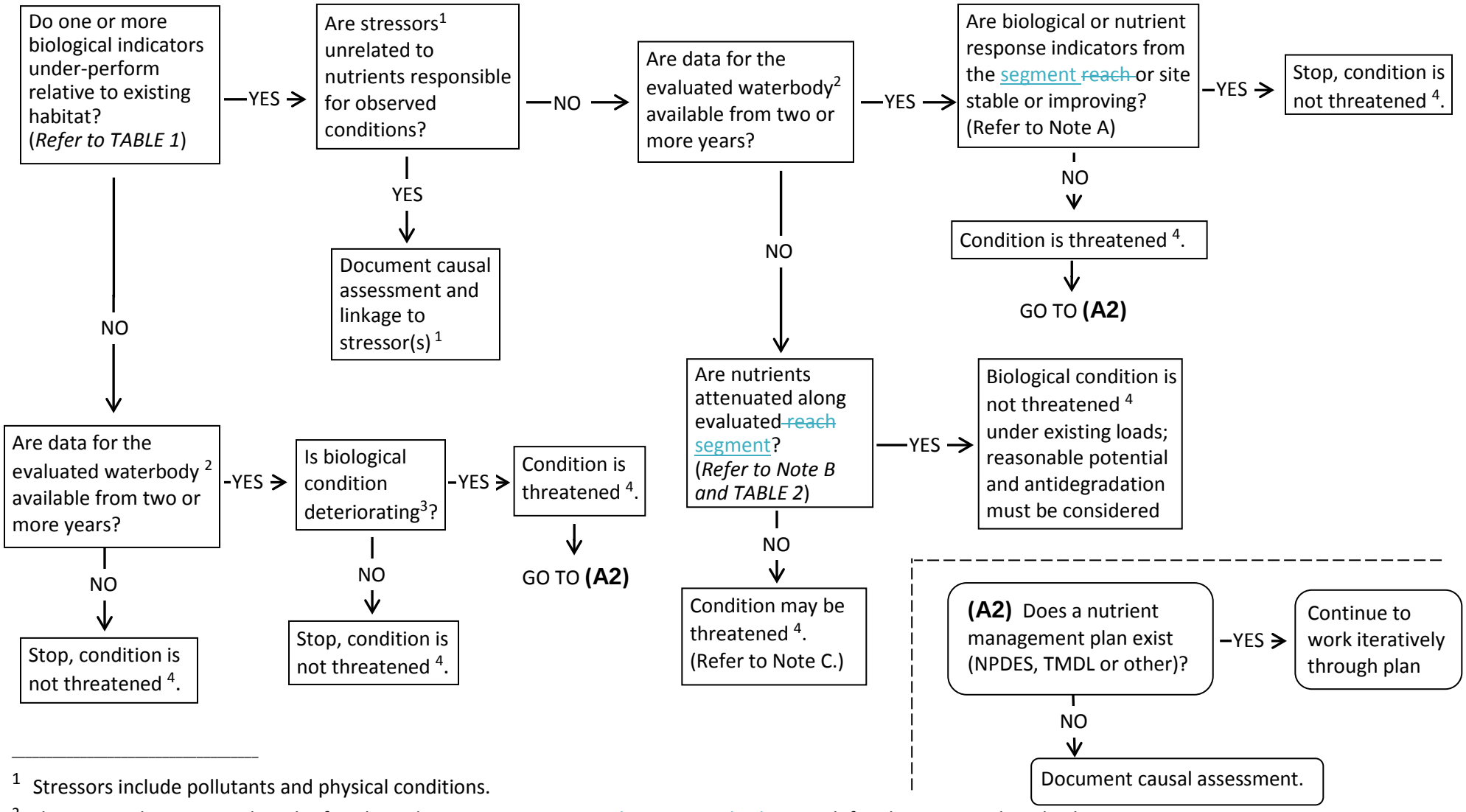
[1] Non-significant departure from biocriteria values accounts for background variability in measurements for biological indices. In accordance with "*Biological Criteria for the Protection of Aquatic Life: Volume II: Users Manual for Biological Field Assessment of Ohio Surface Waters*", Ohio EPA (1987, updated 1988, 1989, 2006), non-significant departure is 4 points for IBI and ICI, and 0.5 point for MIwb.

