Calcasieu Estuary Remedial Investigation/Feasibility Study (RI/FS): Baseline Ecological Risk Assessment (BERA)

Appendix A2: Baseline Problem Formulation
Volume II - Appendices

Prepared For:

CDM Federal Programs Corporation
8140 Walnut Hill Lane, Suite 1000
Dallas, Texas 75231

Under Contract To:

Mr. John Meyer, Regional Project Manager
U.S. Environmental Protection Agency, Region 6
1445 Ross Avenue
Dallas, Texas 75202

Prepared – October 2002 – By:

MacDonald Environmental Sciences Ltd.
#24 - 4800 Island Highway North
Nanaimo, British Columbia V9T 1W6

In Association With:

The Cadmus Group, Inc.
411 Roosevelt Street, Suite 204
Ottawa, Ontario K2A 3X9

CONTRACT NO. 68-W5-0022
DOCUMENT CONTROL NO. 3282-941-RTZ-RISKZ-14858

Calcasieu Estuary Remedial Investigation/Feasibility Study (RI/FS): Baseline Ecological Risk Assessment (BERA)

Appendix A2:
Baseline Problem Formulation
Volume II - Appendices

Prepared For:

[1]CDM Federal Programs Corporation
8140 Walnut Hill Lane, Suite 1000
Dallas, Texas 75231

Under Contract To:

Mr. John Meyer, Regional Project Manager
U.S. Environmental Protection Agency, Region 6
1445 Ross Avenue
Dallas, Texas 75202

Prepared – October 2002 – By:

D.D. MacDonald[2], D.R.J. Moore[3], A. Pawlisz[3], D.E. Smorong[2], R.L. Breton[3], D.B. MacDonald[3], R. Thompson[3], R.A. Lindskoog[2], M.A. Hanacek[2], and M.S. Goldberg[1]

[2]MacDonald Environmental Sciences Ltd.
#24 - 4800 Island Highway North
Nanaimo, British Columbia V9T 1W6

[3]The Cadmus Group, Inc.
411 Roosevelt Street, Suite 204
Ottawa, Ontario K2A 3X9

CONTRACT NO. 68-W5-0022
DOCUMENT CONTROL NO. 3282-941-RTZ-RISKZ-14858

Table of Contents

List of Tables VI

List of Acronyms VII

Glossary of Terms XI

Appendix A2-1 Goals and Objectives of the Remedial Investigation and Feasibility Study 1

Appendix A2-2 An Overview of the Environmental Fate and Effects of Copper 4
2.1 Identity 4
2.2 Uses 4
2.3 Sources 4
2.4 Physical and Chemical Properties 5
2.5 Environmental Fate and Transport 5
2.6 Bioaccumulation 6
2.7 Effects on Aquatic and Terrestrial Organisms 6
2.8 References 8

Appendix A2-3 An Overview of the Environmental Fate and Effects of Chromium 11
3.1 Identity 11
3.2 Uses 11
3.3 Sources 12
3.4 Physical and Chemical Properties 12
3.5 Environmental Fate and Transport 13
3.6 Bioaccumulation 13
3.7 Effects on Aquatic and Terrestrial Organisms 14
3.8 References 15

Appendix A2-4 An Overview of the Environmental Fate and Effects of Lead 18
4.1 Identity 18
4.2 Uses 18
4.3 Sources 19
4.4 Physical and Chemical Properties 19
4.5 Environmental Fate and Transport 20
4.6 Bioaccumulation 20
4.7 Effects on Aquatic and Terrestrial Organisms 21
4.8 References 23

Appendix A2-5 An Overview of the Environmental Fate and Effects of Mercury 25
5.1 Identity 25
5.2 Uses 25
5.3 Sources 26
5.4 Physical and Chemical Properties 26
5.5 Environmental Fate and Transport 27
5.6 Bioaccumulation 27
5.7 Effects on Aquatic and Terrestrial Organisms 27
5.8 References 29

Appendix A2-6 An Overview of the Environmental Fate and Effects of Nickel 34
6.1 Identity 34
6.2 Uses 34
6.3 Sources 35
6.4 Physical and Chemical Properties 35
6.5 Environmental Fate and Transport 36
6.6 Bioaccumulation 36
6.7 Effects on Aquatic and Terrestrial Organisms 37
6.8 References 38

Appendix A2-7 An Overview of the Environmental Fate and Effects of Zinc 42
7.1 Identity 42
7.2 Uses 42
7.3 Sources 43
7.4 Physical and Chemical Properties 43
7.5 Environmental Fate and Transport 43
7.6 Bioaccumulation 44
7.7 Effects on Aquatic and Terrestrial Organisms 45
7.8 References 46

Appendix A2-8 An Overview of the Environmental Fate and Effects of Polycyclic Aromatic Hydrocarbons 49
8.1 Identity 49
8.2 Uses 49
8.3 Sources 50
8.4 Physical and Chemical Properties 50
8.5 Environmental Fate and Transport 51
8.6 Bioaccumulation 51
8.7 Effects on Aquatic and Terrestrial Organisms 53
8.8 References 55

Appendix A2-9 An Overview of the Environmental Fate and Effects of Polychlorinated Biphenyls 62
9.1 Identity 62
9.2 Uses 62
9.3 Sources 63
9.4 Physical and Chemical Properties 63
9.5 Environmental Fate and Transport 64
9.6 Bioaccumulation 64
9.7 Effects on Aquatic and Terrestrial Organisms 65
9.8 References 67

Appendix A2-10 An Overview of the Environmental Fate and Effects of Polychlorinated Dibenzo-*p*-Dioxins and Polychlorinated Dibenzofurans 75
10.1 Identity 75
10.2 Uses 75
10.3 Sources 76
10.4 Physical and Chemical Properties 76
10.5 Environmental Fate and Transport 77
10.6 Bioaccumulation 78
10.7 Effects on Aquatic and Terrestrial Organisms 79
10.8 References 81

Appendix A2-11 An Overview of the Environmental Fate and Effects of Chlorinated Benzenes: Hexachlorobenzene and Hexachlorobutadiene 88
11.1 Introduction 88
11.2 Hexachlorobenzene 88
11.2.1 Identity 88
11.2.2 Uses 88
11.2.3 Sources 89
11.2.4 Physical and Chemical Properties 89
11.2.5 Environmental Fate and Transport 90
11.2.6 Bioaccumulation 90
11.2.7 Effects on Aquatic and Terrestrial Organisms 91
11.3 Hexachlorobutadiene 93
11.3.1 Identity 93
11.3.2 Uses 93
11.3.3 Sources 93
11.3.4 Physical and Chemical Properties 94
11.3.5 Environmental Fate and Transport 94
11.3.6 Bioaccumulation 95
11.3.7 Effects on Aquatic and Terrestrial Organisms 95
11.4 References 97

Appendix A2-12 An Overview of the Environmental Fate and Effects of Bis(2-ethylhexyl)phthalate 107
12.1 Identity 107
12.2 Uses 107
12.3 Sources 107
12.4 Physical and Chemical Properties 108
12.5 Environmental Fate and Transport 108
12.6 Bioaccumulation 109
12.7 Effects on Aquatic and Terrestrial Organisms 110
12.8 References 111

Appendix A2-13 An Overview of the Environmental Fate and Effects of Dichloroethane 117
13.1 Identity 117
13.2 Uses 117
13.3 Sources 117
13.4 Physical and Chemical Properties 118
13.5 Environmental Fate and Transport 118
13.6 Bioaccumulation 119
13.7 Effects on Aquatic and Terrestrial Organisms 119
13.8 References 121

Appendix A2-14 An Overview of the Environmental Fate and Effects of Trichloroethane 126
14.1 Identity 126
14.2 Uses 126
14.3 Sources 126
14.4 Physical and Chemical Properties 127
14.5 Environmental Fate and Transport 127
14.6 Bioaccumulation 128
14.7 Effects on Aquatic and Terrestrial Organisms 128
14.8 References 129

Appendix A2-15 An Overview of the Environmental Fate and Effects of Carbon Disulfide 133
15.1 Identity 133
15.2 Uses 133
15.3 Sources 134
15.4 Physical and Chemical Properties 134
15.5 Environmental Fate and Transport 135
15.6 Bioaccumulation 135
15.7 Effects on Aquatic and Terrestrial Organisms 136
15.8 References 136

Appendix A2-16 An Overview of the Environmental Fate and Effects of Acetone 139
16.1 Identity 139
16.2 Uses 139
16.3 Sources 139
16.4 Physical and Chemical Properties 140
16.5 Environmental Fate and Transport 140
16.6 Bioaccumulation 140
16.7 Effects on Aquatic and Terrestrial Organisms 141
16.8 References 142

Appendix A2-17 An Overview of the Environmental Fate and Effects of Organochlorine Pesticides: Aldrin and Dieldrin 143
17.1 Aldrin 143
17.1.1 Identity 143
17.1.2 Uses 143
17.1.3 Sources 144
17.1.4 Physical and Chemical Properties 144
17.1.5 Environmental Fate and Transport 144
17.1.6 Bioaccumulation 145
17.1.7 Effects on Aquatic and Terrestrial Organisms . . . 146
17.2 Dieldrin 147
17.2.1 Identity 147
17.2.2 Uses 148
17.2.3 Sources 148
17.2.4 Physical and Chemical Properties 149
17.2.5 Environmental Fate and Transport 149
17.2.6 Bioaccumulation 150
17.2.7 Effects on Aquatic and Terrestrial Organisms . . . 150
17.3 References 152

Appendix A2-18 Profiles for Wildlife Species That Are Part of the Calcasieu Estuary Ecosystem 159
18.1 Wildlife Species That are Proposed Receptors at Risk . . . 159
18.1.1 Non-Perching Birds 159
18.1.2 Mammals 168
18.2 Other Species Resident to the Calcasieu Estuary Ecosystem 173
18.2.1 Birds Resident to the Calcasieu Estuary 173
18.2.2 Mammals Resident to the Calcasieu Estuary Ecosystem 199
18.2.3 Amphibians Resident to the Calcasieu Estuary Ecosystem 230
18.2.4 Reptiles Resident to the Calcasieu Estuary Ecosystem 242
18.3 References 264

List of Tables

Table A2-1 World Health Organization toxic equivalency factors (TEFs) for humans, mammals, fish, and birds (from van den Berg *et al.* 1998) 74

List of Acronyms

%	percent
ACGIH	American Conference of Governmental Industrial Hygienists, Inc
AQUIRE	Aquatic Toxicity Information Retrieval System
AHH	aryl hydrocarbon hydroxylase
ASTM	American Society for Testing and Materials
ATSDR	Agency for Toxic Substances and Disease Registry
BCF	bioconcentration factor
BEHP	bis(2-ethylhexyl)phthalate
BERA	baseline ecological risk assessment
BI	bioavailability index
BSAF	biota-sediment bioaccumulation factor
BW	body weight
CA	California
CAS	Chemical Abstracts Service
CCC	criterion continuous concentration
CCME	Canadian Council of Ministers of the Environment
CCREM	Canadian Council of Resource and Environment Ministers
CDM	CDM Federal Programs Corporation
CERCLIS	Comprehensive Environmental Response, Compensation, and Liability Information System
CERCLA	Comprehensive Environmental Response, Compensation, and Liability Act of 1980, 42 U.S.S. 9601 et seq.
CIS	Camford Information Services, Inc.
CLP	Contract Laboratory Program
cm	centimeter
CMA	Chemical Manufacturer's Association
CMC	criteria maximum concentration
CO	Colorado
COPC	chemical of potential concern
Cr	chromium
Cr(III)	trivalent chromium
Cr(IV)	hexavalent chromium
Cu	copper
DCE	1,2-dichloroethane
DEHP	di(2-ethylhexyl)phthalate; synonym of BEHP
DELT	deformities, fin erosion, lesions, and tumors
DL	detection limit
DNA	deoxyribonucleic acid
DO	dissolved oxygen
DQO	data quality objectives
DW	dry weight
EC_{50}	median effect concentration
EDC	ethylene dichloride

EDTA	ethylenediaminetetraacetic acid
Eh	oxidation/reduction potential
ERA	ecological risk assessment
ERM	effects range median
EROD	ethoxyresorufin *O*-deethylase
ETAG	Ecological Technical Assistance Group
FDA	Food and Drug Administration
FS	feasibility study
g/L	grams per liter
g/m^3	grams per cubic meter
g/mole	grams per mole
g/kg	grams per kilogram
HASP	health and safety plan
HCB	hexachlorobenzene
HCBD	hexachlorobutadiene
Hg	mercury
HMW-PAHs	high molecular weight polycyclic aromatic hydrocarbons
HSDB	hazardous substance databank
IARC	International Agency for Research on Cancer
IPCS	International Program on Chemical Safety
IRIS	Integrated Risk Information System
ITEF	international toxicity equivalency factor
kg	kilogram
K_{oc}	organic carbon partition coefficient
K_{ow}	octanol/water partition coefficient
LA	Louisiana
LC_{50}	median lethal concentration
LCL	lower confidence limit
LD_{50}	median lethal dose
LDEQ	Louisiana Department of Environmental Quality
LMW-PAHs	low molecular weight polycyclic aromatic hydrocarbons
LNHP	Louisiana Natural Heritage Program
LOAEL	lowest observed adverse effect level
LOEL	lowest observed effect level
LOEC	lowest observed effect concentration
MESL	MacDonald Environmental Sciences Ltd.
mg	milligram
mg/kg	milligrams per kilogram
mg/L	milligrams per liter
mg/m^3	milligrams per cubic meter
mm	millimeter
MFO	mixed function oxidase
mPa	millipascals (standard international unit for pressure)
MS	matrix spike
MSD	matrix spike duplicate
NAS	National Academy of Sciences
ng	nanogram

NG	no guideline
Ni	nickel
NIOSH	National Institute for Occupational Safety and Health
NOAEL	no observed adverse effect level
NOAA	National Oceanic and Atmospheric Administration
NOEL	no observed effect level
NPDES	National Pollutant Discharge and Elimination System
NPL	National Priorities List
NRC	National Research Council
NRCC	National Research Council of Canada
NTP	National Toxicology Program
OC	organic carbon
OH^-	hydroxide
Pa	pascals (standard international unit for pressure)
PAH	polycyclic aromatic hydrocarbon
Pb	lead
PCB	polychlorinated biphenyl
PCDD	polychlorinated dibenzo-*p*-dioxin
PCDF	polychlorinated dibenzofuran
PCS	Permit Compliance System
PEC	probable effect concentration
PEL	probable effect level
ppb	parts per billion
ppm	parts per million
QA/QC	quality assurance/quality control
QAPP	quality assurance project plan
QMP	quality monitoring program
QP	quality procedure
RCRA	Resource Conservation and Recovery Act
RI	remedial investigation
RNA	ribonucleic acid
ROI	receptors of interest
RTECS	Registry of Toxic Effects of Chemical Substances
SAP	sampling and analysis plan
SD	standard deviation
SERA	screening level ecological risk assessment
SMDP	scientific management decision point
SO_4^-	sulfate
SPF	specific pathogen free
SRI	Stanford Research Institute
SQG	sediment quality guideline
STORET	Storage and Retrieval System for water quality data
SVOCs	semi-volatile organic compounds
TAL	target analyte list
TCA	trichloroethane
TEF	toxic equivalency factor
TEQ	toxic equivalents

TOC	total organic carbon
TRI	Toxic Release Inventory
TU	toxic units
UCL	upper confidence limit
USDA	United States Department of Agriculture
USEPA	United States Environmental Protection Agency
USFWS	United States Fish and Wildlife Service
µg/kg	micrograms per kilogram
µg/L	micrograms per liter
µmol/g	micromoles per gram
VOCs	volatile organic compounds
WHO	World Health Organization
WQC	water quality criteria
WW	wet weight
Zn	zinc

Glossary of Terms

Acute toxicity threshold – The concentration of a substance above which adverse effects are likely to be observed in short-term toxicity tests.

Acute toxicity – The immediate or short-term response of an organism to a chemical substance. Lethality is the response that is most commonly measured in acute toxicity tests.

Adverse effects – Any injury (i.e., loss of chemical or physical quality or viability) to any ecological or ecosystem component, up to and including at the regional level, over both long and short terms.

Ambient – Of or relating to the immediate surroundings.

Aquatic organisms – The species that utilize habitats within aquatic ecosystems (e.g., aquatic plants, invertebrates, fish, amphibians and reptiles).

Aquatic-dependent species – Species that are dependent on aquatic organisms and/or aquatic habitats for survival.

Aquatic-dependent wildlife – Wildlife species that are dependent on aquatic organisms and/or wildlife habitats for survival, including fish, amphibians, reptiles, birds, and mammals (e.g., egrets, herons, kingfishers, osprey, racoons, mink, otter; see Figure A1-9).

Aquatic ecosystem – All the living and nonliving material interacting within an aquatic system (e.g., pond, lake, river, ocean).

Aquatic invertebrates – Animals without backbones that utilize habitats in freshwater, estuaries, or marine systems.

Benchmarks – Guidelines that are intended to define the concentration of a contaminant that is associated with a high or a low probability of observing harmful biological effects or unacceptable levels of bioaccumulation.

Benthic invertebrate community – The assemblage of sediment-dwelling organisms that are found within an aquatic ecosystem.

Bioaccumulation – The net accumulation of a substance by an organism as a result of uptake from all environmental sources.

Bioaccumulative substances – The chemicals that tend to accumulate in the tissues of aquatic and terrestrial organisms.

Bioavailability – Degree to which a chemical can be absorbed by and/or interact with an organism.

Bioconcentration – The accumulation of a chemical in the tissues of an organism as a result of direct exposure to the surrounding medium (i.e., it does not include food web transfer).

Biological half-life – The time required for one-half of the total amount of a particular substance in a biological system to be consumed or broken down by biological processes.

Biomagnification – The accumulation of a chemical in the tissues of an organism as a result of food web transfer.

Brackish marsh – A marsh of low salinity, usually up to 5 parts per thousand during the period of average annual low flow.

Brood – The young animals produced during one reproductive cycle.

Calanoid (copepods) – Small crustaceans, 1-5 mm in length, commonly found as part of the free-living zooplankton in freshwater lakes and ponds.

Catabolism – The phase of metabolism which consists in breaking down of complex substances into simpler substances.

Chelating agent – An organic chemical that can bond with a metal and remove it from a solution.

Chronic toxicity – The response of an organism to long-term exposure to a chemical substance. Among others, the responses that are typically measured in chronic toxicity tests include lethality, decreased growth, and impaired reproduction.

Chronic toxicity threshold – The concentration of a substance above which adverse effects on sediment-dwelling organisms are likely to occur in longer-term toxicity tests.

Colloids – Very small, finely divided solids (that do not dissolve) that remain dispersed in a liquid for a long time due to their small size and electrical charge.

Confluence – The location where two waterways meet.

Congener – A member of a group of chemicals with similar chemical structures (e.g., PCDDs generally refers to a group of 75 congeners that consist of two benzene rings connected to each other by two oxygen bridges).

Chemicals of potential concern – The substances that occur in environmental media at levels that pose a potential risk to ecological receptors or human health.

Contaminated sediment – Sediment that contains chemical substances at concentrations that could harm sediment-dwelling organisms, wildlife, or human health.

Cracking catalysts – Substances that speed-up petroleum refining processes (used to "crack" crude oil into gasoline, jet fuel, kerosene, diesel fuel, and other petroleum products).

Degradation – A breakdown of a molecule into smaller molecules or atoms.

Demethylated – Removal of a methyl group from a chemical compound.

Diagenesis – The sum of the physical and chemical changes that take place in sediments after its initial deposition (before they become consolidated into rocks, excluding all metamorphic changes).

Dimorphic – Existing in two forms (e.g., male and female individuals in animals).

Endpoint – A measured response of a receptor to a stressor. An endpoint can be measured in a toxicity test or a field survey.

Estivate – To pass the summer or dry season in a dormant condition.

Fumarolic – Describes a vent in or near a volcano from which hot gases, especially steam are emitted.

Gavage – Forced feeding by means of a tube inserted into the stomach through the mouth.

Genotoxic – Describes the toxic effects of a substance which damages DNA.

Half-life – The length of time required to reduce the concentration of a substance by 50% in a particular medium.

Halogenated aliphatic compound – A chemical compound with a halogen atom (F, Cl, Br, I) associated with an alkane chain.

Hepatomegaly – A condition in which the liver is enlarged beyond its normal size.

Hepatotoxic – Refers to anything which poisons the liver.

Hibernate – To pass the winter in a dormant condition, in which metabolism is slowed down.

Homeostasis – The maintenance of metabolic equilibrium within an animal.

Hyperplasia – An abnormal multiplication or increase in the number of normal cells in a tissue.

Hypertrophy – Enlargement of an organ resulting from an increase in the size of the cells.

Lethal dose – The amount of a chemical necessary to cause death.

Littoral (vegetation) – Pertaining to or along the shore.

Marine – Relating to the sea.

Mast – The fruit of forest trees.

Microsomal – Describing the membrane-bound vesicles that result from the fragmentation of the endoplasmic reticulum.

Miscible – Capable of being mixed.

Morphometry (bone) – The quantitative study of the geometry of bone shapes.

Necrosis – Necrosis is the death of plant or animal cells or tissue.

Neoplastic – Refers to abnormal new growth.

Neotenic (salamander) – The retention of juvenile characteristics in the adult individual.

Nephrotoxic – Refers to anything that poisons the kidney.

Order of magnitude – A single exponential value of the number ten.

Organogenesis – The basic mechanisms by which organs and tissues are formed and maintained in an animal or plant.

Osmoregulation – The control of the levels of water and mineral salts in the blood

Pannes – Bare, exposed, or water-filled depressions in marshes

Partition coefficient – A variable that is used to describe a chemical's lipophilic or hydrophobic properties.

Petechial (hemorrhages) – A minute discolored spot on the surface of the skin or mucous membrane, caused by an underlying ruptured blood vessel.

Photolysis – Chemical decomposition caused by light or other electromagnetic radiation.

Porphyria – A hereditary disease of body metabolism that is caused by a change in the amount of porphyrins (nitrogen-containing substances) found in the blood.

Pyrolysis – Decomposition of a chemical by extreme heat.

Ranid (frog) – The family of true frogs of the order Anura.

Receiving water – A river, ocean, stream or other watercourse into which wastewater or treated effluent is discharged.

Receptor – A plant or animal that may be exposed to a stressor.

Sediment – Particulate material that usually lies below water.

Sediment-associated contaminants – Contaminants that are present in sediments, including whole sediments or pore water.

Sediment-dwelling organisms – The organisms that live in, on, or near bottom sediments, including both epibenthic and infaunal species.

Seminiferous tubules – The glandular part of testicles that contain the sperm producing cells.

Sorption – The process by which one substance takes up or holds another; adsorption or absorption.

Stressor – Physical, chemical, or biological entities that can induce adverse effects on ecological receptors or human health.

Sublethal dose – The amount, or dosage, of a toxin necessary to cause adverse effects, not including death.

Teratogenic – Causing birth defects.

Terrestrial habitats – Habitats associated with the land, as opposed to the sea or air.

Tissue – A group of cells, along with the associated intercellular substances, which perform the same function within a multicellular organism.

Trophic level – A portion of the food web at which groups of animals have similar feeding strategies.

Volatilization – To change or cause to change from a solid or liquid to a vapor.

Wet deposition – The transfer of an element from the atmosphere to land or water through rain or snow.

Appendix A2-1 Goals and Objectives of the Remedial Investigation and Feasibility Study

A remedial investigation and feasibility study (FI/FS) has been initiated as part of the Calcasieu Estuary Initiative. Ths RI/FS is an analytical process that is designed to support risk management decision-making at contaminated sites. The remedial investigation component of the RI/FS is intended to provide the information needed to characterize the nature and extent of contamination at the site and to estimate risks to human health and the environment that are posed by contaminants at the site. Such risks are evaluated by conducting human health and ecological risk assessments at the site. The feasibility study component of the RI/FS is intended to support the evaluation of remedial options that can be applied to manage any risks that are identified at the site.

Ecological risk assessments (ERAs) represent essential components of the overall RI/FS process. In accordance with the guidance that has been developed by the United States Environmental Protection Agency (USEPA 1997), two types of ERAs are typically conducted in the course of implementing a RI/FS. The screening-level ecological risk assessment (SERA) is conducted first and is designed to provide the information needed to determine if ecological risks are likely to occur at the site. If the results of the SERA indicate that ecological threats at the site are negligible, then no further investigations are required to support decision-making activities. In contrast, a baseline ecological risk assessment (BERA) is needed if the results of the SERA indicate that insufficient information exists to determine if a risk exists or that the potential for adverse ecological effects exists at the site.

In the Calcasieu Estuary, the results of the SERA indicated that there is potential for risk to ecological receptors from exposure to contaminated water and sediment (CDM

1999). As such, it was recommended that the remedial investigation proceed to the initiation of a BERA (CDM 1999). Workshop participants recognized that each of the participants in the ERA process has specific needs that must be met during the course of implementing the BERA. To increase the likelihood that these needs will be met during the remedial investigation, each of the workshop participants was asked to reflect on their organization's interests and needs and to refine the goals and objectives for the project. The input that was provided by workshop participants included:

- Establish and maintain an open, transparent process that provides opportunities for participation by regulated interests and the public;
- Communicate effectively with the public such that people fully understand the issues, appreciate the scope of the problems within the study area, and recognize that many areas do not pose a risk to public health, welfare, and the environment;
- Build a process that is scientifically-defensible;
- Identify the sources and pathways of contaminants to the aquatic systems;
- Determine if adverse effects on aquatic organisms are occurring as a result of exposure to contaminants;
- Determine if adverse effects are associated with contaminated sediments (i.e., the risks to aquatic organisms and aquatic-dependent wildlife);
- Formalize the screening process that has been used to identify the chemicals of potential concern (COPCs; i.e., roughly 25 substances);
- Identify the areas within the study area that represent problems with respect to environmental contamination;
- Identify the areas within the study area that do not represent problems with respect to environmental contamination;

- Develop remediation goals that will protect wildlife;
- Develop scientifically-defensible clean-up targets for each habitat type and environmental medium;
- Build cooperative solutions to the problems that exist in the study area;
- Generate information on recovery times for the no action remedial option (i.e., using modeling efforts based on sedimentation rates; focus on key areas such as Bayou d'Inde; note: natural recovery must occur within a reasonable time frame to represent a viable option);
- Develop a rationale for action and remedial alternatives that the public will understand and support; and,
- Identify the long-term monitoring that is needed to evaluate recovery in the study area and restoration of the uses of the ecosystem.

Appendix A2-2 An Overview of the Environmental Fate and Effects of Copper

2.1 Identity

Copper is a heavy metal that is widely distributed in the environment. Copper has an atomic number of 29 and an atomic weight of 63.546 g/mole. The divalent form of copper (i.e., Cu^{++}) can combine with chlorides, sulfates, or nitrates to form salts that are highly soluble in water (McNeely *et al.* 1979). Other copper salts, such, as carbonates, hydroxides, and oxides, are not readily soluble in water (McNeely *et al.* 1979).

2.2 Uses

Metallic copper and copper compounds have been used by humans for thousands of years. Modern uses of copper include electrical wiring and electroplating, the production of alloys (e.g., bronze and brass), photography, utensils, antifouling paint, art designs, pesticide formulations, and textiles (CCREM 1987). Copper is also used in construction, in the production of roofing materials, and in the manufacturing of brass and copper plumbing (Demayo and Taylor 1981). United States is the third largest producer of copper worldwide, accounting for roughly 19% of the world production (CCREM 1987).

2.3 Sources

Copper is released into the environment from both natural and anthropogenic sources. Natural sources of copper include weathering of sulphide and carbonate ores under

oxidizing conditions and from deposits of native copper. However, little of the copper found in water is of natural origin because copper minerals tend to be relatively insoluble. It is estimated that releases of copper into receiving water systems from anthropogenic sources represents 33 to 60% of the total global annual input (Demayo and Taylor 1981). These sources include corrosion of brass and copper pipe by acid waters, sewage treatment plant effluents, the use of copper compounds as aquatic algicides, runoff and groundwater contamination from agricultural uses of copper as fungicides and pesticides in the treatment of soils, and effluents and atmospheric fallout from industrial sources (CCREM 1987). Major industrial sources include mining, smelting and refining industries, copper wire mills, coal-burning industries, and iron- and steel-producing industries (USEPA 1980).

2.4 Physical and Chemical Properties

In nature, copper (Cu) can occur in four oxidation states (Cu, Cu^{+}), Cu^{++}, and Cu^{+++}), with the cuprous (Cu^{+}) and cupric (Cu^{++}) forms being the most common. As cuprous copper is unstable in aqueous solutions, it is normally oxidized to the cupric form in water (CCREM 1987). The solubility of copper is influenced by both pH and alkalinity, with lower pHs and alkalinities favoring increased solubilities (Spear and Pierce 1979). Organically-complexed copper tends to be more soluble than inorganic copper complexes (Spear and Pierce 1979).

2.5 Environmental Fate and Transport

As copper is an element, it is neither created nor destroyed in the environment. Rather, the fate of this substance in the environment is determined by the processes that influence the cycling of copper. In aqueous media, copper ions coordinate with water molecules to form aquo ions [$Cu(H_2O)_6^{++}$; CCREM 1987]. The presence of

other ligands [e.g., hydroxide (OH^-) or sulfate (SO_4^{--})], can result in formation of copper complexes by successively displacing water molecules [e.g., $Cu(OH)^+(H_2O)_5$ and $CuSO_4(H_2O)_5$; Spear and Pierce 1979). Such complexes tend to be relatively unstable and, hence, potent toxicants in the environment (i.e., the copper is readily available to aquatic organisms; CCREM 1987). Copper ions can also form associations with organic ligands (i.e., chelating agents), such as fulvic acid, tannic acid, and Ethylenediaminetetraacetic acid (EDTA; Spear and Pierce 1979). Such complexes tend to have much higher stability (Stumm and Morgan 1970) and lower toxicity. Sorption and precipitation also play major roles in determining the aquatic fate of copper; nevertheless, copper that becomes associated with suspended or bottom sediments can be remobilized under reducing acidic conditions (CCME 1999). Photolysis, volatilization, and biodegradation are minor fate processes for copper in aquatic ecosystems (CCREM 1987).

2.6 Bioaccumulation

As an essential trace nutrient, copper is readily accumulated by plants and animals, with bioconcentration factors of 100 to 26,000 reported for various aquatic species (Spear and Pierce 1979). However, whole body concentrations tend to decrease with increasing trophic level due to organ specific accumulation and metabolic regulation in higher organisms (CCREM 1987). There is little evidence that copper biomagnifies to any significant extent in aquatic or terrestrial food webs.

2.7 Effects on Aquatic and Terrestrial Organisms

The toxicity of water-borne copper to aquatic organisms varies depending on the form of copper under consideration and the physical and chemical characteristics of the water (e.g., dissolved oxygen, temperature, water hardness, turbidity, presence of

chelating agents; Demayo and Taylor 1981). The results of laboratory studies indicate that copper salts are acutely toxic to aquatic plants, invertebrates, and fish, with LC_{50}s (median lethal concentrations) as low as 6.5 µg/L reported in the literature (CCREM 1987). In long-term exposures, copper has been shown to adversely affect the behavior, growth, reproduction, and survival of aquatic organisms, with copper concentrations as low as 3.9 µg/L were found to be toxic to fish (Spear and Pierce 1979; CCREM 1987; Sauter *et al.* 1976). Even lower concentrations of copper (i.e., 1 µg/L) inhibited the growth of plant species (USEPA 1980; 1985).

Sediment-associated copper can be acutely or chronically toxic to sediment-dwelling organisms. In freshwater sediments, 10 to 14-day LC_{50}s of 380 to 1078 mg/kg dry weight (DW) have been reported for the amphipod, *Hyalella azteca* (Cairns *et al.* 1984; Milani *et al.* 1996). The midge, *Chironomus tentans*, appears to less sensitive to the effects of this substance, with 10- to 14-day LC_{50}s of 857 to 2296 mg/kg DW reported (Cairns *et al.* 1984; Milani *et al.* 1996). Adverse effects on the growth of these species have been noted at copper concentrations of 89.8 and 496 mg/kg DW, respectively (Milani *et al.* 1996). In marine sediments, long-term exposure (i.e., to 48-day) to copper-spiked sediments (13.6 to 38.2 mg/kg DW) resulted in delayed predator avoidance response (i.e., increased reburial time) and reduced survival rates in clams, *Protothaca staminea* and *Mya arenaria* (Phelps *et al.* 1983; 1985).

In birds and mammals, consumption of contaminated prey items represents the primary route of exposure to environmental copper. While no information is available on the toxicity of copper to avian wildlife species, laboratory studies have shown that the survival and growth were reduced when domestic turkeys were fed 50 mg Cu/kg food for three weeks (Eisler 1997). Adverse effects on the growth of chickens were observed at even lower levels of dietary exposure to copper. In mammals, long-term exposure to elevated levels of dietary copper can cause degeneration of liver, kidney, brain, and muscle tissues, anemia, impaired growth, and reduced survival rates.

2.8 References

Cairns, M.A., A.V. Nebeker, J.H. Gakstatter, and W.L. Griffis. 1984. Toxicity of copper-spiked sediments to freshwater invertebrates. Environmental Toxicology and Chemistry 3:435-445.

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian sediment quality guidelines for the protection of aquatic life: Copper. Canadian Environmental Quality Guidelines. Winnipeg, Manitoba.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian water quality guidelines. Task Force on Water Quality Guidelines. Ottawa, Ontario.

Demayo, A. and M.C. Taylor. 1981. Copper. *In:* Guidelines for Surface Water Quality. Volume 1. Inorganic Chemical Substances. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

Eisler, R. 1997. Copper hazards to fish, wildlife, and invertebrates: A synoptic review. Biological Science Report USGS/BRD/BSR–1997-0002. Contaminant Hazard Reviews. United States Geological Survey. Laurel, Maryland.

McNeely, R.N., V.P. Neimanis, and L. Dwyer. 1979. Water quality sourcebook: A guide to water quality parameters. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

Milani, D., K.E. Day, D.J. McLeay, and R.S. Kirby. 1996. Recent intra- and inter-laboratory studies related to the development and standardization of Environment Canada's biological test methods for measuring sediment toxicity using freshwater amphipods (*Hyalella azteca*) or midge larvae (*Chironomus riparius*). Environmental Protection Service. Environment Canada. Burlington, Ontario.

Phelps, H.L., J.T. Hardy, W.H. Pearson, and C.W. Apts. 1983. Clam burrowing behaviour: Inhibition by copper-enriched sediment. Marine Pollution Bulletin 14(12):452-455.

Phelps, H.L., W.H. Pearson, and J.T. Hardy. 1985. Clam burrowing behaviour and mortality related to sediment copper. Marine Pollution Bulletin 16(8):309-313.

Sauter, S., K.S. Buxton, K.J. Macek, and S.R. Petrocelli. 1976. Effects of exposure to heavy metals on selected freshwater fish. Toxicity of copper, cadmium, chromium, and lead to eggs and fry of seven fish species. EPA-600/3-76-105. United States Environmental Protection Agency. Duluth, Minnesota. (As cited in CCREM 1987).

Spear, P.A. and R.C. Pierce. Copper in the Aquatic environment: Chemistry, distribution, and toxicology. NRCC No. 15454. Associate Committee on Scientific Criteria for Environmental Quality. National research Council of Canada. Ottawa, Ontario.

Stumm, W. and J.J. Morgan. 1970. Aquatic Chemistry: An Introduction Emphasizing Chemical Equilibria in Natural Waters. Wiley-Interscience. John Wiley and Sons, Inc. New York, New York.

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for copper. EPA-440/5-80-036. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1985. Ambient water quality criteria for copper - 1984. EPA-440/5-84-031. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

Appendix A2-3 An Overview of the Environmental Fate and Effects of Chromium

3.1 Identity

Chromium, in the crystalline form, is a steel-gray, lustrous metal that is extremely resistant to most corrosive agents (Eisler 1986). Chromium has an atomic number of 24 and an atomic weight of 52.0 g/mole. Although chromium is the seventh most abundant element in the earth as a whole and more than 40 chromium-containing minerals have been identified, it is usually present in only trace amounts (i.e., < 1000 mg/kg in surface rocks and soils (Government of Canada 1994).

3.2 Uses

In the United States, chromium is used principally in the metallurgy and chemical industries. Ferrochromium, a chromium alloy, is used in the production of stainless steal and heat-resistant steels that are employed in petrochemical processing, in high-temperature environments (e.g., turbines and furnaces), and in consumer goods (e.g., cutlery and decorative trim (Phillips 1988). Copper-chromium alloys are used in electrical applications that require high strength and good conductivity, while copper-nickel-chromium alloys are used in marine equipment that require corrosion resistance (Government of Canada 1994). In the automotive industry, chromium alloys are used in stainless steel components, catalytic converters, chrome trim, and other applications. Chromium salts are used in paints, dyes, explosives, leather tanning, wood preservatives, copy machine toners, drilling muds, textiles, water treatment, magnetic tapes, ceramics, paper, and photography. Chromite ores and concentrates are used to make refractory products, such as bricks, mortars, and ramming mixtures for the production of iron and steel, portland cement, glass, and non-ferrous metals.

3.3 Sources

Chromium is released into the environment from both natural and anthropogenic sources. The weathering of chromium-bearing rocks, primarily chromium oxides, represents the principal natural source of chromium (Taylor *et al.* 1979). Weathering processes have been estimated to contribute 200,000 tonnes of chromium annually to the environment. By comparison, an estimated 77,700 tonnes of chromium are released worldwide as a result of human activities (CCREM 1987). The major sources of atmospheric chromium emissions are the chromium alloy and metal producing industries; coal combustion, municipal incinerators, cement production, and cooling towers represent less important sources (Eisler 1986). The major sources of chromium to aquatic ecosystems include atmospheric deposition, electroplating and metal finishing industries, and publicly-owned treatment plants. Locally, but less globally, important chromium sources to water include iron and steel foundries, inorganic chemical plants, tanneries, textile manufacturing, and runoff from urban and residential areas (McNeely *et al.* 1979; CCREM 1987).

3.4 Physical and Chemical Properties

Data on the physical and chemical properties of chromium provides important information for evaluating the environmental fate of this substance. While elemental chromium tends to be a very stable substance, it is only rarely found in nature. It has a density of 7.14, melting point of 1900ºC and a boiling point of 2642ºC (Budavari *et al.* 1989). Of the nine possible oxidation states of chromium (i.e., ranging from -II to VI), only trivalent chromium [Cr (III)] and hexavalent [Cr (VI)] are commonly encountered in the environment (Government of Canada 1994). Chromium (II), (IV), and (V) are unstable and are rapidly converted to chromium (III). Both of the primary environmental forms of chromium form salts when released into water; however, the

chromium (III) salts tend to be insoluble, whereas the chromium (VI) salts are readily soluble in water.

3.5 Environmental Fate and Transport

The fate of chromium that is discharged into the environment depends on the nature of the release and the chemical form of the chromium. Chromium released into the atmosphere tends to be chemically stable, with dry fallout and wet precipitation representing the most important fate processes. In water, dissolved trivalent chromium tends to form insoluble salts (e.g., oxides, hydroxides, phosphates), which adsorb to particulate matter and are rapidly removed from the water column by settling (Government of Canada 1994). However, chromium (III) can also form stable complexes with dissolved or colloidal ligands. This complexed chromium is relatively unaffected by adsorption and precipitation reactions and, thus, can remain for extended periods in the water column. Nevertheless, most of the chromium (III) that is discharged to surface water is ultimately transferred to sediments. By comparison, most hexavalent chromium salts are quite soluble in water and are not sorbed to any significant extent by sediment or other particulate matter; hence, this form of chromium tends to remain in the water column. Under anaerobic conditions, chromium (VI) can be reduced to chromium (III); however, the reverse reaction in more common in natural waters (CCREM 1987). Photolysis, volatilization, and biodegradation are minor fate processes for chromium in aquatic ecosystems (CCREM 1987).

3.6 Bioaccumulation

Chromium is readily accumulated by aquatic organisms, with bioconcentration factors of 100 to 1000 commonly reported in the literature (CCREM 1987). However, there

is little evidence that chromium biomagnifies to any significant extent in aquatic or terrestrial food webs (Outridge and Scheuhammer 1993). Due to differences in their properties, chromium (VI) is absorbed more readily than chromium (III); however, the Cr(VI) species is reduced to the Cr(III) form in most organisms (Government of Canada 1994). Algae tends to bioconcentrate chromium to a greater extent than other aquatic organisms, with BCFs (bioconcentration factors) of up to 8500 reported for these aquatic plants (CCREM 1987).