[2] Threshold value for 24-hour DO swing based upon a change point of 6.5 mg/l between DO swing and minimum DO. "Low to normal" DO swing is ≤6.5 mg/l. "Wide" DO swing is >6.5 mg/l. Data used for analysis from *Technical Support Document for Nutrient Water Quality Standards for Ohio Rivers and Streams*, Ohio EPA (2011).

[3] Threshold values for benthic chlorophyll *a* are based upon change points between benthic chlorophyll *a* and DO swings or Invertebrate Community Index (ICI). "Low" chlorophyll *a* is ≤182 mg/m^2. "Moderate" chlorophyll *a* is >182 and ≤320 mg/m^2. "High" chlorophyll *a* is >320 mg/m^2. Data used for analysis from *Technical Support Document for Nutrient Water Quality Standards for Ohio Rivers and Streams*, Ohio EPA (2011).

FLOW CHART A. – DECISION TREE FOR DETERMINING WHEN BIOLOGICALLY ATTAINING CONDITION STATUS IS THREATENED BY NUTRIENTS

For application when biological criteria are attaining, but one or both nutrient response indicators (DO swing or benthic chlorophyll) are elevated.



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[1] Stressors include pollutants and physical conditions.

[2] The geographic scope or length of evaluated stream segments ~~reaches or waterbodies~~ are defined in approved study plans.

[3] For a given ~~site~~location, a decrease of 5 or more IBI or ICI points, or 0.6 or more MIWb points between sampling years ~~can~~ represents a significant change. Trends for waterbodies are formally evaluated in Biological and Water Quality Technical Support Documents.

[4] As recommended by US EPA in its integrated reporting guidance (*Guidance for 2006 Assessment, Listing and Reporting Requirements Pursuant to Sections 303(d), 305(b) and 314 of the Clean Water Act*), "threatened" waters are currently attaining WQSs but are expected to not meet WQSs by the next listing cycle (every two years). For example, a declining trend may indicate threatened status, whereas a stable or improving trend would not.

Flow Chart A (continued) – Additional Notes:

Note A. Two set of circumstances result in a determination of "threatened" by nutrient impacts – (1) when biological indicators are underperforming relative to habitat and biological or nutrient response indicators are not stable or improving; and (2) when (although biological indicators are not underperforming) the biological condition is deteriorating. For such cases, the Flow Chart at "A2" provides a conditional evaluation for a subset of cases where existing nutrient management plans exist, either via NPDES permit, TMDL, or other. In such cases, the Flow Chart indicates that the nutrient management plan shall continue to be implemented iteratively, reviewing and reassessing the results of implementation.

~~The top row provides a conditional evaluation for a subset of cases where existing nutrient management plans exist, either via NPDES permits or a TMDL. To enter this row, the determination has already been made that nutrient response indicators are elevated, and biological indicators are under-performing relative to habitat. This evaluation identifies cases where biology may be under-performing, but is on an improving trajectory due to management. An existing management plan implies historic data exist, and that the reach was likely flagged as impaired; therefore, in most cases, to get to the right hand side of this row presupposes that the biological condition has already improved.~~

Note B. Attenuation of nutrients in an evaluated ~~reach~~ segment is demonstrated by nutrient concentrations measured at two or more successive sites downstream from a defined source decreasing through uptake, sequestration or dilution such that concentrations fall to either background levels or levels where risk of eutrophication to downstream waters is minimal (see Table 2). Where there are no historic data on which to base trends, attenuation of nutrients within the ~~reach~~ segment implies assimilation within what the waterbody can handle under existing conditions, and that stress from the nutrient load is spatially transient (i.e., localized to the immediate ~~reach~~segment).