3.7 Effects on Aquatic and Terrestrial Organisms

Exposure to elevated levels of water-borne chromium is known to adversely affect aquatic organisms, with toxicity influenced by chromium species present, water hardness, and pH (CCREM 1987. Of the species tested, crustaceans (water fleas, *Daphnia magna* and *Ceriodaphnia dubia*) appear to be among the most sensitive to the effects of chromium, with acute toxicity thresholds (i.e., 96-hour LC_{50}s (median lethal concentrations) as low as 15.3 µg/L reported for Cr(VI) and 2000 reported for Cr(III) (Eisler 1986). In long-term toxicity tests, exposure to concentrations as low as 2.5 µg/L of Cr(VI) resulted in impaired reproduction, growth, and/or survival of water fleas (Call *et al.* 1981). Chronic toxicity thresholds for Cr(III) were somewhat higher in these species, however (i.e., 66 to 445 µg/L; USEPA 1985). Algae appear to be nearly as sensitive to the effects of chromium as are water fleas (CCREM 1987).

Although the available data from spiked-sediment toxicity tests are limited, it is apparent that sediment-associated chromium is acutely toxic to sediment-dwelling organisms (CCME 1999). In freshwater sediments, 48-hour LC_{50}s for water fleas of 195 and 167 mg/kg dry weight (DW) have been reported for trivalent and hexavalent chromium, respectively (Dave 1992). In marine sediments, 24 week exposure of blue mussels, *Mytilus edulis*, to 150 mg/kg DW of chromium caused reduced filtration

rates. In the long-term, reduced filtration rates are likely to translate into reduced growth rates in bivalves.

Dietary exposure to chromium has the potential to adversely affect avian and mammalian wildlife species. In laboratory studies, short- and long-term consumption of chromium contaminated foods resulted in alterations in kidney and liver function, reduced spleen and liver weights, genotoxicity, and histological changes in the ovaries in mammals (Government of Canada 1994). Reduced egg production, growth and survival have been observed in birds in birds with elevated dietary exposure to chromium. (Eisler 1986). Insufficient data are available to evaluate the potential carcinogenicity of either species of chromium [i.e., Cr(III) or Cr(VI); Government of Canada 1994].

3.8 References

Budavari, S., M.J. O'Neil, A. Smith, and P.E. Heckelman (Eds.). 1989. The Merck Index: An Encyclopedia of Chemicals, Drugs, and Biologicals. Eleventh Edition. Merck and Company, Inc. Rahway, New Jersey.

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian sediment quality guidelines for the protection of aquatic life: Copper. Canadian Environmental Quality Guidelines. Winnipeg, Manitoba.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian water quality guidelines. Task Force on Water Quality Guidelines. Ottawa, Ontario.

Call, D.J., L.T. Brooke, N. Ahmad, and D.D. Vaishnav. 1981. Aquatic pollutant hazard assessments and development of a hazard prediction technology by quantitative structure activity relationships. Second Quarterly Report to the USEPA Center for Lake Superior Environmental Studies. University of Wisconsin. Superior, Wisconsin. (As cited in CCREM 1987).

Dave, G. 1992. Sediment toxicity in lakes along the river Kolbacksan, central Sweden. Hydrobiologia 235/236:419-433. (As cited in CCME 1999).

Eisler, R. 1986. Chromium hazards to fish, wildlife, and invertebrates: A synoptic review. Biological 85(1.6). Contaminant Hazard Reviews Report Number 6. Fish and Wildlife Service. United States Department of the Interior. Laurel, Maryland.

Government of Canada. 1994. Chromium and its compounds. Canadian Environmental Protection Act Priority Substances List Assessment Report. Environment Canada. Health Canada. Ottawa, Ontario.

McNeely, R.N., V.P. Neimanis, and L. Dwyer. 1979. Water quality sourcebook: A guide to water quality parameters. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

Outridge, P.M. and A.M. Scheuhammer. 1993. Bioaccumulation and toxicology of chromium: Implications for wildlife. Reviews of Environmental Contamination and Toxicology 130:31-77. (As cited in Government of Canada 1994).

Phillips, D.R. 1988. Chromium. *In:* Canadian Mineral Yearbook. Mineral Resources Branch. Department of Energy, Mines, and Resources. Ottawa, Ontario. (As cited in Government of Canada 1994).

Taylor, M.C., S.W. Reeder, and A. Demayo. 1979. Chromium. *In:* Guidelines for Surface Water Quality. Volume 1. Inorganic Chemical Substances. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

USEPA (United States Environmental Protection Agency). 1985. Ambient water quality criteria for chromium - 1984. EPA-440/5-84-029. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

Appendix A2-4 An Overview of the Environmental Fate and Effects of Lead

4.1 Identity

Lead (Pb) is a heavy metal that is a constituent of over 200 minerals, most of which are very rare. Only three of these minerals, galena, angelside, and cerusite, are sufficiently abundant to form mineral deposits (Eisler 1988). Galena (PbS), the most abundant lead-based mineral, is often found in association with sphalerite (ZnS), pyrite (FeS_2), chalopyrite ($CuFeS_2$), and other sulfur salts. The main sources of lead-based minerals are igneous, metamorphic, and sedimentary rocks (CCREM 1987).

4.2 Uses

Lead is used in a variety of applications in the United States. The primary use of lead in the production of acid-storage batteries, while the second largest use is in the production of chemicals, such as tetramethyllead and tetraethyllead (Eisler 1988). Lead and its compounds are also used in electroplating, metallurgy, construction materials, coatings and dyes, electronic equipment, plastics, veterinary medicines, fuels, and radiation shielding (CCREM 1987. Other uses of lead include ammunition, corrosive-liquid containers, paints, glassware, storage tank lining, transporting radioactive materials, solder, piping, cable sheathing, roofing, and sound attenuators (CCREM 1987).

4.3 Sources

Lead is released into the environment from both natural and anthropogenic sources. The weathering of sulfide ores represents the principal natural pathway by which lead is released into the environment. There are a wide variety of anthropogenic sources of lead, with the relative importance of each changing over the past 20 years. In the early 1980's, for example, emissions of lead from gasoline-powered motor vehicles accounted for most of the total annual lead emissions. However, reductions in the use of leaded gasolines and improved emission controls in the automotive industry have dramatically reduced releases from this source. The major industrial sources of lead to the environment include emissions from copper, nickel, and lead smelters, from operations involved in the mining, milling, and concentrating of lead-bearing ores, and from iron and steel production facilities (McNeely *et al.* 1979). Discharges of liquid effluents from chemical manufacturing facilities, pulp and paper mills, and municipal wastewater treatment plants also result in the release of lead to aquatic ecosystems (Garrett 1985). Until recently, the use of lead shot in shotgun ammunition represented an important source of lead to aquatic systems that support hunting of waterfowl.

4.4 Physical and Chemical Properties

Elemental lead is a bluish-gray metal, with an atomic number of 82 and an atomic weight of 207.19 (Budavari *et al.* 1989). Lead can occur in four oxidation states, including elemental lead (Pb), monovalent lead (Pb^{+}), divalent lead (Pb^{2+}), and tetravalent lead (Pb^{4+}), with the divalent form being the most stable and prevalent in the environment. While elemental lead is sparingly soluble in water (i.e., to 30 to 500 µg/L), certain lead salts are highly soluble in water (i.e., 443 g/L for lead acetate, 565 g/L for lead nitrate, and 9.9 g/L for lead chloride; Eisler 1988). Other lead salts, such as lead sulfate (42.5 mg/L) and lead oxide (17 mg/L), are only moderately soluble in

water. Organoleads, such as tetraethyllead (0.18 mg/L) and tetramethyllead (18 mg/L), are relatively less soluble in water than many of the inorganic lead salts (Eisler 1988). The solubility of lead can be influenced by water pH and dissolved CO_2 levels (CCREM 1987).

4.5 Environmental Fate and Transport

Because lead is an element, its fate and transport in the environment is determined by the processes that influence the cycling of lead. Lead exists in three main forms in surface waters, including the dissolved labile form (e.g., Pb^2+ and $PbOH^+$), the dissolved bound form (i.e., colloids or strong complexes), and the particulate form (Benes *et al.* 1985). While photolysis (i.e., decomposition caused by light) is an important fate process for atmospheric lead (i.e., for converting lead halides into more soluble forms of the substance), there is no evidence that photolysis plays a significant role in the removal of lead from the water column. Rather, sorption is the dominant mechanism influencing the distribution of lead in aquatic ecosystems, with most of the lead entering natural waters forming associations with suspended particulates and, ultimately bottom sediments (CCREM 1987). Sediment-associated lead can be re-mobilized and released into the water column when pH decreases suddenly or when the ionic composition of the water changes (Demayo *et al.* 1982). Some of the lead in sediments can be transformed into organoleads.

4.6 Bioaccumulation

Exposure to elevated levels of lead can lead to bioaccumulation in the tissues of aquatic organisms. The accumulation of lead in aquatic organisms is influenced by water pH, with lower pHs favoring the accumulation of this substance (i.e., presumably due to increases in the abundance of divalent lead, Pb^{2+}). For example,

water-to-biota bioconcentration factors (BCF) of over 10^6 have been observed in algae, while BCFs of 20 to 360 have been reported for aquatic invertebrates and fish (CCREM 1987). In one study conducted in Oklahoma, the levels of lead in water, surficial sediments, plankton, benthos, and mosquito fish were 0.013 mg/L, 529 mg/kg dry weight (DW), 281 mg/kg DW, 37 mg/kg DW, and 11 mg/kg DW, respectively (Demayo *et al.* 1982). Similarly, frog tadpoles, crayfish, and bluegills from a tailings pond in Missouri had 4139, 500, and 128 mg/kg DW of lead in their tissues (Gale 1976). Collectively, these results suggest that the concentrations of lead in biological tissues generally decreases at higher trophic levels in the food web (i.e., emphasizing that bioconcentration, rather than biomagnification, is an important process in the accumulation of lead).

4.7 Effects on Aquatic and Terrestrial Organisms

Water-borne lead is highly toxic to aquatic organisms, with toxicity varying depending on the species and life stage tested, duration of exposure, the form of lead tested, and the physical and chemical characteristics of the water. Among the species tested, aquatic invertebrates, such as amphipods and water fleas, were the most sensitive to the effects of lead; species mean acute values ranged from 143 to 448 µg/L for these taxa (USEPA 1985). Higher species mean acute values were reported for rainbow trout (2,448 µg/L) and brook trout (4,820 µg/L; USEPA 1985). Lead tends to be more toxic in longer-term exposures, with chronic toxicity thresholds for reproduction in water fleas ranging as low as 30 µg/L reported (CCREM 1987). In general, organoleads compounds are more toxic than inorganic lead compound, with tetraethyllead reported more toxic than tetramethyllead (Eisler 1988). Water hardness plays a key role in determining the toxicity of this substance, with the lowest toxicity thresholds reported in soft water exposure systems (CCREM 1987).

Exposures to sediment-associated lead can be toxic to sediment-dwelling organisms. In freshwater sediments, 48-hour exposure of water fleas to 7,000 mg/kg DW significantly reduced mobility, while exposure to 13,400 mg/kg DW for 24-hours produced the same effect (Dave 1992a; 1992b). Longer-term (i.e., 14-day) exposure of midges, *Chironomus tentans*, to sediments containing 31,900 mg/kg DW of lead resulted in 100% mortality. Field-derived toxicity thresholds (i.e., probable effect levels) were much lower than those that were derived from the results of spiked-sediment toxicity tests (CCME 1999).

In birds, consumption of lead shot probably represents a primary route of exposure to environmental lead. Lethality associated with the consumption of lead shot had been documented in a variety of bird species, including waterfowl, raptores, and others. In some cases, ingestion of a single lead shot pellet (i.e.,1 to 1.6 g of lead) resulted in increased mortality in mallard ducks (i.e., by 9 to 19%; Longcore *et al.* 1974). Sub-lethal effects associated with the ingestion of lead shot include damage to the nervous system, muscular paralysis, damage to liver and kidneys, impaired reproduction, and increased susceptibility to predation (Mudge 1983). Other forms of lead may be more toxic than lead shot, as single oral LD_{50}s (i.e., the dose that causes 50% mortality in test organisms) as low as 107 mg/kg body weight (BW) have been reported for tetraethyllead (Eisler 1988).

Lead has been documented to be toxic to a variety of mammals. Acute oral LD_{50}s of 5 to 108 mg Pb/kg BW have been reported in rats (Eisler 1988). Longer-term dietary exposure to this substance has been shown to cause headaches, fatigue, muscle atrophy, muscle paralysis, convulsions, coma, and death. Chronic oral lethality thresholds as low as 0.32 mg/kg BW/day have been reported in dogs; however, higher thresholds have been observed in several other species (Eisler 1988). As is the case for birds, the organolead compounds tend to be more toxic than the inorganic lead salts.

4.8 References

Benes, P., M. Cejchanova, and B. Havlik. 1985. Migration and speciation of lead in a river system heavily polluted from a smelter. Water Research 19:1-6. (As cited in Eisler 1988).

Budavari, S., M.J. O'Neil, A. Smith, and P.E. Heckelman (Eds.). 1989. The Merck Index: An Encyclopedia of Chemicals, Drugs, and Biologicals. Eleventh Edition. Merck and Company, Inc. Rahway, New Jersey.

Demayo, A., M.C. Taylor, K.W. Taylor, and P.V. Hodson. 1982. Toxic effects of lead and lead compounds on human health, aquatic life, wildlife, plants, and livestock. CRC Critical Rev. Environ. Control 12:257-305. (As cited in Eisler 1988).

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian sediment quality guidelines for the protection of aquatic life: Lead. Canadian Environmental Quality Guidelines. Winnipeg, Manitoba.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian water quality guidelines. Task Force on Water Quality Guidelines. Ottawa, Ontario.

Dave, G. 1992a. Sediment toxicity and heavy metals in eleven lime reference lakes of Sweden. Water, Air and Soil Pollution. 63:187-200. (As cited in CCME 1999).

Dave, G. 1992b. Sediment toxicity in lakes along the river Kalbachsan, central Sweden. Hydrobiologia 235/236:419-433. (As cited in CCME 1999).

Eisler, R. 1988. Lead hazards to fish, wildlife, and invertebrates: A synoptic review. Biological Report 85(1.14). Contaminant Hazard Reviews Report 14. United States Fish and Wildlife Service. Laurel, Maryland.

Gale, N.L., E. Bolter, and B.G. Wixson. 1976. Investigation of Clearwater Lake as a potential sink for heavy metals from lead mining in southeast Missouri. *In:* D.D.Hemphill (Ed.). Trace Substances in the Environment. Volume X. University of Missouri. Columbia, Missouri. (As cited in Eisler 1988).

Garrett, C.L. 1985. Chemicals in the environment: Pacific and Yukon Region. IV. Lead. Environmental Protection Service. Department of the Environment. Vancouver, British Columbia. 32 pp.

Longcore, J.R., R. Andrews, L.N. Locke, G.E. Bagley, and L.T. Young. 1974. Toxicity of lead and proposed substitute shot to mallards. Special Scientific Report - Wildlife 183. United States Fish and Wildlife Service. 23 pp. (As cited in Eisler 1988).

McNeely, R.N., V.P. Neimanisand L. Dwyer. 1979. Pesticides. *In:* Water Quality Sourcebook. A Guide to Water Quality Parameters. Water Quality Branch. Inland Waters Directorate. Environment Canada. Ottawa. pp. 34-45.

Mudge, G.P. 1983. The incidence and significance of ingested lead pellet poisoning in British waterfowl. Biological Conservation. 27:333-372.

USEPA (United States Environmental Protection Agency). 1985. Ambient water quality criteria for lead - 1984. EPA-440/5-84-027. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

Appendix A2-5 An Overview of the Environmental Fate and Effects of Mercury

5.1 Identity

Mercury (Hg) is an elemental metal that belongs to group 2B (12) of the periodic table. It has a characteristic liquid form and silver-white color in its metallic state. Its atomic number is 80 with a molecular weight of 200.59. The water solubility of mercury is 0.28 µmoles/L at 25°C. It dissolves to some extent in lipids. Mercury has a melting point of -38°C, a boiling point of 357°C, and a vapor pressure of 0.002 mm Hg at 25°C (HSDB 2000).

5.2 Uses

Mercury is used in the chlor-alkali industry to produce chlorine, caustic soda (sodium hydroxide), and hydrogen. The metal is also used in the paint industry to produce paint pigments and preservatives (Hocking 1979; Health and Welfare Canada 1980). Other uses of mercury include production of thermometers, switches, batteries, fluorescent bulbs, dental amalgams, and pharmaceuticals. In the past, mercury was used to produce pesticides, but those were banned and subsequently phased out of production (McNeely *et al.* 1979; Reeder *et al.*1979; Health and Welfare Canada 1980; USEPA 1980).

5.3 Sources

Mercury is a natural element and, thus, is subject to ordinary biogeological cycling. Natural mercury deposits occur in all types of rocks and minerals (Jonasson and Boyle 1979). Thus, terrestrial environments appear to be large sources of atmospheric mercury, with contributions from evapo-transpiration of leaves, decaying vegetation, and degassing of soils (Kothny 1973). Volcanic activity, weathering, and releases from the oceans (Beyer *et al.* 1996) contribute a steady stream of mercury into the environment. However, volcanic, fumarolic, and thermal spring activities probably make only small contributions on a global basis (Jonasson and Boyle 1979). Far greater amounts of mercury are released due to anthropogenic activities. Coal combustion, non-ferrous metal production, waste incineration, chemical production (e.g., chlorine and alkali production from chlor-alkali plants), and the dumping of sewage sludge (Beyer *et al.* 1996) are responsible for the majority of current contamination problems.

5.4 Physical and Chemical Properties

Chemically, mercury can exist in three oxidation states: Hg^0 or elemental (metallic) mercury, Hg^+ or mercurous ion (monovalent mercury), and Hg^{2+} or mercury II mercuric ion (divalent mercury; USEPA 1997a). All three forms of mercury can be found in air, water, and sediments, albeit in different proportions. In air, mercury occurs mostly (95 to 99%) as Hg^0. In water, mercury occurs mostly as Hg^{2+} in particulate and dissolved form. Methylmercury usually comprises less than 20% of total mercury. In sediments, Hg^{2+} forms complexes with particles that have a high organic or sulfur content. These complexes are stable and tend to immobilize mercury in the sediment layer (USEPA 1997a). In fact, Hg^{2+} sorption to sediments is probably the most important process for determining its abiotic fate in the aquatic environment (USEPA 1979).

5.5 Environmental Fate and Transport

Mercury can be transformed to mono- and dimethylmercury by microorganisms under aerobic and anaerobic conditions (Bisogni and Lawrence 1975; Wood 1976; McNeely *et al.* 1979). Bacteria that are common in most natural waters are capable of this action (Jensen and Jernelov 1969; Bisogni and Lawrence 1975). Both forms of methylmercury may also be demethylated by bacteria in sediments (Fagerstrom and Jernelov 1972; NAS 1977; McNeely *et al.* 1979; Ramamoorthy *et al.* 1982).

5.6 Bioaccumulation

Even though most of the mercury present in the water column is in the divalent inorganic form, methylated forms constitute most of the mercury residues in the tissues of aquatic organisms (Hattula *et al.* 1978). Bioconcentration factors for aquatic organisms are usually high (104) due to rapid uptake and slow depuration. The biological half-life for mercury in fish is estimated to be approximately 2 years (Lockhart *et al.* 1972; McKim *et al.* 1976).

5.7 Effects on Aquatic and Terrestrial Organisms

Mercury has no known biological function in vertebrate physiology (Beyer *et al.* 1996). However, it is a potent systemic toxin and acts primarily on the central nervous and reproductive systems of animals (USEPA 1997b). Although it has no teratogenic or carcinogenic activity, it may disrupt endocrine activity (Fynn-Aikins *et al.* 1998). Methylmercury is very toxic due to its high affinity for sulfur-containing organic compounds i.e. proteins. Methylmercury forms covalent bonds with sulfhydryl groups of proteins and other macromolecules. This allows methylmercury to migrate readily through biological membranes (USEPA 1997a). Thus,

methylmercury can easily reach sensitive tissues such as the brain or migrate across the placenta to reach the sensitive fetus.

In aquatic animals, methylmercury rapidly diffuses across the gills (7 to 12% of MeHg) and enters the blood stream. Methylmercury is also rapidly accumulated through the gut (65 to 80%) and transferred to the blood stream. It is subsequently transported to all organs including the liver, kidney, and the brain. Methylmercury is ultimately deposited in the muscle tissue. Inorganic mercury is also absorbed through the gut and the fish gills, but at a much lower efficiency. It is also excreted much more readily (Beyer *et al.* 1996). As mercury is a natural toxicant, fish and terrestrial mammals have developed depuration pathways, however, they are much slower than uptake.

Effects of mercury poisoning in fish include death, reduced reproduction, impaired growth and development, behavioral abnormalities, altered blood chemistry, impaired osmoregulation, reduced feeding rates and predatory success, and altered respiration (USEPA 1997a). USEPA's AQUIRE (Aquatic Toxicity Information Retrieval) database (USEPA 2001) indicates that total mercury concentrations as low as 20 µg/L causes median hatchability effects in fish (orangethroat darter). Median lethality concentrations to other fish, including bass, killifish and carp, range from 60 to 200 µg/L.

Mercury has also been observed to induce adverse effects in benthic organisms. The adverse effects include reduced fertilization, impaired development, and lethality. Jaagumagi (1988) and Jaagumagi *et al.* (1989) reported significant decreases in abundance of Gastropoda and Chironomidae at sites in Toronto Harbor compared to reference sites. Mercury concentrations at the test site were five times higher than interim sediment quality guidelines and twice as high as probable effect levels set out

by Canadian Sediment Quality Guidelines for the Protection of Aquatic Life (CCME 1999).

Adverse effects reported for terrestrial species exposed to mercury include death, decreased reproduction, decreased growth, decreased immune response, inhibition of induction of enzymes, change in behavior, and change in respiration (USEPA 1997a). Toxicity data for mammals suggests that a mercury residue of 30 mg/kg WW (wet weight) in the liver or kidney is harmful and lethal to most mammals. Also, dietary methylmercury concentrations in the range of 2 to 6 mg/kg WW are sufficient to cause mercury poisoning.

The analysis of published data on mercury toxicity to birds reveals that mercury concentrations in the liver and the kidney higher than 30 mg/kg WW are lethal to several species. Mercury concentrations in eggs above 0.5 mg/kg WW (2.5 mg/kg dry weight (DW) at egg water content of 80%) begin to have detrimental effect on reproduction. Whereas dietary concentrations above 1 mg/kg WW begin to have detrimental effects on reproduction. Dietary mercury concentrations of 10 mg/kg WW are lethal to birds of prey, passerines, and pheasants.

5.8 References

Beyer, W.N., G.H. Heinz, and A.W. 1996. Environmental Contaminants in Wildlife. *In*: Wiener, J.G. and D.J. Spry (eds.). Toxicological significance of mercury in freshwater fish. Redmon Norwood. pp. 297-339.

Bisogni, J.J., Jr. and A.W. Lawrence. 1975. Kinetics of mercury methylation in aerobic and anaerobic aquatic environments. Journal of the Water Pollution Control Federation 47:135-152.

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian sediment quality guidelines for the protection of aquatic life: Mercury. Canadian Environmental Quality Guidelines. Winnipeg, Manitoba.

Fagerstrom, T. and A. Jernelov. 1972. Some aspects of the quantitative ecology of mercury. Water Research 6:1193-1202.

Fynn-Aikins, K., E. Gallagher, S. Ruessler, J. Wiebe,, and T.S. Gross. 1998. An evaluation of methyl mercury as an endocrine disruptor in largemouth bass. SETAC 19th Annual Meeting. Charlotte, North Carolina.

Hattula, M.L., J. Sarkka, J. Janatuinen, J. Paasivirta, and A. Roos. 1978. Total mercury and methylmercury contents in fish from Lake Paijanne. Environmental Pollution 17:19-39.

Health and Welfare Canada. 1980. Mercury. *In*: Guidelines for Canadian drinking water quality 1978. Supporting Documentation. Supply and Services Canada. Ottawa. pp. 387-407.

Hocking, M.B. 1979. Uses and emissions of mercury in Canada. *In*: Effects of Mercury in the Canadian Environment. Associate Committee on Scientific Criteria for Environmental Quality. National Research Council of Canada. Ottawa, Ontario. NRCC No. 16739. pp. 50-75.

HSDB (Hazardous Substances Databank). 2000. Profile for mercury. National Library of Medicine. Bethesda Maryland.

Jaagumagi, R. 1988. The in-place pollutants program. Volume V, Part B. Benthic invertebrates studies results. Ontario Ministry of the Environment. Water Resources Branch. Aquatic Biology Section. Toronto, Ontario.

Jaagumagi, R., D. Persaud, and T. Lomas. 1989. The in-place pollutants program. Volume V, Part A. A synthesis of benthic invertebrate studies. Ontario Ministry of the Environment. Water Resources Branch. Aquatic Biology Section. Toronto, Ontario.

Jensen, S. and A. Jernelov. 1969. Biological methylation in aquatic organisms. Nature (London) 223:753-754.

Jonasson, I.R. and R.W. Boyle. 1979. The biogeochemistry of mercury. *In*: Effects of Mercury in the Canadian Environment. NRCC No.16739. Associate Committee on Scientific Criteria for Environmental Quality. National Research Council of Canada. Ottawa, Ontario. pp. 28-49.

Kothny, E.L. 1973. The three-phase equilibrium of mercury in nature. *In*: Trace Elements in the Environment. E.L. Kothny (Ed.). Advances in Chemistry Series Number 123:48-80. (As cited in Jonasson and Boyle 1979).

Lockhart, W.L., J.F. Uthe, A.R. Kennedy, and P.H. Mehrle. 1972. Methylmercury in northern pike (*Esox lucius*): Distribution, elimination, and some biochemical characteristics of contaminated fish. Journal of Fisheries Research Board of Canada 29:1519-1523.

McKim, J.M., G.F. Olson, G.W. Holcombe, and E.P. Hunt. 1976. Long-term effects of methylmercuric chloride on three generations of brook trout (*Salvelinus fontinalis*): Toxicity, accumulation, distribution, and elimination. Journal of Fisheries Research Board of Canada 33:2726-2739.

McNeely, R.N., V.P. Neimanis, and L. Dwyer. 1979. Mercury. *In*: Water Quality Sourcebook. A Guide to Water Quality Parameters. Water Quality Branch. Inland Waters Directorate. Environment Canada. Ottawa, Ontario. pp. 23-24.

NAS (National Academy of Sciences). 1977. An Assessment of mercury in the environment. United States National Research Council. Washington, District of Columbia.

Ramamoorthy, S., T.C. Cheng, and D.J. Kushner. 1982. Effect of microbial life stages on the fate of methylmercury in natural waters. Bulletin of Environmental Contamination and Toxicology 29:167-173.

Reeder, S.W., A. Demayo, and M.C. Taylor. 1979. Mercury. *In*: Guidelines for Surface Water Quality. Vol. 1: Inorganic chemical substances. Water Quality Branch. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

USEPA (United States Environmental Protection Agency). 1979. Mercury. *In*: Water-related Environmental Fate of 129 Priority Pollutants. Vol. 1: Introduction, technical background, metals and inorganics, pesticides, polychlorinated biphenyls. EPA-440/4-79-029a. Office of Water Planning and Standards. Washington, District of Columbia. pp. 14-1 to 14-15.

USEPA (United States Environmental Protection Agency). 1980. Ambient Water Quality Criteria for Mercury. EPA-440/5-80-058. Office of Water Regulations and Standards. Criteria and Standards Division. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1997a. Mercury study report to congress. Volume VI: An ecological assessment for anthropogenic mercury emissions in the United States. EPA-452/R-97-008.

USEPA (United States Environmental Protection Agency). 1997b. Mercury study report to congress. Volume VII: Characterization of human health and wildlife risks from mercury exposure in the United States. EPA-452/R-97-009.

Wood, J.M. 1976. Les metaux toxiques dans I'environnement. Recherche 7:711. (As cited in USEPA 1980).

Appendix A2-6 An Overview of the Environmental Fate and Effects of Nickel

6.1 Identity

Nickel (Ni) is the 24th most abundant element in the earths crust, occurring at an average concentration of about 75 µg/g (Government of Canada 1994). Nickel is found in many minerals and ores, primarily in combination with sulfur, arsenic, and antimony (CCREM 1987). Some of the more common nickel-bearing minerals include niccolite (NiAs), pentlandite [$(FeNi)_9S_8$], and millerite (NiS; McNeely *et al.* 1979). The principal sources of nickel-based minerals are igneous and serpentine rocks.

6.2 Uses

Due to its resistence to corrosion, high strength, and favorable alloying properties, nickel has a wide range of uses. The production of stainless steel, nickel plating, and nickel alloys represent the principal uses of this substance (CCREM 1987). Nickel-containing stainless steel is used in the chemical manufacturing, food processing, and health care industries (CCREM 1987). It has been estimated that nickel is used in the production of over 3000 alloys that have more than 250,000 applications (Mining Association of Canada 1991). High-nickel alloys are used in electrical, chemical, marine, electronic, nuclear, and aerospace applications. Nickel is also used as a catalyst in industrial processes and oil refining, in colors and glazes for ceramics, and in electrolyte solutions, jewellery, batteries, and gas masks (Government of Canada 1994).

6.3 Sources

Nickel is released into the environment from both natural sources and human activities. The weathering and erosion of soils and bedrock, forest fires, and volcanoes represent the principal natural sources of lead to the environment. The processing of nickel-bearing ores, metal plating, burning of fossil fuels, and waste incineration are the most important anthropogenic sources of nickel, with releases from the combustion of petroleum, coal, and coke accounting for the majority of total releases (CCREM 1987). In areas that have substantial nickel deposits, the mining of nickel-bearing ores and the smelting and refining of concentrates also represent important sources of nickel (Government of Canada 1994). The electroplating, alloy casting, and iron and steel processing industries are also major contributors of nickel to the environment (CCREM 1987). As the concentrations of nickel in crude oil are relatively high (i.e., 0.6 to >300 mg/kg; CCREM 1987), chemical manufacturing industries that utilize crude oil or petroleum products in their processes are likely to release nickel into the environment.

6.4 Physical and Chemical Properties

Elemental nickel is a silver-white metal, with an atomic number of 28 and an atomic weight of 58.71 (Budavari *et al.* 1989). Although nickel can occur in six oxidation states (i.e., -1, 0, +1, +2, +3, and +4), the divalent form (Ni^{2+}) is the most common in the environment. While elemental nickel is relatively insoluble in water, certain nickel salts are highly soluble in water (e.g., nickel chloride hexahydrate - 2500 g/L; nickel sulfate hexahydrate - 660 g/L; nickel sulfate heptahydrate -760 g/L; nickel nitrate hexahydrate - 2400 g/L; Lide 1992). Other nickel salts, such as hexaamine nickel nitrate (45 g/L), nickel hydroxide (0.13 g/L, and nickel carbonate (0.09 g/L) are only moderately soluble in water (Lide 1992). Nickel oxide and nickel subsulfide are considered to be insoluble in water (Government of Canada 1994).

6.5 Environmental Fate and Transport

Because nickel is an element, it is neither created nor destroyed. Rather, its fate and transport in the environment is determined by the processes that influence the its cycling. The nickel that is released to the atmosphere is generally attached to small particles that are removed from the air by wet or dry precipitation. In aquatic systems, nickel occurs primarily in the form of hydroxide, carbonate, and sulfate salts, which may become associated with suspended organic or inorganic materials (Government of Canada 1994). While sorption to particulates and subsequent deposition can result in the enrichment of bottom sediments with this substance (particularly at higher pHs; i.e., > 9), microbial activity or changes in the physical and chemical characteristics of the receiving water body (e.g., decreasing pH, increasing dissolved organic carbon) can remobilize sediment-associated nickel (CCREM 1987). Hence, nickel is considered to be highly mobile in aqueous systems. There is no evidence that photolysis or volatilization play significant roles in the removal of nickel from the water column (Government of Canada 1994).

6.6 Bioaccumulation

While nickel is bioaccumulated in the tissues of aquatic organisms, there is no evidence that this substance is biomagnified in the food web. In aquatic plants, such as algae, lichens, mosses, and macrophytes, bioconcentration factors (BCFs) range between 100 and 10,000 (CCREM 1987). The range of BCFs is narrower for aquatic invertebrates, with BCFs of 200 to 1000 reported for clams, zooplankton, and benthos (Government of Canada 1994). Somewhat higher BCFs (i.e., 2,000 to 4,500) were observed in the water flea, *Daphnia spp*. (Cowgill 1976). Fish had the lowest BCFs, typically ranging from 220 to 330 (Hutchinson *et al.* 1976). Collectively, these results suggest that the concentrations of nickel in biological tissues generally decreases at higher trophic levels in the food web (i.e., emphasizing that bioconcentration, rather

than biomagnification, is the dominant process influencing the bioaccumulation of nickel).

6.7 Effects on Aquatic and Terrestrial Organisms

The toxicity of water-borne nickel varies depending on the species and life stage tested, duration of exposure, and water hardness (USEPA 1980). Overall, acute and chronic toxicity thresholds for nickel ranged from 24 to 10,000 µg/L (Government of Canada 1994). In short-term toxicity tests (i.e., 96-hour) in soft waters, median lethal concentrations (i.e., median lethal concentrations; LC_{50}s) of 102 µg/L and 190 µg/L were reported for the snail, *Juga plicifera*, and the mussel, *Anodonta imbecilis*, respectively (Government of Canada 1994). Exposure to water-borne nickel for longer periods of time caused an avoidance response in rainbow trout (*Oncorhynchus mykiss*; i.e., at 24 µg/L; Giattina et al. 1982), reduced longevity in water fleas (*Daphnia magna*; i.e., at 40 µg/L; Munzinger 1990), impaired growth in algae (Scenedesmus acuminatus; i.e., at 50 µg/L; Stokes 1981), and reduced embryo survival in rainbow trout, and toads *(Gastrophryne carolinensis*; i.e., at 50 µg/L; Birge 1978). Therefore, exposure to water-borne metal can adversely affect the survival, growth, and reproduction of aquatic organisms. The toxicity of nickel to aquatic organisms tends to increase with decreasing water hardness.

No data from spiked-sediment toxicity tests were available to assess the effects of sediment-associated nickel on benthic invertebrates (Long and Morgan 1991; Government of Canada 1994; CCME 1999).

Information from controlled laboratory experiments and epidemiological studies indicates that prolonged exposure to nickel has the potential to adversely affect mammalian and avian receptors. Based on the results of occupational health studies

in humans, the respiratory system represents the critical target for adverse effects following exposure to nickel by inhalation (Government of Canada 1994). As ambient levels of nickel tend to be low in air, the dietary exposure route is likely to be more important in wildlife species. In mallard ducks, long-term dietary exposure (i.e., up to 90 days) to nickel caused tremors, ataxia, and death in newly-hatched ducklings (i.e., at dietary levels of 800 mg/kg dry weight (DW) or higher; Cain and Pafford 1981). Similarly, growth and survival were reduced in newly-hatched chickens maintained on nickel-contaminated diets (i.e., at dietary levels of 300 mg/kg DW or higher and 500 mg/kg DW or higher, respectively (Ling and Leach 1979). While dietary exposure to nickel (i.e., in drinking water) has been linked with bladder and lung cancer in mammalian studies; this linkage has not been conclusively demonstrated (Government of Canada 1994; Young 1995).

6.8 References

Birge, W.J. 1978. Aquatic toxicology of trace elements of coal and fly ash. In: Energy and Environmental Stress in Aquatic Systems. J.H. Thorp and J.W. Gibbons (Eds.). Department of Energy Symposium Series (CONF-771114). Washington, District of Columbia. (As cited in Government of Canada 1994).

Budavari, S., M.J. O'Neil, A. Smith, and P.E. Heckelman (Eds.). 1989. The Merck Index: An Encyclopedia of Chemicals, Drugs, and Biologicals. Eleventh Edition. Merck and Company, Inc. Rahway, New Jersey.

Cain, B.W. and E.A. Pafford. 1981. Effect of dietary nickel on survival and growth of mallard ducklings. Archives of Environmental Contamination and Toxicology 10:737-745. (As cited in Government of Canada 1994).

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian sediment quality guidelines for the protection of aquatic life: Copper. Canadian Environmental Quality Guidelines. Winnipeg, Manitoba.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian water quality guidelines. Task Force on Water Quality Guidelines. Ottawa, Ontario.

Cowgill, U.M. 1976. The chemical composition of two species of *Daphnia*, their algal food, and their environment. Science of the Total Environment 6:79-102. (As cited in Government of Canada 1994).

Giattina, J.D., R.R. Garton, and D.G. Stevens. 1982. Avoidance of copper and nickel by rainbow trout as monitored by a computer-based data acquisition system. Transactions of the American Fisheries Society 111:491-504. (As cited in Government of Canada 1994).

Government of Canada. 1994. Nickel and its compounds. Canadian Environmental Protection Act Priority Substances List Assessment Report. Environment Canada. Health Canada. Ottawa, Ontario.

Hutchinson, T.C., A. Fedorenko, F. Fitchko, A. Kuja, J. VanLoon, and J. Lichwa. 1976. Movement and compartmentation of nickel and copper in aquatic ecosystems. *In:* Environmental Biogeochemistry: Metals Transfer and Ecological Mass Balances. Volume 2. J.O. Nriagu (Ed.). Ann Arbor Science Publishers Inc. Ann Arbor, Michigan. (As cited in Government of Canada 1994).

Lide, D.R. (Ed.). 1992. CRC handbook of Chemistry and Physics. 73rd Edition. CRC Press. Boca Raton, Florida.

Ling, J.R. and R.M. Leach. 1979. Studies on nickel metabolism: Interaction with other mineral elements. Poultry Science. 58:5910596. (As cited in Government of Canada 1994).

Long. E.R. and L. Morgan. 1991. The potential for biological effects of sediment-sorbed contaminants tested in the National Status and Treands Program. NOAA Technical Memorandum NOS OMA 52. National Oceanic and Atmospheric Administration. Seattle, Washington.

McNeely, R.N., V.P. Neimanis, and L. Dwyer. 1979. Water quality sourcebook: A guide to water quality parameters. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

Mining Association of Canada. 1991. Mining in Canada: Facts and figures. Ottawa, Ontario. 48 pp. (As cited in Government of Canada 1994).

Munzinger, A. 1990. Effects of nickel on *Daphnia magna* during chronic exposure and alterations in the toxicity to generations pre-exposed to nickel. Water Research 24:845-852. (As cited in Government of Canada 1994).

Stokes, P.M. 1981. Multiple Metal Tolerance in Copper-tolerant Green Algae. Journal of Plant Nutrition. 3:667-678. (As cited in Government of Canada 1994).

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for nickel. EPA-440/5-80-060. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia. (As cited in CCREM 1987).

Young, R.A. 1995. Toxicity profile: Toxicity summary for nickel and nickel compounds. Risk Assessment Information System: Nickel and Nickel Compounds (7440-02-0). Oak Ridge National Laboratory. Oak Ridge, Tennessee.

Appendix A2-7 An Overview of the Environmental Fate and Effects of Zinc

7.1 Identity

Zinc is a heavy metal that is a constituent of a variety of minerals. The most common ores of zinc are sulfides, such as sphalerite (cubic ZnS) and wurtzite (hexagonal ZnS), carbonates, such as smithsonite (or calamine; $ZnCO_3$), and silicates, such as willemite (Zn_2SiO_4). Zincite, franklinite [$(ZnMnFe)O(FeMn_2)O_3$], and gahnite ($ZnAl_2O_4$; McNeely *et al.* 1979; Budavari *et al.* 1989)). In sulfides, zinc usually occurs in combination with other elements, particularly iron, copper, and lead (CCREM 1987).

7.2 Uses

Zinc is used in a number of industrial and agricultural application. Worldwide, the production of galvanized metals and metal alloys represents the primary uses of zinc, accounting for roughly 75% of the global production (i.e., 35% is used to produce coatings for steel and iron, 25% is used in alloys for dye casting, and 20% is used to produce brass; CCREM 1987). In addition, zinc is used as a rubber vulcanization activator and accelerator, heat conductor, pigment, UV stabilizer, supplement in animal feeds, catalyst, chemical intermediate, and mildew inhibitor (Opresko 1992). Zinc-based compounds are also used in rayon manufacturing, wood preservatives, parchment paper, smoke bombs, cements for metals, artificial silk, deodorants, antiseptics, and pesticides (Opresko 1992).

7.3 Sources

Zinc is released into the environment from both natural and anthropogenic sources, with the weathering of zinc-bearing rock representing the principal natural pathway by which this substance is released into the environment. According to the CCREM (1987), discharges from primary zinc production facilities and municipal wastewater treatment plants represent the principal sources of zinc from human activities. Other sources of this substance include wood combustion, waste incineration, iron and steel production, and other releases to the atmosphere (CCREM 1987).

7.4 Physical and Chemical Properties

Elemental zinc is a bluish-white, lustrous metal, with an atomic number of 30 and an atomic weight of 65.38 (Budavari *et al.* 1989). Zinc can occur in two oxidation states, including elemental zinc (Zn) and divalent zinc (Zn^{2+}). Elemental zinc is sparingly soluble in water; however, certain zinc salts are highly soluble in water (i.e., 435 g/L for zinc acetate, 4,320 g/L for zinc chloride, 1667 g/L for zinc sulfate, and 3,333 g/L for zinc iodide; Budavari *et al.* 1989). Other zinc salts, such as zinc phosphate, zinc sulfide, and zinc oxide, are virtually insoluble in water (Budavari *et al.* 1989).

7.5 Environmental Fate and Transport

The fate and transport of zinc in the environment is determined by the processes that influence its cycling. Zinc can exist in four main forms in surface waters, including the simple hydrated ion (i.e., Zn^{2+}), inorganic compounds (e.g., $ZnCO_3$), stable organic complexes (e.g., Zn-cysteinate), or adsorbed to organic (e.g., Zn2+-humic acids) or inorganic (Zn2+-clay) colloids (CCREM 1987). The fraction of the total zinc that is present in each of these forms depends on the pH of the water, the

concentration of zinc, and the presence of other metal ions, with low pH, low alkalinity, and high ionic strength favoring the ionic form of the substance (Taylor and Demayo 1980). Sorption of zinc by hydrous metal oxides, clay minerals, and organic materials appears to be an important process influencing the distribution of zinc in aquatic ecosystems (USEPA 1987). Above pH 7.0, greater than 90% of the zinc is likely to be bound, subject to precipitated, and, ultimately, associated with bottom sediments (CCREM 1987). In contrast, little adsorption is likely to occur below pH 6.0 (CCREM 1987). Sediment-associated zinc can be re-mobilized and released into the water column under anaerobic conditions, when pH decreases suddenly, or when the ionic composition of the water increases (James and MacNaughton 1977).

7.6 Bioaccumulation

As zinc is an essential micronutrient, it is accumulated in the tissues of aquatic organisms. As many organisms have the capability to regulate the concentrations within relatively tight homeostatic limits, bioconcentration factors (BCFs) are influenced by the concentration of zinc in the water (i.e., high BCFs are observed when zinc concentrations are low and lower BCFs are reported at elevated levels of environmental zinc). As the ionic zinc is the most bioavailable form of the substance, BCFs are also influenced by the factors that influence chemical equilibrium among its four primary forms. In general, BCFs in the order of 1,000 have been reported for freshwater invertebrates, while higher BCFs (i.e., in the order of 10,000) have been observed in fish and aquatic plants. While zinc does bioconcentrate in aquatic organisms, there is no evidence that it biomagnifies in aquatic food webs.

7.7 Effects on Aquatic and Terrestrial Organisms

Water-borne zinc is highly toxic to aquatic organisms, with the respiratory organs being the primary site of toxic action (Eisler 1993). The toxicity of water-borne zinc varies depending on the species and life stage tested, duration of exposure, and the physical and chemical characteristics of the water. According to Eisler (1993), freshwater fish tend to be more sensitive than marine species and embryos and larvae are the most sensitive development stages. For fish and aquatic invertebrates, acute toxicity thresholds ranged from 90 to 58,100 µg/L (CCREM 1987). While acute toxicity to zinc is modified by water hardness (i.e., zinc is more toxic in soft water than in hard water), chronic toxicity is not (USEPA 1980). Zinc is also more toxic at low dissolved oxygen concentrations, high sodium levels, low levels of organic complexing agents, and low pH. The results on long-term toxicity tests indicate that zinc toxicity increases with duration of exposure. Adverse effects on the survival, growth, and reproduction of aquatic organisms start at roughly 30 µg/L to 70 µg/L, depending on the life stage and species tested.

Exposures to sediment-associated zinc can be toxic to sediment-dwelling organisms. In freshwater sediments, an LC_{25} of 3531 mg/kg dry weight (DW) was reported for the amphipod, *Hyalella azteca* (Borgmann and Norwood 1997). By comparison, Oakden *et al.* (1984) reported >50% mortality in amphipods, *Rhepoxynius abronius*, exposed to 613 mg/kg DW of zinc for 72-hours in marine sediments. Swartz *et al.* (1988) reported a 10-day LC_{50} (median lethal concentration) of 276 mg/kg DW for the same species of amphipod. Collectively, these data suggest the toxicity of zinc may vary depending on the species tested, duration of exposure, and the physical-chemical characteristics of the receiving water.

Zinc is an essential micronutrient in birds and mammals. For this reason, the balance between excess and insufficient zinc is important (Eisler 1993). Zinc deficiency can

occur in many wildlife species and is associated with severe effects on the growth, development, reproduction, and survival. The diets of birds and mammals should contain > 25 and > 30 mg Zn/kg DW of ration, respectively, to prevent zinc deficiency (Eisler 1993).

Exposure to elevated levels of dietary zinc can be toxic to birds and mammals, with effects including impaired survival, growth, and health. For example, survival was reduced in ducks fed single oral doses of > 742 mg Zn/kg body weight (BW) or diets containing 2,500-3,000 mg Zn/kg ratio for 30 days (NAS 1979). While adult chickens appear to be less sensitive than ducks, dietary exposure of chicks to 100 and 178 mg Zn/kg ration (i.e., 5 to 9 mg Zn/kg BW/day) caused increased pancreas histopathology and immunosuppression, respectively (Eisler 1993). Mammals appear to be as sensitive as birds to zinc, with acute oral LD50s (median lethal dose) of 350 to 800 mg Zn/kg BW reported for rats (Eisler 1993). Sub-lethal effects, such as weight loss, pancreas histopathology, digestive problems, and immunosuppression, were observed in various mammalian species fed 80 to 300 mg Zn/kg ration (i.e., 4 to 15 mg Zn/kg BW/day) for extended periods of time (i.e., several months; Eisler 1993).

7.8 References

Budavari, S., M.J. O'Neil, A. Smith, and P.E. Heckelman (Eds.). 1989. The Merck Index: An Encyclopedia of Chemicals, Drugs, and Biologicals. Eleventh Edition. Merck and Company, Inc. Rahway, New Jersey.

Borgmann, U., and W.P. Norwood. 1997. Toxicity and bioaccumulation of zinc and copper in *Hyalella azteca* exposed to metal-spiked sediments. Canadian Journal of Fisheries and Aquatic Science 54(5):1046-1054.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian water quality guidelines. Task Force on Water Quality Guidelines. Ottawa, Ontario.

Eisler, R. 1993. Zinc hazards to fish, wildlife, and invertebrates: A synoptic review. Biological Report 10. Contaminant Hazard Reviews Report 26. United States Fish and Wildlife Service. Laurel, Maryland.

James, RO. and MG MacNaughton. 1977. The absorption of aqueous heavy metals on inorganic minerals. Gatum. Cosmochin. Acta 41:1549-1555.

McNeely, R.N., V.P. Neimanis, and L. Dwyer. 1979. Water quality sourcebook: A guide to water quality parameters. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

NAS (National Academy of Sciences). 1979. Zinc. Subcommittee on Zinc. National Research Council. University Parl Press. Baltimore, Maryland.

Oakden, J.M., J.S. Oliver, and A.R. Flegal. 1984. Behavioural responses of a phoxocephalid amphipod to organic enrichment and trace metals in sediments. Marine Ecology-Progress Series 14:253-257.

Opresko, D.M. 1992. Toxicity summary for zinc and zinc compounds. Health and Safety Research Division. Oak Ridge National Laboratory. Oak Ridge, Tennessee.

Swartz, R.C., P.F. Kemp, D.W. Schults, and J.O. Lamberson. 1988. Effects of mixtures of sediment contaminants on the marine infaunal amphipod, *Rhepoxynius abronius*. Environmental Toxicology and Chemistry 7:1013-1020.

Taylor, M.C. and A. Demayo. 1980. Zinc. *In:* Guidelines for Surface Water Quality. Volume 1. Inorganic Chemical Substances. Inland Waters Directorate. Environment Canada. Ottawa, Ontario.

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for zinc. EPA-440/5-80-079. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1987. Ambient water quality criteria for zinc - 1987. EPA-440/5-87-003. Criteria and Standards Division. Office of Regulations and Standards. Washington, District of Columbia.

Appendix A2-8 An Overview of the Environmental Fate and Effects of Polycyclic Aromatic Hydrocarbons

8.1 Identity

Polycyclic aromatic hydrocarbons (PAHs) are a diverse class of organic compounds that includes about one hundred individual compounds containing two or more fused benzene, or aromatic, rings (McElroy *et al.* 1989). The term low molecular weight (LMW) PAHs is applied to the group of PAHs with fewer than four rings, while high molecular weight (HMW) PAHs have four or more rings. The LMW PAHs are considered to include acenaphthene, acenaphthylene, anthracene, fluorene, naphthalene, 2-methylnaphthalene, and phenanthrene. The HMW PAHs are considered to include benz[a]anthracene, benzo[a]pyrene, chrysene, dibenz[a,h]anthracene, fluoranthene, and pyrene. While these 13 parent PAHs are composed entirely of carbon and hydrogen atoms, nitrogen, sulphur, and oxygen atoms may be substituted on one or more of the benzene rings to form a variety of heterocyclic aromatic compounds (HACs; McElroy *et al.* 1989; Wilson and Jones 1993).

8.2 Uses

While many PAHs do not have any significant commercial applications, several are important in various industrial processes. For example, acenaphthene, anthracene, and pyrene are used as intermediates in the chemical (i.e., soap, pesticide, and dye production), photographic and pharmaceutical industries (Fidler *et al.* 1991). Certain PAHs, such as anthracene and phenanthrene, are also used in the production of

explosives, including pyrotechnic materials, and in the processing of certain foods. Importantly, naphthalene is used extensively in the production of phthalic anhydride, which is a precursor in the production of dyes, plasticisers, resins, and insect repellents (Government of Canada 1994).

8.3 Sources

A variety of natural and anthropogenic activities result in the production of PAHs. Most commonly, PAHs are produced as a result of the incomplete combustion of wood and fossil fuels. However, pyrolysis of organic materials (e.g., coal tar, creosote, anthracene oil, coal tar pitch, and carbon black) and diagenesis of sedimentary organic materials (e.g., which form fossil fuels) can also result in the formation of PAHs (LGL 1993). Other sources of these substances include biosynthesis by microbes and plants, tire wear, cigarette smoke, asphalt production, and wood preservatives (Slooff *et al.* 1989). Oil spills represent an important source of the PAHs that are released into marine and estuarine waters.

8.4 Physical and Chemical Properties

As might be expected based on the differences in their structures, the physical and chemical properties of the substances within these groups are highly variable. In general, PAHs tend to have high melting points, high boiling points, low water solubilities, and low vapour pressures. The LMW PAHs are generally more soluble in water (45 to 31 700 ：gL^{-1} at 25°C) than the HMW PAHs (0.5 to 140 ：gL^{-1} at 25°C; Southworth 1979; NRCC 1983). The solubility of PAHs tends to increase with increasing water temperature and to decrease with increasing salinity (NRCC 1983). The octanol/water (K_{ow}) and organic carbon (K_{oc}) partition coefficients of all PAHs are relatively high; however, those of HMW PAHs (5.32 - 6.04 and 5.23 - 5.94,

respectively) exceed those of LMW PAHs (3.37 - 4.46 and 3.32 - 4.39, respectively). As a result, these compounds tend to be lipophilic (i.e., they have an affinity for fatty organic substances) and readily adsorb to both organic and inorganic particulate matter (Government of Canada 1994).

8.5 Environmental Fate and Transport

The behavior of PAHs in surface waters depends on a variety of chemical-specific and site-specific factors, with physicochemical properties playing an important role in determining their fate in aquatic systems. While PAHs with high solubilities (such as naphthalene) may remain dissolved in surface waters, those with lower solubilities are likely to form associations with colloidal material (Wijayarante and Means 1984) or suspended particulates (Varanasi 1989). Hence, PAHs are commonly associated with suspended particulates in aquatic systems (Harrison *et al.* 1975; Germain and Langlois 1988). While PAHs associated with suspended particulates may be photochemically degraded (David and Boule 1993), biodegraded (Hall *et al.* 1986), transported to other areas (Murphy *et al.* 1988), and incorporated into aquatic biota (Baker *et al.* 1991), deposition and consolidation with bedded sediments (often within several hundred meters of the source) probably represents the most important environmental fate process (Murphy *et al.* 1988; Herrmann and Thomas 1984). Hence, sediments represent the major environmental sink for these compounds (Government of Canada 1994).

8.6 Bioaccumulation

Bioaccumulation may occur as a result of exposures to PAHs in water, through contact with suspended and bedded sediments, and through consumption of contaminated food organisms (Ringuette *et al.* 1993). Bioaccumulation of PAHs is

generally evaluated by calculating the ratio of the concentration of a chemical in the organisms tissues (expressed on a wet weight basis) to its concentration in the exposure medium. When water represents the exposure medium, this ration is termed a bioconcentration factor (BCF); the comparable ratio for sediment exposures is the biota-sediment bioaccumulation factor (BSAF).

Information from laboratory studies suggest that water-to-tissue BCFs are directly correlated with the K_{ow} (octanol/water partition coefficient) of the PAH under investigation. For example, BCFs in the cladoceran, *Daphnia pulex*, after one hour of exposure ranged from 131 for naphthalene to 10,100 for benz[a]anthracene (Southworth *et al.* 1978). Similar results (i.e., K_{ow} -dependent bioaccumulation) have been observed when benthic invertebrates were exposed to PAH-contaminated sediments (Eadie *et al.* 1982). In addition to an array of physical and chemical factors, the bioaccumulation of sediment-associated PAHs may be influenced by the physiology and feeding strategy of the species under consideration. Fish, for example, have well developed mixed function oxidase (MFO) systems which rapidly transform PAHs into substances that are more readily excreted (Stegeman 1981; Varanasi *et al.* 1989). In contrast, certain benthic invertebrates (e.g., bivalve mollusks) have much less efficient MFO systems and, therefore, accumulate higher concentrations of PAHs in their tissues (Naf *et al.* 1993; Germain *et al.* 1993). Invertebrate species and tissues with high lipid contents tend to accumulate higher levels PAHs than those with lower lipid contents (Neff 1985). Furthermore, lipid-associated PAHs may be mobilized, transferred to lipid-rich eggs, and released during spawning activities (Rossi and Anderson 1977; NAS 1985). Therefore, overall condition and reproductive state are important considerations when assessing bioaccumulation.

8.7 Effects on Aquatic and Terrestrial Organisms

The acute toxicity of PAHs is primarily associated with their action as non-polar narcotics (i.e., narcosis; Eisler 1987). That is, PAHs tend to enter the organisms and bind irreversibly to lipophilic sites within the cell. Binding to sites on cell membranes tends to disrupt surface membrane processes, inhibit ion and gas exchange, and increase osmosis across the membrane (Van Overbeek and Blondeau 1954). In fish and other organisms that rely on gills for respiration, hypoxia and osmotic imbalances may result from impaired membrane function (Sims and Overcash 1983). In the tissues, changes in membrane permeability may disrupt neurological and muscular function (Neff 1985).

While non-polar narcosis is the primary mode of toxicity for PAH with three or fewer aromatic rings, certain HMW-PAHs may also be associated with mutagenic, carcinogenic, and teratogenic effects (Eisler 1987). In particular, methyl-substituted PAHs tend to be much more mutagenic than the parent compound (Government of Canada 1994). Interestingly, however, PAHs with longer alkyl-substitution tend to be less toxic due to their decreased ability to cross cell membranes (NRCC 1983). In addition, the metabolic degradation products of HMW-PAHs, particularly epoxide derivatives, tend to be highly mutagenic (NRCC 1983). These metabolites tend to be much more electrophilic and reactive than the parent compound, which increases the likelihood that they will bind covalently to DNA (deoxyribonucleic acid), RNA (ribonucleic acid), and other cellular proteins (Varanasi 1989). In turn, these complexes may be associated with alterations in normal cellular processes (e.g., cell division, protein synthesis). Such alterations may be expressed by tumour formation, developmental abnormalities, and/or other related effects in aquatic animals (Heidelberger 1976; Larson *et al.* 1976; 1977). In plants, PAH metabolites may bind with various components of the chloroplast and, thereby, inhibit photosynthesis (Neff 1979).

Exposure to PAHs has been associated with a wide range of adverse effects in aquatic organisms, including effects on survival, growth, reproduction, metabolism, and health (Eisler 1987). The responses of aquatic biota vary significantly among taxonomic groups and depend, at least in part, on their ability to metabolize and excrete PAHs (Fidler *et al.* 1991). In addition, biotic responses may be affected by the duration of exposure to PAHs, by the substance or group of substances under consideration, and by ambient environmental conditions. Among the various PAHs, the lowest observed effect levels of PAHs ranged from 5 µg/L for benzo[a]pyrene (96-hour LC_{50} (median lethal concentration) for the water flea, *Daphnia pulex*) to 125 µg/L for fluorene (which resulted in reduced fecundity in the water flea, *Daphnia magna* in a 14-day test; Government of Canada 1994).

In the field, PAHs always occur as complex mixtures of the individual PAHs, commonly in association with other contaminants. Toxic effects on sediment-dwelling organisms are likely to result from the cumulative effects of these mixtures of contaminants. For this reason, it is important to evaluate the interactive effects of the individual PAHs using a toxic units model (Swartz 1999). Such models provide a basis for determining the overall toxicity of the contaminant mixture by summing the toxic units (i.e., TU = measured concentration/toxicity reference value) for the substances that have a similar mode of toxicity (i.e., the individual PAHs). Acute and chronic toxicity thresholds (i.e., for narcosis) of 50.2 and 9.9 mg/kg dry weight (DW) @ 1%OC, respectively, have been determined for total PAHs using equilibrium partitioning (DiToro and McGrath 2000). By comparison, a 10-day LC_{50} of 23.9 mg tPAH/kg DW @ 1%OC has been reported for the amphipod, *Rhepoxynius abronius* (Swartz *et al.* 1997).

A variety of adverse effects have been observed in birds and mammals exposed to PAHs, including non-neoplastic effects and carcinogenicity (Eisler 1987). While metabolic dysfunction, immobility, and death have been reported in birds and

mammals fed PAH-contaminated diets, the doses that cause such effects tend to be much higher than those that induce neoplasms (i.e., up to an order or magnitude higher; ATSDR 1990). In mice, ingestion of diets containing 50 to 250 mg/kg benzo[a]pyrene (i.e., 6.5 to 32.5 mg/kg body weight; BW/day) for 70 to 197 days resulted in a > 70% incidence of stomach tumors (ATSDR 1990). Long-term dietary to benz[b]fluoranthene, benz[j]fluoranthene, benz[k]fluoranthene, and indeno[1,2,3-cd]pyrene have also been shown to induce tumors in mice when exposed via other exposure routes (IARC 1983).

8.8 References

ATSDR (Agency for Toxic Substances and Disease Registry) 1990. Toxicological profiles for PAHs, Group III final reports. Agency for Toxic Substances and Disease Registry. United States Public Health Service. 230 pp. + apps.

Baker, J.E., S.J. Eisenreich, and B.J. Eadie. 1991. Sediment trap fluxes and benthic recycling of organic carbon, polycyclic aromatic hydrocarbons, and polychlorobiphenyl congeners in Lake Superior. Environmental Science and Technology 25(3):500-509.

David, B. and P. Boule. 1993. Phototransformation of hydrophobic pollutants in aqueous medium I-PAHs adsorbed on silica. Chemosphere 26, 9:1617-1630.

DiToro, D.M. and J.A. McGrath. 2000. Technical basis for narcotic chemicals and polycyclic aromatic hydrocarbon criteria. II Mixtures and sediments. Environmental Toxicology and Chemistry 19:1971-1982.

Eadie, B.J., W. Faust, W.S. Gardner, and T. Nalepa. 1982. Polycyclic aromatic hydrocarbons in sediments and associated benthos in Lake Erie. Chemosphere 11:185-191.

Eisler, R. 1987. Polycyclic aromatic hydrocarbon hazards to fish, wildlife, and invertebrates: A synoptic review. Biological Report. Publication No. 85(1.11). Contaminant Hazard Reviews Report No. 11. U.S. Department of the Interior. Fish and Wildlife Service. Patuxent Wildlife Research Center. Laurel, Maryland.

Fidler L.E., J.B. Miller and D.D. MacDonald. 1991. Canadian water quality guidelines for polycyclic aromatic compounds. Prepared for the Canadian Council of Ministers for the Environment. Environment Canada. Hull, Quebec. 229 p + appendices.

Germain, A. et C. Langlois. 1988. Contamination des eaux et des sédiments en suspension du fleuve Saint-Laurent par les pesticides organochlorés, les biphényles polychlorés et d'autres contaminants organiques prioritaires. Water Pollution Research Journal of Canada 23:603-614.

Germain, A., F. Perron, and R. Van Coillie. 1993. PAHs in the environment: Fate, ecotoxicity, and regulations. Support Document No. 3. Assessment Report on PAHs under the Canadian Environmental Protection Act Priority Substances List. Prepared for: Environment Canada. Quebec Region. Environmental Protection Branch. Prevention and Pollution Abatement Division. Montreal, Quebec. 104 pp.

Government of Canada (Environment Canada and Health Canada). 1994. Canadian Environmental Protection Act. Priority Substances List Assessment Report: Polycyclic Aromatic Hydrocarbons. Ottawa, Ontario. 61pp.

Hall, W.S., T.J. Leslie, and K.L. Dickson. 1986. Effects of suspended solids on the biotransformation of acenaphthene. Bulletin of Environmental Contamination and Toxicology 36:286-293.

Harrison, R.M., R. Perry, and R.A. Wellings. 1975. Polynuclear aromatic hydrocarbons in raw, potable and waste waters. Water Research 9:331-346.

Heidelberger, C. 1976. Studies on the mechanisms of carcinogenesis by polycyclic aromatic hydrocarbons and their derivatives. *In:* Carcinogenesis - A Comprehensive Survey, Volume 1. Polycyclic Aromatic Hydrocarbons. R. Freudenthal and P.W. Jones (Eds.). Raven Press. New York, New York.

Herrmann, R. and W. Thomas. 1984. Behaviour of some PAH, PCB and organochlorine pesticides in an estuary, a comparison - Exe, Devon. Fresenius Z Anal Chem 319:152-159

IARC (International Agency for Research on Cancer). 1983. Polynuclear aromatic compounds. Part 1: Chemical, Environmental and Experimental Data. IARC Monographs on the Evaluation of the Carcinogenic Risk of Chemicals to Humans. Vol. 32. Lyon, France. pp. 33-91.

LGL (Lalonde, Girouard, Letendre and Associates). 1993. PAH emissions into the Canadian environment - 1990. Support Document No. 1 for the National Evaluation Report on PAHs. Prepared for: Environment Canada. Conservation and Protection. Quebec Region. Montreal, Quebec.

Larson, R.A., D.W. Blankenship, and L.L Hunt. 1976. Toxic hydroperoxides: Photochemical formation from petroleum constituents. *In:* Sources, Effects, and Sinks of Hydrocarbons in the Aquatic Environment. American Institute of Biological Sciences. Washington, District of Columbia.

Larson, R.A., L.L. Hunt and D.W. Blankenship. 1977. Formation of toxic products from a #2 fuel oil by photooxidation. Environmental Science and Technology 11:492-496.

McElroy, A.E., J.W. Farrington and J.M. Teal. 1989. Bioavailability of PAHs in the aquatic environment. *In:* Metabolism of PAHs in the Aquatic Environment. U. Varanasi (Ed.). CRC Press, Inc. Boca Raton, Florida.

Murphy, P.P., T.S. Bates, H.C. Curl Jr., R.A. Freely, and R.S. Burger. 1988. The transport and fate of particulate hydrocarbons in an urban fjord-like estuary. Estuarine, Coastal and Shelf Science 27:461-482.

Naf, C., D. Broman, J. Axelman, C. Rolff, P-A. Bergqvist, K. Lundgren, K. Strandberg, B. van Bavel and C. Rappe. Accumulation of sediment-associated PAHs, non-ortho PCBs and ortho-PCBs in situ by the amphipod *Monoporeia affinis* in the Gulf of Bothnia. University of Stockholm. Stockholm, Sweden.

NAS (National Academy of Sciences). 1985. Oil in the Sea: Inputs, Fates and Effects. US National Academy of Science, National Research Council. National Academy Press, Washington, District of Columbia. 601 pp.

NRCC (National Research Council of Canada). 1983. Polycyclic Aromatic Hydrocarbons in the Aquatic Environment: Formation, Sources, Fate and Effects on Aquatic Biota. NRC Associate Committee on Scientific Criteria for Environmental Quality. Publication No. NRCC 18981. 209 pp.

Neff, J.M. 1985. Polycyclic aromatic hydrocarbons. *In:* Fundamentals of Aquatic Toxicology, Methods and Applications. G.M. Rand and S.R. Petrocelli (Eds.). Hemisphere Publishing Corporation. New York, New York. pp. 416-454.

Ringuette, S., A. Germain, C. Gonthier, and F. Perron. 1993. Presence of PAHs in the Canadian environment. An overview. Supporting Document No. 2. Prepared for: Environment Canada. Conservation and Protection. Quebec Region. Montreal, Quebec.

Rossi, S.S. and J.W. Anderson. 1977. Accumulation and release of fuel oil-derived diaromatic hydrocarbons by the polychaete *Neanthes arenaceodentata*. Marine Biology 39:51-55.

Sims, R.C. and M.R. Overcash. 1983. Fate of polynuclear aromatic compounds (PNAs) in soil plant systems. Residue Revues 88: 1-68.

Sloof, W., J.A. Janus, A.J. Matthijsen, G.K. Montizaan and J.P.M. Ros (Eds). 1989. Integrated criteria document PAHs. Report no. 758474011. National Institute of Public Health and Environmental Protection, Biltoven The Netherlands. (As cited in Government of Canada (Environment Canada and Health Canada) 1994).

Southworth, G.R. 1979. The role of volatilization in removing PAHs from aquatic environments. Bulletin of Environmental Contamination and Toxicology 21:507-514.

Southworth, G.R., J.J. Beauchamp, and P.K. Schmieden. 1978. Bioaccumulation Potential of Polycyclic Aromatic Hydrocarbons in Daphnia pulex. Water Research 12: 973-977 (As cited in Government of Canada 1994).

Stegeman, J. 1981. Polynuclear aromatic hydrocarbons and their metabolism in the marine environment. *In:* Polycyclic hydrocarbons and cancer. H.V. Gelboin and P.O. Ts'o (Eds.). Volume 3. Academic Press. New York, New York. (As cited in Eisler 1987).

Swartz, R.C. 1999. Consensus sediment quality guidelines for polycyclic aromatic hydrocarbon mixtures. Environmental Toxicology and Chemistry 18:780-787.

Swartz, R.C., S.F. Ferraro, J.O. Lamberson, F.A. Cole, R.J. Ozretich, B.L. Boese and D.W. Schults. 1997. Photoactivation and toxicity of mixtures of polycyclic aromatic hydrocarbon compounds in marine sediment. Environmental Toxicology and Chemistry 16:2151-2157.

Van Overbeek, J. and R. Blondeau. 1954. Mode of action of phytotoxic oils. Weeds 3:55-65. (As cited in Neff 1985).

Varanasi, U. (Ed.). 1989. Metabolism of Polycyclic Aromatic Hydrocarbons in the Aquatic Environment. CRC Press Inc., Boca Raton, Florida. 341 pp.

Varanasi, U., J.E. Stein, and M. Nishimoto. 1989. Biotransformation and disposition of polycyclic aromatic hydrocarbons (PAH) in fish. *In:* Metabolism of Polycyclic Aromatic Hydrocarbons in the Aquatic Environment. U. Varanasi, Ed. CRC Press, Inc. Boca Raton, Florida. pp. 93-149.

Wijayaratne, R.D. and J.C. Means. 1984. Adsorption of polycyclic aromatic hydrocarbons by natural esturine colloids. Marine Environmental Research 11:77-89.

Wilson, S.C. and K.C. Jones. 1993. Bioremediation of soil contaminated with polynuclear aromatic hydrocarbons (PAHs): a review. Environmental Pollution. 81(3):229-249.

Appendix A2-9 An Overview of the Environmental Fate and Effects of Polychlorinated Biphenyls

9.1 Identity

Polychlorinated biphenyls (PCBs) is the generic term applied to a group of 209 chlorinated organic compounds that have similar molecular structures and properties. PCBs are synthetic chemicals that were produced commercially in the United States between 1929 and 1977, at which time their production was banned. The principal manufacturer of PCBs in the United States was the Monsanto Chemical Company; the PCBs sold by Monsanto were marketed under the trade name of Aroclor.

9.2 Uses

As a class of compounds, PCBs are fire-resistant and chemically stable. In addition, PCBs conduct electricity poorly. These and other properties made PCBs useful in a range of industrial and consumer applications. The vast majority of the PCBs used in North America were employed as cooling and insulating fluids in industrial transformers and capacitors. However, PCBs have also been used in several other applications, including as hydraulic fluids, heat transfer fluids, and plasticizers. As a result of their widespread use, PCBs have been released into the environment worldwide. The results of various monitoring programs indicate that PCBs are ubiquitous environmental contaminants and are commonly detected in air, precipitation, soil, surface water, groundwater, sediment, and living organisms.

9.3 Sources

PCBs are synthetic substances and, as such, they are released into the environment solely as a result of human activities. In recent years, restrictions on the use and disposal of PCBs has greatly reduced the magnitude and extent of PCB inputs into the environment. Nonetheless, a variety of activities resulted in significant losses of these substances prior to the implementation of those regulations, including uncontrolled past use, past disposal practices, illegal disposal, and accidental releases.

9.4 Physical and Chemical Properties

The majority of PCB congeners tend to have low solubilities, low vapour pressures, high dielectric constants (i.e., low conductivity), low flammability, and high resistance to chemical breakdown. Commercial PCB formulations are usually light coloured, viscous liquids that have a slippery or oily appearance. The density of all PCB formulations is greater than that of water (WHO 1993) and increases with increasing chlorine content. As a group, PCBs are sparingly soluble in water. However, aqueous solubilities vary substantially among the various PCB mixtures and congeners. In contrast to water, PCBs are readily soluble in oils and other organic solvents (WHO 1993). The vapour pressure of PCBs is variable, spanning two orders of magnitude for PCB mixtures and eight orders of magnitude for PCB congeners. As is the case for solubility, vapour pressures tend to decrease with the addition of each chlorine atom (Mackay *et al.* 1983). The available data indicate that PCBs are highly lipophilic (Mackay *et al.* 1992).

9.5 Environmental Fate and Transport

Many of the same physical and chemical properties that made PCBs useful in commercial and industrial applications dictate their fate upon release into the environment. Due to the density of PCBs, once released to aquatic systems they tend to sink to the bottom of the water body. As a result of their hydrophobicity, as well as their generally low water solubilities, PCBs tend to accumulate in sediments and soils that contain organic carbon. Together, these properties give PCBs a high potential for uptake by aquatic and terrestrial organisms, including fish, birds, mammals, and other wildlife. Due to chemical stability, PCBs are highly persistent in the environment. Hence, cycling, rather than degradation, represents the most important process affecting PCBs once they have been released into the environment.

9.6 Bioaccumulation

Information from both field and laboratory studies indicates that PCBs bioaccumulate in the tissue of benthic invertebrate species (van der Oost *et al.* 1988; Lester and McIntosh 1994). For total PCBs, laboratory-derived sediment-to-biota bioaccumulation factors (BSAF = [tissue] ÷ [sediment]; Ferraro *et al.* 1990) for clams, shrimp, and sandworms ranged from 0.02 to 1.89 (Pruell *et al.* 1990; Boese *et al.* 1995; Lester and McIntosh 1994). PCBs are also known to accumulate to elevated levels in the tissues of freshwater fish species (Schwartz *et al.* 1987; van der Oost *et al.* 1988; Macdonald *et al.* 1993; Lores *et al.* 1993), with the partitioning of PCBs depending on fish lipid content, the trophic level of the fish species, and the trophic structure of the food web (Rowan and Rasmussen 1992). As a result, calculated BSAFs and bioavailability indices (BI = [tissue] ÷ lipid content x TOC (total organic carbon) content ÷ [sediment]; Carey *et al.* 1990; Foster *et al.* 1987) vary substantially between species and between lake systems (Macdonald *et al.* 1993). For example, BIs ranged from 0.72 to 259 for chinook salmon in the Great Lakes basin

(Rowan and Rasmussen 1992). Intermediate BSAFs and BIs were calculated for the fish in Lake Ontario, Lake St. Clair, and Lake Superior (Evans *et al.* 1991; Rowan and Rasmussen 1992). Fish-eating birds (e.g., bald eagles) and mammals (e.g., dolphins) at the highest trophic levels in the food web tend to accumulate PCBs to the highest levels in their tissues (Eisler 1986).

9.7 Effects on Aquatic and Terrestrial Organisms

Exposure to PCBs is known to cause a broad range of adverse effects in aquatic organisms. In controlled laboratory studies, both short- (i.e., # 96 hrs) and longer- (i.e., > 96 hrs) term exposure of aquatic organisms (plants, invertebrates, fish, and amphibians) to water-borne PCBs has been shown to cause a variety of adverse effects, including increased mortality, reduced metabolic rates, reduced growth rates, and impaired reproduction (CCREM 1987; Moore and Walker 1991; CCME 1999). In aquatic invertebrates, acute toxicity thresholds range from 10 µg/L for amphipods (*Gammarus fasciatus*) to 400 µg/L for damselflies (*Ischnura verticalis*; CCREM 1987). Freshwater fish appear to be more sensitive that invertebrates to the effects of PCBs, with 96-hour LC_{50}s (median lethal concentrations) of 2.0 and 2.3 µg/L reported for rainbow trout and largemouth bass, respectively (Birge *et al.* 1979). In long-term tests, chronic toxicity thresholds for fish and invertebrates ranged from 0.2 to 15 µg/L (USEPA 1980). Exposure to relatively low levels of PCBs (i.e., # 1 µg/L) inhibited photosynthesis in phytoplankton (CCREM 1987).

Exposure to sediment-associated PCBs can adversely affect sediment-dwelling organisms. For example, Swartz *et al.* (1988) demonstrated that PCBs (Aroclor 1254) were acutely toxic to amphipods (*Rhepoxynius abrionius*), with a 10-day LC_{50} of 8.8 mg/kg dry weight (DW) reported. Impaired reproduction in copepods (*Microarthridion littorale*) was observed at PCB concentrations as low as 4 mg/kg

DW (DiPinto *et al.* 1993). Because acute-to-chronic ratios tend to be large for PCBs (i.e., 11 to 58; DiPinto *et al.* 1993; MacDonald *et al.* 2000), sublethal effects on sensitive species and life stages atre likely to occur at concentrations below 1 mg/kg DW.

In aquatic-dependent avian and mammalian wildlife, consumption of contaminated prey items (i.e., fish and invertebrates) represents the primary source of exposure to PCBs (Moore *et al.* 1999). The Ah-receptor mechanism is considered to be the primary mode of action for PCBs (Coulston and Kolbye 1994; Metcalfe and Haffner 1995). The biological effects associated with the induction of enzyme systems by PCBs arise because these enzymes are critical to the homeostasis of the organism. Induction of MFO (mixed function oxidase) enzyme systems, including the aryl hydrocarbon hydroxylase (AHH) and ethoxyresorufin *O*-deethylase (EROD) systems, can result in altered activity of gonadal (e.g., estrogens), pituitary (growth hormone), thyroid (e.g., thyroxine), and adrenocortical (e.g., cortisol) hormones. As many bodily functions are regulated by hormonal (or endocrine) systems, alteration of these systems can lead to adverse effects on the physiology of the organism. For example, suppression of various gonadal hormones is likely to result in direct effects on reproduction, such as reduced fertility. Similarly, reduction in the production of hormones from the pituitary gland can result in adverse effects on growth. Carcinogenic, mutagenic, and teratogenic effects have also been observed in terrestrial organisms exposed to sub-lethal levels of certain PCBs in field and laboratory studies (Eisler 1986). Consumption of higher doses of PCBs has been shown to be lethal in many wildlife species, with mink being among the most sensitive species tested (MESL 1996). In this species, long-term dietary exposure to daily doses as low as 1.5 mg/kg body weight (BW) resulted in > 50% mortality, while reduced growth was observed at much lower doses of PCBs (i.e., 0.17 mg/kg BW/day; Bleavens 1980).

The results of mammalian toxicity tests suggest that a relatively small group of congeners are highly toxic and may account for most of the toxicity (Safe *et al.* 1985). The non-ortho-, mono-ortho-, and di-ortho-PCBs are considered to have a similar receptor-mediated mechanism of action as the polychlorinated dibenzo-*p*-dioxins and polychlorinated dibenzofurans (Safe 1990; 1994). For this reason, a method has been developed to facilitate assessments of the cumulative effects of dioxins, furans, PCBs, and other related halogenated aromatic compounds that occur in complex mixtures. This method is based on the determination of the relative toxicities of dioxin-like substances in relation to 2,3,7,8-TCDD. Specifically, toxic equivalency factors (TEFs) are assigned to each chemical based on the results of both *in vivo* and *in vitro* studies. The most recent TEFs that have been established for co-planar PCBs using mammalian data are presented in Table A2-1 (van den Berg *et al.* 1998).

9.8 References

Ahlborg, U.G., G.C. Becking, L.S. Birnbaum, A. Brouwer, H.J. Derks, M.Feeley, G.Golor, A. Hanberg, J.C. Larsen, A.K.D. Liem, S.H. Safe, C. Schlatter, F.Waern, M. Younes, and E. Yrjanheikki. 1994. Toxic equivalency factors for dioxin-like PCBs. Chemosphere 28(6):1049-1067.

Birge, W.J., J.A. Black and D.M. Bruser. 1979. Toxicity of Organic Chemicals to Embryo-larval Stages of Fish. U.S. Enviromental Protection Agency. EPA-560/11-79-007 (As cited in CCREM 1987).

Bleavins, M.R, R.J Aulerich, and R.K. Ringer. 1980. Polychlorinated biphenyls (Aroclor 1016 and 1242): Effects on survival and reproduction in mink and ferrets. Archives of Environmental Contamination and Toxicology 9:627-635. (As cited in USEPA 1993).

Boese, B.L., M. Winsor, H. Lee II, S. Echols, J. Pelletier, and R. Randall. 1995. PCB congeners and hexachlorobenzene biota sediment accumulation factors for *Macoma nasuta* exposed to sediments with different total organic carbon contents. Environmental Toxicology and Chemistry 14(2):303-310.

Carey, A.E., N.S. Shifrin, and A.P. Briefer. 1990. Derivation of a Lake Ontario bioaccumulation factor for 2,3,7,8-TCDD. *In:* Lake Ontario TCDD Bioaccumulation Study: Final Report. U.S. Environmental Protection Agency. New York State Department of Environmental Conservation. New York State Department of Health. Occidental Chemical Corporation. Albany, New York.

CCME (Canadian Council of Ministers of the Environment). 1999. Canadian water quality guidelines for the protection of aquatic life. CCME Task Group on Water Quality Guidelines. Ottawa, Ontario.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. PCB action plan: Final report. 22 pp.

CCREM (Canadian Council of Resource and Environment Ministers). 1987. Canadian Water Quality Guidelines. Task Force on Environment Ministers. Ottawa, Ontario.

Coulston, F. and A.C. Kolbye [Ed.]. 1994. Interpretive review of the potential adverse effects of chlorinated organic chemicals on human health and the environment: Report of an expert panel. Chapter 5: Polychlorinated Biphenyls. Regulatory Toxicology and Pharmacology 20(1):S187-307.

DiPinto, L.M., B.C. Coull, and G.T. Chandler. 1993. Lethal and sublethal effects of the sediment-associated PCB Aroclor 1254 on a meiobenthic copepod. Environmental Toxicology and Chemistry 12:1909-1918.

Eisler, R. 1986. Polychlorinated biphenyl hazards to fish, wildlife, and invertebrates: A synoptic review. Biological Report 85. Fish and Wildlife Service. United States Department of the Interior. Laurel, Maryland. 72 pp.

Evans, M.S., G.E. Noguchi, and C.P. Rice. 1991. The biomagnification of polychlorinated biphenyls, toxaphene, and DDT compounds in a Lake Michigan offshore food web. Archives of Environmental Contamination and Toxicology 20:87-93.

Ferraro, S.P., H. Lee II, R.J. Ozretich, and D.T. Specht. 1990. Predicting bioaccumulation potential: A test of fugacity-based model. Archives of Environmental Contamination and Toxicology 19:386-394.