Note C. If attenuation appears ambiguous or cannot be determined because of an insufficient number of downstream sampling points between the source in question and the next downstream receiving water or the next downstream major source contributor, additional sampling is needed to determine condition status.

TABLE 1 – Equations used as guidance to help determine whether biological indicators are underperforming relative to existing habitat.

To assist in determining whether measured biological indicator values at the site being assessed underperform relative to the existing habitat, the measured value(s) are compared with the 25th and 15th percentile values of all data classified as unimpaired in the Ohio EPA assessment database and stratified by the designated classification (EWH, WWH or MWH) within the specific ecoregion for the site. The 25th and 15th percentiles represent levels that most sites equal or exceed. If the respective measured biological indicator value is less than the 15th percentile value then the site is likely underperforming relative to what could be expected given the local habitat quality (QHEI). If the indicator value is between the 15th and 25th percentile values, additional information or observations should be used to determine whether ~~or not~~ the site is underperforming with respect to its habitat. If the indicator value is above the 25th percentile value, the site ~~would be considered~~ is performing within the range expected for the existing habitat.

The following equations calculate the 25th and 15th percentile values as determined by regression analysis for the respective biological indicators for a given QHEI score, or a combination of QHEI score and drainage area. For small streams where insufficient stream flow prevents collection of a quantitative sample, thereby precluding calculation of an ICI score, the number of EPT taxa is used as the macroinvertebrate indicator. Such small streams are typically less than 20 square miles in drainage area, or larger if stream velocity is insufficient to collect a quantitative sample.

Class / Ecoregion		Percentile	IBI (fish)	MIWb (fish)	EPT Taxa (macroinvertebrates)	ICI (macroinvertebrates)
EWH / All Ecoregions		25th	40.67 + 0.118·QHEI	8.21 + 0.006·QHEI + 0.385·Log10(DA)	4.65 + 0.123·QHEI + 1.182·Log10(DA)	= 46
		15th	39.60 + 0.113·QHEI	NA	1.47 + 0.151··QHEI + 1.084·Log10(DA)	NA
WWH & MWH	HELP	25th	23.65 + 0.150··QHEI	5.64 + 0.959·Log10(DA)	4.26 + 2.585·Log10(DA)	All Ecoregions: 25th percentile: 25.60 + 0.160·QHEI 15th percentile: 19.32 + 0.213·QHEI
		15th	22.00 + 0.121·QHEI	NA	2.54 + 2.659·Log10(DA)	
	EOLP	25th	22.00 + 0.316·QHEI	4.76 + 0.043·QHEI + 0.491·Log10(DA)	NA	
		15th	18.24 + 0.336·QHEI	4.55 + 0.045·QHEI + 0.397·Log10(DA)	= 9 taxa	
	WAP	25th	31.30 + 0.200·QHEI	7.94 + 0.537·Log10(DA)	3.94 + 0.114·QHEI	
		15th	27.78 + 0.225·QHEI	7.58 + 0.543·Log10(DA)	2.14 + 0.113·QHEI	
	ECBP & IP	25th	29.96 + 0.157·QHEI	4.94 + 0.036·QHEI + 0.388·Log10(DA)	-0.95 + 0.147·QHEI + 0.927·Log10(DA)	
		15th	29.47 + 0.133·QHEI	4.96 + 0.034·QHEI + 0.362·Log10(DA)	-2.19 + 0.138·QHEI + 1.010·Log10(DA)	

NA = Not Available. Could not be determined because of limited data or data distribution.
DA = Drainage Area (in square miles)

TABLE 2 – Concentrations of total phosphorus (TP) and dissolved inorganic nitrogen (DIN) arrayed by narrative levels of ecological risk.

Table 2 presents narrative descriptions of various levels of ecological condition and potential risk, arrayed with ranges of nutrient concentrations commonly observed at the respective ecological condition levels. This information may be useful reference for nutrient assessment using Charts A or C. **Chart A**: Attenuation from a defined source may be inferred by nutrient concentrations measured at successive stations within an evaluated decreasing from a higher risk level to a lower risk level. **Chart C**: Table 2 may be used as a general reference in assessing impairment risk. Actual risks and the potential benefits of abatement are site-specific determinations.