Foster, G.D., S.M. Baksi, and J.C. Means. 1987. Bioaccumulation of trace organic contaminants from sediment by baltic clams (*Macoma balthica*) and soft-shell clams (*Mya arenaria*). Environmental Toxicology and Chemistry 6:969-976.

Lester, D.C. and A. McIntosh. 1994. Accumulation of polychlorinated biphenyl congeners from Lake Champlain sediments by *Mysis relicta*. Environmental Toxicology and Chemistry 13(11):1825-1841.

Lores, E.M., J.M. Patrick, and J.K. Summers. 1993. Humic acid effects on uptake of hexachlorobenzene and hexachlorobiphenyl by sheepshead minnows in static sediment/water systems. Environmental Toxicology and Chemistry 12:541-550.

Macdonald, C.R., C.D. Metcalfe, G.C. Balch, and T.L. Metcalfe. 1993. Distribution of PCB congeners in seven lake systems: Interactions between sediment and food-web transport. Environmental Toxicology and Chemistry 12:1991-2003.

MacDonald, D.D., L.M. Dipinto, J. Field, C.G. Ingersoll, E.R. Long, and R.C. Swartz. 2000. Development and evaluation of consensus-based sediment effect concentrations for polychlorinated biphenyls. Environmental Toxicology and Chemistry 19(5): 1403-1413.

Mackay, D., W.Y. Shiu, and J.C. Ma. 1992. Illustrated Handbook of Physical-Chemical Properties and Environmental Fate for Organic Chemicals Vol. I. Lewis Publishers Inc. Chelsea, Michigan. 697 pp.

Mackay, D., W.Y. Shiu, J. Billington, and G.L. Huang. 1983. Physical and chemical properties of polychlorinated biphenyls. *In*: Mackay, D., S. Paterson, S. J. Eisenreich, and M. S. Simmons (Ed). Physical Behavior of PCBs in the Great Lakes. Ann Arbor, Michigan, Ann Arbor Science Publishers. pp. 59-69.

MESL (MacDonald Environmental Sciences Ltd.). 1996. Canadian tissue residue guidelines for polychlorinated biphenyls. Prepared for Guidelines Division. Environment Canada. Ottawa, Ontario.

Metcalfe, C.D. and G.D. Haffner. 1995. The ecotoxicology of coplanar polychlorinated biphenyls. Environmental Review 3:171-190

Moore, D. R.J., B.E. Sample, G.W. Suter, B.R. Parkhurst and R. Scott Teed. 1999. A probabilistic risk assessment of the effects of methylmercury and PCBs on mink and kingfishers along east fork Poplar Creek, Oak Ridge, Tennessee, USA. Environmental Toxicology and Chemistry 18(12): 2941-2953.

Moore, D.R. and S.L. Walker. 1991. Canadian Water Quality Guidelines for Polychlorinated Biphenyls in Coastal and Estuarine Waters. Inland Waters Directorate, Water Quality Branch. Ottawa, Ontario. 61 pp.

Pruell, R.J., N.I. Rubinstein, B.K. Taplin, J.A. LiVolsi, and C.B. Norwood. 1990. 2,3,7,8-TCDD, 2,3,7,8-TCDF and PCBs in marine sediments and biota: Laboratory and field studies. EPA/ 600/8-90/068. United States Environmental Protection Agency. Environmental Research Laboratory. Narragansett, Rhode Island. 72 pp.

Rowan, D.J. and J.B. Rasmussen. 1992. Why don't Great Lakes fish reflect environmental concentrations of organic contaminants?-An analysis of between-lake variability in the ecological partitioning of PCBs and DDT. Journal of Great Lakes Research 18:724-741.

Safe, S. 1994. Polychlorinated biphenyls (PCBs): Environmental impact, biochemical and toxic responses and implications for risk assessment. CRC Critical Reviews of Toxicology 24:1-63 (As cited in Ahlborg et al. 1994).

Safe, S. 1990. Polychlorinated biphenyls (PCBs), dibenzo-p-dioxins (PCDDs), and related compounds: Environmental and mechanistic considerations which support the development of toxic equivalency factors (TEFs). Toxicology 21:51-88.

Safe, S., S. Bandiera, T. Sawyer, B. Zmudzka, G. Mason, M. Romkes, M.A. Denomme, J. Sparling, A.B., Okey, and T. Fujita. 1985. Effects of structure on binding to the 2,3,7,8-TCDD receptor protein and AHH induction-halogenated biphenyls. Environmental Health Perspectives 61:21-33. (As cited in Coulston and Kolbye 1994).

Schwartz, T.R., D.L. Stalling and C.L. Rice. 1987. Are polychlorinated biphenyl residues adequately described by Aroclor mixture equivalents? Isomer-specific principal components analysis of such residues in fish and turtles. Environmental Science and Technology 21:72-76.

Swartz, R.C., P.F. Kemp, D.W. Schults, and J.O. Lamberson. 1988. Effects of mixtures of sediment contaminants on the marine infaunal amphipod, *Rhepoxynius abronius*. Environmental Toxicology and Chemistry 7:1013-1020.

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for polychlorinated biphenyls. EPA 440/5-80-068. Office of Water Regulations and Standards Criteria and Standards Division. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1993. Great Lakes water quality initiative criteria documents for the protection of wildlife (Proposed). DDT, Mercury, 2,3,7,8-TCDD, PCBs. EPA-822-R-93-007. Office of Water. Washington, District of Columbia.

van den Berg, M., L. Birnbaum, A.T. Bosveld, B. Brunström, P. Cook, M. Feeley, J. Giesy, A. Hanberg, R. Hasegawa, S.W. Kennedy, T. Kubiak, J.C. Larsen, F.X. van Leeuwen, A.K. Liem, C. Nolt, R.E. Peterson, L. Poeliinger, S. Safe, D. Schrenk, D. Tillitt, M. Tysiind, M. Younes, F. Waern, and T. Zacharewski. 1998. Toxic equivalency factors (TEFs) for PCBs, PCDDs, and PCDFs for humans and wildlife. Environmental Health Perspectives 106:775-792.

van der Oost, R., H. Heida, and A. Opperhuizen. 1988. Polychlorinated biphenyl congeners in sediments, plankton, molluscs, crustaceans, and eel in a freshwater lake: Implications of using reference chemicals and indicator organisms in bioaccumulation studies. Archives of Environmental Contamination and Toxicology 17:721-729.

WHO (World Health Organization). 1993. Environmental Health Criteria 140: Polychlorinated Biphenyls and Terphenyls. World Health Organization, Geneva, Switzerland. 682 pp.

Table A2-1. World Health Organization toxic equivalency factors (TEFs) for humans, mammals, fish, and birds (from van den Berg *et al.* 1998)

Congener	TEF		
	Humans/Mammals	Fish	Birds
2,3,7,8-TetraCDD	1	1	1
1,2,3,7,8-PentaCDD	1	1	1
1,2,3,4,7,8-HexaCDD	0.1	0.5	0.05
1,2,3,6,7,8-HexaCDD	0.1	0.01	0.01
1,2,3,7,8,9-HexaCDD	0.1	0.01	0.1
1,2,3,4,6,7,8-HeptaCDD	0.01	0.001	<0.001
OctaCDD	0.0001	<0.0001	0.0001
2,3,7,8-TetraCDF	0.1	0.05	1
1,2,3,7,8-PentaCDF	0.05	0.05	0.1
2,3,4,7,8-PentaCDF	0.5	0.5	1
1,2,3,4,7,8-HexaCDF	0.1	0.1	0.1
1,2,3,6,7,8-HexaCDF	0.1	0.1	0.1
1,2,3,7,8,9-HexaCDF	0.1	0.1	0.1
2,3,4,6,7,8-HexaCDF	0.1	0.1	0.1
1,2,3,4,6,7,8-HeptaCDF	0.01	0.01	0.01
1,2,3,4,7,8,9-HeptaCDF	0.01	0.01	0.01
OctaCDF	0.0001	<0.0001	0.0001
3,4,4',6-TetraCB (81)	0.0001	0.0005	0.1
3,3',4,4'-TetraCB (77)	0.0001	0.0001	0.05
3,3',4,4',5-PentaCB (126)	0.1	0.005	0.1
3,3',4,4',5,5'-HexaCB (169)	0.01	0.00005	0.001
2,3,3',4,4'-PentaCB (105)	0.0001	<0.000005	0.0001
2,3,4,4',5-PentaCB (114)	0.0005	<0.000005	0.0001
2,3',4,4',5-PentaCB (118)	0.0001	<0.000005	0.00001
2',3,4,4',5-PentaCB (123)	0.0001	<0.000005	0.00001
2,3,3',4,4',5-HexaCB (156)	0.0005	<0.000005	0.0001
2,3,3',4,4',5'-HexaCB (157)	0.0005	<0.000005	0.0001
2,3',4,4',5,5'-HexaCB (167)	0.00001	<0.000005	0.00001
2,3,3',4,4',5,5'-HeptaCB (189)	0.0001	<0.000005	0.00001

CDD = Chlorinated dibenzodioxins; CDF = Chlorinated denzofurans; CB = Chlorinated biphenyls.

Appendix A2-10 An Overview of the Environmental Fate and Effects of Polychlorinated Dibenzo-p-Dioxins and Polychlorinated Dibenzofurans

10.1 Identity

Polychlorinated dibenzo-*p*-dioxins (PCDDs) and polychlorinated dibenzofurans (PCDFs) represent two groups of aromatic compounds with very similar properties (WHO 1989). The term PCDDs generally refers to a group of 75 congeners that consist of two benzene rings connected to each other by two oxygen bridges. The term PCDFs refers to a group of 135 aromatic compounds that are similar to PCDDs in their chemical behaviour and toxicity. Structurally, these substances are not unlike PCDDs in that they are comprised of two benzene rings, however, PCDFs contain only one oxygen atom. As few as one or as many as eight chlorine atoms may be attached to the benzene rings in PCDDs and PCDFs. It is both the number of chlorine atoms and their position in the molecule that determines the physical and chemical properties and the toxicity of each congener (Boddington *et al*. 1990).

10.2 Uses

PCDDs and PCDFs are not produced intentionally, and there are no known uses of these compounds (WHO 1989; Fiedler *et al*. 1990).

10.3 Sources

PCDDs and PCDFs are undesirable contaminants that are formed largely as a result of various anthropogenic activities, including chemical manufacturing, waste incineration, pulp and paper production, and petroleum refining. Trace quantities of PCDDs and PCDFs occur in many formulated chemical products, mainly as a result of the high-temperatures and chlorinated solvents that are used in manufacturing processes (Kuehl *et al.* 1987a; 1987b). Substances known to be contaminated with PCDDs and/or PCDFs include various pesticides (e.g., 2,4-D and 2,4,5-T), wood preservatives (e.g., pentachlorophenol), 1,2,4-trichlorobenzene, hexachlorobenzene, tetrachlorobenzoquinones, askarels (i.e., the PCB (polychlorinated biphenyl) mixtures used in electric transformers), perchloroethylene, and recycled oils (Fiedler *et al.* 1990). Natural sources of PCDDs and PCDFs include forest fires and volcanoes.

10.4 Physical and Chemical Properties

The number of chlorine atoms that are substituted on the benzene rings appears to be the one of the most important factors influencing the physical and chemical properties of PCDDs and PCDFs. For example, aqueous solubilities decrease with increasing chlorine substitution, ranging from 0.07 - 0.97 ng•L^{-1} for O_8CDD to 7.91 - 483 ng•L^{-1} for T_4CDD (Shiu *et al.* 1988; Friesen *et al.* 1985; Adams and Blaine 1986; Lodge 1989). Likewise, vapour pressures decrease with increasing chlorine substitution (Rordorf *et al.* 1990). Octanol/water partition coefficients (log K_{ow}) provide an estimate of the hydrophobicity of a substance and, hence, are good indicators of the potential for uptake by aquatic organisms. The available data on PCDDs suggest that these compounds tend to be highly lipophilic, with log K_{ow} ranging from 5.50 - 8.93 for T_4CDD (Fiedler *et al.* 1990) to 7.53 - 12.26 for O_8CDD (Webster *et al.* 1986; Doucette 1985). Increasing hydrophobicity with increasing chlorine substitution was also reported for PCDFs (Broman *et al.* 1991).

10.5 Environmental Fate and Transport

PCDDs and PCDFs are released into the environment from a variety of sources, which can be classified as chemical sources, combustion sources, natural sources, and industrial sources. Upon release, these substances are distributed into the various environmental compartments in accordance with the nature of the source (i.e., to air, water, or soils), their physico-chemical properties and local environmental conditions. When released into the atmosphere, these substances may be rapidly degraded by photolysis or through reactions with hydroxide (OH^-) radicals, principally to the lower chlorinated congeners. In many situations, however, these substances are transported significant distances by the wind and subsequently deposited in terrestrial and/or aquatic ecosystems.

PCDDs and PCDFs that are released into aquatic systems tend to be more persistent that those that are released into the atmosphere. While photolysis and volatilization may result in some degradation of these compounds (particularly in shallow, warm water systems), biodegradation is considered to be a relatively minor fate process in water (NRCC 1981; Howard 1991). The majority of the PCDDs and PCDFs that are released into water form associations with dissolved and/or particulate organic matter in the water column. Within days, these substances become associated with suspended and bed sediments (MacDonald 1993).

Aquatic sediments provide a major sink for the PCDDs and PCDFs that enter the water column. The results of various studies indicate that most of the PCDDs and PCDFs that are added to model aquatic ecosystems (i.e., mesocosms) partition almost entirely into the sediment phase (Corbet *et al.* 1988; Tsushimoto *et al.* 1982; Muir *et al.* 1985). As PCDDs and PCDFs tend to be very stable in sediments, they tend to persist for extended periods in this environmental matrix. Importantly, PCDDs and

PCDFs that are associated with bed sediments may represent long term sources to the aquatic food web (Kuehl *et al.* 1987c; Muir 1988).

10.6 Bioaccumulation

The bioconcentration of T_4CDD in aquatic biota has been assessed in a variety of laboratory studies using fish, invertebrates, and aquatic plants. Bioconcentration is generally evaluated by calculating the ratio of the concentration of a chemical in an organism's tissues (expressed on a wet weight basis) to its concentration in water, with this ratio termed the bioconcentration factor (BCF). Bioconcentration factors of up to 900 000 have been predicted from the physical/chemical properties (K_{ow}) of this substance (USEPA 1984). By comparison, BCFs ranging from 490 to 159 000 have been determined in laboratory studies on fish, depending on the species tested, the duration of exposure, levels of dissolved organic carbon, and several other factors (Isensee 1978; Rabert 1990; Cook *et al.* 1990a; 1990b). However, most of these measurements were made under conditions that did not approach equilibrium or steady state (Cook *et al.* 1991).

While direct uptake from the water column may be important for certain ecosystem components (e.g., aquatic plants; zooplankton), sediments probably represent the most significant long-term source of PCDDs and PCDFs that are transferred into the food web (Carey *et al.* 1990). For this reason, sediment to biota bioaccumulation factors (BSAFs) provide valuable information for assessing the potential effects of sediment-associated PCDDs and PCDFs to wildlife consumers of aquatic organisms. While few data are available to derive bioaccumulation factors in fish and invertebrates, the results of a study conducted in Lake Ontario provides some very relevant information. In this study, Carey *et al.* (1990) reported lakewide BSAFs ranging from 0.04 to 1.13 for five species of fish (median = 0.23), including brown trout, lake trout, smallmouth

bass, white perch, and yellow perch. These investigators suggested that much of the observed variability in the estimated of the BSAFs may result from differences in tissue lipid levels and/or sediment total organic carbon concentrations. For this reason, lipid- and TOC (total organic carbon)-normalized BAF (or bioavailability indices - BI) were calculated using the data from Lake Ontario. These BI ranged from 0.03 to 0.24 on a lakewide basis (median = 0.06) for the five species of fish considered. Normalization of the data to lipid and TOC levels significantly reduced the apparent variability in the calculated bioaccumulation factors for T_4CDD, confirming the influence of tissue lipid and sediment TOC levels on the uptake and retention of this contaminant.

10.7 Effects on Aquatic and Terrestrial Organisms

Toxicological evidence indicates that all of the PCDDs and PCDFs congeners have the same mode of toxicity as T_4CDD; however their actual toxicity may differ by several orders of magnitude (O'Brien 1990). While differences in the toxicity of many PCDDs and PCDFs congeners are well established, the available toxicological information does not provide an adequate basis for precisely establishing the relative toxicity of each substance. As such, it is difficult to accurately evaluate the environmental hazards posed by complex mixtures of PCDDs and PCDFs (Eadon *et al.* 1986). For this reason, an internationally accepted system for comparing the toxicity of 17 of the most important PCDDs and PCDFs congeners has been established to facilitate assessments of the cumulative effects of these compounds assuming that the effects of these substances are additive (Boddington *et al.* 1990).

The International Toxicity Equivalency Factor (ITEF) method provides a basis for evaluating the toxicity of complex mixtures of PCDDs, PCDFs, and several related compounds (e.g., coplanar PCB; Safe *et al.* 1988). To date, ITEF have been

developed for the 17 PCDDs and PCDFs congeners that contribute most to the toxicity of complex mixtures of PCDDs and PCDFs (i.e., those that are substituted with chlorine in the 2,3,7, and 8 positions; Boddington *et al.* 1990). These ITEF have been assigned to individual PCDDs and PCDFs on the basis of their toxicity relative to the most toxic PCDDs congener, T_4CDD, where the ITEF for this substance is equal to 1.0 (van den Berg 1998). The ITEF method of risk assessment has been officially adopted by the scientific and regulatory communities in eight countries, including Canada, Denmark, the Federal Republic of Germany, Italy, the Netherlands, Norway, the United Kingdom and the United States (Safe *et al.* 1988). The ITEF provide a means of expressing concentrations of PCDDs and PCDFs on a common basis (i.e., as T_4CDD toxic equivalents or TEQs) and, therefore, provide a basis for evaluating the significance of mixtures of PCDDs and PCDFs.

The ITEF method is based on the fact that PCDDs, PCDFs, and several other groups of compounds (including polychlorinated biphenyls, polybrominated biphenyls, and polychlorinated naphthalenes) have very similar modes of toxic action. For each of these substances, the mechanism of toxic action is linked to their binding to the aryl hydrocarbon (Ah) receptor in the cytoplasm of a cell (Safe 1990). Once a PCDDs or PCDFs congener attaches to the Ah receptor, the cytosolic receptor complex migrates into the cell's nucleus, where it binds to specific sites (i.e., the Ah locus) on the genetic material (DNA - deoxyribonucleic acid; Safe 1990). This complex then causes specific regulatory and structural genes to be transcribed, resulting in the production (or induction) of a variety of proteins (O'Brien 1990). However, the exact mechanism that causes toxicity in test organisms has not been definitively established.

PCDDs and PCDFs have been demonstrated to cause a variety of adverse effects in aquatic organisms. The available toxicity data indicate that freshwater fish can be adversely affected by exposure to relatively low levels of water-borne PCDD and PCDFs (i.e., as low as 0.038 ng TEQ/L; Merle *et al.* 1988). In addition to lethality,

documented effects on fish include growth inhibition, impaired reproductive success, fluid build-up in tissues (i.e., edema), and fin necrosis (MacDonald 1993). In general, freshwater invertebrates are less sensitive to the PCDDs and PCDFs than are fish; however, impaired reproduction has been observed in snails following long-term exposure to 200 ng TEQ/L (Miller et al. 1973). Adverse effects on aquatic plants were not observed in response to exposure to up to 1330 ng TEQ/L of PCDDs and/or PCDFs.

The compound T_4CDD and, to a lesser extent, related substances are extremely toxic to birds and mammals. In addition to acute toxicity, exposure to these substances may result in a number of sub-lethal effects. Long-term exposure to T_4CDD has been associated with enzyme induction, growth impairments, tissue damage, immunotoxicity, developmental abnormalities, and cancer (WHO 1989). However, this group of substances does not appear to damage genetic material or chromosomes in laboratory animals (Boddington *et al.* 1990). In laboratory animals, death results from a single exposure to amounts ranging from less than one microgram to a few milligrams T_4CDD TEQs per kilogram of body weight (i.e., <1 ng/kg body weight (BW) to > 1 mg/kg BW; Boddington 1990). The no-observed-effect-level of T_4CDD TEQs for chronic exposure in rats, relative to carcinogenic and reproductive effects, is in the order of 1 ng/kg BW/day (Boddington 1990).

10.8 References

Adams, W.J. and K.M. Blaine. 1986. A water solubility determination of TCDD. Chemosphere 15(9-12):1397-1400.

Boddington, M.J., A.P. Gilman, R.C. Newhook, B.M. Braune, D.J. Hay, and V. Shantora. 1990. Canadian Environmental Protection Act: Priority substances list assessment report No 1. Polychlorinated dibenzodioxins and polychlorinated dibenzofurans. Canadian Environmental Proctection Act. Environment Canada and Health and Welfare Canada. 56 pp.

Broman, D., C. Naef, C. Rolff, and Y. Zebuehr. 1991. Occurrence and dynamics of polychlorinated dibenzo-*p*-dioxins and dibenzofurans and polycyclic aromatic hydrocarbons in the mixed surface layer of remote coastal and offshore waters of the Baltic. Environmental Science and Technology 25(11):1850-64.

Carey, A.E., N.S. Shifrin, and A.P. Briefer. 1990. Derivation of a Lake Ontario bioaccumulation factor for 2,3,7,8-TCDD. *In:* Lake Ontario TCDD Bioaccumulation Study: Final Report. United States Environmental Protection Agency. New York State Department of Environmental Conservation. New York State Department of Health. Occidental Chemical Corporation. Albany, New York.

Cook, P.M., M.K. Walker, D.W. Kuehl, and R.E. Peterson. 1990a. Bioaccumulation and toxicity of 2,3,7,8-tetrachlorodibenzo-*p*-dioxin and related compounds in aquatic ecosystems. Prepared in cooperation with Wisconsin University. PC A03/MF A01. EPA/600/D-91/045. Environmental Research Laboratory-Duluth. Duluth, Minnesota. 38 pp.

Cook, P.M., A.R. Batterman, B.C. Butterworth, K.B. Lodge, and S.W. Kohlbry. 1990b. Laboratory study of TCDD bioaccumulation by lake trout from Lake Ontario sediments, food chain and water. EPA Report for Region 2 Superfund. Lake Ontario, from revised draft of Lake Ontario modeling studies. Chapter 6. (As cited in NCASI 1991).

Cook, P.M., D.W. Kuehl, M.K. Walker, and R.E. Peterson. 1991. Bioaccumulation and toxicity of TCDD and related compounds in aquatic ecosystems. *In:* Banbury Report 35: Biological Basis for Risk Assessment of Dioxins and Related Compounds. Cold Spring Harbor Laboratory Press. Cold Spring Harbor, New York.

Corbet, R.L., G.R.B. Webster, and D.C.G. Muir. 1988. Fate of 1,3,6,8-tetrachlorodibenzo-*p*-dioxin in an outdoor aquatic system. Environmental Toxicology and Chemistry 7:167-180.

Doucette, W.J. 1985. Ph.D. Thesis. University of Wisconsin. Madison, Wisconsin (As cited in Shiu *et al.* 1988).

Eadon, G., L. Kaminsky, J. Silkworth, K. Aldous, D. Hilker, P. O'Keefe, R. Smith, J. Gierthy, J. Hawley, N. Kim, and A. DeCaprio. 1986. Calculation of 2,3,7,8-TCDD equivalent concentrations of complex environmental contaminant mixtures. Environmental Health Perspectives 70:221-227.

Fiedler, H., O. Hutzinger, and C.W. Timms. 1990. Dioxins: sources of environmental load and human exposure. Toxicological and Environmental Chemistry 29:157-234.

Friesen, K.J., L.P. Sarna, and G.R. Webster. 1985. Aqueous solubility of polychlorinated dibenzo-*p*-dioxins determined by high pressure liquid chromatography. Chemosphere. 14:1267-1274.

Howard, P.H. 1991. Handbook of environmental degradation rates. Lewis Publishers Inc. 121 South Main Street. Chelsea, Michigan. pp. 657-658.

Isensee, A.R. 1978. Bioaccumulation of 2,3,7,8 - tetrachlorodibenzo- *p*- dioxin. *In:* Ramel, C. (ed.) Chlorinated Phenoxy Acids and Their Dioxins: Mode of Action, Health Risks and Environmental Effects. Ecological Bulletin 27:255-262. Swedish Natural Science Research Council. Stockholm, Sweden.

Kuehl, D.W., P.M. Cook, A.R. Batterman, and B.C. Butterworth. 1987a. Isomer dependent bioavailability of polychlorinated dibenzo-*p*-dioxins and dibenzofurans. Chemosphere. 16(4):657-666.

Kuehl, D.W., B.C. Butterworth, W.M. DeVita, and C.P. Sauer. 1987b. Environmental contamination by polychlorinated dibenzo-*p*-dioxins and dibenzofurans associated with pulp and paper mill discharge. Biomedical and Environmental Mass Spectrometry. 14:443-447.

Kuehl, D.W., P.M. Cook, A.R. Batterman, D. Lothenback, and B.C. Butterworth. 1987c. Bioavailability of polychlorinated dibenzo-*p*-dioxins and dibenzofurans from contaminated Wisconsin River sediment to carp. Chemosphere. 16(4):667-679.

Lodge, K.B. 1989. Solubility studies using a generator column for 2,3,7,8-tetrachlorodibenzo-*p*-dioxin. Chemosphere. 18(1-6):933.

MacDonald, D.D. 1993. Canadian environmental quality guidelines for polychlorinated dibenzo-para-dioxins and polychlorinated dibenzofurans. Prepared for EcoHealth Branch. Environment Canada. Ottawa, Ontario.

Mehrle, P.M., D.R. Buckler, E.E. Little, L.M. Smith, J.D. Petty, P.H. Peterman, D.L. Stalling, G.M. De Graeve, J.J. Coyle, and W.J. Adams. 1988. Toxicity and bioconcentration of TCDD and TCDF in rainbow trout. Environmental Toxicology and Chemistry 7:47-62.

Miller, R.A., L.A. Norris, and C.L. Hawkes. 1973. Toxicity of TCDD in aquatic organisms. Environmental Health Perspectives pp. 177-186.

Muir, D.C. 1988. Bioaccumulation and effects of chlorinated dibenzodioxins and furans in fish, shellfish and crustacea. A brief review. Internal Report. Prepared for Oceanography and Contaminants Branch. Department of Fisheries and Oceans. Ottawa, Ontario. 8 pp.

Muir, D.C., A.L. Yarechewski, R.L. Corbet, G.R. Webster, and A.E. Smith. 1985. Laboratory and field studies on the fate of 1,3,6,8-tetrachlorodibenzo-*p*-dioxin in soil and sediments. Journal of Agricultural and Food Chemistry 33(3):518-523.

NCASI (National Council of the Paper Industry for Air and Stream Improvement, Inc). 1991. Observations on the bioaccumulation of 2,3,7,8-TCDD and 2,3,7,8-TCDF in channel catfish and largemouth bass and their survival or growth during exposure to biologically treated bleached kraft mill effluent in experimental streams. NCASI Technical Bulletin No. 611. New York, New York. 53 pp + appendices.

NRCC (National Research Council of Canada). 1981. Polychlorinated dibenzo-*p*-dioxins: Criteria for their effects on man and his environment. NRCC Report No. 18574. Ottawa, Ontario. 251 pp.

O'Brien, M. 1990. A crucial matter of cumulative impacts: toxicity equivalency factors. Journal of Pesticide Reform 10(2): 23-27.

Rabert, W.S. 1990. An update on the environmental effects of TCDD and TCDF releases from pulp and paper mills on aquatic and terrestrial animals. 1990. Environmental Effects Branch. Office of Toxic Substances. United States Environmental Protection Agency. Washington, District of Columbia. 648-668.

Rordorf, B.F., L.P. Sarna, G.R. Webster, S.H. Safe, L.M. Safe, D. Lenoir, K.H. Schwind, and O. Hutzinger. 1990. Vapor pressure measurements on halogenated dibenzo-*p*-dioxins and dibenzofurans. An extended data set for a correlation method. Chemosphere. 20(10-12):1603-1607.

Safe, S. 1990. Polychlorinated biphenyls (PCBs), dibenzo-*p*-dioxins (PCDDs), and related compounds: Environmental and mechanistic considerations which support the development of toxic equivalency factors (TEFs). Toxicology 21:51-88.

Safe, S.H., F.W. Katz, and D.P. Bottimore. 1988. Pilot study on international information exchange on dioxins and related compounds. Scientific basis for the development of international toxicity equivalency factor (ITEF) method of risk assessment for complex mixtures of dioxins and related compounds. EPA/600/6-90/015. Committee on the Challenges of Modern Society. 56 pp.

Shiu, W.Y., W. Doucette, F.A. Gobas, A. Andren, and D. MacKay. 1988. Physical-Chemical properties of chlorinated dibenzo-*p*-dioxins. Environmental Science and Technology 22:651-658.

Tsushimoto, G., F. Matsumura, and R. Sago. 1982. Fate of TCDD in an outdoor pond and in model aquatic ecosystems. Environmental Toxicology and Chemistry 1:61-68.

USEPA (United States Environmental Protection Agency). 1984. Ambient water quality criteria for 2,3,7,8-tetrachlorodibenzo-*p*-dioxin. EPA 440/5-807. Office of Water Regulations and Standards. Washington, District of Columbia. 277 pp.

van den Berg, M., L. Birnbaum, A.T. Bosveld, B. Brunström, P. Cook, M. Feeley, J. Giesy, A. Hanberg, R. Hasegawa, S.W. Kennedy, T. Kubiak, J.C. Larsen, F.X. van Leeuwen, A.K. Liem, C. Nolt, R.E. Peterson, L. Poeliinger, S. Safe, D. Schrenk, D. Tillitt, M. Tysiind, M. Younes, F. Waern, and T. Zacharewski. 1998. Toxic equivalency factors (TEFs) for PCBs, PCDDs, and PCDFs for humans and wildlife. Environmental Health Perspectives 106:775-792.

Webster, G.R., D.H. Muldrew, J.J. Graham, L.P. Sarna, and D.C. Muir. 1986. Dissolved organic matter mediated aquatic transport of chlorinated dioxins. Chemosphere. 15(9-12):1379-1386.

WHO (World Health Organization). 1989. Polychlorinated dibenzo-para-dioxins and dibenzofurans. United Nations Environment Programme. Geneva, Switzerland.

Appendix A2-11 An Overview of the Environmental Fate and Effects of Chlorinated Benzenes: Hexachlorobenzene and Hexachlorobutadiene

11.1 Introduction

Chlorinated benzenes belong to a subset of semi-volatile organic compounds that includes chemicals with one to six chlorine substitutions on the benzene parent molecule, such as hexachlorobenzene and hexachlorobutadiene. Much of the information contained in this section is based on Environment Canada's Priority Substances List Assessment Report on Hexachlorobenzene (Government of Canada 1993).

11.2 Hexachlorobenzene

11.2.1 Identity

Hexachlorobenzene (HCB) is a cyclic aromatic hydrocarbon with six chlorine atoms substituting for hydrogen atoms in the benzene ring. At ambient temperature, hexachlorobenzene is a white crystalline solid.

11.2.2 Uses

HCB started to be used on a commercial scale in 1940's as an antifungal agent for use on seeds of wheat, barley, oats, and rye. However, the fungicidal application of HCB was halted in the 1970's due to concerns regarding its impacts on the environment and

human health. Other uses of HCB include production of pyrotechnics, tracer bullets, and aluminum. HCB has also been used as a wood preservative, a porosity control agent in graphite anodes, and as a peptizing agent in the production of synthetic rubber (Mumma and Lawless 1975).

11.2.3 Sources

The main releases of hexachlorobenzene to the environment include incidental emissions associated with the manufacture and use of chlorinated solvents (Quinlivan *et al.* 1975; Jacoff *et al.* 1986), use of HCB-contaminated pesticides (Tobin 1986), waste incineration, and long-range transport. HCB has been detected in emissions from paint manufacturers, coal and steel producers, pulp and paper mills, textile mills, pyrotechnics producers, aluminum smelters, soap producers, and wood-treatment sites (Quinlivan *et al.* 1975). Municipal and industrial waste water facilities also discharge HCB-contaminated effluents (King and Sherbin 1986). Releases from chlor-alkali and sodium chlorate plants that used graphite anodes were historically an important source of HCB (Quinlivan *et al.* 1975; Mumma and Lawless 1975). However, present releases from this source are negligible because new types of anodes are used that do not produce HCB (Brooks and Hunt 1984). Incinerators release HCB as a result of incomplete thermal breakdown of substances such as kepone, mirex, chlorobenzenes, PCBs (polychlorinated biphenyls), pentachlorophenol, polyvinyl chloride, and chlorinated solvents (Ahling *et al.* 1978).

11.2.4 Physical and Chemical Properties

HCB is not very soluble in water (0.005 mg/L at 25/C). However, it is soluble in ether, benzene, chloroform, and ethanol. HCB has a relatively high octanol/water partition coefficient (1og K_{ow} = 5.5), low vapor pressure (0.0023 Pa at 25/C), and low flammability. Its Henry's Law constant is 131 Pa/m^3/mol (USEPA 1985; ATSDR 1990; Mackay *et al.* 1992).

11.2.5 Environmental Fate and Transport

Volatilization and sedimentation are two major processes for removal of HCB from water. Oliver (1984) and Oliver and Charlton (1984) reported that 80% of the HCB entering Lake Ontario was lost through volatilization. The remainder was removed by sedimentation (15%) and outflow to the St. Lawrence River (5%). In the troposphere, HCB is transported long distances. There, it undergoes a slow photolytic degradation ($t_{1/2}$ approximately 80 days; Mill and Haag 1986) or is removed through atmospheric deposition to water and soil (Lane *et al.* 1992).

HCB tends to be trapped in sediments and accumulate over time (Oliver and Nicol 1982). However, disturbed sediments may release some HCB back into the water column. This may represent an important secondary source of HCB to aquatic environments. Such inputs may continue long after direct inputs to the system cease (Oliver *et al.* 1989). Chemical and biological degradation are unlikely to be important removal processes of HCB from water or sediments (Mill and Haag 1986). However, slow aerobic ($t_{1/2}$ = 2.7 to 5.7 years) and anaerobic biodegradation ($t_{1/2}$ = 10.6 to 22.9 years) are important removal processes of HCB from the soil matrix (Howard *et al.* 1991).

11.2.6 Bioaccumulation

Aquatic and terrestrial organisms accumulate HCB from food and water. Benthic organisms, on the other hand, may accumulate HCB directly from sediments (Gobas *et al.* 1989). The relative importance of food versus water uptake is not well understood. However, field studies indicate that exposure via food rather than via water is important for organisms at higher trophic levels. Oliver and Niimi (1988) pointed out that several studies have shown that higher trophic level organisms in natural aquatic ecosystems accumulated HCB to levels greater than those at lower trophic levels.

11.2.7 Effects on Aquatic and Terrestrial Organisms

Exposure of aquatic and terrestrial biota to HCB causes a wide range of effects. These include acute and chronic toxicities, carcinogenicity, reproductive toxicity, and genotoxicity. Aquatic toxicity testing of HCB is difficult due to the limited aqueous solubility and high volatility of HCB. The existing studies with freshwater algal species tested at 1 µg/L revealed adverse effects in only one species. *Chloroella pyrenoiosa* experienced reduced production of chlorophyll, carbohydrate, dry matter, and nitrogen after 46 h static-closed system exposures (Geike and Parasher 1976). Acute exposures of water flea (Nebecker *et al.*1989) and sheepshead minnow (Parrish *et al.* 1974) resulted in no lethality at 5 and 300 µg/L, respectively. Ninety-six hour flow-through tests on pink and grass shrimp resulted in 13% mortality in the pink shrimp group at 7 µg/L and 10% mortality in grass shrimp at 17 µg/L (Parrish *et al.* 1974). McLeese and Metcalfe (1980) tested for acute effects of HCB in sediments to the marine shrimp *Crangon septemspinos*, but observed no mortality at levels as high as 300 µg/L.

Longer exposures of water flea (14 days) to HCB resulted in a 50% reduction in fertility at an exposure level of 16 µg/L (Calamari *et al.* 1983). Largemouth bass experienced liver necrosis from a 10-day exposure at 3.5 µg/L (Laseter *et al.* 1976b). The acute toxicity of HCB to laboratory mammals is relatively low. Reported oral LD50s (median lethal doses) range from more than 1,000 mg/kg for the guinea pig to 10,000 mg/kg for the rat. Inhalation LC_{50}s (median lethal concentrations) range from 1,600 mg/m^3 for the cat to 4,000 mg/m^3 for the mouse (IARC 1979).

The effects of short-term, repeated exposures to HCB are primarily hepatotoxic and neurologic. Responses of orally-dosed rats (30 to 250 mg/kg/day) include altered body weight, cutaneous lesions, tremors, hepatomegaly, liver damage and, in some cases, early alterations in porphyrin or heme metabolism (USEPA 1985). Short-term exposures also induce a variety of Phase I and Phase II enzymes (USEPA 1985). Reported effect levels for this endpoint in rats were as low as 50 mg/kg feed (approximately 2.5 mg/kg body weight; BW/day; den Tonkelaar and van Esch 1974).

The effects produced by subchronic exposure to HCB are similar to those observed in short-term studies, but are generally evident at lower doses (USEPA 1985; ATSDR 1990). The lowest dose producing effects on pigs was 0.5 mg/kg BW/day.

The carcinogenicity of HCB was assessed in several studies on rats, mice, and hamsters. Cabral *et al.* (1977) reported a statistically-significant increase of "liver cell tumors (hepatomas)" in male and female Syrian golden hamsters fed 50 mg/kg (4 mg/kg BW/day) HCB in their diets for life. Results from several studies suggest that HCB is a co-carcinogen or a promoter of cancer (Shirai *et al.* 1978). Dietary exposure of rats to HCB after exposure to iron induced liver tumors (Smith *et al.* 1989) and produced hepatocellular carcinomas (Stewart *et al.* 1989).

HCB has not been shown to be genotoxic (Khera 1974; Kitchin and Brown 1989). However, it can be gonadotoxic. Relatively low doses of HCB (0.1 mg HCB/kg BW/day for 90 days) affected the reproductive tissues in female monkeys (Babineau *et al.* 1991).

Placental and lactational transfer of HCB can adversely affect the foetus and nursing offspring. Maternal doses in the range from 1.4 to 4 mg/kg to rats and cats were hepatotoxic and affected the survival or growth of the nursing offspring. Comparable doses reduced litter sizes and increased the number of stillbirths (Arnold *et al.* 1985). Mink are particularly sensitive to the effects of prenatal and perinatal exposure to HCB. The offspring of mink fed diets containing concentrations as low as 1 mg/kg of HCB (approximately 0.16 mg/kg BW/day) for 47 weeks (prior to mating and throughout gestation and nursing) had reduced birth weights and increased mortality (Bleavins *et al.* 1984).

Data show that HCB is not a developmental toxicant (Khera 1974). However, HCB can affect the immune system. Rats and monkeys exposed to 5 mg HCB/kg BW/day show adverse effects in their thymus, spleen, lymph nodes, and/or lymphoid tissues of the lung (Kimbrough and Linder 1974; Iatropoulos *et al.* 1976; Goldstein *et al.* 1978; Vos *et al.* 1979). Gralla *et al.* (1977) reported that chronic exposure to as little as 1 mg/day of HCB caused nodular hyperplasia of the gastric lymphoid tissue in beagle dogs.

11.3 Hexachlorobutadiene

11.3.1 Identity

Hexachlorobutadiene (HCBD; CAS; Chemical Abstracts Service Number 87-68-3) is a colorless liquid at room temperature. It has a molecular weight of 260.76, a melting point of -21/C, and a boiling point of 215/C (USEPA 1979; Verschueren 1983). Synonyms for HCBD include hexachloro-1,3-butadiene, 1,1,2,3,4,4-hexachloro-1,3-butadiene, perchlorobutadiene, and perchloro-1,3-butadiene.

11.3.2 Uses

HCBD is used primarily as a solvent for many organic substances, especially elastomers. HCBD is very useful in recovering chlorine from waste gases and in the production of rubber compounds, lubricants, and hydraulic fluids. Its also used as a heat transfer fluid in electrical transformers (USEPA 1980).

11.3.3 Sources

HCBD is typically a by-product of tetrachloroethylene, trichloroethylene, and carbon tetrachloride production streams. Waste holding areas that contain HCBD are the most significant emission sources through volatilization into air and solubilization into runoff water. Wastewater from industrial processes may also be a significant source of HCBD to the environment (Li *et al.* 1976). In addition to waste streams, releases can also come from HCBD use as a solvent for elastomers, heat transfer liquid, transformer and hydraulic fluid, and as a wash liquor for removing C4 and higher hydrocarbons (Hawley 1981). According to the United States Toxic Release Inventory (TRI), 75% of total releases went into air, 15% to water, and 10% to underground injection (TRI 1997). Note that recent changes in manufacturing processes and improvements in waste treatment facilities have reduced HCBD emissions (Environment Canada 2001).

11.3.4 Physical and Chemical Properties

HCBD is rather soluble. Its water solubility is 2 mg/L at 20/C. HCBD is not very volatile with a vapor pressure of 20 Pa at 20/C (Pearson and McConnell 1975) and a Henry's law constant of 1,044 Pa×m^3/mol (Shen 1982). Hexachlorobutadiene has a log octanol/water partition coefficient of 4.9 (Chiou 1985) suggesting that it tends to partition into biota and sediments.

11.3.5 Environmental Fate and Transport

Sorption to sediments is an important removal mechanism of HCBD from the water column (USEPA 1979). Photolysis, oxidation, and hydrolysis play only a minor role (USEPA 1979). Unlike other short-chain, halogenated aliphatic compounds, hexachlorobutadiene has a relatively low vapor pressure suggesting a low potential for volatilization (USEPA 1979). No information was found on the biodegradation of hexachlorobutadiene in the aquatic environment (USEPA 1979).

HCBD can exhibit atmospheric long-range transport or transboundary movement. Evidence for long-range transport of HCBD was provided by Mudroch *et al.* (1992), who reported HCBD at concentrations ranging from 0.01 to 0.23 µg/kg at various sediment depths in samples taken from Great Slave Lake.

Some of the released HCBD may be available for direct uptake from the aquatic environment. However, most HCBD will partition to sediments given its affinity for sediment adsorption. Biological degradation of HCBD in sediments under anaerobic conditions is slow (Howard 1991). As a result, HCBD is expected to persist in anaerobic soils and sediments. The main route of exposure of receptors of interest to HCBD is through direct contact with, and ingestion of, soils and sediments, as well as trophic transfer of the contaminant through consumption of benthic and soil organisms.

11.3.6 Bioaccumulation

Bioconcentration factors in algae and animals are generally below 300 for short-term exposures (USEPA 1979). For example, bioconcentration factors of 160 for algae (*Oedogonium cardiacum*; 7-day exposure), 60 for crayfish (*Procambarus clarki*; 10-day exposure), and 29 for largemouth bass (*Micropterus salmoides*; 10-day exposure) were reported by Leeuwangh *et al.* (1975). Bioconcentration factors for goldfish (*Carassius auratus*) ranged from 920 to 2,300 in longer exposures (49 days; USEPA 1976). Exposure of rainbow trout (*Salmo gairdneri*) to 1 ng HCBD/L resulted in a mean bioconcentration factor of 5,800 (Oliver and Niimi 1983). Biomagnification in aquatic food chains has not been demonstrated (Environment Canada 1984).

11.3.7 Effects on Aquatic and Terrestrial Organisms

In aquatic ecosystems, HCBD accumulates in the livers of fish (Pearson and McConnell 1975) where it is transformed into polar metabolites, which adversely affect the kidneys (Anders and Jakobson 1985; Yang 1988; IPCS 1994).

In general, freshwater fish and marine Crustacea are more sensitive than their marine and freshwater counterparts. The lowest reported chronic toxicity value was a 28-day Lowest-Observed-Effect Concentration (LOEC; survival and growth) of 13 mg/L reported for the fathead minnow (*Pimephales promelas*; Benoit *et al.* 1982). No chronic data on toxicity were identified for aquatic invertebrates. The lowest reported acute toxicity value was a 96-hour LC_{50} of 32 mg/L for a marine mysid shrimp (*Mysidopsis bahia*; USEPA 1980). For fish, the lowest acute value was a 96-hour LC_{50} of 90 mg/L for the goldfish (Leeuwangh *et al.* 1975). Many other studies reported acute toxicities well above 100 mg/L (Pearson and McConnell 1975; Laseter *et al.* 1976a; Dow Chemical Co. 1978; Juhnke and Lüdemann 1978; Laska *et al.* 1978; Slooff 1979; USEPA 1980; Walbridge *et al.* 1983; Geiger *et al.* 1985; Mayer and Ellersieck 1986). The most sensitive freshwater invertebrate identified was the aquatic sowbug, *Asellus aquaticus*, with a 96-hour LC_{50} of 130 mg/L (Leeuwangh *et al.* 1975). Toxicity studies of HCBD to benthic organisms could not be found.

Short-term and subchronic studies in rats and mice suggest that the renal proximal tubules appear to be the principal site of injury following oral or inhalation exposures. Increased relative kidney weight and histopathological changes, including degeneration of the proximal tubular epithelial cells, necrosis and regeneration, and alterations in biochemical parameters in the blood and urine (renal damage) were reported in short-term studies on Wistar and Sprague-Dawley rats exposed to HCBD in the diet for 2 to 4 weeks at doses as low as 2.5 mg/kg BW/day (Kociba *et al.* 1971; Harleman and Seinen 1979; Stott *et al.* 1981; Jonker *et al.* 1993). A subchronic study with rats given doses of 6.3 and 15.6 mg HCBD/kg BW/day for 13 weeks revealed a significant increase in relative kidney weight. Histopathological changes in the kidney, consisting of large, prominent hyperchromatic nuclei and focal necrosis of epithelial cells and nuclear detritus, were observed in the renal proximal tubules in females at 2.5 mg/kg BW/day.

Short-term and subchronic exposures (15 days) of rats to HCBD by inhalation (25 ppm or 267 mg/m^3) resulted in renal proximal tubular degeneration and adrenal cortical degeneration in male or female Alderley Park SPF (specific pathogen free) rats (Gage 1970).

In a long-term study, groups of male and female Sprague-Dawley rats were exposed to doses as high as 20 mg HCBD/kg BW/day in the diet for two years. The occurrence of renal tumors (adenomas, adenocarcinomas and carcinomas, combined) was significantly increased in rats of both sexes (Kociba *et al.* 1977). However, no adverse effects on testes, ovaries, estrous cycle, or sperm parameters were observed (Kociba *et al.* 1977; NTP 1991).

11.4 References

Ahling, B., A. Bjorseth, and G. Lunde. 1978. Formation of chlorinated hydrocarbons during combustion of poly(vinyl chloride). Chemosphere 10:799-806.

Anders, M.W. and I. Jakobson. 1985. Biotransformation of halogenated solvents. International Conference on Organic Solvent Toxicity, Stockholm, Sweden, October 15–17, 1984. Scandanavian Journal of Work and Environmental Health 11(Suppl. 1):23–32.

Arnold, D.L., C.A. Moodie, S.M. Charbonneau, H.C. Grice, P.F. McGuire, F.R. Bryce, B.T. Collins, Z.Z. Zwadizka, D.R. Krewski, E.A. Nera, and I.C. Munro. 1985. Long-term toxicity of hexachlorobenzene in the rat and the effect of dietary vitamin A. Food and Chemical Toxicology 23:779-793.

ATSDR (Agency for Toxic Substances and Disease Registry). 1990. Toxicological profile for hexachlorobenzene. Pub. TP-90-17. Public Health Service. United States Department of Health and Human Services. Atlanta, Georgia.

Babineau, K.A., A. Singh, J.F. Jarrell, and D.C. Villeneuve. 1991. Surface epithelium of the ovary following oral administration of hexachlorobenzene to the monkey. Journal of Submicroscopic Cytology and Pathology 23:457-464.

Benoit, D.A., F.A. Puglisi, and D.L. Olson. 1982. A fathead minnow *(Pimephales promela*s) early life stage toxicity test method evaluation and exposure to four organic chemicals. Environmental Pollution, Series A 28:189–198.

Bleavins, M.R., R.J. Aulerich, and R.K. Ringer. 1984. Effects of chronic dietary hexachlorobenzene exposure on the reproductive performance and survivability of mink and European ferrets. Archives of Environmental Contamination and Toxicology 13:357-365.

Brooks, G.W. and G.E. Hunt. 1984. Source Assessment for Hexachlorobenzene, Final Report, Prepared for U.S. Environmental Protection Agency, Pollutant Assessment Branch, Research Triangle Park, North Carolina.

Cabral, J.R.P., P. Shubik, T. Mollner, and F. Raitano. 1977. Carcinogenic activity of hexachlorobenzene in hamsters. Nature 269:510-511.

Calamari, D., S. Galassi, F. Setti, and M. Vighi. 1983. Toxicity of selected chlorobenzenes to aquatic organisms. Chemosphere 12:253-262.

Chiou, C.T. 1985. Partition coefficients of organic compounds in lipid–water systems and correlations with fish bioconcentration factors. Environmental Science and Technology 19(1):57–62.

den Tonkelaar, E.M. and G.J. van Esch. 1974. No-effect levels of organochlorine pesticides based on induction of microsomal liver enzymes in short-term toxicity experiments. Toxicology 2:371-380.

Dow Chemical Co. 1978. The acute fish toxicity of hexachlorobutadiene and hexachloroethane to the sheepshead minnow *(Cyprinodon variegatu*s). The Dow Chemical Company. Midland, Michigan (TWC 103).

Environment Canada. 1984. 1980-1981 Cornwall Industrial Survey. Draft. Pollution Control Division. Environmental Protection Service. Ontario Region. Toronto, Ontario.

Environment Canada. 2001. Hexabutadiene Assessment Report: Draft for Public Comments. Environmental Protection Service. Ottawa.

Gage, J.C. 1970. The subacute inhalation toxicity of 109 individual chemicals. British Journal of Industrial Medicine 27:1–8.

Geiger D.L., Poirier S.H., Brooke L.T., and Call D.J., (Eds). 1985. Acute Toxicities of Organic Chemicals to Fathead Minnows (*Pimephales promelas*). Vol. II. University of Wisconsin-Superior. Superior, Wisconsin.

Geike, F. and C.D. Parasher. 1976. Effect of hexachlorobenzene on some growth parameters of *Chlorella pyrenoidosa*. Bulletin of Environmental Contamination and Toxicology 15:670-677.

Gobas, F.A., D.C. Bedard, J.J.H. Ciborowski, and G.D. Haffner. 1989. Bioaccumulation of chlorinated hydrocarbons by mayfly (*Hexagenia limbata*) in Lake St. Clair. Journal of Great Lakes Research 15:581-588.

Goldstein, J.A., M. Friesen, T.M. Scotti, P. Hickman, J.R. Hass, and H. Bergman. 1978. Assessment of the contribution of chlorinated dibenzo-p-dioxins and dibenzofurans to hexachlorobenzene-induced toxicity, porphyria, changes in mixed function oxygenases, and histopathological changes. Toxicology and Applied Pharmacology 46:633-649.

Government of Canada. 1993. Hexachlorobenzene. Canadian Environmental Protection Act Priority Substances List Assessment Report. Environment Canada. Health Canada. Ottawa, Ontario.

Gralla, E.J., R.W. Fleischman, Y.K. Luthra, M. Hagopian, J.R. Baker, H. Esber, and W. Marcus. 1977. Toxic effects of hexachlorobenzene after daily administration to beagle dogs for one year. Toxicology and Applied Pharmacology 40:227-239.

Harleman, J.H. and W. Seinen. 1979. Short-term toxicity and reproduction studies in rats with hexachloro-(1,3)-butadiene. Toxicology and Applied Pharmacology 47:1–14.

Hawley G.G. 1981. The Condensed Chemical Dictionary 10th Edition. Van Nostrand Reinhold Co. New York, New York.

Howard, P. 1991. Handbook of environmental fate and exposure data for organic chemicals. Lewis Publishers. London, United Kingdom. pp. 360–369.

Howard, P.H., R.S. Boethling, W.F. Jarvis, W.M. Meylan, E.M. Michalenko. 1991. Handbook of environmental degradation rates. H. Taup. (Ed.) Lewis Publishers. Chelsea, Michigan.

IARC (International Agency for Research on Cancer). 1979. Hexachlorobenzene. IARC Monograph 20:155-178.

Iatropoulos, M.J., W. Hobson, V. Knauf, and H.P Adams. 1976. Morphological effects of hexachlorobenzene toxicity in female rhesus monkeys. Toxicology and Applied Pharmacology 37:433-444.

IPCS (International Programme on Chemical Safety). 1994. Environmental Health Criteria. Hexachlorobutadiene (No. 156). World Health Organization. Geneva.

Jacoff, F.S., R. Scarberry, and D. Rosa. 1986. Source assessment of hexachlorobenzene from the organic chemical manufacturing industry. *In:* Morris, C.R. and J.R.P. Cabral (Eds.) Hexachlorobenzene: Proceedings of an International Symposium. International Agency for Research on Cancer. IARC Scientific Publications No. 77. Lyon, France. pp. 31-37.

Jonker, D., R.A. Woutersen, P.J. van Bladeren, H.P. Til, and V.J. Feron. 1993. Subacute (4-wk) toxicity of a combination of four nephrotoxicants in rats: compared with the toxicity of the individual compounds. Food Chemistry and Toxicology 31:125–136.

Juhnke, I. and D. Lüdemann. 1978. Results of the study of 200 chemical compounds on acute fish toxicity using the golden orfe test. Zeitschrift Fur Wasser Und Abwasser Forschung 11(5):161–164.

Khera, K.S. 1974. Teratogenicity and dominant lethal studies on hexachlorobenzene in rats. Food and Cosmetics Toxicology 12:471-477.

Kimbrough, R.D., and R.E. Linder. 1974. The toxicity of technical hexachlorobenzene in the Sherman rat strain. A preliminary study. Research Communications in Molecular Pathology and Pharmacology 8:653-664.

King, L. and G. Sherbin. 1986. Point sources of toxic organics to the upper St. Clair River. Water Pollution Research Journal of Canada 21:433-446.

Kitchin, K.T. and J.L. Brown. 1989. Biochemical studies of promoters of carcinogenesis in rat livers. Teratogenesis Carcinogenesis and Mutagenesis 9:273-285.

Kociba, R.J., D.G. Keyes, G.C. Jersey, J.J. Ballard, D.A. Dittenber, J.F. Quast, C.E. Wade, C.G. Humiston, and B.A. Schwetz. 1977. Results of a two year chronic toxicity study with hexachlorobutadiene in rats. American Industrial Hygeine Association Journal 38:589–602.

Kociba, R.J., P.G. Gehring, C.G. Humniston, and G.L. Sparschu. 1971. Toxicologic study of female rats administered hexachlorobutadiene or hexachlorobenzene for thirty days. The Dow Chemical Company. Midland, Michigan [cited in IPCS, 1994].

Lane, D.A., W.H. Schroeder, and N.D. Johnson. 1992. On the spatial and temporal variations in atmospheric concentrations of hexachlorobenzene and hexachlorocyclohexane isomers at several locations in the province of Ontario, Canada. Atmospheric Environment 26A:31-42.

Laseter, J.L., C.K. Bartell, A.L. Laska, D.G. Holmquist, and D.B. Condie. 1976a. Ecological study of hexachlorobutadiene (HCBD). United States Environmental Protection Agency. Washington, District of Columbia. (EPA-68-01-2689).

Laseter, J.L., C.K. Bartell, A.L. Laska, D.G. Holmquist, D.B. Condie, J.W. Brown, and R.L. Evans. 1976b. An ecological study of hexachlorobenzene (HCB). Department of Biological Sciences, University of New Orleans, prepared for Office of Toxic Substances. United States Environmental Protection Agency. Washington, District of Columbia. (EPA560/6-76-009).

Laska, A.L., C.K. Bartell, D.B. Condie, J.W. Brown, R.L. Evans, and J.L. Laseter. 1978. Acute and chronic effects of hexachlorobenzene and hexachlorobutadiene in red swamp crayfish *(Procambarus clark*i) and selected fish species. Toxicology and Applied Pharmacology 43(1):1–12.

Leeuwangh, P., H. Bult, and L. Schneiders. 1975. Toxicity of hexachlorobutadiene in aquatic organisms. *In:* Sublethal Effects of Toxic Chemicals on Aquatic Animals. Proceedings of the Swedish-Netherlands Symposium. J.H. Koeman and J.J. Strik (Eds.). Wageningen, The Netherlands. (As cited in Environment Canada 1983.)

Li, R.T., J.E. Going, and J.L. Spigarelli. 1976. Sampling and analysis of selected toxic substances. Task IB. Hexachlorobutadiene. EPA-560/6-76-015. United States Environmental Protection Agency. Washington, District of Columbia.

Mackay, D., W.Y. Shiu, and K.C. Ma. 1992. Illustrated handbook of physical-chemical properties and environmental fate for organic chemicals. Volume I. Monoaromatic hydrocarbons, chlorobenzenes, and PCBs. Lewis Publishers. Chelsea, Michigan.

Mayer, F.L. and M.R. Ellersieck. 1986. Manual of acute toxicity: Interpretation and data base for 410 chemicals and 66 species of freshwater animals. U.S. Fish Wildlife Service Resource Publication 160:1–226.

McLeese, D.W. and C.D. Metcalfe. 1980. Toxicities of eight organochlorine compounds in sediment and seawater to *Cragnon septamspinosa*. Bulletin of Environmental Contamination and Toxicology 25:921-928.

Mill, T. and W. Haag. 1986. The environmental fate of hexachlorobenzene. *In:* Morris, C.R. and J.R.P. Cabral (Eds.) Hexachlorobenzene: Proceedings of an International Symposium. International Agency for Research on Cancer. IARC Scientific Publications No. 77. Lyon, France. pp. 61-66.

Mudroch, A., R.J. Allan, and S.R. Joshi. 1992. Geochemistry and organic contaminants in the sediments of Great Slave Lake, Northwest Territories, Canada. Arctic 45(1):10–19.

Mumma, C.F. and E.W. Lawless. 1975. Survey of industrial processing data. Task I - Hexachlorobenzene and hexachlorobutadiene pollution from chlorocarbon processes. EPA 560/3-75-003. Office of Toxic Substances. United States Environmental Protection Agency. Washington, District of Columbia.

Nebecker, A.V., W.L. Griffis, C.M. Wise, E. Hopkins, and J.A. Barbitta. 1989. Survival, reproduction and bioconcentration in invertebrates and fish exposed to hexachlorobenzene. Environmental Toxicology and Chemistry 8:601-611.

NTP (National Toxicology Program). 1991. Toxicity studies of hexachloro-1,3-butadiene in $B6C3_{F1}$ mice (feed studies). Research Triangle Park, North Carolina (NTP TOX 1; NIH Publication No. 91-3120).

Oliver, B.G. 1984. Distribution and pathways of some chlorinated benzenes in the Niagra River and Lake Ontario. Water Pollution Research Journal of Canada 19:47-59.

Oliver, B.G. and A.J. Niimi. 1983. Bioconcentration of chlorobenzenes from water by rainbow trout: correlations with partition coefficients and environmental residues. Environmental Science and Technology 17:287-291.

Oliver, B.G. and A.J. Niimi. 1988. Trophodynamic analysis of polychlorinated biphenyl congeners and other chlorinated hydrocarbons in the Lake Ontario ecosystem. Environmental Science and Technology 22:388-397.

Oliver, B.G. and K.D. Nicol. 1982. Chlorobenzenes in sediments, water, and selected fish from Lakes Superior, Huron, Erie, and Ontario. Environmental Science and Technology 16:532-536.

Oliver, B.G. and M.N. Charlton. 1984. Chlorinated organic contaminants on settling particulates in the Niagara River vicinity of Lake Ontario. Environmental Science and Technology 18:903-908.

Oliver, B.G., M.N. Charlton, and R.W. Durham. 1989. Distribution, redistribution, and geochronology of polychlorinated biphenyl congeners and other chlorinated hydrocarbons in Lake Ontario sediments. Environmental Science and Technology 23:200-208.

Parrish, P.R., G.H. Cook, and J.M. Patrick Jr. 1974. Effects on several estuarine animals. Proceedings of the 28th Annual Conference, White Sulphur Springs, West Virginia, 17-20 November, 1974. W.A. Rogers, ed. Southeastern Association of Game and Fish Commissioners, Sulphur Springs, West Virginia.

Pearson, C.R. and G. McConnell. 1975. Chlorinated C_1, and C_2 hydro- carbons in the marine environment. Proceedings of the Royal Society of London, Proceedings B, Biological Sciences 189:305-332.

Quinlivan, S., M. Ghassemi, and M. Santy. 1975. Survey of methods used to control wastes containing hexachlorobenzene. EPA 530/SW-120c. Office of Solid Waste Management Programs. United States Environmental Protection Agency. Washington, District of Columbia.

Shen, T.T. 1982. Air quality assessment for land disposal of industrial wastes. Environmental Management 6:297–305.

Shirai, T., Y. Miyata, K. Nakanishi, G. Murasaki, and N. Ito. 1978. Hepatocarcinogenicity of polychlorinated terphenyl (PCT) in ICR mice and its enhancement by hexachlorobenzene. Cancer Letters 4:271-275.

Slooff, W. 1979. Detection limits of a biological monitoring system based on fish respiration. Bulletin of Environmental Contamination and Toxicology 23(4–5):517–523.

Smith, A.G., J.R.P. Cabral, P. Carthew, J.E. Francis, and M.M. Manson. 1989. Carcinogenicity of iron in conjunction with a chlorinated environmental chemical, hexachlorobenzene, in C57BL/10ScSn mice. International Journal of Cancer 43:492-496.

Stewart, F.P., M.M. Manson, J.R.P. Cabral, and A.G. Smith. 1989. Hexachlorobenzene as a promoter of diethylnitrosamine-initiated hepatocarcinogenesis in rats and comparison with induction of porphyria. Carcinogenesis 10(7):1225-1230.

Stott, W.T., J.F. Quast, and P.G. Watanabe. 1981. Differentiation of the mechanism of oncogenicity of 1,4-dioxane and 1,3-hexachlorobutadiene in the rat. Toxicology and Applied Pharmacology 60:287–300.

Tobin, P. 1986. Known and potential sources of hexachlorobenzene. *In:* Morris, C.R. and J.R.P. Cabral (Eds.) Hexachlorobenzene: Proceedings of an International Symposium. International Agency for Research on Cancer. IARC Scientific Publications No. 77. Lyon, France. pp. 3-11.

TRI (Toxic Release Inventory). 1997. Toxic Chemical Release Inventory. National Toxicology Information Program, National Library of Medicine. Bethesda, Maryland.

USEPA (United States Environmental Protection Agency). 1976. An Ecological Study of Hexachlorobutadiene. EPA 580/6-76-010. Office of Toxic Substances. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1979. Hexachlorobutadiene.in water-related environmental fate of 129 priority pollutants. Volume II. Halogenated Aliphatic Hydrocarbons, Halogenated Ethers, Monocyclic Aromatics, Phthalate Esters, Polycyclic Aromatic Hydrocarbons, Nitrosomonas and Miscellaneous Compounds. EPA-440/4-79-029b. Office of Water Planning and Standards. Washington, District of Columbia. pp. 56-1 to 56-7.

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for hexachlorobutadiene. EPA 440/5-80-053. Office of Water Regulations and Standards, Criteria and Standards Division. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1985. Health Assessment Document for Chlorinated Benzenes. EPA/600/8-84/015F. Office of Health and Environmental Assessment. Washington, District of Columbia.

Verschueren, K. 1983. Handbook of environmental data on organic chemicals. 2nd edition. Van Nostrand Reinhold Co. New York, New York. 1310 pp.

Vos, J.G., M.J. van Logten, J.G. Kreeftenberg, and W. Kruizinga. 1979. Hexachlorobenzene-induced stimulation of the humoral immune response in rats. Annals of the New York Academy of Sciences 320:535-550.

Walbridge, C.T., J.T. Fiandt, G.L. Phipps, and G.W. Holcombe. 1983. Acute toxicity of ten chlorinated aliphatic hydrocarbons to the fathead minnow *(Pimephales promela*s). Archives of Environmental Contamination and Toxicology 12(6):661–666.

Yang, R.S.H. 1988. Hexachloro-1,3-butadiene: toxicology, metabolism, and mechanisms of toxicity. Reviews in Environmental Contamination and Toxicology 101:121-137.

Appendix A2-12 An Overview of the Environmental Fate and Effects of Bis(2-ethylhexyl) phthalate

12.1 Identity

Bis(2-ethylhexyl)phthalate (BEHP) belongs to the group of chemicals called semivolatile organic compounds (SVOCs). This group of chemical compounds includes chemicals that are moderately volatile and may be present as liquids or solids. Bis(2-ethylhexyl)phthalate, a phthalic acid ester, has the CAS; Chemical Abstracts Service Registry Number 117-81-7 and a molecular weight of 390.6. Synonyms include DEHP (di(2-ethylhexyl)phthalate), 1,2-benzenedicarboxylic acid, bis(2-ethylhexyl) ester, phthalic acid, bis(2-ethylhexyl) ester, and di(2-ethylhexyl)phthalate. Much of the information contained in this section is based on Environment Canada's Priority Substances List Assessment Report on BEHP (Government of Canada 1994).

12.2 Uses

BEHP is used as a plasticizer in PVC films, sheets, flooring, and other vinyl products (CIS 1992). The quantity of BEHP used in PVC resins depends on the type of a product (CIS 1992). BEHP is also used widely in insect repellant formulations, cosmetics, rubbing alcohol, liquid soap, detergents, decorative inks, lacquers, munitions, industrial and lubricating oils, and as defoaming agents during paper and paperboard manufacturing (NRCC 1980).

12.3 Sources

The release of BEHP into the atmosphere is the most important route of entry to the environment. The sources of such releases include emissions associated with the

production and use of BEHP as well as the incomplete combustion of plastic materials (IPCS 1992). Peakall (1975) estimated that articles containing phthalate-plasticized materials may lose about 1 % per year of their phthalate content when in contact with liquids and 0.1 % per year when in contact with air. Phthalates may be leached from hazardous waste landfills (Ghassemi *et al.* 1984). Spills are also potential sources of BEHP contamination.

12.4 Physical and Chemical Properties

BEHP is a colorless, oily liquid (Montgomery and Welkom 1990) with vapor pressures ranging between 8.3 x 10^{-6} Pa (Montgomery and Welkom 1990) and 8.6 x 10^{-4} Pa @ 25/C (Howard *et al.* 1985) and Henry's Law Constant of 3.0 x 10-2 Pa m^3/mol (Volskay and Grady 1988). Its log octanol-water partition coefficient (Log K_{ow}) ranges from 5.11 (Geyer *et al.* 1984) to 9.61 (USEPA 1982). BEHP solubility in water is from 270 to 400 µg/L @ 25/C (DeFoe *et al.* 1990; Volskay and Grady 1988).

12.5 Environmental Fate and Transport

The most important processes influencing the distribution and transformation of BEHP in the environment include atmospheric photo-oxidation, partitioning to soil, sediment, and biota, and aerobic degradation (Howard 1989).

More than 50% of the BEHP present in the atmosphere occurs as a vapor phase (Giam *et al.* 1980). This vapor is subject to photo-oxidation. Howard *et al.* (1991) reported an estimated photo-oxidation half-life of gaseous BEHP of 2.9 to 29 h. Washout by precipitation and dry deposition are believed to play significant roles in the removal of BEHP from the atmosphere (Eisenreich *et al.* 1981). On the basis of experimental data on photolysis of dimethyl phthalate, Howard *et al.* (1991) estimated the photolysis half-life of BEHP in the atmosphere to be more than 144 days.

In water, aerobic biodegradation half-lives of BEHP range from five days to one month (Howard *et al.* 1991). In anaerobic conditions, BEHP persists between 42 and 389 days. The

photolysis half-life of BEHP in water is at least 144 days. Volatilization of BEHP from water is considered to be very slow. The estimated evaporative half-life is 15 years from a pond 1-m deep (Branson 1978). However, Klöpfer *et al.* (1982) determined an evaporative half-life for BEHP of about 140 days in a 21-cm deep vessel. Chemical hydrolysis of BEHP in water is extremely slow, with an estimated half-life of over 100 years (Wams 1987).

Bis(2-ethylhexyl)phthalate has a strong tendency to partition to sediments from the water column (Al-Omran and Preston 1987). Some BEHP may desorb from the sediments back into the water column (Atwater *et al.* 1990). Biodegradation (ring cleavage) of BEHP occurs at a higher rate in aerobic (13.8% degradation) than in anaerobic sediments (9.9% degradation; Johnson *et al.* 1984).

Bis(2-ethylhexyl)phthalate also has a strong tendency to adsorb to soil. Thus, it is not expected to evaporate from soil or leach into groundwater (Howard 1989). Though, BEHP may form a complex with water-soluble fulvic acid that may increase its mobilization and reactivity in soil (Khan 1980). The half-life of BEHP in soil ranges from 5 to 23 days (aerobic biodegradation rates; Howard *et al.* 1991).

12.6 Bioaccumulation

Bioconcentration factors for BEHP for aquatic algae and invertebrates range from 6.9 for the oyster, *Crassostrea virginica* (24-hour exposure period; Wofford *et al.* 1981) to 5,400 for the alga, *Chlorella fusca* (24-hour exposure; Geyer *et al.* 1984). Bioconcentration factors for fish range from 8.9 for rainbow trout (*Oncorhynchus mykiss*; 4-day exposure; Tarr *et al.* 1990) to 1,380 for the fathead minnow (*Pimephales promelas*; 28-day exposure; Mayer and Sanders 1973). In general, bioconcentration factors appeared to be higher for algae and aquatic invertebrates than for fish. This is because fish metabolize BEHP quite readily (Wofford *et al.* 1981). For example, Mayer (1976) reported that the fathead minnow (*Pimephales promelas*) metabolized BEHP with a biological half-life averaging 12.2 days. Due to the metabolism of BEHP, the biomagnification of BEHP through the aquatic food is unlikely (ATSDR 1991).

12.7 Effects on Aquatic and Terrestrial Organisms

For aquatic organisms, the lowest identified acutely toxic concentration was a 48-hour LC_{50} (median lethal concentration) of 133 µg/L for the cladoceran, *Daphnia pulex* (Passino and Smith 1987). No other studies were identified in which acute toxicity values were less than the solubility limit of 400 µg/L. The lowest reported chronic toxicity value was a 21-day LOEL (lowest observed effect level; survival reduced by 25%) of 160 µg/L for *Daphnia magna* and a 21-day NOEL (no observed effect level) of 77 µg/L for the same organism (Springborn Bionomics 1984).

The Chemical Manufacturer's Association (CMA; 1990) reported 96-hour LC_{50} values of 320 µg/L and 670 µg/L for the rainbow trout (*Oncorhynchus mykiss*) and the fathead minnow (*Pimephales promelas*). DeFoe *et al.* (1990) reported a 96-hour LC_{50} of 327 µg/L for the fathead minnow. Chronic toxicity of BEHP in sediments to frog eggs was investigated by Larson and Thuren (1987). A no observed effects level (NOEL) of 10 mg/kg fresh weight was determined for the hatchability of frog eggs over a 60 day exposure.

The acute toxicity of BEHP to mammals is relatively low. Oral LD50s (median lethal doses) generally exceed 25,000 mg/kg (body weight; BW) in mice and rats (Woodward 1988), 33,900 mg/kg (BW) in rabbits (Shaffer *et al.* 1945), and 26,000 mg/kg (BW) in guinea pigs (Krauskopf 1973).

Short-term, acute toxicity investigations of orally-administered phthalates to rats show decreases in body weight at concentrations greater than 625 mg/kg BW/day (NTP 1982). Increases in liver weight and transient mitotic bursts were observed at doses greater than 50 mg/kg BW/day in rats (Morton 1979; Lake *et al.* 1991). Alterations in the activity of hepatic enzymes consistent with peroxisome proliferation or increases in peroxisome numbers have been observed at doses greater than 25 mg/kg BW/day in rats (Morton 1979; Lake *et al.* 1991; Dostal *et al.* 1987). Effects on the kidneys, including increases in organ weight and changes in renal enzymes have also been observed following exposure to doses of BEHP ranging from 1,000 to 2,000 mg/kg BW/day (Dostal *et al.* 1987).

Subchronic studies with rats and mice revealed reductions in body weight gain at doses starting at 100 mg/kg BW/day (Shaffer *et al.* 1945; NTP 1982). Hepatomegaly and adverse

effects on the testes have also been observed at doses around 143 mg/kg BW/day (Gray *et al.* 1977). Clinical signs and mortality were observed only at high doses (370 mg/kg BW/day (NTP 1982). Chronic toxicity studies on rats and mice reported increases in peroxisome proliferation and alterations in hepatic enzymes at doses as low as 12 mg/kg BW/day (Ganning *et al.* 1991).

Reproductive toxicity was observed in one study (NTP 1982). Degeneration of the seminiferous tubules was observed in male rats in the high-dose group. Hypertrophy of cells in the anterior pituitary was increased in male rats in the high-dose group.

The potential carcinogenicity of BEHP was examined in a study sponsored by the National Toxicology Program (NTP 1982; Kluwe *et al.* 1982). Rats exposed a dose of 322 mg/kg BW/day were associated with a significantly higher (P=0.01) incidence of neoplasmic nodules of the liver and hepatocellular carcinomas.

12.8 References

Al-Omran, L.A. and M.R. Preston. 1987. The Interactions of phthalate esters with suspended particulate material in fresh and marine waters. Environmental Pollution 46:177-186.

ATSDR (Agency for Toxic Substances and Disease Registry). 1991. Toxicological profile for DEHP. United States Department of Health and Human Services. Public Health Service. Atlanta, Georgia.

Atwater, J.W., S.E. Jasper, P.D. Parkinson, and D.S. Mavinic. 1990. Organic contaminants in Canadian coal wastewaters and associated sediments. Water Pollution Research Journal of Canada 25:187-200.

Branson, D.R. 1978. Predicting the fate of chemicals in the aquatic environment from laboratory data. *In:* Estimating the Hazard of Chemical Substances to Aquatic Life. J. Cairns, Jr., K.L. Dickson, and A.W. Maki (Eds.) ASTM STP 657. American Society for Testing and Materials. pp. 55-70.

CIS (Camford Information Services, Inc.). 1992. CPI product profiles - Dioctyl phthalate. Camford Information Services Inc. Don Mills, Ontario. 4 pp.

CMA (Chemical Manufacturers Association). 1990. Comments of the phthalate esters panel of the chemical manufacturers association on EPA's proposed ambient water quality criteria for DEHP. Docket OW-FRL-3762-9. Chemical Manufacturers Association. Arlington, Virginia.

DeFoe, D.L., G.W. Holcombe, D.E. Hammermeister, and K.E. Biesinger. 1990. Solubility and Toxicity of eight phthalate esters to four aquatic organisms. Environmental Toxicology and Chemistry 9:623-636.

Dostal, L.A., W.L. Jenkins, and B.A. Schwetz. 1987. Hepatic peroxisome proliferation and hypolipidemic effects of DEHP in neonatal and adult rats. Toxicology and Applied Pharmacology 87:81-90.

Eisenreich, S.J., B.B. Looney, and J.D. Thornton. 1981. Airborne organic contaminants in the great lakes ecosystem. Environmental Science and Technology 15:30-38.

Ganning, A.E., M.J. Olsson, U. Brunk, and G. Dallner. 1991. Effects of prolonged treatment with phthalate ester on rat liver. Pharmacology and Toxicology 68:392-401.

Geyer, H., G. Politzki, and D. Freitag. 1984. Prediction of ecotoxicological behavior of chemicals: Relationship between n-octanol/water partition coefficient and bioaccumulation of organic chemicals by alga, *Chlorella*. Chemosphere 13:269-284.

Ghassemi, M., S. Quinlivan, and J. Bachmaier. 1984. Characteristics of leachates from hazardous waste landfills. Journal of Environmental Science and Health Al9:579-620.

Giam, C.S., E. Atlas, H.S. Chan, and G.S. Neff. 1980. Phthalate esters, PCB and DDT residues in the Gulf of Mexico atmosphere. Atmospheric Environment 14:65-69.

Government of Canada. 1994. Bis(2-ethylhexyl) Phthalate. Canadian Environmental Protection Act Priority Substances List Assessment Report. Environment Canada. Health Canada. Ottawa, Canada.

Gray, T.J.B., K.R. Butterworth, I.F. Gaunt, P. Grasso, and S.D. Gangolli. 1977. Short-term toxicity study of di(2-ethylhexyl)phthalate in rats. Food and Cosmetic Toxicology 15:389-399.

Howard, P.H. 1989. Handbook of environmental fate and exposure data for organic chemicals. Volume 1. Large Production and Priority Pollutants. Lewis Publishers Inc. Chelsea, Michigan.

Howard, P.H., R.S. Boethling, W.F. Jarvis, W.M. Meylan, and E.M. Michalenko. 1991. Handbook of environmental degradation rates. Lewis Publishers Inc. Chelsea, Michigan.

Howard, P.H., S. Banerjee, and K.H. Robillard. 1985. Measurement of water solubilities, octanol/water partition coefficients and vapor pressures of commercial phthalate esters. Environmental Toxicology and Chemistry 4:653-661.

IPCS (International Programme on Chemical Safety). 1992. Diethylhexyl phthalate. Environmental Health Criteria 131. World Health Organization. Geneva, Switzerland. 141 pp.

Johnson, B.T., M.A. Heitkamp, and J.R. Jones. 1984. Environmental and chemical factors influencing the biodegradation of phthalic acid esters in freshwater sediments. Environmental Pollution (Ser. B) 8:101-118.

Khan, S.U. 1980. Role of humic substances in predicting fate and transport of pollutants in the environment. *In:* Dynamics, Exposure and Hazard Assessment of Toxic Chemicals, Symposium. Miami Beach, Florida. September 11-13, 1978. R. Haque (Ed.). Ann Arbor, Science Publishers Inc. Ann Arbor, Michigan. pp. 215-230.

Klöpfer, W., G. Kaufmann, G. Rippen, and H.-J. Poremski. 1982. A laboratory method for testing the volatility from aqueous solution: First results and comparison with theory. Ecotoxicology and Environmental Safety 6:545-559.

Kluwe, K.M., J.K. Haseman, J.F., Douglas, and J.E. Huff. 1982. The carcinogenicity of dietary DEHP in Fischer 344 rats and B6C3FL mice. Journal of Toxicology and Environmental Health 10:797-815.

Krauskopf, L.G. 1973. Studies on the toxicity of phthalates via ingestion. Environmental Health Perspectives 3:61-72.

Lake, B.G., W.M. Cook, N.R. Worrell, M.E. Cunninghame, J.G., Evans, R.J. Price, P.J. Young, and F.M.B. Carpanini. 1991. Dose-response relationships for induction of hepatic peroxisome proliferation and testicular atrophy by phthalate esters in the rat. Human and Experimental Toxicology 10:67 (abstract).

Larson, P. and A. Thuren. 1987. Di-2-ethylhexylphthalate inhibits the hatching of frog eggs and is bioaccumulated by tadpoles. Environmental Toxicology and Chemistry 6:417-422.

Mayer, F.L. 1976. Residue dynamics of di-2-ethylhexyl phthalate in fathead minnows (*Pimephales promelas*). Journal of Fisheries Research Board of Canada 33:2610-2613.

Mayer, F.L., Jr. and H.O. Sanders. 1973. Toxicology of phthalic acid esters in aquatic organisms. Environmental Health Perspectives 3:153-157.

Montgomery, J.H. and L.M. Welkom. 1990. Groundwater chemicals desk reference. Lewis Publishers Inc. Chelsea, Michigan.

Morton, S.J. 1979. The hepatic effects of dietary di-2-ethyl-hexyl phthalate. Ph.D. Dissertation. The Johns Hopkins University. UMI Dissertation Information Service. Ann Arbor, Michigan. 120 pp.

NRCC (National Research Council Canada). 1980. Phthalate esters in the aquatic environment. NRCC No 17583. p. 27.

NTP (National Toxicology Program). 1982. Carcinogenesis bioassay of di(2-ethylhexyl)phthalate (CAS No.117-81-7) in F344 rats and B6C3F1 mice (feed study).

NTP-80-37. NIH Publication No. 82-1773. Research Triangle Park, North Carolina. 127 pp.

Passino, D.R.M. and S.B. Smith. 1987. Acute bioassays and hazard evaluation of representative contaminants detected in Great Lakes fish. Environmental Toxicology and Chemistry 6:901-907.

Peakall, D.B. 1975. Phthalate esters: Occurrence and biological effects. Residue Reviews 54:1- 41.

Shaffer, C.B., C.P. Carpenter, and H.F. Smyth. 1945. Acute and subacute toxicity of DEHP with note upon its metabolism. Journal Industrial Hygiene and Toxicology 27:130-135.