		← *DECREASING RISK*				
	TP Conc. (mg/l)	**DIN Concentration** (mg/l)				
		<0.44	0.44 < 1.10	1.10 < 3.60	3.60 < 6.70	≥6.70
DECREASING RISK →	<0.040	background levels typical of least disturbed conditions	levels typical of developed lands; little or no risk to beneficial uses	levels typical of modestly enriched condition in phosphorus limited systems; low risk to beneficial use if allied responses are within normal ranges	levels typical of enriched condition in phosphorus limited systems; moderate risk to beneficial use if allied responses are elevated	characteristic of tile-drained lands; otherwise atypical condition with moderate risk to beneficial use if allied responses are elevated (1.1% of observations)
	0.040-<0.080	levels typical of developed lands; little or no risk to beneficial uses	levels typical of developed lands; little or no risk to beneficial uses	levels typical of working landscapes; low risk to beneficial use if allied responses are within normal ranges	levels typical of enriched condition in phosphorus limited systems; moderate risk to beneficial use if allied responses are elevated	characteristic of tile-drained lands; moderate risk to beneficial use if allied responses are elevated (1.1% of observations)
	0.080-<0.131	levels typical of modestly enriched condition in nitrogen limited systems; low risk to beneficial use if allied responses are within normal ranges	levels typical of working landscapes; low risk to beneficial use if allied responses are within normal ranges	levels typical of working landscapes; low risk to beneficial use if allied responses are within normal ranges	characteristic of tile-drained lands; moderate risk to beneficial use if allied responses are elevated; increased risk with poor habitat	characteristic of tile-drained lands; moderate risk to beneficial use if allied responses are elevated (1.0% of observations)
	0.131-<0.400	levels typical of modestly enriched condition in nitrogen limited systems; low risk to beneficial use if allied responses are within normal ranges	levels typical of enriched condition; low risk to beneficial use if allied responses are within normal ranges	levels typical of enriched condition; low risk to beneficial use if allied responses are within normal ranges; increased risk with poor habitat	enriched condition; generally high risk to beneficial uses; often co-occurring with multiple stressors; increased risk with poor habitat	enriched condition; generally high risk to beneficial uses; often co-occurring with multiple stressors
	≥0.400	atypical condition (1.3% of observations)	atypical condition (1% of observations);	enriched condition; generally high risk to beneficial uses; often co-occurring with multiple stressors; increased risk with poor habitat	enriched condition; generally high risk to beneficial uses; often co-occurring with multiple stressors ; increased risk with poor habitat	enriched condition; generally high risk to beneficial uses; often co-occurring with multiple stressors

"allied responses" = allied response indicators (24-hour DO swing, benthic chlorophyll)

TABLE 2 (continued)

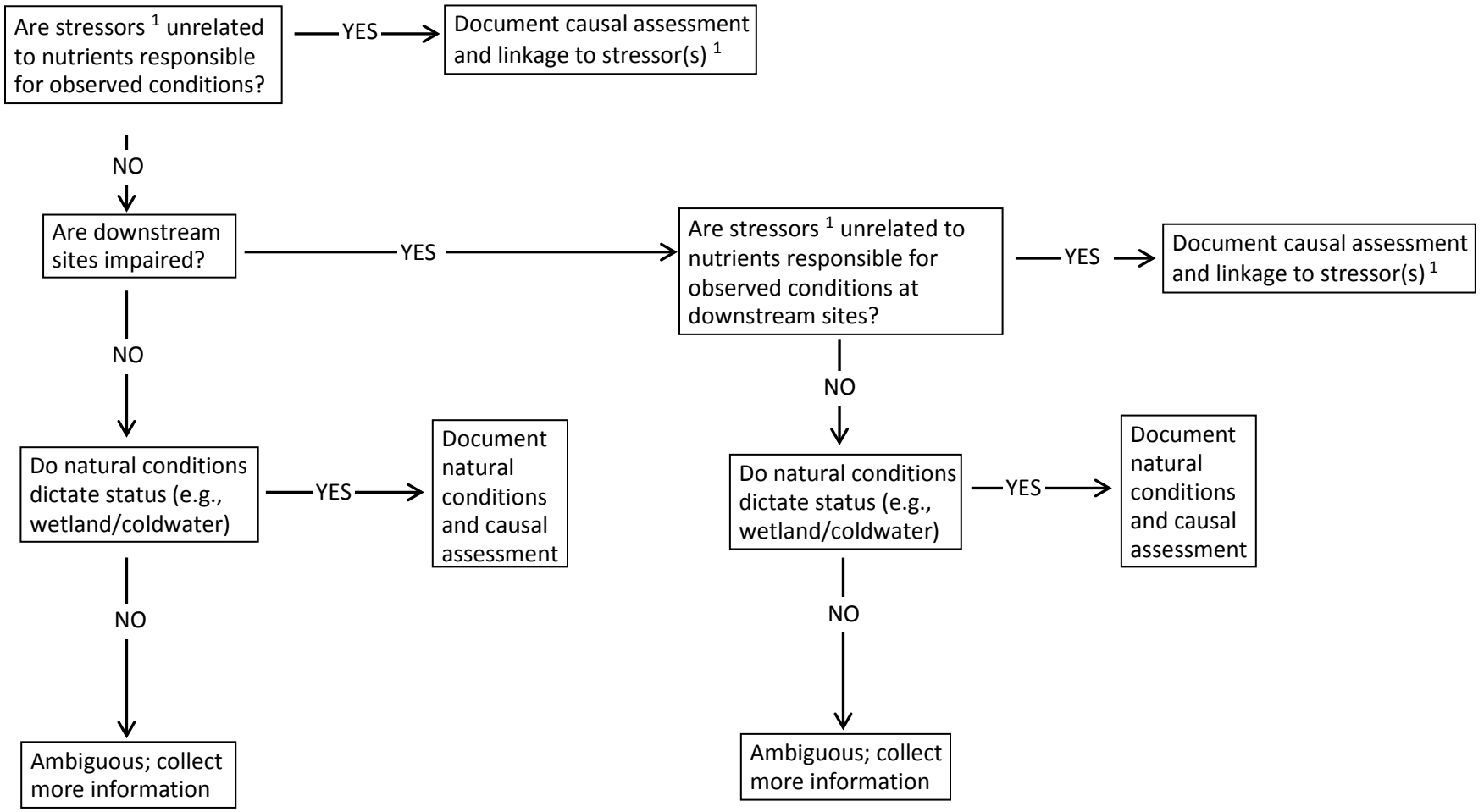
Ohio EPA's monitoring data for the years 1981 through 2011 (n = 16,870), from index period samples (June-October) and all stream sizes, was used to derive the information presented in Table 2. Following is the frequency of occurrence in the database for each nutrient concentration range, expressed as percent of total data values.

Frequency of Occurrence in Database, as Percent of Total (n=16,870)

Total Phosphorus (TP) [mg/l]	**Dissolved Inorganic Nitrogen (DIN)** [mg/l]				
	<0.44	0.44 < 1.10	1.10 < 3.60	3.60 < 6.70	≥6.70
<0.040	18.14%	5.00%	4.26%	1.13%	0.66%
0.040 < 0.080	6.50%	5.66%	4.87%	1.11%	0.29%
0.080 < 0.131	3.30%	3.77%	5.20%	1.01%	0.31%
0.131 < 0.400	3.62%	4.31%	11.39%	3.01%	1.45%
≥0.400	1.33%	0.99%	4.84%	4.07%	3.78%