Springborn Bionomics. 1984. Chronic toxicity of fourteen phthalate esters to *Daphnia magna.* Toxicity test report submitted to Chemical Manufacturers Association. Washington, District of Columbia. Report No. BW-84-5-1567.

Tarr, B.D., M.G. Barron, and W.L. Hayton. 1990. Effect of body size on the uptake and bioconcentration of di-2-ethylhexyl phthalate in rainbow trout. Environmental Toxicology and Chemistry 9:989-995.

USEPA (United States Environmental Protection Agency). 1982. Aquatic fate processes data for organic priority pollutants. EPA 440/4-81-014. Office of Water Regulations and Standards. Washington, District of Columbia.

Volskay, V.T., Jr. and C.P.L. Grady Jr. 1988. Toxicity of selected RCRA compounds to activated sludge microorganisms. Journal of the Water Pollution Control Federation 60:1850-1856.

Wams, T.J., 1987. Diethylhexylphthalate as an environmental contaminant - A review. Science of the Total Environment 66:1-16.

Wofford, H.W., C.D. Wilsey, G.S. Neff, C.S. Giam, and J.M. Neff. 1981. Bioaccumulation and Metabolism of phthalate esters by oysters, brown shrimp, and sheepshead minnows. Ecotoxicology and Environmental Safety 5:202-210.

Woodward, K.N. 1988. Phthalate esters toxicity and metabolism. Volume I & II. CRC Press Inc. Boca Raton, Florida.

Appendix A2-13 An Overview of the Environmental Fate and Effects of Dichloroethane

13.1 Identity

Dichloroethane (DCE) belongs to a subgroup of volatile organic compounds that include chemicals with one to six chlorine substitutions on the ethane parent molecule. The Chemical Abstracts Service (CAS) registry number for dichloroethane is 107-06-2. DCE is a colorless liquid with the empirical formula CH2ClCH2CI. Other common and trade names of DCE include ethylene dichloride, 1,2-bichloroethane, dichloroethylene, ethylene chloride, glycol dichloride, symdichloroethane, and ethene dichloride (Archer 1979; Konemann 1981; Gossett *et al.* 1983; Verschueren 1983; Sax and Lewis 1987).

13.2 Uses

DCE is used for vinyl chloride production. A small amount (0.4%) is used as an antiknock additive in unleaded fuel. Other minor applications include adhesives, coatings, solvent extractants, and cleaning solutions (ZENON 1982). In 1991, DCE was used primarily for the synthesis of vinyl chloride monomer (approximately 88%) and in the synthesis of other chlorinated solvents and ethyleneamines (2%; Chemical Marketing Reporter 1992).

13.3 Sources

DCE can enter the environment during production, storage, disposal, and secondary processing of the chemical. In the production stage, DCE may be released to the environment via the atmosphere, wastewater releases, and land disposal (Environment Canada 1988). Atmospheric emissions of DCE account for the majority of releases. Wastewater releases are the second largest source. Other dispersive environmental

releases include activities such as lead scavenging, paints, coatings, grain fumigation, and cleaning (USEPA 1985).

13.4 Physical and Chemical Properties

DCE is a highly volatile chemical with a vapor pressure of 8.5 kPa (at 20°C). It is very soluble with a reported water solubility of 8,690 mg/L (at 20°C). It has a Henry's Law constant of 111.5 PaAm^3 /mol (at 25°C) and a log partition coefficient (K_{ow}, octanol/water partition coefficient; K_{oc}, organic carbon partition coefficient) less than 2 (log K_{ow} 1.76 and log K_{oc} = 1.28; Archer 1979; Konemann 1981; Warner *et al.* 1987; Chiou *et al.* 1979).

13.5 Environmental Fate and Transport

There is little information regarding the fate of DCE in the aquatic environment. Based on the limited information available, volatilization appears to be the major removal mechanism of DCE from the aquatic environment (Dilling *et al.* 1975). Dilling *et al.* (1975) determined the half- life of a 1 mg/L solution of 29 min when stirred at 200 rpm in water in an open container. However, the authors commented that these data are not readily transferable to the environment because natural concentrations of DCE are expected to be much lower and because other factors such as wind speed and wave action are highly variable. No studies were found that investigated photolysis, oxidation, or hydrolysis of DCE in water or sediment.

Studies on analogous compounds (e.g., dichloromethane, trichloroethane, dibromoethane) indicate that the above processes are unlikely to be important in the removal of DCE from the aquatic environment (Dilling *et al.* 1975; Radding *et al.* 1977). Portier and Meyers (1984) reported that aerobic biodegradation may be an important process for the removal of DCE from the aquatic environment. In sediment/water microcosms, they found that DCE had a half-life of 48 h in fresh water. In saline conditions (10 to 24 g/L), the degradation rate was reduced by a factor of 4 to 5 times. Continuous-flow microcosms promoted either co-metabolic or

co-oxidative biotransformation, resulting in 71% degradation of DCE after 48 h (Portier and Meyers 1984).

In the atmosphere, DCE will react with hydroxyl radicals to form chloracetyl chloride (Howard and Evenson 1976; Radding *et al.* 1977). Radding *et al.* (1977) indicate an atmospheric half-life of 234 h for this photo-oxidation reaction. USEPA (1975) and Howard and Evenson (1976) predicted that DCE will have an atmospheric lifetime of one to four months. Because this reaction is relatively rapid, little DCE is expected to reach the stratosphere from the troposphere. Similarly, chloracetyl chloride will be rapidly hydrolyzed to hydrochloric and carboxylic acids in the troposphere (Morrison and Boyd 1973). Despite the relatively short residence time of DCE in the atmosphere, Pearson and McConnell (1975) suggest that DCE has the potential for long-range transport and that this process accounts for its presence in upland waters.

13.6 Bioaccumulation

Given that DCE has a high water solubility and low octanol/water partition coefficient, the accumulation of this substance from aquatic environments and through trophic transfer would be expected to be low. The EPA's AQUIRE (Aquatic Toxicity Information Retrieval) database (USEPA 2001) lists the experimentally-derived bioconcentration factor at 2. Thus, DCE is not expected to biomagnify in Calcasieu Estuary foodwebs. However, the principal route of exposure to this substance would be through direct contact and ingestion of contaminated waters. As the substance has a low bioaccumulative potential, food sources are unlikely to contribute significantly to the exposure.

13.7 Effects on Aquatic and Terrestrial Organisms

The 96-hour LC_{50} (median lethal concentration) values for the most sensitive fish species ranged from 116 mg/L for the fathead minnow (*Pimephales promelas*; Walbridge *et al.* 1983) to 225 mg/L for rainbow trout (*Oncorhynchus mykiss*; Mayer and Ellersieck 1986). Other reported LC_{50} values ranged from 106 mg/L for guppies

(*Poecilia reticulata*) after a 7-day exposure (Konemann 1981) to 550 mg/L for sunfish (*Lepomis macrochirus*) after a 4-day exposure (Dawson *et al.* 1975/1977).

Acute toxicity tests with a marine polychaete, *Ophryotrocha labronica*, have shown low toxicity. Rosenberg *et al.* (1975) reported lethal median concentrations of 400 to 900 mg/L and decreased numbers of eggs hatching at concentrations of 400 mg/L.

1,2-Dichloroethane is not acutely toxic to experimental mammals. For example, LC_{50}s for rats exposed via inhalation for 6 or 7.25 h ranged from 1,000 ppm (4,000 mg/m^3 ; Spencer *et al.* 1951) to 1,650 ppm (6,600 mg/m^3 ; Bonnet *et al.* 1980). Oral LD50s (median lethal doses) for rats, mice, dogs, and rabbits ranged from 413 to 2,500 mg/kg (body weight; BW; WHO 1987; NIOSH 1977; Munson *et al.* 1982).

The results of short-term and subchronic studies with several species of experimental animals indicate that the liver and kidneys are the target organs. In studies with rats, mice, rabbits, guinea pigs, cats, dogs, and monkeys exposed to 1,2-dichloroethane by inhalation, there were morphological changes in the liver and kidneys at concentrations as low as 200 ppm (800 mg/m^3). No effects were observed at 100 ppm (400 mg/m^3; Spencer *et al.* 1951; Hofmann *et al.* 1971). Administration of 1,2-dichloroethane in corn oil by gavage to rats resulted in more severe toxic effects than similar doses administered in drinking water. These effects included hyperplasia and inflammation of the fore stomach at doses as low as 240 mg/kg (BW) per day (National Toxicology Program 1991).

Reproduction was not affected in rats exposed to up to 150 ppm 1,2-dichloroethane by inhalation 6 h/day for 176 days. DCE was not teratogenic in rats inhaling 100 ppm or in rabbits inhaling 100 or 300 ppm for 7 h/day throughout the period of major organogenesis (Hayes and Laws 1991). A dose of 250 or 500 ppm of 1,2-dichloroethane was administered in feed mash to rats for a 2 year period. Approximately 60 to 70% of the dose was consumed. No significant decrease in fertility, litter size, or fetal weight was observed.

DCE is classified as a probable human carcinogen based on the induction of several tumor types in rats and mice treated by gavage and lung papillomas after topical application (IRIS 2000). 1,2-Dichloroethane has been consistently positive for

genotoxic activity (Prodi *et al.* 1986; Hellman and Brandt 1986; Baertsch *et al.* 1991) during in vivo studies in rats, mice, and insects (Drosophila).

13.8 References

Archer, W.J. 1979. Chlorocarbons and chlorohydrocarbons. *In:* Encyclopedia of Chemical Technology, J. Kirk and D.F. Other (Eds.). Volume 5, 3rd Edition. John Wiley and Sons. New York, New York.

Baertsch, A., W.K. Lutz, and C. Schlatter. 1991. Effect of inhalation exposure regimen on DNA binding potency of 1,2-dichloroethane in the rat. Archives of Toxicology 65:169-176.

Bonnet, P., J.M. Francin, D. Grakiski, G. Raoult, and D. Zissu. 1980. Determination de la concentration lethale 50 des principaux hydrocarbures aliphatiques chlores chez le rat. Archives des maladies professionnelles de medecine du travail et de securite sociale 41(6-7):317-321.

Chemical Marketing Reporter. 1992. Chemical profile: Ethylene dichloride. Chemical Marketing Reporter Magazine 241(19):42.

Chiou, C.T., L.J. Peters, and V.H. Freed. 1979. A physical concept of soil-water equilibria for nonionic organic compounds. Science 206:831-832.

Dawson, G.W., A.L. Jennings, D. Drozdowski, and E. Rider. 1995/1977. The acute toxicity of 47 industrial chemicals to fresh and saltwater fishes. Journal of Hazardous Materials 1(4):303-318.

Dilling, W.L., N.B. Tefertiller, and G.J. Kallos. 1975. Evaporation rates and reactivities of methylene chloride, chloroform, trichloroethylene, tetrachloroethylene, and other chlorinated compounds in dilute aqueous solutions. Environmental Science and Technology 9(9):833-837.

Environment Canada. 1988. Chlorinated hydrocarbon use pattern update (unpubl.). Use Pattern Section. Chemical Contaminants Branch. Ottawa. (As cited in CCME 1994).

Gossett, R.W., D.A. Brown, and D.R. Young. 1983. Predicting the bioaccumulation of organic compounds in marine organisms using octanol/water partition coefficients. Marine Pollution Bulletin 14(10):387-392.

Hayes, W.J., Jr. and E.R. Laws, Jr., (Eds.). 1991. Handbook of pesticide toxicology. Volume 2. Classes of Pesticides. Academic Press, Inc. New York, New York.

Hellman, B. and I. Brandt. 1986. Effects of carcinogenic halogenated aliphatic hydrocarbons on [3 h]thymidine incorporation into various organs of the mouse. A comparison between 1,2-dibromoethane and 1,2-dichloroethane. Mutation Research 163:193 - 199.

Hofmann, H.T., H. Birnsteil, and P. Jobst. 1971. Zur inhalations toxicitat von 1,1- and 1.2-dichloroathan. Archives of Toxicology 27:248 - 265 (in German; As cited in WHO 1987).

Howard, C.J. and K.M. Evenson. 1976. Rate constants for the reactions of -OH with ethane and some halogen substituted ethanes at 296/K. Journal of Chemical Physics 64(11) :4303-4306.

IRIS (Integrated Risk Information System). 2000. Substance file list on 1,2-dichloroethane (107-06-2). Obtained from: http://www.epa.gov/ngispgm3/iris.

Konemann, H. 1981. Quantitative structure-activity relationships in fish toxicity studies, part 1: Relationship for 50 industrial pollutants. Toxicology 19:209-221.

Mayer, F.L. and M.R. Ellersieck. 1986. Manual of acute toxicity: Interpretation and data base for 410 chemicals and 66 species of freshwater animals. Resource Publication 160. Fish and Wildlife Service. United States Department of the Interior. Washington, District of Columbia.

Morrison, R.T. and R.N. Boyd. 1973. Organic chemistry. 3rd edition. Allyn and Bacon. Boston, Massachusetts.

Munson, A.E., V.M. Sanders, K.A. Douglas, L.E. Sain, B.M. Kauffmann, and K.L. White Jr. 1982. In vivo assessment of immunotoxicity. Environmental Health Perspectives 43:41-52.

NIOSH (National Institute for Occupational Safety and Health). 1977. Registry of toxic effects of chemical substances. Volume 2. DHEW Entry NKI05250. Cincinnati, Ohio.

National Toxicology Program. 1991. Toxicity studies of 1,2-dichloroethane (ethylene dichloride) in F344/N rats, Sprague Dawley rats, Osborne-Mendel rats, and B6C3F1 mice (drinking water and gavage studies). NIH publication Number 91-3123. Research Triangle Park, North Carolina.

Pearson, C.R. and G. McConnell. 1975. Chlorinated C, and C2 hydro- carbons in the marine environment. Proceedings of the Royal Society of London. Proceedings B. Biological Sciences 189:305-332.

Portier, R.J. and S. P. Meyers. 1984. Chitin/toxicant interactions and microbial degradation within estuarine ecosystems. GERBAM-Deuxieme Colloque International de Bacteriologie Marine-CNRS, Brest. 1-5 October. IFREMER. Actes de Colloques 3:579-587.

Prodi, G., G. Arfelini, A. Colacci, S. Grilli, and M. Mazzulo. 1986. Interaction of halocompounds with nucleic acids. Toxicologic Pathology 14:438-444.

Radding, S.B., D.H. Liu, M.L. Johnson, and T. Mill. 1977. Review of the environmental fate of selected chemicals. EPA-560/5-77-003. Office of Toxic Substances. United States Environmental Protection Agency. Washington, District of Columbia.

Rosenberg, R., O. Grahn, and L. Johansson. 1975. Toxic effects of aliphatic chlorinated by-products from vinyl chloride production on marine animals. Water Research 9(7):607-612.

Sax, N.J. and R.J. Lewis (Eds.). 1987. Hazardous chemicals desk reference. Van Nostrand Reinhold. New York, New York.

Spencer, H.C., V.K. Rowe, E.M. Adams, D.D. McCollister, and D.D. Irish. 1951. Vapor toxicity of ethylene dichloride determined by experiments on laboratory animals. American Medical Association Archives of Industrial Hygiene and Occupational Medicine 4:482-493.

USEPA (United States Environmental Protection Agency). 1975. Report on the problem of halogenated air pollutants and stratospheric ozone. EPA-600/9-75-008. Office of Research and Development. Research Triangle Park, North Carolina.

USEPA (United States Environmental Protection Agency). 1985. Quantification of toxicological effects of 1,2-dichloroethane . Office of Drinking Water. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 2001. Ecotox Database System. Washington, District of Columbia. (Obtained from http://.epa.gov/ecotox/).

Verschueren, K. (Ed.). 1983. Handbook of environmental data on organic chemicals. Van Nostrand Reinhold, New York.

Walbridge, C.T., J.T. Fiandt, G.L. Phipps, and G.W. Holcombe. 1983. Acute toxicity of ten chlorinated aliphatic hydrocarbons to the fathead minnow (*Pimephales promelas*). Archives of Environmental Contamination and Toxicology 12(6):661-663.

Warner, H.P., J.M. Cohen, and J.C. Ireland. 1987. Determination of Henry's law constants of selected priority pollutants. EPA-600/D-87/229. Office of Science and Development. United States Environmental Protection Agency.

WHO (World Health Organization). 1987. Environmental Health Criteria 62. 1,2-dichloroethane. IPCS International Programme on Chemical Safety. Geneva.

ZENON. 1982. Sources of persistent organic toxic substances in municipal waste water. ZENON Environmental Enterprises, Ltd. Hamilton, Ontario.

Appendix A2-14 An Overview of the Environmental Fate and Effects of Trichloroethane

14.1 Identity

The CAS or Chemical Abstracts Service registry number for trichloroethane (TCA) is 71-55-6. TCA is a colorless liquid with the empirical formula CH_3CCl_3. Other common and trade names by which TCA is known include methyl chloroform, chloroethene, and alpha-trichloroethane (Archer 1979; Konemann 1981; Verschueren 1983; Konietzko 1984).

14.2 Uses

TCA is widely used as an industrial solvent (Verschueren 1983). The majority of applications include cleaning of metals, armatures of electric motors, generators, switch gears, and in electronic equipment. The remaining uses include production of adhesives, as a propellant modifier in aerosols, textile finishing operations, office supplies, dry lubricants, and laboratory solvents (Environment Canada 1988).

14.3 Sources

The majority of releases during the manufacturing process are to surface water, followed by air, and land. During the consumption of TCA in degreasing operations, the majority of releases are into air, then soil and water (USEPA 1982).

14.4 Physical and Chemical Properties

TCA is a highly volatile chemical, with a vapor pressure of 16.5 kPa (124 mm mercury; Hg) at 25 °C (Lide 1994-1994). TCA is soluble in all common organic solvents and has a water solubility of 4,400 mg/L. The octanol-water partition coefficient is reported at 2.49 (HSDB 2000).

14.5 Environmental Fate and Transport

Volatilization is the major process for the removal of TCA from aquatic ecosystems (Dilling *et al.* 1975; Wakeham *et al.* 1983). Wakeham *et al.* (1983) studied the volatilization of TCA in seawater microcosms under conditions simulating winter, spring, and summer in a moderately polluted estuary (2.7 to 4.3 µg/L TCA). They found that TCA had a half-life that ranged from 11 days in winter to 24 days in spring. Subsequent experiments that compared TCA removal in microcosms poisoned with mercuric chloride (to retard biological activity) with that in non-poisoned microcosms indicated that 83.5% of TCA removal could be attributed to volatilization. The remainder was due to microbial degradation. Other studies have indicated that photolysis, oxidation, and elimination reactions are not important in the removal of TCA from aquatic systems (Dilling *et al.*1975; USEPA 1979; Vogel and McCarty 1987b). Hydrolysis of TCA was shown to occur in aquatic ecosystems. The half-life was 0.5 to 0.75 years (Dilling *et al.* 1975; Pearson and McConnell 1975; Haag *et al.* 1986). Therefore, this process may be important in TCA removal from groundwater. However, in groundwater that is anaerobic and conducive to methanogenesis, TCA can be biotransformed by reductive dehalogenation to 1,1-dichloroethane and chloroethene. The half-life for this process may be less than six days (Vogel and McCarty 1987a).

At present, there is no clear evidence to suggest that TCA is selectively concentrated in sediments. Dilling *et al.* (1975) showed that bentonite clay, dolomitic limestone, and peat moss adsorbed TCA, but the adsorption and desorption rates were approximately equal after 10 to 30 min. TCA is long-lived in the atmosphere, with a photo oxidative half-life of more than six years in the troposphere. Consequently,

12 to 25% of TCA in the troposphere will reach the stratosphere (McConnell and Schiff 1978; USEPA 1982). The presence of TCA in pristine waters is believed to be due to long-range transport (Pearson and McConnell 1975).

Given that TCA has a high water solubility and low octanol/water partition coefficient, the accumulation of this substance from aquatic environments and through trophic transfer would be expected to be low. The EPA's AQUIRE (Aquatic Toxicity Information Retrieval) database (USEPA 2001) lists the experimentally-derived bioconcentration factor for bluegill fish at 9. Thus, DCE is not expected to biomagnify in Calcasieu Estuary foodwebs.

14.6 Bioaccumulation

Once released to aquatic environments, it will tend to volatilize, but a significant portion will remain in the water column. Thus, the principal route of exposure to this substance is through direct contact and ingestion of contaminated waters. As the substance has a moderate to low bioaccumulative potential, food sources may also contribute to the exposure.

14.7 Effects on Aquatic and Terrestrial Organisms

Geiger *et al.* (1985) found that fathead minnow (*Pimephales promelas*) had a 96-hour LC_{50} (median lethal concentration) of 42.3 mg/L and a 96-hour EC50 (median effect concentration; behavioral changes, increased respiration, loss of equilibrium) of 28.8 mg/L in flow-through tests. Alexander *et al.* (1978) found that exposure to TCA in a static test resulted in a 96-hour LC_{50} of 105.0 mg/L. In a flow-through test, Alexander *et al.* (1978) found that *P. promelas* had a 96-hour EC50 of 11.1 mg/L and a 96-hour EC10 of 9.0 mg/L. The only other freshwater fish species examined was sunfish (*Lepomis macrochirus*), it was found to have an LC_{50} of 72.0 mg/L in a static test (Buccafusco *et al.* 1981).

TCA appears to have low potential for acute toxicity to mammals. The median lethal concentration range for acute exposures to mammals tested is 5 to 11 g/kg. Inhalation toxicity studies for TCA administered to mice and rats resulted in LC_{50}s of 24,000 ppm for a 1-hour exposure to 13,500 ppm for a 10-hour exposure (Verschueren 1983).

Male and female rats given TCA for 78 weeks at 750 mg/kg BW (body weight)/day exhibited early mortality when compared with the untreated controls. Only 3% of the treated rats survived to termination of experiment (IARC 1979).

Pregnant mice and rats were exposed to concentrations of 875 ppm. Both were exposed for 7 h daily periods on days 6 through 15 of gestation. No fetal toxicity or teratogenicity was found (Shepard 1986). Male and female ICR Swiss mice received 1,1,1-trichloroethane at concentrations up to 5.83 mg/mL in drinking water (daily dose 1,000 mg/kg). No taste aversion was evident and there were effects on fertility, gestation, viability, or lactation indices. Pup survival and weight gain were not adversely affected. TCA failed to produce significant dominant lethal mutations or teratogenic effects in either of the two generations tested (Lane *et al.*1982).

There is inadequate evidence for the carcinogenicity of TCA in experimental animals (IARC 1979). TCA showed mutagenic activities when tested by salmonella/microsome test, the basic test on drosophila, and the micronucleus test on mouse bone marrow (Gocke *et al.* 1981).

14.8 References

Alexander, H.C., W.M. McCarty, and E.A. Bartlett. 1978. Toxicity of perchloroethylene, trichloroethylene, 1, 1, 1-trichloroethane, and methylene chloride to fathead minnows. Bulletin of Environmental Contamination and Toxicology 20:344-352.

Archer, W.J. 1979. Chlorocarbons and chlorohydrocarbons. *In:* Encyclopedia of Chemical Technology. Volume 5. 3rd edition. J. Kirk and D.F. Othmer (Eds.). John Wiley and Sons. New York.

Buccafusco, R.J., S.J. Ells, and G.A. LeBlanc. 1981. Acute toxicity of priority pollutants to bluegill (*Lepomis macrochirus*). Bulletin of Environmental Contamination and Toxicology 26:446-452.

Dilling, W.L., N.B. Tefertiller, and G.J. Kallos. 1975. Evaporation rates and reactivities of methylene chloride, chloroform, trichloroethylene, tetrachloroethylene, and other chlorinated compounds in dilute aqueous solutions. Environmental Science and Technology 9(9):833-837.

Environment Canada. 1988. Chlorinated hydrocarbon use pattern update (unpubl.). Use Pattern Section. Chemical Contaminants Branch. Ottawa, Ontario. (As cited in CCME 1994)

Geiger, D.L., S.H. Poirier, L.T. Brooke, and D.J. Call (Eds.). 1985. 1, l'1-trichloroethane. *In:* Acute Toxicities of Organic Chemicals to Fathead Minnow (*Pimephales promelas*). Volume 3. Center for Lake Superior Environmental Studies. University of Wisconsin-Superior. pp. 31-32.

Gocke, E., M.-T. King, K. Eckhardt, and D. Wild. 1981. Mutagenicity of cosmetics ingredients licensed by the European communities. Mutation Research 90:91-109.

Haag, N.R., T. Mill, and A. Richardson. 1986. Effect of subsurface sediment on hydrolysis reactions. In: Extended Abstracts. 192nd National Meeting of the American Chemical Society. Anaheim, California. American Chemical Society. Washington, District of Columbia.

HSDB (Hazardous Substances Databank). 2000. Profile for 1,1,1-trichloroethane. National Library of Medicine. Bethesda, Maryland.

IARC (International Agency for Research on Cancer). 1979. Monographs on the evaluation of the carcinogenic risk of chemicals to man. World Health Organization. Geneva, Switzerland. p. V20-523.

Konemann, H. 1981. Quantitative structure-activity relationships in fish toxicity studies. Part 1: Relationship for 50 industrial pollutants. Toxicology 19:209-221.

Konietzko, H. 1984. Chlorinated ethanes: Sources, distribution, environmental impact, and health effects. *In:* Hazard Assessment of Chemicals. Current Developments. J. Saxena (Ed.). Academic Press. New York, New York.

Lane, R.W., Riddle, B.L., and Borzelleca, J.F. 1982. Effects of 1,2-dichloroethane and 1,1,1- trichloroethane in drinking water on reproduction and development in mice. Toxicology and Applied Pharmacology 63:409-421.

Lide, D.R. (Ed.). 1994-1995. CRC handbook of chemistry and physics. 75th ed. CRC Press Inc. Boca Raton, Florida. p. 6-80.

McConnell, J.C. and M.I. Schiff. 1978. Methyl chloroform: Impact on stratospheric ozone. Science 199:174-177.

Pearson, C.R. and G. McConnell. 1975. Chlorinated C, and C_2 hydrocarbons in the marine environment. Proceedings of the Royal Society of London, Proceedings B, Biological Sciences 189:305-332.

Shepard, T.H. 1986. Catalog of teratogenic agents. 5th edition. The Johns Hopkins University Press. Baltimore, Maryland.

USEPA (United States Environmental Protection Agency). 1979. Water-related environmental fate of 129 priority pollutants. II. Halogenated Aliphatic Hydrocarbons, Halogenated Ethers, Monocyclic Aromatics, Phthalate Esters, Polycyclic Aromatic Hydrocarbons, Nitrosamines, and Miscellaneous Compounds. EPA-440/4- 79-029b. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1982. An exposure and risk assessment for trichloroethanes. EPA-440/4-85-018. Office of Water Regulations and Standards. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 2001. Ecotox Database System. Washington, District of Columbia. (Obtained from http://www.epa.gov/ecotox/).

Verschueren, K. (Ed.). 1983. Handbook of environmental data on organic chemicals. Van Nostrand Reinhold, New York, New York.

Vogel, T.M. and P.L. McCarty. 1987a. Abiotic and biotic transformations of 1,1,1-trichloroethane under methanogenic conditions. Environmental Science and Technology 21:1208-1213.

Vogel, T.M. and P.L. McCarty. 1987b. Rate of abiotic formation of 1,1-dichloroethylene from 1,1,1-trichloroethane in ground water. Journal of Contaminant Hydrology 1:299-308.

Wakeham, S.G., A.C. Davis, and J.L. Karas. 1983. Mesocosm experiments to determine the fate and persistence of volatile organic compounds in coastal seawater. Environmental Science and Technology 17:611- 617.

Appendix A2-15 An Overview of the Environmental Fate and Effects of Carbon Disulfide

15.1 Identity

Carbon disulfide is a highly refractive, mobile, and very flammable liquid. Pure carbon disulfide is a colorless liquid that is comprised of one carbon atom and two sulfur atoms (i.e., CS_2; USEPA 1994a). While pure carbon disulfide has a pleasant chloroform-like aroma, the impure carbon disulfide that is used in most industrial processes is a yellowish liquid with an unpleasant odor (like that of rotting radishes; ATSDR 1997). Common synonyms for this substance include carbon bisulfide and dithiocarbonic anhydride (Budavari 1989).

15.2 Uses

Carbon disulfide is used in a number of industrial applications. Roughly 40% of the carbon disulfide that is used in the United States is employed in the production of rayon fibers (USEPA 1994b). Carbon disulfide is also used in the production of agricultural fumigants (18% of domestic use), cellophane and other regenerated cellulose (12% of domestic use), rubber chemicals (16% of domestic use), as an agent in metal treatment and plating, as a solvent for cleaning and extraction, and as an extractant in olive oil production (collectively 14% of domestic use; USEPA 1994b). In the past, carbon disulfide was also used a s a precursor in carbon tetrachloride production; however, this use has been phased out. Demand for carbon disulfide is expected to continue to decline in the future (Mannsville 1993).

15.3 Sources

As of 1994, there are three major producers of carbon disulfide in the United States, including Akzo Chemicals (in LeMoyne, Alabama), Atochem North America Green Bauou, Texas), and PPG Industries (Natrium, West Virginia; USEPA 1994b). Akzo Chemicals closed a second plant in 1991 (Delaware City, Delaware; 350 million pound per annum capacity). The aggregate annual capacity of the three operating plants is 350 million pounds of carbon disulfide; however, only 207 million pounds of carbon disulfide were produced in 1992 (USEPA 1994b). In the same year, four million pounds of carbon disulfide were imported and 32 million pounds of this substance were exported (Mannsville 1993). Carbon disulfide also occurs naturally in the environment and is formed primarily during the anaerobic biodegradation of organic material (i.e., in oceans, lakes, and wetlands; USEPA 1994b). This substances has also been detected in the air over volcanoes (ATSDR 1992).

15.4 Physical and Chemical Properties

Information on the physical and chemical properties of carbon disulfide provides a basis for evaluating the fate of this substance upon release into the environment. At room temperature, carbon disulfide is a liquid that is denser than water (i.e., specific gravity of 1.263 at 20°C) and is moderately soluble in water (i.e., 2.3 g/L at 22°C; Verschueren 1983). With a K_{oc} (organic carbon partition coefficient) of 54 and a log K_{ow} (octanol/water partition coefficient) of 2.16, carbon disulfide is does not form strong associations with organic carbon in soils or sediments or with lipids in biological tissues (USEPA 1986). As carbon disulfide has a high vapor pressure (i.e., 260 mm mercury; Hg at 20°C) and Henry's Law Constant (i.e., 0.0101 atm m^3/mol), it is likely to readily partition to the atmosphere upon release to water or soils.

15.5 Environmental Fate and Transport

The oceans represent the largest source of carbon disulfide that is released into the environment, with annual emissions estimated at 270 million pounds (HSDB 1994). According to the Toxic Chemical Release Inventory, releases of carbon disulfide from United States industries totaled 93 million pounds in 1992 (USEPA 1994b). Of this total, 92.9 million pounds were released into the atmosphere, 48,000 pounds were released into surface waters, 3,000 pounds were released to underground injection sites, and 21 pounds were released to land (TRI92 1994). Upon release to aquatic systems, volatilization represents the most important fate process for carbon disulfide, with a half-life of 2.6 hours reported for a model river system (HSDB 1994). By comparison, hydrolysis and biodegradation are relatively minor fate processes. Oxidation and hydroxylation represent the most important fate processes in the atmosphere, with a half-life of 12 days reported for this substance in air (ATSDR 1992).

15.6 Bioaccumulation

Studies on animals have demonstrated that carbon disulfide is readily absorbed via the lungs, the gastrointestinal tract, and the skin. Following uptake, this substance is distributed throughout the body, with the highest concentrations found in lipid-rich tissues (ATSDR 1992). In most organisms, bioaccumulation appears to be limited by the rapid metabolism and excretion of carbon disulfide, with roughly 10 to 30% of absorbed doses excreted through the lungs unmetabolized and 70 to 90% of absorbed doses metabolized and excreted through the kidneys (ATSDR 1992). A bioconcentration factor in fish of 626 was calculated for carbon disulfide (USEPA 1986). Due to the low K_{ow} and rapid excretion of this substance, bioaccumulation in the food web is predicted to be low (USEPA 1996).

15.7 Effects on Aquatic and Terrestrial Organisms

Short-term (i.e., # 96 hours) exposure to water-borne carbon disulfide can adversely affect aquatic organisms. For example, 96-hour LC_{50}s (i.e., median lethal concentrations) of carbon disulfide ranged from 3.0 mg/L in guppies (*Poecelia reticulata*) to 135 mg/L in mosquitofish (*Gambusia affinis*; USEPA 1994c). Aquatic invertebrates tended to be more sensitive to the toxic effects of carbon disulfide than fish, with 48-hour LC_{50}s of 1.9 to 2.2 mg/L reported for the water flea, *Daphnia magna*(USEPA 1994c). Growth of the alga, *Chlorella pyrenoidosa*, was impaired following a 96-hour exposure to 21.0 mg/L of carbon disulfide (USEPA 1994c). No information was located on the chronic toxicity of this substance.

The results of toxicological studies show that exposure to carbon disulfide can cause a wide range of effects in mammals. Short-term exposure to moderate to high levels of carbon disulfide can be fatal to animals, with oral LD_{50} (median lethal dose) values of 3188, 2780, 2550, and 2125 mg/kg body weight (BW) reported for rats, mice, rabbits, and guinea pigs, respectively (USEPA 1986). In air, 2-hour LC_{50}s of 25 g/m^3 and 10 mg/m^3 were reported for rats and mice, respectively (RTECS 1994). The neurotoxicity, cardiovascular system impairment, and kidney dysfunction are the principal effects that have been reported for longer-term exposure to carbon disulfide (USEPA 1994b). Although neither carcinogenic nor genotoxic effects have been documented in association with exposure to carbon disulfide, prolonged oral exposure to this substance resulted in impaired reproduction in rabbits(i.e., fetal resorption; USEPA 1994a).

15.8 References

ATSDR (Agency for Toxic Substances and Disease Registry). 1997. Carbon Disulfide. ToxFAQs. (Obtained from http://www.atsdr.cdc.gov/tfacts82.html)

ATSDR (Agency for Toxic Substances and Disease Registry). 1992. Toxicological profile for carbon disulfide. Public Health Service. United States Department of Health and Human Services. Atlanta, Georgia. (As cited in USEPA 1994b).

Budavari, S., M.J. O'Neil, A. Smith, and P.E. Heckelman (Eds.). 1989. The Merck Index: An Encyclopedia of Chemicals, Drugs, and Biologicals. Eleventh Edition. Merck and Company, Inc. Rahway, New Jersey.

HSDB (Hazardous Substances Data Bank). 1994. MEDLARS Online Information Retrieval System. National Library of Medicine. (As cited in USEPA 1994b).

Mansville. 1993. Chemical products synopsis, carbon disulfide. Mansville Chemical Products Corporation. (As cited in USEPA 1994b).

RTECS (Registry of Toxic Effects of Chemical Substances). 1994. MEDLARS online information retrieval system. National Library of Medicine. (As cited in USEPA 1994a).

TRI92. 1994. 1992 Toxics release inventory. Public data release. Office of Pollution Prevention and Toxics. United States Environmental Protection Agency. Washington, District of Columbia. (As cited in USEPA 1994b).

USEPA (United States Environmental Protection Agency). 1994a. Chemicals in the environment: Carbon disulfide (CAS No. 75-15-0). OPPT Chemical Fact Sheet. EPA 749-F-94-008. Office of Pollution Prevention and Toxics. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1994b. Chemical summary for carbon disulfide. EPA 749-F-94-008a. Office of Pollution Prevention and Toxics. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1994c. Acquire: EPA's aquatic ecotoxicological data system. Duluth Environmental Research Laboratory. Duluth, Michigan.

USEPA (United States Environmental Protection Agency). 1986. Health and environmental effects profile for carbon disulfide. ECAO-CIN-P170. Office of Health and Environmental Assessment. Environmental Criteria and Assessment Office. Washington, District of Columbia. (As cited in USEPA 1994b).

Verschueren, K. 1983. Carbon disulfide. *In*: Handbook of Environmental Data on Organic Chemicals. Second Edition. Van Nostrand Reinhold Company. New York, New York. (As cited in USEPA 1994b).

Appendix A2-16 An Overview of the Environmental Fate and Effects of Acetone

16.1 Identity

Acetone belongs to the group of chemicals called volatile organic compounds. This group includes many industrial chemicals and solvents that readily volatilize into the atmosphere. Acetone (CAS; Chemical Abstracts Service Number 67-64-1) is a clear, colorless liquid with a distinct smell and taste. Alternate names include dimethyl ketone, 2-propanone, and beta-ketopropane.

16.2 Uses

Acetone's primary use is as a solvent. The largest solvent use is in surface coatings, including paint thinners and wash solvents. Other uses include the production of acetone cyanohydrin (an intermediate in the production of acrylics), bisphenol, pharmaceuticals, cosmetics, and as a flavoring agent (IPCS 1998).

16.3 Sources

Acetone enters air, water, and soil as a result of natural processes and human activities. Acetone occurs naturally in plants, trees, and is produced by animals during fat catabolism. It is also released via forest fires, volcanic gases, and other geophysical processes (ATSDR 1994). Anthropogenic releases of acetone are associated with acetone synthesis manufacture and use, exhaust from automobiles, tobacco smoke, landfills, and the burning of certain waste materials (IPCS 1998).

16.4 Physical and Chemical Properties

Acetone evaporates readily into the air and is completely miscible with water, as well as many common organic solvents (IPCS 1998). Acetone has a high vapor pressure, a boiling point of 56.2°C at STP, and a melting point of -95°C (Weast 1987).

16.5 Environmental Fate and Transport

Acetone is relatively stable in air. Thus, it is commonly found in air samples, especially from indoor and urban locations. Expired human breath typically contains acetone as do human urine and blood (IPCS 1998). Acetone has also been detected in surface, ground, rain, waste and drinking water samples, and in soil samples associated with landfill facilities (IPCS 1998).

A large percentage (97%) of the acetone released during its manufacture or use goes into the air. In air, about one-half of the total amount breaks down as a result of sunlight or other chemicals every 22 days. It moves from the atmosphere into the water and soil by rain and snow. It also moves quickly from soil and water back to air. Acetone does not bind to soil or build up in animals. It is broken down by microorganisms in soil and water. It can move into groundwater from spills or landfills. Acetone is broken down in water and soil, but the time required for this to happen varies (ATSDR 1994).

16.6 Bioaccumulation

Once released to aquatic environments, acetone will tend to volatilize, but a significant portion will remain in the water column. Thus, the principal route of exposure to this substance is through direct contact and ingestion of contaminated waters. As the substance has a low bioaccumulative potential, food sources are unlikely to contribute significantly to the exposure.

16.7 Effects on Aquatic and Terrestrial Organisms

The acute toxicity of acetone to aquatic organisms (fish, daphnia, shrimp) is in the range of 5,000 to 20,000 mg/L. One 48 h LC50 (median lethal concentration) value for *Daphnia magna* was reported at 10 mg/L (Verschueren 1996). Other studies have reported the acute toxicity of acetone to aquatic invertebrates in the 10 to 50 mg/L range over a 24 h exposure (Dowden and Bennett 1965; Snell *et al.* 1991).

In mammals, kidney, liver, and nerve damage, increased birth defects, and lowered ability to reproduce (males only) occurred in animals exposed to acetone for extended periods of time (ATSDR 1994). Animals given large amounts of acetone for short periods of time had bone marrow hypoplasia (fewer new cells being made), degeneration of kidneys, heavier than normal livers, and bigger liver cells. Pregnant mice exposed to acetone through diet had lower body weights and produced fewer newborn mice. The newborns of mice that had swallowed acetone had a higher mortality rate. Male rats that swallowed or drank even small amounts of acetone had anemia and kidney disease. The rats also had abnormal sperm. Acetone caused effects on their nervous systems (ATSDR 1994).

Inhalation studies conducted on six female rats (per group) observed an LC50 of 50.1 mg/L over 8 h of inhalation exposure. The LD50 (median lethal dose) for mice was found to be 3,000 mg/kg/BW (EUCLID 1996).

Acetone does not cause skin cancer in animals when it is applied to their skin. The Department of Health and Human Services and the International Agency for Research on Cancer have not classified acetone for carcinogenic effects. The USEPA has determined that acetone is not classifiable as to its human carcinogenicity (ATSDR 1994).

16.8 References

ATSDR (Agency for Toxic Substances and Disease Registry). 1994. Toxicological profile for acetone. United States Department of Health and Human Services. Public Health Service. Atlanta, Georgia.

Dowden, B.F. and H.O. Bennett. 1965. Toxicity of selected chemicals to certain animals. Journal of the Water Control Federation 37(9):1308-16.

IPCS (International Programme on Chemical Safety). 1998. Environmental Health Criteria 207. Acetone. World Health Organization. Geneva, Switzerland.

EUCLID (International Uniform Chemical Information Database). 1996. Data Sheet. CD-Rom European Chemicals Bureau. Acetone. Feb 1996. 79 pp.

Snell, T.W., B.D. Moffat, C.Janssen, and G.Persoone. 1991. Acute toxicity tests using rotifers IV. Effects of cyst age, temperature, salinity on the sensitivity of *Barachionus calyciflorus*. Ecotoxicology and Environmental Safety 21(3):308-17.

Verschueren, K. 1996. Handbook of environmental data on organic chemicals. 3rd Edition. Van Nostrand Reinhold Co. New York, New York. (As cited in HSDB 2000)

Weast, R.C. (Ed). 1987. Handbook of chemistry and physics, 67th Edition. CRC Press. Boca Raton, Florida. p 51. (As cited in IPCS 1998)

Appendix A2-17 An Overview of the Environmental Fate and Effects of Organochlorine Pesticides: Aldrin and Dieldrin

17.1 Aldrin

17.1.1 Identity

Aldrin (309-00-2) is an organochlorine pesticide whose pure form is a white powder with a mild chemical odor. The technical compound is light tan to brown solid or powder. It has a molecular weight of 364.9. The technical grade aldrin contains 95% active ingredient (Ashworth *et al.* 1970). Aldrin is also known as Alarite, Aldrec, Aldrex, Aldrine, Aldrosor, Algran, Compound 118, Aldrite, Drinox, HHDN [(1R,4S,4aS,5S,8R,8aR)-1,2,3,4,10,10-hexachloro-1,4,4a,5,8,8a-hexahydro- 1,4:5,8-dimethanonaphthalene)], Octalene, Seedrin Liquid, and Soildrin (McNeely *et al.* 1979).

17.1.2 Uses

Aldrin has been used as a soil insecticide to control root worms, beetles, and termites. Its original use included control of soil, fruit, and vegetable pests. Specific targets included grasshoppers, locusts, and termites. Aldrin's current use in the United States is restricted to those situations in which there is no effluent discharge, e.g. in ground injection for termite control (USEPA 1980).

In 1974, the USEPA banned most of the uses of aldrin due to its suspected carcinogenicity. Ultimately, all uses on food crops were banned. The use of aldrin as a subterranean termiticide continued after 1974, but the sole importer ceased importation in 1985 and cancelled its registration in 1987. All other termiticide registrations of aldrin have been either cancelled or suspended. Two minor uses that were still allowed, mothproofing in manufacturing processes and dipping roots and

tops of non-food plants, have been voluntarily cancelled by industry. In 1981, a labeling improvement program (LIP) was initiated by the USEPA to attempt to avoid errors of misapplication to buildings for termite control. In accordance with an agreement with the United States Department of Agriculture (USDA) and the Food and Drug Administration (FDA), the USEPA is currently reevaluating recommended tolerances for unavoidable residue levels of aldrin or dieldrin in food products.

17.1.3 Sources

Since aldrin is not currently produced or imported in the United States, its use and release into the environment is minimal. Possible new releases may come from the use of old stockpiles for the underground control of termites. Aldrin is applied to soil and vegetation by injection or aerial spraying. Leaching of aldrin is thought to be minimal, with soil erosion and sediment transport the major pathways for entering the aquatic environment (USEPA 1980).

17.1.4 Physical and Chemical Properties

Aldrin has a water solubility of 27 µg/L at 25/C (Park and Bruce 1968). Its log K_{ow} (octanol/water partition coefficient) is quite high at 6.9 (Hansch *et al.* 1995). Aldrin's vapor pressure is 3.1 mPa at 25/C (Martin 1972).

17.1.5 Environmental Fate and Transport

Aldrin is rapidly transformed to dieldrin in the environment (USEPA 1979, 1980). One study reported that 60% of aldrin added to river water has dissipated after four weeks of incubation (Eichelberger and Lichtenberg 1971). Trace concentrations of 1 to 2 ng/L were found in rain and snow (Strachan and Huneault 1979). Some studies report concentrations of aldrin in surface waters ranging from 0.1 to 85 ng/L (Lichtenberg *et al.* 1970; USEPA 1976; USEPA 1982).

Biotransformation, volatilization, bioaccumulation, and indirect photolysis play significant roles in the removal of aldrin from the water column (USEPA 1979). Sunlight and bacteria change aldrin to dieldrin. As a consequence, one mostly finds dieldrin in environmental samples. Aldrin binds tightly to soil and slowly evaporates into the air. Plants take up aldrin from soil and store it in their tissues, where it is rapidly transformed to dieldrin.

Hydrolysis is not expected to be an important process in the aquatic environment. Although very few data are available, it is expected that sorption processes play a relatively minor role (USEPA 1979). In general, soil sorption coefficients are small (400; Kenaga and Goring 1978).

The half-life of aldrin in a sample of natural water in the presence of sunlight is about 24 h (Singmaster 1975). Studies with non-aqueous systems showed that aldrin is converted to photoaldrin (Rosen and Carey 1968; Ivie and Casida 1971). The photolysis of aldrin in sterile paddy water yielded 25% dieldrin in 36 h (Ross and Crosby 1975).

Laboratory measures of volatilization under simulated wind and temperature conditions have yielded volatilization half-lives ranging from as short as 0.4 h (Singmaster 1975) to a maximum of 7.7 days (Mackay and Wolkoff 1973; Mackay and Leinonen 1975).

17.1.6 Bioaccumulation

In terms of exposure potential, some of the released aldrin will be available for direct uptake from the aquatic environment, but given its affinity for sediment adsorption, most will partition to sediments. Thus, the principal route of exposure to aldrin would be through a direct contact with, and ingestion of, sediment, soil, and through trophic transfer of the contaminant through consumption of benthic and soil organisms.

Short term bioconcentration factors measured in terrestrial-aquatic microcosm studies range from 103 to 104 (Metcalf *et al.* 1973). Biomagnification is not considered to be important because aldrin is rapidly converted to dieldrin in aquatic biota (USEPA

1979). Biotransformation appears to be the most important process governing the fate of aldrin in the aquatic environment (USEPA 1979). The transformation process occurs in virtually all organisms, from microbes, algae, invertebrates, fish to birds and mammals (Rosenblatt *et al.*1975; Sanborn *et al.*1977). A biological half-life of 7 dayshas been reported for aldrin in the Atlantic salmon (*Salmo salar*; Addison *et al.* 1976).

17.1.7 Effects on Aquatic and Terrestrial Organisms

Aldrin is highly toxic to aquatic organisms. Acute exposures to a variety of fish species (salmon, trout, minnow, catfish, bass, and bluegill) have resulted in lethal concentrations in the range of 2 to 50 µg/L. Invertebrates, such as *Daphnia magna* have experienced adverse effects (EC50; median effect concentration) at concentration ranges of 20 to 30 µg/L (HSDB 2000a). As aldrin rapidly transforms to dieldrin in the environment, aquatic and sediment toxicity for dieldrin may be considered for this substance. See the dieldrin portion of this appendix for more information.

Aldrin is moderately to highly toxic to mammals. Acute oral LD50 (median lethal dose) ranges from 38 to 67 mg/kg (HSDB 2000a). Studies with animals fed aldrin have shown that the liver can be damaged and the ability of the immune system to protect against infections can be suppressed. It is thought to exert its hepatotoxicity by increasing the activity of microsomal biotransformation enzymes. This appears to be associated with the occurrence of reversible hypertrophy of centrilobular hepatocytes with cytoplastic changes and hepatomegaly in the liver of rodents (IPCS 1989). Goats administered 50 mg aldrin/kg body weight showed mild degenerative changes, congestion, and petechial hemorrhages in various organs. In the kidneys, degenerative changes of the proximal convoluted tubules were found (IPCS 1989). Cats fed aldrin at 1 mg/kg/day or made to inhale 0.1 µg/L of air had marked lowering of conditioned reflexes and of unconditioned food and orientation reflexes, which required up to 8 days to return to normal (NRC 1977). Acute symptoms were observed in ducks, pheasants, and bobwhite quail following oral exposure, including ataxia, low carriage, nictitating membrane closed for long periods, fluffed feathers, tremors, phonation, violent wing-beat convulsions, seizures, and opisthotonos. Death

occurred ½ hours to 10 days post-treatment. Weight losses occurred among survivors of higher levels. Gross autopsies revealed occasional liver adhesions to parietal peritoneum (USFWS 1970)

For chronic exposures, groups of male and female Osborne-Mendel rats were fed diets containing 0, 0.5, 2, 10, 50, 100, or 150 ppm recrystallized aldrin for 2 years. Considering together the groups given 0.5, 2, or 10 ppm (i.e., the groups showing survival rates at 2 yr comparable to those of controls), number of tumor-bearing animals was 25/60 compared with 3/17 controls. Among those treated, 12 developed lymphomas (9 located in lungs), 13 had mammary tumors (malignant in 4 rats), 2 had fibrosarcomas and 3 had tumors at other sites (IARC 1974). Microscopic exam of costochondral junction of goats after chronic aldrin intoxication revealed drastic reduction in width of proliferating, maturing and degenerating cartilage cells (Singh and Jha 1982). Administered in single, 50 mg/kg doses to hamsters during the period of organogenesis, high incidence of fetal deaths, congenital abnormalities, and growth retardation were observed (ACGIH 1991).

The Environmental Protection Agency (EPA) considers aldrin a probable carcinogen, as a result of animal testing. Further, aldrin is classified as a probable human carcinogen. This is based on the observations where orally-administered aldrin produced significant increases in tumor responses in three different strains of mice in both males and females. Tumor induction has been observed for structurally related chemicals, including dieldrin, a metabolite of aldrin (USEPA 2000).

17.2 Dieldrin

17.2.1 Identity

Dieldrin (CAS; Chemical Abstracts Service Number 60-57-1) is an organochlorine insecticide that is closely related structurally and chemically to aldrin. Pure dieldrin is a white powder with a mild chemical odor. The less pure commercial powders have a tan color. Dieldrin is also known as HEOD (1,2,3,4,10,10-hexachloro-6,7-epoxy-1,4,4a,5,6,7,8,8a-octahydro-endo-1,4-exo-5,8-dimethanonaphthalene),

Compound 497, Octalox, Insecticide no.497, ENT 16225, Alvit, Dieldrex, Dieldrite, and Panoram D31 (USEPA 1979; McNeely *et al.* 1979; Windholtz *et al.* 1983; Agriculture Canada 1984). Its toxicity does not differ significantly from that of aldrin.

17.2.2 Uses

Dieldrin has been used in agriculture for soil and seed treatment and in public health to control disease vectors such as mosquitoes and tsetse flies. Dieldrin has also had veterinary use as a sheep dip and has been used in treatment of wood and mothproofing of woolen products (Marth 1965).

Dieldrin used to be one of the most widely used domestic pesticides (Lykken 1971; Waldbott 1978; USEPA 1980). The original uses of dieldrin were as a pesticide for control of soil, fruit, and vegetable pests, as well as for control of grasshoppers, locusts and termites. However, its use was restricted in the United States in 1974 to those situations in which there is no effluent discharge (USEPA 1980). The United States no longer manufactures dieldrin as a result of a ban in 1974, but instead imports the insecticide from the Shell Chemical Company which manufactures it in Holland (USEPA 1980).

17.2.3 Sources

The pathways for environmental contamination by dieldrin include atmospheric dispersion, wind and water erosion of soil, and transport while sorbed onto soil particles in the silt of streams and lakes. Dieldrin can also move through the environment as residues in plants and animals, especially in fish and wildfowl (Lykken 1971).

Dieldrin is everywhere in the environment, but at very low levels. Since its use was banned, most foods contain very little, if any, dieldrin. Air, surface water, or soil near waste sites may contain higher levels.

17.2.4 Physical and Chemical Properties

Dieldrin has a melting point of 175 to 176/C and a vapor pressure of 400 µPa at 20/C (Worthing 1983). The pesticide does not dissolve in water to a great extent (0.186 mg/L at 20/C; Park and Bruce 1968). Also, it tends to be hydrophobic (log K_{ow} of 4.55; Brooke *et al.* 1986).

17.2.5 Environmental Fate and Transport

Dieldrin is considered to be persistent in the environment. Sorption volatilization and bioaccumulation are the important processes determining its fate (USEPA 1979). Dieldrin binds to soil particles and slowly evaporates into the air. Although dieldrin is persistent in soil, environmental background levels are known to be decreasing slowly. Plants take up dieldrin from the soil and store it in their tissues. When animals take up dieldrin, it is stored in the fat and leaves the body very slowly.

The hydrolysis of dieldrin in the aquatic environment is very slow (Eichelberger and Lichtenberg 1971). However, sorption to sediments containing organic matter is appreciable. An organic carbon partition coefficient (K_{oc}) of approximately 104 at 15/C has been reported (Weil *et al.* 1973). Experimental studies suggest that direct photolysis of dieldrin does occur; its photolytic half-life is approximately 2 months (Henderson and Crosby 1968). Volatilization of dieldrin from aquatic systems is also an important removal process. Half-lives in the order of a few hours to a few days have been determined from laboratory experiments (Singmaster 1975).

Sorption, volatilization, and bioaccumulation are the important processes determining its fate (USEPA 1979). Dieldrin binds to soil particles and slowly evaporate into the air. Although dieldrin is persistent in soil, environmental background levels are known to be decreasing slowly. Plants take up dieldrin from the soil and store it in their tissues. When animals take up dieldrin, it is stored in the fat and leaves the body very slowly.

17.2.6 Bioaccumulation

Some of the released dieldrin will be available for direct uptake from the aquatic environment, but given its affinity for sediment adsorption, most dieldrin will partition to sediments. The principal route of exposure to dieldrin would be through direct contact with, and ingestion of, soils and sediments, and through trophic transfer of the contaminant through consumption of benthic and soil organisms. Thus, the most likely route of exposure to dieldrin is through the consumption of contaminated food and drinking water.

Dieldrin may be bioaccumulated by various organisms in the aquatic environment. Bioconcentration factors ranging from 102 to 104 for bacteria (Grimes and Morrison 1975) and averaging 104 for freshwater algae (Neudorf and Khan 1975) have been reported. Data from microcosm experiments also suggest significant bioaccumulation (Sanborn and Vu 1973; Metcalf *et al.* 1973). Bioconcentration factors of 102 to 103 for algae, 104 to 105 for snails, and 103 for fish were reported. Biological half-lives in fish vary from 7 days in bluegill sunfish (*Lepomis macrochirus*; Gakstatter and Weiss 1967) to 40 days in rainbow trout (*Salmo gairdneri*; Macek *et al.* 1970). Very little microbial biotransformation of dieldrin occurs in the aquatic environment (Bohonos and Francis 1975; Sanborn *et al.* 1977).

17.2.7 Effects on Aquatic and Terrestrial Organisms

Dieldrin has also shown to be highly toxic to aquatic organisms. Acute exposures to a variety of fish species (salmon, trout, minnow, catfish, bass, bluegill) have resulted in lethal concentrations in the range of 1 to 20 µg/L (USEPA 2001). Dieldrin has also been observed to illicit adverse effects to benthic organisms, such as reduced populations and species richness. Jaagumagi (1988) and Jaagumagi *et al.* (1989) reported significant decreases in abundance of benthic organisms at sites in Humber Bay, Lake Huron compared to reference sites. Toxicity to the marine amphipod, *Rhepoxynius abronius* was observed at a concentration of 1.17 µg/g from sediments from Puget Sound, WA (Pastorok and Becker 1990). The USEPA (Ingersoll 1995) published proposed freshwater quality criterion of 0.00625 µg/L and sediment quality criterion (at 1% OC; organic carbon) of 0.166 µg/g.

Dieldrin is moderately to highly toxic to mammals. Acute oral LD50 for rats has been reported in the range of 24 to 87 mg/kg, and in rhesus monkeys, as low as 3 mg/kg (HSDB 2000b). Studies with animals fed dieldrin have shown that the liver can be damaged and the ability of the immune system to protect against infections can be suppressed. An oral exposure study fed 1,500 CF1 mice at concentrations of 0.1 to 10 ppm dieldrin. Fifty percent of the mice fed dieldrin at 10 ppm were dead at 15 months and 50% in the other groups were at 20 months. Statistically-significant and dose-related increases in liver tumors occurred in dieldrin-exposed mice in both sexes. Incidences of pulmonary adenomas and pulmonary carcinomas in males and females exposed to dieldrin at 0.1 and 1 ppm were increased above those in controls (Walker *et al.* 1972). Dieldrin at 1, 2, or 4 mg/kg/day produced hyperplastic goiters in the thyroids of pigeons. Visual exam indicated that the thyroids were significantly enlarged and microscopic exam revealed small follicles with decreased amount of colloid, epithelial hyperplasia, and vascular congestion (NIOSH 1978). Dieldrin affects the central nervous system. It inhibits gamma amino butyric acid-induced chloride ion uptake into skeletal muscles and the binding of tritiated dihydropicrotoxinin (anion channel probe) to the membrane. This results in central nervous system excitation and convulsions due to the blocking of gamma amino butyric acid transmitter (IPCS 1989). Ingesting moderate levels of dieldrin over a longer period may also cause convulsions as dieldrin builds up in tissues. Dieldrin also caused immunosuppression in mice. Levels of 1 or 5 mg dieldrin/kg diet were fed to BALB/c mice for 3.5 or 10 weeks, this resulted in decreased antibody formation to PVP, a T-independent antigen (IPCS 1989).

Lifetime feeding studies were conducted with Syrian golden hamsters. Groups of nearly equal size (i.e. 32 to 41 per group) of male and female hamsters were fed a diet containing 0, 20, 60, or 80 mg/kg for up to 120 weeks at which time the remaining survivors were killed. While there was no decrease in survival at 50 weeks, the numbers of females remaining at 70 weeks was one-half or less than that of the males. At 90 weeks the survival rate was about 10% for all groups except the males of the 180 mg/kg level which had 32% survivors. Both males and females at the low and high doses demonstrated a marked retardation of growth and it was also noted that there was a dose-related increase in the incidence of hepatic cell hypertrophy in the dieldrin-treated hamsters (USEPA 1980). Purified dieldrin was administered at concentrations between 0.08 and 40 ppm in the diet to Wistar rats for up to 2 years.

Nonspecific neural lesions, cranial edema, convulsions, and dieldrin residues in the brain were reported in most exposed rats (NIOSH 1978).

In a reproductive toxicity study, 39 to 140 day old female Wistar rats were fed dieldrin at 2.5 to 10.0 ppm in the diet. Parental mortality and reduced fecundity was noted at 10 ppm. Convulsions in pups was observed at 2.5 ppm (NIOSH 1978). In a teratogenic study, dieldrin was administered in doses of 1.5, 3.0, and 6.0 mg/kg/day on days 7 to 16 of gestation, to DCI mice and CD rats. In mice, the highest dose produced an increased percentage of supernumerary ribs and a decrease in the number of caudal ossification centers (Clayton and Clayton 1994).

Oral doses of dieldrin have caused liver cancer in mice, but not in rats. The Environmental Protection Agency (EPA) considers dieldrin a probable carcinogen based on experiments with animals. A series of experiments on mice involving continuous feeding of recrystallized (>99% pure) dieldrin was found to produce liver-cell tumors, while the incidence of tumors at other sites was either unaffected or decreased in relation to the shorter life span of animals with liver tumors (IARC 1974).

17.3 References

ACGIH (American Conference of Governmental Industrial Hygienists, Inc.). 1991. Documentation of the threshold limit values and biological exposure indices. 6th Edition. Volumes I, II, III. Cincinnati, Ohio.

Addison, R.F., M.E. Zinck, and J.R. Leahy. 1976. Metabolism of single and combined doses of 14C-aldrin and 3H-p,p'-DDT by Atlantic salmon (*Salmo salar*) fry. Journal of Fisheries Research Board of Canada 33:2073-2076.

Agriculture Canada. 1984. Compendium of pest control products registered in Canada. Registered pest control products. Volume Code: RP. Publication Number1654 RP184. Pesticides Division. Plant Health and Plant Products Directorate. Ottawa, Ontario.

Ashworth, R. de B., J. Henriet, and J.F. Lovett. 1970. CIPAC Handbook. Volume 1. Analysis of Technical and Formulated Pesticides. G.R. Raw (Ed.). Collaborative International Pesticides Analytical Council Limited. W. Heifer and Sons Ltd. Cambridge, England. 1079 pp.

Bohonos, N. and A.J. Francis. 1975. Microbiological degradation of military standard pesticide formulations. Final Report. SRI (Stanford Research Institute) to the United States Army Medical Research Development Command. Contract No. DADA17-73-C-3124. (As cited in USEPA 1979.)

Brooke, D.N., A.J. Dobbs, and N. Williams. 1986. Octanol:water partition coefficients (P): Measurement, estimation, and interpretation, particularly for chemicals with P>E+5. Ecotoxicology and Environmental Safety 11:251-60.

Clayton, G.D. and F.E. Clayton (Eds.). 1994. Patty's industrial hygiene and toxicology. Volumes 2A, 2B, 2C, 2D, 2E, 2F: Toxicology. 4th edition. John Wiley & Sons Inc. New York, New York.

Eichelberger, J.W. and J.J. Lichtenberg. 1971. Persistence of pesticides in river water. Environmental Science and Technology 5:541-544.

Gakstatter, J.H. and C.M. Weiss. 1967. The elimination of 14C-DDT and 14C-dieldrin from fish following a single sublethal exposure in aquaria. Transactions of the American Fisheries Society 96:201-207.

Grimes, D.J. and Morrison, S.M. 1975. Bacterial bioconcentration of chlorinated hydrocarbon insecticides from aqueous systems. Microbial Ecology 2:43-59.

Hansch, C., A. Leo, and D. Hoekman. 1995. Exploring QSAR - hydrophobic, electronic, and steric constants. American Chemical Society. Washington, District of Columbia. p. 96.

Henderson, G.L. and D.G. Crosby. 1968. The photodecomposition of dieldrin residues in water. Bulletin of Environmental Contamination and Toxicology 3:131-134.

HSDB (Hazardous Substances Databank). 2000a. Profile for Aldrin. National Library of Medicine. Bethesda Maryland.

HSDB (Hazardous Substances Databank). 2000b. Profile for Dieldrin. National Library of Medicine. Bethesda Maryland.

IARC (International Agency for Research on Cancer). 1974. Monographs on the evaluation of the carcinogenic risk of chemicals to man. World Health Organization. Geneva, Switzerland. 1972-present. (Multivolume work). Volume 5

Ingersoll, C.G. 1995. Sediment Tests. *In:* Fundamentals of Aquatic Toxicology, Second Edition, G.M. Rand, ed. Taylor and Francis, Washington, D.C.

IPCS (International Program on Chemical Safety). 1989. Environmental Health Criteria 91: Aldrin and dieldrin. p. 217.

Ivie, G.W. and J.E. Casida. 1971. Sensitized photodecomposition and photosensitizer activity of pesticide chemicals exposed to sunlight on silica gel chromatoplates. Journal of Agriculture and Food Chemistry 19:405-409. (As cited in USEPA 1979).

Jaagumagi, R. 1988. The in-place pollutants program. Volume V, Part B. Benthic invertebrates studies results. Ontario Ministry of the Environment, Water Resources Branch, Aquatic Biology Section, Toronto, Ontario.

Jaagumagi, R., D. Persaud, and T. Lomas. 1989. The in-place pollutants program. Volume V, Part A. A synthesis of benthic invertebrate studies. Ontario Ministry of the Environment, Water Resources Branch, Aquatic Biology Section. Toronto, Ontario.

Kenaga, E.E. and C.A.I. Goring. 1978. Relationship between water solubility, soil-sorption, octanol-water partitioning and bioconcentration of chemicals in biota. *In:* Proceedings of the American Society of Testing Materials. 3rd Aquatic Toxicology Symposium. New Orleans, Louisiana. p. 63.

Lichtenberg, J.J., J.W. Eichelberger, R.C. Dressman, and J.E. Longbottom. 1970. Pesticides in surface waters of the United States - A five-year summary, 1964-1968. Pesticides Monitoring Journal 4:71-86. (As cited in USEPA 1980).

Lykken, L. 1971. Chemical control of pests. Chemistry 44:18-21.

Macek, K.J., C.R. Rodgers, D.L. Stalling, and S. Korn. 1970. The uptake, distribution and elimination of dietary 14C-DDT and 14C- dieldrin in rainbow trout Transactions of the American Fisheries Society 99:689-695.

Mackay, D. and A.W. Wolkoff. 1973. Rate of evaporation of low-solubility contaminants from water bodies to atmosphere. Environmental Science and Technology 7:611-614.

Mackay, D. and P.J. Leinonen. 1975. Rate of evaporation of low-solubility contaminants from water bodies to atmosphere. Environmental Science and Technology 9:1178-1180.

Marth, E.H. 1965. Residues and some effects of chlorinated hydrocarbon insecticides in biological material. Residue Reviews 9:1-89.

Martin, H. 1972. Pesticide manual. 3rd edition. British Crop Protection Council. Worcester, England.

McNeely, R.N., V.P. Neimanisand, and L. Dwyer. 1979. Pesticides. *In:* Water Quality Sourcebook. A Guide to Water Quality Parameters. Water Quality Branch. Inland Waters Directorate. Environment Canada. Ottawa, Ontario. pp. 34-45.

Metcalf, R.L., I.P. Kapoor, P.- V. Lu, C.K. Schuth, and P. Sherman. 1973. Model ecosystem studies of the environmental fate of six organochlorine pesticides. Environmental Health Perspectives 4:35-44.

Neudorf, S. and M.A.Q. Khan. 1975. Pickup and metabolism of DDT, dieldrin and photoaldrin by a freshwater alga (*Ankistrodesmus amalloides*) and a microcrustacean (*Daphnia pulex*). Bulletin of Environmental Contamination and Toxicology 13:443-450.

NIOSH. 1978. Special occupational hazard review: Aldrin/dieldrin p. 46-47 DHEW Publication NIOSH 78-201 (As cited in HSDB 2000).

NRC (National Research Council). 1977. Drinking water and health: Volume 1. National Academy Press. Washington, District of Columbia.

Park, K.S and W.N. Bruce. 1968. The determination of the water solubility of aldrin, dieldrin, heptachlor and heptachlor epoxide. Journal of Economic Entomology 61:770-774.

Pastorok, R.A. and D.S. Becker. 1990. Comparative sensitivity of sediment toxicity bioassays at three Superfund sites in Puget Sound. *In:* Aquatic toxicology and risk assessment, W.G. Landis and W.H. van der Schalie, eds. American Society for Testing and Materials.

Rosen, J.D. and W.F. Carey. 1968. Preparation of the photoisomers of aldrin and dieldrin. Journal of Agriculture and Food Chemistry 16:536-537. (As cited in USEPA 1979).

Rosenblatt, D.H., T.A. Miller, J.C. Dacre, I. Muul, and D.R. Cogley (Eds.). 1975. Appendix K: Aldrin/dieldrin. *In:* Problem Definition Studies on Potential Environmental Pollutants. II. Physical, Chemical, Toxicological, and Biological Properties of 16 Substances. Technical Report 7509. United States Army Medical Bioengineering Research and Development Laboratory. Frederick, Maryland. 290 pp.

Ross, R.D. and D.G. Crosby. 1975. The photo oxidation of aldrin in water. Chemosphere 4:277-282.

Sanborn, J.R. and C. Vu. 1973. The fate of dieldrin in a model eco-system. Bulletin of Environmental Contamination and Toxicology 10:340-346.

Sanborn, J.R., B.M. Francis, and R.L. Metcalf. 1977. The degradation of selected pesticides in soil: A review of published literature. EPA/600/9-77/022. United States Environmental Protection Agency. Washington, District of Columbia. (As cited in USEPA 1979).

Singh K.K., and G.J. Jha. 1982. Bone growth during chronic aldrin intoxication in goats. Research in Veterinary Science 32 (3):283-8.

Singmaster, J.A., III. 1975. Environmental behavior of hydrophobic pollutants in aqueous solutions. Ph.D. Thesis. University of California. Davis, California. University Microfilms. Ann Arbor, Michigan. Orcjer No.76-14, 237. [Diss. Abstr. Int. B 1976, 36 (12, pt. 1):6206-6207; As cited in USEPA 1979].

Strachan, W.M.J. and H. Huneault. 1979. Polychlorinated biphenyls and organochlorines in the Great Lakes region. Journal of Great Lakes Research 5:61-68.

USEPA (United States Environmental Protection Agency). 1976. National interim primary drinking water regulations. EPA-570/9-76-003. (As cited in USEPA 1980.)

USEPA (United States Environmental Protection Agency). 1979. Aldrin. *In:* Water-related environmental fate of 129 priority pollutants. Volume I. Introduction, Technical Background, Metals and Inorganics, Pesticides, Polychlorinated Biphenyls. EPA-440/4-79- 029a. Office of Water Planning and Standards. Washington, District of Columbia. pp. 21-1 to 21-13; 26-1 to 26-12.

USEPA (United States Environmental Protection Agency). 1980. Ambient water quality criteria for aldrin/dieldrin. EPA-440/5-80-019. Office of Water Regulations and Standards. Criteria and Standards Division. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1982. STORET. Storage and retrieval of water quality data: A computerized information system. Washington, District of Columbia. (As cited in Strachan and Edwards 1984.)

USEPA (United States Environmental Protection Agency). 2000. Integrated risk information system (IRIS) on aldrin (309-00-2) (Obtained from from: http://www.epa.gov/ngispgm3/iris).

USEPA (United States Environmental Protection Agency). 2001. Ecotox Database System. Washington, District of Columbia. (Obtained from http://www.epa.gov/ecotox/).

USFWS (United States Fish and Wildlife Service), Bureau of Sport Fisheries and Wildlife. 1970. Handbook of toxicity of pesticides to Wildlife. United States Government Printing Office. Washington, District of Columbia.

Waldbott, G.L. 1978. Health effects of environmental pollutants. The C.V. Mosby Co. St. Louis, Missouri. 316 pp.

Walker, A.I.T., Thorpe E., and Stevenson D.E. 1972. The toxicology of dieldrin (HEOD). I. Long-term oral toxicity studies in mice. Food and Cosmetic Toxicology 11: 415-432

Weil, L., G. Dure, and K.E. Quentin. 1973. Adsorption of chlorinated hydrocarbons to organic particles and soils. Zeitschrift Fur Wasser Und Abwasser Forschung. 6:107-112.

Windholtz, M., S. Budavari, R.F. Blumetti, and E.S. Otterbein (Eds.). 1983. The Merck Index. An Encyclopedia of Chemicals, Drugs and Biologicals. 10th edition. Merck and Co., Inc. Rahway, New Jersey.

Worthing, C.D. (Ed). 1983. The Pesticide Manual: A World Compendium, 7th ed. 1983. The British Crop Protection Council, Croydon, England. 695 pp.

Appendix A2-18 Profiles for Wildlife Species that are Part of the Calcasieu Estuary Ecosystem

18.1 Wildlife Species that are Proposed Receptors at Risk

18.1.1 Non-Perching Birds

Order Ciconiiformes
Family Ardeidae
Great egret (*Casmerodius albus*)

The great egret can be found in Maine and southern Canada, as far west as the Great Lakes. In the southern United States, they are found along the Atlantic coast, Florida, the Gulf Coast, and Texas (CDEP 2000). The great egret is a large bird with white plumage, a long thin body, a yellow bill, and legs and feet that are glossy black. The sexes are similar in size, males being slightly heavier (CDEP 2000). Adults average 81 cm in length with a 140 cm wingspan and weigh between 2 and 2.5 kg (Knopf 1977; USGS 2000). This heron is larger than any other except for the great blue heron. In flight, the great egret holds its neck in a more open S than do other white herons.

The preferred habitat of great egrets is along freshwater and saltwater marshes, ponds, streams, lakes, wooded swamps, mud flats, and urban environments (Ehrlich *et al.* 1988). Great egrets are admirable fishermen, standing motionless in the water waiting for their prey. Their diet primarily consists of aquatic invertebrates and fish. But, they are also known to eat reptiles, amphibians and small mammals (USGS 2000). Great egrets forage very similarly to great blue herons; walking slowly through shallow water and snapping up prey as it crosses their path. Young are usually fed frogs, crayfish, and small fish that are regurgitated into their mouths by a parent (Erwin 1985).

Family Threskiornithidae
Roseate spoonbill (*Ajaia ajaja*)

The roseate spoonbill is a pink and white bird with scarlet tinted wings. It has a white neck with red eyes and legs. Its tail feathers are grey and green and its head is bald. A distinctive feature of the roseate spoonbill is its 15 to18 cm dark grey bill, which is shaped like a spatula (University of Michigan 2001; USGS 2000). Roseate spoonbills average 71 cm in length with a 135 cm wingspan. Both sexes are similar in weight, ranging from 1.2 to 1.8 kg (USGS 2000). Roseate spoonbills are year round inhabitants of southwestern Louisiana and can also be found along the coastlines of Florida, Texas, and Central and South America (eNature Field Guide 2001; McLaren/Hart-Chemrisk 1998). Their distribution is rare and local.

Roseate spoonbills prefer estuarine environments. Mudflats edged by mangrove forests are ideal. They will also inhabit fresh water marshes, shallow lakes, and rivers (eNature Field Guide 2001). During low tide, roseate spoonbills will move on to flats and sandy bars in search of prey. They are tactile feeders, preying almost exclusively on fish and to a lesser extent aquatic invertebrates (USGS 2000). Roseate spoonbills feed in shallow water no greater than 20 cm deep. Swinging their partially open bill back and forth through the water, nerves inside their bill sense prey at which point their bill quickly shuts. Throwing back their head, they swallow their prey (University of Michigan 2001). This bird is usually silent and flies with its neck outstretched.

White ibis (*Eudocimus albus*)

White ibis breed in the southeastern United States, and live year-round in most of Florida and the Gulf Coast. It is locally abundant in coastal locations. The adult has a red face and bill. The small, black wing tips are usually hidden when at rest. Young birds show their white rumps when flying, and from below, the dark neck contrasts with the white belly. Ibis are often seen in large flocks flying in long lines or in a "V" formation. Its calls are low and harsh grunts and growls (Robbins *et al.* 1983). White ibis predominantly feed on fish and aquatic invertebrates. Other prey items include reptiles and amphibians (USGS 2000).

Order Charadriiformes
Family Laridae
Least tern (*Sterna antillarum*)

Least terns are the smallest North American tern. They are common on the east, west, and Gulf Coasts, but less common in inland areas (McLaren Hart 1996; NOAA 2001). Interior populations (more than 80 km from the coast) are listed as endangered by the state of Louisiana and the United States. They average 22 cm in length and have a wingspan of 51 cm. Body weight ranges from 39.0 to 47.6 g with a mean of 43.1 g (standard deviation; SD=2.12 g; Dunning 1984). Distinctive characteristics of these birds include their very rapid wingbeats, short legs, and short, forked tail (USGS 2000). Least terns have a black cap, a white forehead, a yellow bill, and yellowish legs. The call is a rapid series of paired notes.

Least terns are a colonial ground nesting species. They prefer nesting on nearly bare ground, such as beaches and sandbars. Shallow water that is close by is also necessary (USGS 2001a). Least terns feed almost exclusively on fish, but also on lesser quantities of aquatic invertebrates (USGS 2000). Least terns dive for fish in water close to beaches or in quiet ponds and bays.

Forster's tern (*Sterna forsteri*)

It was not until 1831 that these terns were recognized as a species separate from the common tern. These medium sized terns have a grey back and wings. They also have a black cap which turns white in the winter, leaving black marks behind the eyes. They have orange legs and an orange bill with a black tip. The sexes are similar in appearance and size. Forster's terns average 36 cm in length and have 76 cm wingspans. Their body weight ranges from 127 to 193 g with a mean of 158 g (SD=16.8 g; Dunning 1984). These birds range from their breeding grounds along the Atlantic coast from Massachusetts to Texas and, in the west, from Alberta to California, to their wintering grounds along the coasts of California and Virginia, and southward (USGS 2000; eNature Field Guide 2001). Forster's terns are summer residents in the Calcasieu region (McLaren/Hart-Chemrisk 1998).

Forster's terns prefer to inhabit fresh and salt water marshes. It is in these environments that they feed on fish, crustaceans, aquatic insects, and mollusks. These terns often catch their prey by aerial diving. Forster's terns are colonial nesters. They have 1 brood each year, consisting of 2 to 3 buff, spotted eggs (McLaren/Hart-Chemrisk 1998; USGS 2000; eNature Field Guide 2001).

Family Scolopacidae
Spotted sandpiper (*Actitis macularia*)

The spotted sandpiper is a small shorebird commonly found in Northern Alaska, Canada, and the southern United States (USEPA 1993). Coastal areas in the southern United States, including Louisiana, serve as wintering grounds, while northern regions are used for breeding (USEPA 1993; McLaren/Hart-Chemrisk 1998). The average weight of a female sandpiper is 50 g, significantly heavier than their male counterparts, which average 40 g. Also, they average 19 cm in length (USEPA 1993).

Spotted sandpipers inhabit areas along the edges of bodies of water. Inland habitats include lakes, ponds, and rivers. They also inhabit coastal environments, where they search beaches, inlets, and creeks for food (USEPA 1993). They also require open water to bathe in. Sandpipers prefer to nest in semi-open vegetation. Their nests are usually well concealed on the ground, lined with vegetation and hidden by grasses, or among rocks and driftwood. While breeding, sandpipers seek the densest vegetation (USEPA 1993). Sandpipers will walk slowly along the shores of sandy beaches, and the muddy edges of inlets and creeks, picking up food along the way (USEPA 1993). Inland, the sandpiper feeds along the shores of sandy ponds, streams, and mountain torrents. They will sometimes stray into meadows, fields or gardens in agricultural areas, where they find their food in low vegetation or off the ground (USEPA 1993).

Spotted sandpipers generally feed on prey that swim in the first 4 cm of the water column. They feed almost exclusively on small invertebrates, small fish, and both terrestrial and aquatic insects, such as beetles, grasshoppers, and ants. The sandpiper has the ability to capture flying insects, however, it prefers to catch its prey by probing and gleaning it from substrate (USEPA 1993). Spotted sandpipers also have remarkable grasping power in their feet, which they use to perch on small branches

over the water when searching for prey. Young sandpipers begin feeding themselves almost immediately after hatching (USEPA 1993).

Willet (*Catoptrophorus semipalmatus*)

The willet is a pigeon-sized bird. The sexes are very similar in size, females being slightly heavier than males. Willets weigh between 200 and 300 g and average 38 cm in length from bill tip to tail tip (Knopf 1977). Willets have a gray-brown head, neck, back and upper wing. Their wings have a distinctive black and white pattern when in flight (USGS 2000, USGS 2001b). Standing birds are very plain and plump with bluish legs and a long, dark bill. Willets breed locally in Canada, the United States, and the West Indies, wintering in the southern United States and South America (Knopf 1977; USGS 2001b). Willets are year-round inhabitants of the Calcasieu Estuary (McLaren/Hart-Chemrisk 1998).

Good willet habitat includes coastal marshes, sand dunes, mud flats and rocky areas (Hayman *et al.* 1986). Willets nest and feed in separate locations. When nesting, willets often form small, loose, breeding colonies. The female chooses the nesting site, which is often concealed by short, thick vegetation, on a high, dry grassy area along a salt marsh, or on an open beach or flat. The nest itself is usually lined with dry grasses, a few dead rushes, or other materials found nearby (Knopf 1977). Feeding sites for willets are on sandbars, mud flats, and along tidal creeks and pannes of salt marshes.

Willets primarily feed on aquatic invertebrates, as well as lesser quantities of fish. This includes crustaceans, molluscs, marine worms, aquatic insects, and small fish (Ehrlich *et al.* 1988; USGS 2000). Foraging takes place in intertidal areas, salt marshes, sandbars, mudflats, and tidal creeks. The willet catches prey by pecking it from surface water and probing sediment with its bill, as well as by stalking (Hayman *et al.* 1986). The call is a “pill-will-willet.”

Family Recurvirostridae
Black-necked stilt (*Himantopus mexicanus*)

These are medium sized shorebirds that measure between 33 and 40 cm in length. Black-necked stilts are found in the southern and western United States, including the Calcasieu Estuary, and as far south as Peru (Knopf 1977; Hayman *et al.* 1986; McLaren/Hart-Chemrisk 1998). Their bills are long and black and their legs are pink (USGS 2000). The plumage is black above and white below. The wings are black, and the red legs trail far behind the white tail. The call is a monotonous series of loud piping notes.

Habitat preferences of the black-necked stilt include coastal salt marshes, commercial salt pans, inland saltwater and freshwater lakes, mudflats, grassy marshes, and sewage farms. Nesting takes place in small colonies. The nest itself is simply a small depression in the ground (Knopf 1977). The average clutch contains 4 eggs.