FLOW CHART B – DECISION TREE FOR DETERMINING BIOLOGICAL IMPAIRMENT CAUSED BY STRESSORS OTHER THAN NUTRIENTS

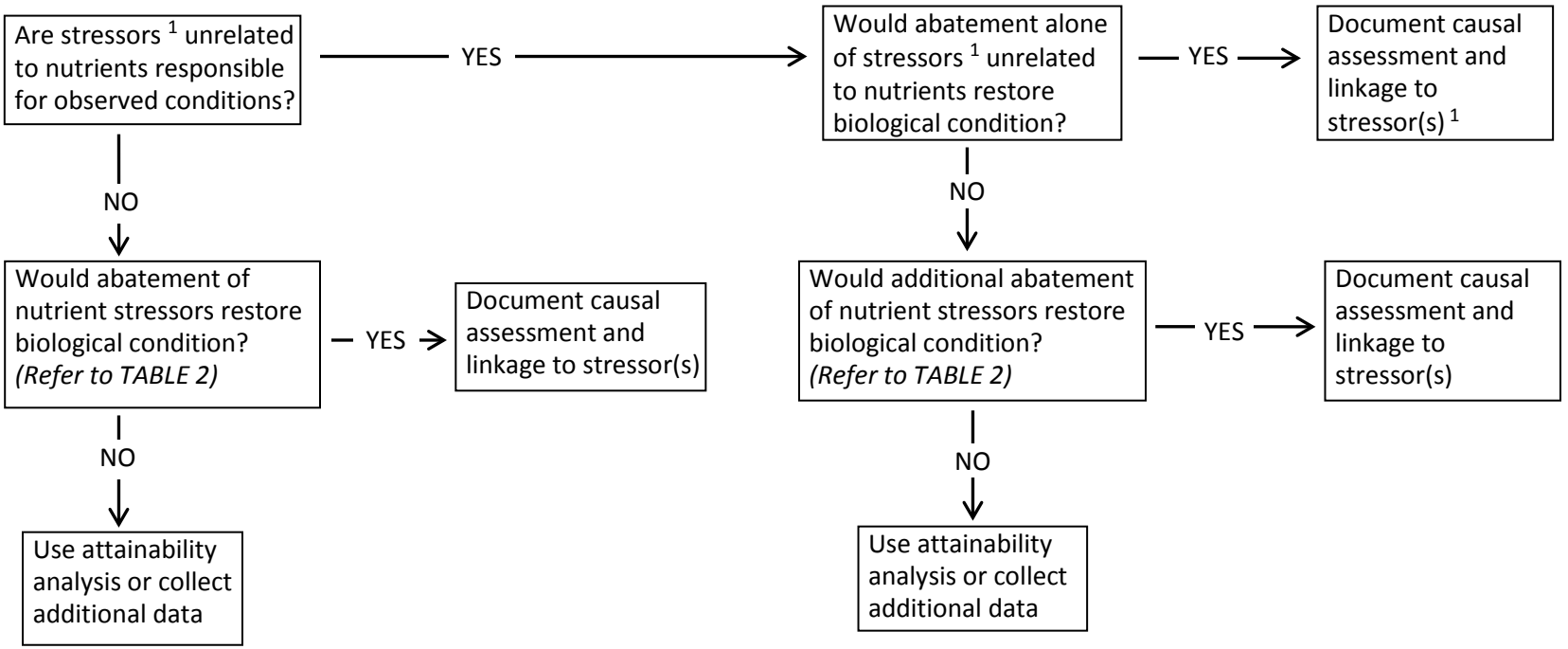
For application when one or more biological criteria are non-attaining, but no nutrient response indicators (DO swing or benthic chlorophyll) are elevated.



[1] Stressors include pollutants and physical conditions.

FLOW CHART C – DECISION TREE FOR CONFIRMING BIOLOGICAL IMPAIRMENT CAUSED BY NUTRIENTS

For application when one or more biological criteria are non-attaining, and either nutrient response indicator (DO swing or benthic chlorophyll) is elevated.



[1] Stressors include pollutants and physical conditions.