Black-necked stilts are visual feeders, foraging primarily for aquatic invertebrates in the top 20 cm of the water column. They also eat fish, reptiles, and amphibians. Prey include, but are not limited to, brine flies, brine shrimp, crayfish, snails, tadpoles, and seeds (Ehrlich *et al.* 1988; USGS 2000).

Order Anseriformes
Family Anatidae
Lesser Scaup (*Aythya affinis*)

The lesser scaup is one of the most abundant bay ducks in North America (USEPA 1995). Their average size is 42 cm from bill tip to tail tip. Males are larger and more colorful than females, who have brown plumage (USEPA 1993). The average body weight of both sexes is 0.815 kg +/- 0.013 (n=39; USEPA 1995).

Over the fall and winter, lesser scaup inhabit large lakes and bays. During the months of spring, they often move to smaller bodies of water (USEPA 1995). Lesser scaup prefer areas with deep water. This is a result of their feeding habits (USEPA 1995). They are omnivores and dive for their food, which primarily consists of aquatic invertebrates and lesser amounts of other plants and animals (USEPA 1995).

Invertebrates in their diet include snails, clams, amphipods, midges, chironomids, and leaches (USEPA 1993). The amount of plant material in their diet, almost exclusively seeds, varies seasonally. When seeds are abundant, they typically constitute a significant portion of their diet (USEPA 1993).

Order Pelacaniformes
Family Pelecanidae
Brown pelican (*Pelecanus occidentalis*)

The brown pelican is listed as endangered by the state of Louisiana and the United States. Brown pelicans are large stocky birds between 114 and 137 cm in length. They inhabit areas along the Gulf Coast, the Atlantic coast from North Carolina to Venezuela, and the Pacific coast from British Columbia to Chile (Knopf 1977). The average weight of adult males is usually greater than that of adult females, 3.7 kg compared to 3.2 kg (USGS 2001b). Adults have brown bodies and white heads. The most distinctive feature of the brown pelican is its large bill and throat pouch.

Brown pelicans nest in coastal areas. They prefer to nest on islands, which provide protection from predators. Nests are usually built on the ground, in mangrove trees, or in bushes (Knopf 1977). Brown pelicans are piscivorus birds, primarily fishing in shallow estuarine waters and rarely traveling further than 30 km out to sea. Adult brown pelicans require about 2 kg of fish per day to fulfill their dietary requirements. Young brown pelicans are fed by their parents for 9 weeks, consuming 70 kg of fish over that time (Arnqvist 1992).

Brown pelicans have a powerful stroking flight alternating with short glides. They fly with their head drawn back to the shoulder. Pelicans rarely soar and often fly just centimeters above the water. Small flocks of pelicans fly in long lines. Brown pelicans use their eyesight to spot schools of fish and then dive for them, sometimes completely submerging. The large pouch they have can hold up to 10 liters of water along with prey. The species of fish that brown pelicans most commonly eat are referred to as “rough” fish; meaning they are commercially unimportant. Brown pelicans consume the following “rough” fish species; menhaden, herring, sheepshead, pigfish, mullet, grass minnows, and silver sides (Arnqvist 1992).

Order Falconiformes
Family Accipitridae
Osprey (*Pandion haliaetus*)

Osprey are large birds of prey found world-wide. The mean body weight of a female osprey is 1.57 kg, while the male is slightly smaller at 1.40 kg (USEPA 1993). Body length ranges from 53 to 61 cm (USEPA 1993). It has a conspicuous crook in its long wings and a black wrist mark. The plumage is dark above, light below. In the past, osprey populations experienced sharp declines, as a result of DDT bioaccumulation and its impact on nesting success. More recently, the reduction or elimination of DDT in the environment, along with conservation efforts, have contributed to increasing the osprey population (NGS 1983). Osprey typically use southern regions, such as Louisiana, to overwinter (McLaren/Hart-Chemrisk 1998).

Osprey prefer aquatic environments, such as large inland lakes, rivers, and estuaries. Ideal nesting sites are typically found close to open, shallow water with an abundance of prey (USEPA 1993). They nest in large trees with an open crown, power poles and other man-made structures, and on artificial platforms. Osprey are piscivorus birds. However, nearly all osprey will take other prey, such as birds, frogs, and crustaceans (USEPA 1993). Prey preferences change seasonally with abundance of local fish (USEPA 1993). The ospreys "terminal" position on the food chain makes it a good indicator of toxic contaminants that bioaccumulate (USEPA 1993). Osprey feed twice a day, in the mid-morning hours and again in late afternoon. Each meal is approximately 300 g (University of Michigan 2001). They are most successful at catching medium sized, slow moving benthic feeding fish in shallow waters (e.g., fish length between 11-30 cm; USEPA 1993; USGS 2001b). All parts of the fish are consumed with bones and indigestible parts being eliminated in fecal pellets (USEPA 1993). These birds hover, often 15 to 46 m high, then suddenly plunge, sometimes going completely under the water. The call is a series of loud, clear whistles.

Order Coraciiformes

Kingfishers are large-headed, short-tailed birds that dive for fish, catching them with their long, sharp beaks. They perch motionless in the open, over water. They have

very short legs. They lay from three to eight white eggs in a deep burrow in a steep bank (Robbins *et al.* 1983).

Family Alcedinidae
Belted kingfisher (*Ceryle alcyon*)

Belted kingfishers are medium-sized birds averaging 33 cm in length. These birds are slate blue with a white neckband and belly. Females have a rust-colored band across their belly. Kingfishers have deep, irregular wingbeats, a big head, and a loud, rattling call. They are commonly found throughout most of North America (USEPA 1993). Belted kingfishers are winter inhabitants in the Calcasieu Estuary (McLaren/Hart-Chemrisk 1998). Males and females are similar in size and body weight, averaging 0.15 kg (USEPA 1995). Preferred habitat is found along stream, lake and pond edges, as well as sea coasts and estuaries (USEPA 1993). Since belted kingfishers are sight-feeders, they prefer water that is clear and not obstructed by over hanging tree canopies or aquatic vegetation. Belted kingfishers typically nest in burrows in earthen banks beside rivers, streams, ponds and lakes. Belted kingfishers nest near suitable fishing areas when possible, but will nest away from water and feed in bodies of water other than the one closest to home. During spring and early summer, both male and female kingfishers defend a territory that includes both their nest site and their foraging area (USEPA 1993). By autumn each bird defends an individual feeding territory only. Breeding territories are, on average, more than twice as long as non-breeding territories (*e.g.*, 1,030 ± 219 m vs. 389.29 ± 92.63 m, respectively; USEPA 1993).

The belted kingfisher's diet primarily consists of fish and occasionally invertebrates (USEPA 1993). During shortages of their preferred foods they will consume crabs, lizards, frogs, turtles, mussels, small snakes, insects, salamanders, newts, young birds, mice, and berries (USEPA 1993). Belted kingfishers typically forage on fish in the first 12 to 15 cm of the water column. This includes fish in shallow water, as well as fish in deeper water that swim close to the surface (USEPA 1993). Feeding strategies vary depending upon the availability of perches that overhang the water. When adequate perches are available belted kingfishers will use them to spot prey in the water and then dive from them. Another technique is to fly over the surface of the

water waiting for potential prey to come into view and then striking (USEPA 1993).

Belted kingfishers eat large fish relative to their body size (USEPA 1995). Several field studies have reported on the size preferences of fish caught by belted kingfishers. The USEPA (1993) reported on a study where the average length of fish caught, in a field study in Michigan, was less than 7.6 cm (range: 2.5 to 17.8 cm). Another study found that belted kingfishers in Ohio selected fish ranging from 4 to 14 cm and 88% of the fish were between 6 and 12 cm (USEPA 1993). The trophic level of the prey consumed by belted kingfishers varies slightly between regions. In a survey of field studies examining trophic level of belted kingfisher prey, USEPA (1995) found that 94% of prey were from the aquatic environment with trophic levels ranging between 2.6 and 3.

18.1.2 Mammals

Order Carnivora
Family Procyonidae
Raccoon (*Procyon lotor*)

Raccoons occur in southern Canada, most of the United States with the exception of the Rocky Mountains and desert areas of the southwest, and in Mexico and most of Central America (Lowery 1974; Choate *et al.* 1994). Raccoons are the most abundant and widespread omnivore in North America, including Louisiana (USEPA 1993). Raccoons are stocky, short-legged, grayish to blackish animals (with some yellow and white) that are about the size of a large beagle. The heavily furred tail is ringed alternately with five or six blackish and yellowish rings. A prominent black mask extends across the face from the jowls through the eyes and is bordered above and below by white. Adult males average somewhat larger than females. They range in length from 46 to 71 cm with a 20 to 30 cm tail. Their weight can range from 3 to 9 kg (USEPA 1993). The young grow rapidly and are soon indistinguishable from adults.

Raccoons are usually most abundant near water, especially in bottomland forests along streams, hardwood swamps, flooded timber near reservoirs, marshes, wooded

areas near urban developments, and agricultural areas. They are able to live in a diversity of habitats provided certain requirements are found nearby, such as food, water, and a protected area for denning. Preferred denning sites include hollow trees, logs, stumps, holes in the ground, caves, and rocky ledges (USEPA 1993). The size of a raccoon's home range is dependent on the availability of food and shelter, as well as the space necessary for reproduction. In eastern North America, home ranges between 1 and 4 km^2 are normal. In contrast, home ranges in the prairies can be up to 50 km^2 (Environment Canada 2000).

Raccoons produce only one litter per year. Mating occurs from December to June. The young are generally born in late April or early May after a gestation period of 63 days. The number of young in a litter is usually two to five.

Raccoons eat crayfish, snails, clams, small fishes, frogs, earthworms, and a wide variety of insects such as grasshoppers, crickets, and beetles. Acorns, berries, watermelons, cantaloupe, corn, tomatoes, and the tender shoots and buds of many trees and other plants are also consumed. Raccoons are mainly active in late afternoon and night, but can be seen during the day. They are excellent swimmers and climbers. Their dens are often in trees, in hollows 9 to 12 m above the ground, or on the ground. Raccoons have few enemies, but their babies are killed by coyotes and other large predators (Lowery 1974; Choate *et al.* 1994).

Family Mustelidae
Mink (*Mustela vison*)

Mink occur throughout North America, with the exception of the southwest United States (Lowery 1974; Choate *et al.* 1994). In Louisiana, it is found statewide. The tail consists of approximately one third of the total length of this medium-sized mammal. It is considerably longer, heavier bodied, and darker in color than a weasel. They are long and thin, with short sturdy legs. The mink is chocolate brown above and lighter brown below, with white blotches on the chin, throat, chest, stomach, and anal region. The tail is bushy, and the fir is dense and glossy. This animal leads a semi-aquatic existence and has semi-webbed toes. Males are larger than females. Males are between 33 and 43 cm in length with a 18 to 23 cm tail. Females, on the

other hand, range from 30 to 36 cm in length with a 13 to 20 cm tail (USEPA 1993). Mink typically weigh between 0.7 and 1.1 kg (USGS 2001b).

In Louisiana, mink are particularly numerous in tupelo-gum swamps, freshwater to brackish coastal marshes, along wooded streams, and on the edges of lakes. Mink are never found far from water. Habitat preferences include irregular shorelines covered with brush and woody cover, which creates shelter for prey. Fallen debris on shorelines is also useful, as it creates excellent den habitat. Mink will defend 1 to 4 km of shoreline with scent markings and physical aggression (USEPA 1993).

Mink are predominantly nocturnal feeders and can be described as opportunistic. They will feed on a variety of prey, depending on the season and prey abundance. The majority of hunting takes place along shorelines or in emergent vegetation. As a result of their smaller size, female mink are unable to eat prey as large as males eat, such as, muskrats and rabbits (USEPA 1993). Prey that both males and females consume includes aquatic animals such as fish, amphibians and crustaceans, as well as terrestrial animals like small mammals, birds, reptiles, and insects (USEPA 1993). Mink typically kill their prey with a bite to the neck (USGS 2001b). Feeding habitats can be affected by water level. During high water levels mink feed more on crayfish and voles. When water levels are low, their diet switches to aquatic birds, muskrats, and even ducklings. Winter also affects mink diet; during these months, fish become a more important food source (USEPA 1993).

Mink dens are located under fallen logs, in hollow stumps, and in old abandoned muskrat houses and nutria burrows. The nest is lined with grasses, feathers, fur, and any other soft materials. Mink only produce one litter per year with an average of 4 young. The litter is produced in the early spring and the gestation period may last as long as 75 days. Weaning takes place after about five weeks. Predators of the mink include bobcats, owls, and, especially, alligators (Lowery 1974; Choate *et al.* 1994).

Mink are particularly sensitive to PCBs and similar chemicals. Research has found that they accumulate PCBs in their subcutaneous fat at levels 38 to 200 times dietary concentrations, depending on the PCB congener (USEPA 1993).

River otter (*Lutra canadensis*)

The river otter occurs throughout most of Canada and the continental United States, except for the southwestern United States (Lowery 1974; Choate *et al.* 1994). It occurs throughout the state of Louisiana where suitable habitat is still available along streams and river and in coastal marshes. This long-bodied, short-legged, semi-aquatic animal cannot be confused with any other mammal in Louisiana. It has long whiskers, sleek brown fir, a cylindrical body, thick neck, flattened head, webbed feet, and a long, heavy tail. Males are slightly larger than females. Males range in weight from 5 to 10 kg and females from 4 to 7 kg. Body length ranges from 66 to 76 cm with a 30 to 43 cm tail.

Otters, which are very playful, spend most of their time in and near rivers, creeks, bayous, and lakes, especially those bordered by timber. They possess many adaptations for an aquatic existence such as webbed feet, waterproof fir, eyes near the top of the head, and nostrils and ears that can close when the otter is underwater. River otters prefer habitat that is close to lakes, marshes, streams, and seashores. When selecting habitat, abundance of food is a primary consideration. River otters den in banks and hollow logs (USEPA 1993). The home range of river otters follows the shoreline. Its size is determined by the area needed to meet the demands of foraging and reproduction. Home range size can vary from 10 to 78 km of shoreline. It is common to find 1 river otter for every 10 km of shoreline. Males tend to range more than females, with lactating females ranging the least (USEPA 1993).

River otters are piscivorus and opportunistic feeders. This, in addition to their high trophic level, makes them a good indicator of bioaccumulation. They feed almost entirely on frogs, turtles, snakes, fish, and aquatic invertebrates such as crayfish and crabs. Occasionally, they eat birds, rats, and mice. River otters may probe the bottom of ponds or streams for invertebrates and thus ingest sediment and other debris in the process (USEPA 1993).

The USEPA (1995) reviewed available field studies on river otter diet. They found that river otter diets varied markedly depending on the season. For example, where fish may not be as readily available in some regions during the winter months due to ice cover, river otter diets were more dependent on other organisms. Because river

otters are fairly large mammals and are opportunistic in their feeding habits, fish size can vary greatly from 2 to 50 cm. The majority of fish eaten, however, are on the lower half of this range (USEPA 1995). The trophic level of prey items consumed by the northern river otter varied depending on the source of the prey. Aquatic prey trophic levels ranged from 2.6 to 3.2, wetland prey trophic level ranged from 2.0 to 4.0, and terrestrial prey ranged from 1.0 to 2.6 (USEPA 1995).

Otters breed in the late winter/early spring, and after a gestation period of as long as 380 days (resulting from delayed implantation), an average of two to three kits is born. The den is in the bank of a canal or stream or an old muskrat house or nutria den with the entrance beneath the water surface. The nest is grass lined and above the high-water level. The young remain with the mother until they are nearly full grown. Otters have few enemies but are probably occasionally captured by owls and alligators (Lowery 1974; Choate *et al.* 1994).

Order Cetacea
Family Delphinidae
Atlantic bottlenose dolphin (*Tursiops truncatus*)

The bottlenose dolphin is probably the most familiar species due to its coastal distribution and widespread visibility in films as well as in captivity. In some areas, bottlenose dolphins can reach up to 3.8 m in length. Body weights range from 150 to 200 kg (University of Michigan 2001). Bottlenose dolphins eat a wide assortment of fishes, squid, and shrimp. They use a variety of feeding behaviors, including feeding behind shrimp boats and chasing fish onto mudbanks. Bottlenose dolphins live in open societies, with the strongest bonds being between a mother and her calf. Bottlenose dolphins in the Gulf of Mexico occur mostly on the continental shelf (MMS 2001).

18.2 Other Species Resident to the Calcasieu Estuary Ecosystem

18.2.1 Birds Resident to the Calcasieu Estuary

Non-Perching Birds
Order Podicipediformes

Grebes are swimming and diving birds, smaller than loons, with flat lobes on their toes. The short legs are far back on their bodies, and their tails and wings are short. Their flight is weak and hurried. Grebes dive and pursue small aquatic animals. Elaborate courtship displays occur on the water. They nest in floating marsh vegetation. The clutch averages from two to nine eggs (Robbins *et al.* 1983).

Family Podicipedidae
Pied-billed grebe (*Podilymbus podiceps*)

Pied-billed grebes are small, stocky birds, measuring about 35 cm in length and weighing between 350 and 550 g. The sexes are similar in appearance and are seasonally dimorphic. Breeding adults have brown heads and bodies and black chins. Their tails and bills are always stout; during the breeding season their bills have a black band around them and their black eyes are ringed with blue and grey plumage. Winter differences in appearance include white plumage under the chin and yellowish coloring on their bills and around their eyes (USGS 2000; University of Guelph 2000). It rarely flies but instead dives or slowly sinks below the surface to escape.

Pied-billed grebes breed across southern and central Canada, through most of the United States and Mexico. Wintering grounds range from the Rockies to the southern United States and Mexico (University of Guelph 2000). These birds are year-round inhabitants of the Calcasieu region (McLaren/Hart-Chemrisk 1998).

Ideal nesting habitat occurs in shallow water areas, such as wetlands with thick surface vegetation. Nests are constructed from nearby plants, including dead and decaying reeds and rushes (USGS 2000; University of Guelph 2000). Pied-billed

grebes feed on a variety of aquatic invertebrates, fish, reptiles, and amphibians. Foraging most often occurs in shallow water areas where pied-billed grebes dive for their food (University of Guelph 2000). The call is a series of low, slurred whistles (Robbins *et al.* 1983).

Order Pelecaniformes

Pelicans are large aquatic fish-eating birds with webbed toes. Most nest in large colonies. They lay between three and five eggs. Cormorants and anhingas are fish eaters that swim with their bill tilted upward, dive from the surface, and swim under water. They often perch with wings half open to dry (Robbins *et al.* 1983).

Family Phalacrocoracidae
Double-crested cormorant (*Phalcrocorax auritus*)

Double-crested cormorants are large, dark waterbirds with thin, yellow, hooked bills (USGS 2000, USGS 2001b). Their length ranges from 76 to 89 cm, bill tip to tail tip. The sexes are very similar except for weight. Males are usually heavier, with an average weight of 1.8 kg and females 1.5 kg (USGS 2001b).

These birds can be found in coastal and freshwater areas from Newfoundland to New York, as well as in North Carolina, Virginia, and along the Florida, Atlantic, and Gulf Coasts . Double-crested cormorants use the Calcasieu Estuary for overwintering (McLaren/Hart-Chemrisk 1998). They also breed along the coast in the southeast United States, including the Gulf Coast in Louisiana. They are colonial breeders, nesting on the ground, in trees, and on cliffs (USGS 2001b). Preferred habitat includes bays, estuaries, marine islands, freshwater lakes and islands, ponds, rivers, sloughs, and swamps (USEPA 1995).

Double-crested cormorants are generalist feeders, primarily feeding on schooling fish and, to a lesser degree on aquatic invertebrates. Examples of prey include sculpins, shrimp, sandlance, insects, herring, eel, cod, crustaceans and mollusks. Foraging typically occurs in water less than 30 feet deep (USGS 2000, USGS 2001b). Double-crested cormorants dive from the surface to catch their prey, usually over shoals with rocky gravel bottoms (USEPA 1995).

Family Anhingidae
Anhinga (*Anhinga anhinga*)

Anhingas average 71 cm in length with 119 cm wingspans (USGS 2000). Males have black plumage with white spots on their wings and back. Females have a tan head and neck, and a black body with white edging on their wings (USGS 2000). Anhingas display some distinctive characteristics. Firstly, they swim with only their head showing above the surface of the water. Secondly, they are able to soar for extended periods of time, much like hawks. Finally, because they lack oil glands with which to preen, they can be seen with their wings outstretched in an effort to dry them in the sun (eNature Field Guide 2001; USGS 2000).

Anhingas breed along the Atlantic and Gulf Coasts, from North Carolina to Texas. They also breed in Florida and in the Mississippi Valley north to Missouri and Kentucky. Wintering occurs from the Gulf Coast region north to South Carolina (eNature Field Guide 2001).

Anhingas prefer to inhabit areas around freshwater ponds, swamps, and marshes. They also prefer areas with thick vegetation and large trees (eNature Field Guide 2001). These birds predominantly feed on fish, but also consume lesser amounts of aquatic invertebrates, reptiles and amphibians (USGS 2000). Anhingas often dive from the surface for food, much like cormorants, spearing prey with a snap of their neck (eNature Field Guide 2001).

Order Anseriformes

Waterfowl have webbed feet. They have long necks and narrow pointed wings, and most have short legs. They have flattened bills with tooth-like edges that serve as strainers. Their flattened bodies are insulated with down feathers. The young can walk and swim a few hours after hatching. Surface-feeding ducks feed on aquatic plants. They nest on the ground (except for the wood duck), and they lay between five and twelve eggs (Robbins *et al.* 1983).

Family Anatidae
Mottled duck (*Anas fulvigula*)

The mottled duck is a large dabbling duck. Their plumage is similar to the American black duck (*Anas rubripes*) and the female mallard (*Anas platyrhynchos*); both of these species winter in Louisiana. The sexes are similar in appearance. They have a distinct white border behind the blue speculum. They have a darker tail, paler head, and yellower bill than the mallard. Mottled ducks pair in January (Robbins *et al.* 1983). They range in length from 38 to 53 cm (USGS 2000; eNature Field Guide 2001).

Mottled ducks are predominantly found in the central Gulf Coast region and rarely migrating. This region encompasses Florida, Louisiana, and Texas (eNature Field Guide 2001; McLaren/Hart-Chemrisk 1998). These ducks prefer habitat in freshwater and saltwater marsh areas. They most commonly forage for aquatic invertebrates, eating lesser quantities of seeds, plant matter, and fish (USGS 2000).

Blue-winged teal (*Anas discors*)

This teal is a year-round resident along the Louisiana coast. It is a small, common, shy duck of ponds, marshes, and protected bays. It flies rapidly in small, tight flocks. Both sexes have a light blue area on the forward edge of the wing and a green speculum. Males have a white facial crescent. The male peeps, and the female quacks softly (Robbins *et al.* 1983).

Wood duck (*Aix sponsa*)

Wood ducks are permanent residents of the southeastern United States. They are common in open woodlands around lakes and along streams. They have a large head, short neck, and long tail. No other duck has the long, slicked-back crest. The dull colored female has a white eye ring. Males resemble the female when in eclipse plumage but have lots of white under the chin. Wood ducks fly rapidly and dodge between the trees. They feed on plant materials and some insects. Nesting is in natural tree cavities or in nest boxes. Their call is a distinctive rising whistle (Robbins *et al.* 1983).

Order Falconiformes

Vultures, hawks, and falcons are flesh eaters. Most have a heavy, sharp, hooked bill and strong, curved talons. The sexes are usually alike, with the females averaging larger than males (Robbins *et al.* 1983).

Family Cathartidae
Turkey vulture (*Cathartes aura*)

Turkey vultures breed throughout the United States and are year-round residents of the southern United States. This common carrion-eater scavenges in fields and along roadsides. It soars in wide circles, holding its two-toned wings in a broad "V" and tilting quickly from side to side. Immature birds have a naked, black head, while adults have a red head. Feeding vultures are soon joined by others flying in from far away (Robbins *et al.* 1983).

Black vulture (*Coragyps atratus*)

Black vultures are large soaring birds. They measure about 56 cm in length with a wingspan of 137 cm. The sexes are similar, having almost entirely black plumage with some white on their primary feathers. Their heads are featherless and grey. Black vultures are found in western Texas and Arkansas, northeast to New Jersey and south to Florida (USGS 2000; eNature Field Guide 2001). They are found year-round in the Calcasieu region (McLaren/Hart-Chemrisk 1998).

Black vultures have one brood each year consisting of 2 eggs. Eggs are laid either under a bush, amongst rocks, in a hollow log, or in a cave. Black vultures are scavengers usually found in open areas. An exception to this occurs during breeding, when black vultures move into lightly wooded areas. They can often be seen perched in dead trees or soaring high in the air. Carrion is the most common source of food. However, they will also eat sick, injured, or young birds and mammals. Black vultures usually search for food by soaring high in the air, in groups. They use their keen eyesight to locate prey (USGS 2000; eNature Field Guide 2001).

Family Accipitridae
Mississippi kite (*Ictinia mississippiensis*)

Mississippi kites breed in the south-central United States. This kite can be found in brushlands and open woods near water. Adults are recognized by their plain gray underparts and pale head. Immatures have a graceful, almost swallow-like flight, and notched black tail. They are often seen in flocks when migrating or feeding and eat insects caught in the air and on the ground (Robbins *et al.* 1983).

Swallow-tailed kite (*Elanoides forficatus*)

The swallow-tailed kite breeds in the extreme southeast United States. It is fairly common in swamps, marshes, riverbanks, and open forests. This is the most graceful of all North American hawks, with its striking black and white pattern and swallow tail. Immatures resemble adults but are speckled. When hunting, it drifts along slowly just a few feet above the treetops or low over the ground with outstretched wings; its tail in a constant balancing motion. It often eats in flight. This kite is somewhat gregarious (Robbins *et al.* 1983).

Cooper's hawk (*Accipiter cooperii*)

This hawk is a permanent resident of most of the United States. Cooper's hawks are fairly uncommon. They are found in open woodlands and wood margins where they feed on small mammals. Compared to the sharp-shinned hawk (*Accipiter striatus*), it has a comparatively rounded tail, large head, white terminal tailband, and a slower wingbeat. Immature birds have brown streaking. This hawk is very fast and powerful. The call is a series of 15 to 20 "kaks" (Robbins *et al.* 1983).

Red-tailed hawk (*Buteo jamaicensis*)

This well-known hawk is a year-round resident of most of the United States. It nests in woodlands and feeds in open country on rabbits and rodents. The uniformly colored tail of the adult (reddish above, light pink beneath) and the dark belly band are the best marks for identification. The tail of the immature birds is finely barred. The body is heavier than other buteos, and the plumage is extremely variable. It hunts

alone, often perches on poles or treetops, and rarely hovers. The call is a high scream (Robbins *et al.* 1983).

Broad-winged hawk (*Buteo platypterus*)

Broad-winged hawks breed in the eastern half of the United States. They are a woodland species. Adults are recognized by the broad barred tail. Immature birds have a white underwing surface contrasting with black tips. It lacks the belly band of the much larger red-tailed hawk. The broad-winged hawk hunts from a perch, flashing into action upon the appearance of a large insect, mouse, or small reptile. It characteristically migrates in large flocks. The call is a thin whistle (Robbins *et al.* 1983).

Red-shouldered hawk (*Buteo lineatus*)

Red-shouldered hawks are found year-round in California and the eastern United States, including the Calcasieu Estuary. During the summer they also range north to New England and the Great Lakes. They are large, broad-winged hawks, capable of soaring for long periods of time. They have brown heads and reddish breasts and underwing sections. Their underbelly is light with bars of brown and their beak is short, dark, and hooked. Red-shouldered hawks measure about 41cm in length and have a 102 cm wingspan (USGS 2000; eNature Field Guide 2001).

Red-shouldered hawks primarily inhabit deciduous woodlands and swamps. They rely on their excellent vision and broad wings to aid them in foraging. They will perch in a tree or soar above the ground using their eyes to spot prey. Once they identify food they drop to the ground and seize it. Prey items include small mammals, reptiles, amphibians, and birds. Red-shouldered hawks are usually solitary birds, coming together to breed. Pairs of breeding red-shouldered hawks are monogamous (USGS 2000; eNature Field Guide 2001).

Bald eagle (*Haliaeetus leucocephalus*)

The bald eagle is the national symbol of the United States. It is currently listed as threatened in the lower 48 states and endangered in Louisiana (USFWS 2001; LDFW

2001). Body size increases with latitude (USEPA 1995). The average length from bill tip to tail tip is 81 cm, for more northerly populations (USEPA 1993). Female bald eagles weigh more than males, 5.2 kg (n=37) compared to 4.1 kg (n=35).

Bald eagles are most often found in coastal environments near lakes, rivers, and oceans (USEPA 1995). Ideal nesting sites are located in close proximity to large bodies of water. Nests are typically built in large coniferous trees with sturdy branches and open crowns. Areas of old growth forest with a discontinuous canopy are also preferred (USEPA 1993).
Bald eagles are primarily opportunistic carrion feeders. They will forage for whatever food is plentiful and easiest to capture. Bald eagles will eat dead and dying fish when possible. They will also catch live fish that swim in shallow water, or that swim close to the surface in deeper water. Other examples of prey include birds, rodents, and small terrestrial animals (USEPA 1993; USEPA 1995). In coastal environments, sea birds, such as cormorants, gulls, shearwaters, auklets, and petrels, constitute a greater proportion of the bald eagles diet (USEPA 1995). During the winter, they will eat dead or injured waterfowl shot by hunters, as well as shore birds. They will also steal food from other birds such as osprey, mergansers, hawks, and other bald eagles (USEPA 1995). Bald eagles forage upland during the winter if their water environments freeze. In upland areas the bald eagles diet focuses on carrion such as rabbits, squirrels, and domestic livestock (USEPA 1995).

Family Falconidae
Crested caracara (*Caracara plancus*)

The crested caracara is the national bird of Mexico. These broad winged and long-tailed hawks usually measure 53 cm in length and have a wingspan of 122 cm. They have mostly dark brown plumage and a black cap with a grey hooked beak. Their faces are bare and red. White plumage is found around their throat and on their wingtips. In the southern United States, crested caracaras range from southern Arizona to southern Texas, through southwestern Louisiana, and into central and southern Florida (USGS 2000; eNature Field Guide 2001; McLaren/Hart-Chemrisk 1998).

Crested caracaras are ground inhabiting birds most often found on seashores, in prairies, savannas, or desert scrub. They are scavengers and have quite possibly the most diverse diet of any bird. Prey items include carrion, reptiles, amphibians, and birds (USGS 2000; eNature Field Guide 2001).

American kestrel (*Falco sparverius*)

This is the smallest and most common falcon in open and semi-open country. It is a permanent resident in most of the United States. It is the only small falcon with two "whiskers" on each side of the face and a rusty back. It hunts from poles, wires, or trees and frequently hovers. It eats primarily insects. The call is a sharp "killy killy killy." (Robbins *et al.* 1983)

Order Galliformes

Gallinaceous birds are heavy-bodied, chicken-like, land birds. Their flight is not fast, and all are capable runners that forage on the ground for seeds and insects. Males are more colorful than females and exhibit elaborate courtship displays (Robbins *et al.* 1983).

Family Phasianidae
Northern bobwhite (*Colinus virginianus*)

The bobwhite is a permanent resident of the eastern two-thirds of the United States. It is abundant in brush, abandoned fields, and open pinelands. It avoids deep forests. It is a chunky reddish-brown quail with a gray tail. The male is identified by the white throat and eye line; these areas are buffy in the female. During the winter, they are found in flocks (coveys) of up to 30 birds. The clutch averages seven to fifteen eggs. They all burst into flight at once when disturbed. The call is a whistled "bob-bob-white" (Robbins *et al.* 1983).

Order Ciconiiformes

Herons and their allies are wading birds with long legs, neck, and bill. Most feed on aquatic animal life in shallow water. The wings are broad and rounded, and the tail

is short. The clutch of eggs ranges from two to six. Most are colonial nesters (Robbins *et al.* 1983).

Family Ardeidadae
Great blue heron (*Ardea herodias*)

Great blue herons are the most widely distributed and largest heron found in North America. They are year-round inhabitants of the Calcasieu Estuary (McLaren/Hart-Chemrisk 1998). They are very large birds with long legs and necks. Great blue herons average 2.9 kg when full grown, males being somewhat heavier than females. They are approximately 97 cm in length and have a wingspan of about 178 cm. They also have a long and thick yellow bill (USGS 2000). The head is largely white, and the underparts are dark. Single birds are usually encountered, but they nest in colonies. The alarm call is a series of about four hoarse squawks.

Great blue herons are found in both freshwater and marine environments. Their preferred habitat is freshwater lakes, rivers, brackish marshes, lagoons, mangroves and coastal wetlands (USEPA 1993). Preferable habitat has large quantities of fish and sufficient shallow water areas for foraging, such as tidal flats, sandbars, wet meadows, and pastures (USEPA 1993). The size of heron feeding territories can vary with the seasons. The USEPA (1993) mentions a study in which adult herons had a feeding territory of 1.5 acres in the fall and 20.8 acres in the winter. The foraging distance from a heron colony can be up to 25 km (USEPA 1993).

Great blue herons are opportunistic feeders. Their diet primarily consists of small fish, but they will also eat amphibians, reptiles, crustaceans, insects, birds, and mammals (USEPA 1993). Herons usually feed solitarily, foraging on tidal flats, sandbars, wet meadows, and pastures. An exception to feeding solitarily may occur if a large concentration of prey is located in a small area. The most common foraging technique used by herons involves standing or walking in shallow water and waiting for fish to swim within striking distance (USEPA 1993). They usually fish in shallow waters less than 0.5 m in depth with a firm substrate bottom. After consuming large prey, herons often drink water (USEPA 1993).

Snowy egret (*Egretta thula*)

These are small white herons, distinguishable by their black bills and legs, and yellow feet (USGS 2001b). They average 51 cm in length and have a wingspan of about 97 cm (USGS 2000). The average weight for snowy egrets is 371 g. There does not appear to be sexual dimorphism in the species (USGS 2001b). Breeding occurs colonially in salt marshes, freshwater marshes, ponds, and shallow coastal bays. Snowy egrets prefer nesting in more open areas than other herons, usually about 1.0 to 1.5 m above the ground (USGS 2001b). Breeding occurs along the Atlantic coast and on inland bodies of water such as the Mississippi River. Overwintering takes place in Florida, the Carribean, and South America. Snowy egrets can be year-round inhabitants of the Calcasieu area (McLaren/Hart-Chemrisk 1998).

Snowy egrets are generalist feeders. They primarily feed on aquatic invertebrates, but also eat reptiles, amphibians, fish, and small mammals (USGS 2000). Examples include shrimp, small fish, mollusks, frogs, and aquatic and terrestrial insects. Foraging involves standing or walking in shallow water, watching for prey and striking when the opportunity arises. Feeding takes place on oyster bars, in tidal creeks, freshwater ponds, and salt flats (USGS 2001b).

Cattle egret (*Bubulcus ibis*)

This medium-sized heron measures between 46 and 56 cm in length and has a wingspan between 88 and 96 cm. During the breeding season it has a buffy-orange plumage on its crown, breast, and back; the rest of the year it is pure white. Its bill is orange or yellow and its legs are a pale orange color. The neck is shorter and thicker than in other herons.

This is an Old World species that has colonized North America. Cattle egrets are wide-ranging throughout the world. In North America, they are found in most of the western United States, east to the Great Lakes and Maine, and south to the Gulf Coast (University of Michigan 2001; eNature Field Guide 2001). They are found year-round in the Calcasieu region (McLaren/Hart-Chemrisk 1998).

Cattle egrets are the most terrestrial herons. While they do not require aquatic environments to survive, they still frequent them. Cattle egrets are seasonally monogamous and nest in colonies near water with other wading birds. Aside from breeding they are most often found in fields and pasture lands, feeding on insects that are kicked-up from the grass by grazing animals, such as livestock. Examples of insects eaten are grasshoppers, crickets, spiders, and flies. They will also feed on frogs and toads, but rarely forage on fish.

Tricolor heron (*Egretta tricolor*)

Tricolored herons are medium-sized wading herons with long necks and legs. They are inhabitants of North America, from Massachusetts south to the Gulf Coast (USGS 2001b). In the Calcasieu study area, they are year-round inhabitants (McLaren/Hart-Chemrisk 1998). Their long bills are yellow or grey in color with a black tip. They have predominantly grey-blue plumage with white bellies and forenecks (USGS 2000). Tricolored herons are about 56 cm in length and have a wingspan of approximately 97 cm (USGS 2000). Adult males weigh approximately 415 g and adult females 334 (USGS 2001b).

Tricolored herons can be found in brackish and salt water coastal areas, marshes, swamps, and mudflats. Nests are usually built close to the ground in low tide areas (USGS 2001b). These herons feed mostly on aquatic invertebrates and fish, as well as reptiles and amphibians, within 2 km of their nests (USGS 2000; USGS 2001b). The most common foraging technique involves either standing or walking slowly in shallow water waiting for prey to come within striking distance (USGS 2001b).

Little blue heron (*Egretta caerulea*)

Little blue herons are medium-sized wading birds. They are among the most numerous herons found in the southeastern United States. They breed in the southern United States from southern California, southern New Mexico, Texas, and Oklahoma, east to southern Missouri and southern New England, and south to the Gulf Coast (eNature Field Guide 2001). In the Calcasieu region, little blue herons are year-round residents (McLaren/Hart-Chemrisk 1998). The sexes are similar, measuring about 56 cm in length and having a wingspan of 104 cm. Their plumage is blue-gray and their

beaks are long, pointed and bluish-grey, with a black tip (USGS 2000; eNature Field Guide 2001). The legs are bluish green. Immature little blue herons are white with a dark-tipped bluish bill and greenish legs.

Little blue herons inhabit freshwater swamps and marshes, and lagoons. Adults usually feed solitarily, while young tend to feed in groups. Their foraging technique is similar to that of other herons. They wade through shallow water searching for prey, snapping it up when it crosses their path. Prey includes aquatic invertebrates, fish, reptiles, and amphibians. Breeding occurs in wetland and open water areas. Little blue herons nest in colonies, building their nests in small trees and bushes. They lay one brood each year consisting of 3 to 5 eggs, which are incubated for 20 to 23 days (USGS 2000; eNature Field Guide 2001).

Green-backed heron (*Butorides striatus*)

Green-backed herons breed in the eastern half of the United States and are permanent residents of the coasts of the southeastern United States. It is common and locally abundant in both freshwater and saltwater areas. It is found more than other herons in small ponds and along wooded streams. It is more blue than green, and from a distance, it appears all dark. It can be identified by its small size, dark underparts, and bright orange or yellow legs. Their flight is rapid, with deep wingbeats. The crest is not always visible, and the neck is much shorter than that of other herons. The call is a sharp, descending "kew" (Robbins *et al.* 1983).

Black-crowned night-heron (*Nycticorax nycticorax*)

Night-herons are characterized by heavy bodies, short, thick necks, and short legs. Black-crowned night herons are relatively small herons found year-round in the Calcasieu Estuary (McLaren/Hart-Chemrisk 1998). They have a black crown, bill, and back, yellow legs, a white ventral surface, and grey wings. They range in length from 58 to 65 cm and have an average wingspan of 118 cm (USGS 2000). Body weight for black-crowned night herons typically falls between 0.8 and 0.9 kg (USEPA 1995). It flies in loose flocks. It is often inactive during the day, roosting in trees, and fishes more at night. The call, a single "kwawk," is most often heard at night.

Black-crowned night herons are usually found nesting in freshwater, brackish or saltwater environments. They will build their nests in either tall trees or low shrubs (USEPA 1995). They are opportunistic and general feeders that mostly eat fish, but will also prey on aquatic invertebrates, reptiles, amphibians, and small mammals. For example, frogs, crabs, crayfish, and molluscs (USGS 2000; USEPA 1995). Foraging takes place in shallow water, mudflats, and upland habitats. Their foraging technique is similar to that of great blue herons. They will walk slowly through the water, locating prey by sight and then striking (USGS 2001b).

Yellow-crowned night-heron (*Nycticorax violaceus*)

A year-round resident of the coasts of the southeastern United States, the yellow-crowned night-heron is much less common than the black-crowned night-heron. It primarily hunts at night, but may also be seen hunting during the day. It has a distinct black and white face and a gray body. Immature birds are similar to black-crowned night-herons, but have a shorter, thicker bill, longer legs, and grayer plumage with smaller light spots on the back. The call is slightly higher-pitched than the black-crowned night-heron's (Robbins *et al.* 1983).

American bittern (*Botaurus lentiginosus*)

American bitterns winter throughout most of the southern United States, but there is a year-round population in southwest Louisiana. This bird is common but very elusive in the tall vegetation of freshwater and brackish marshes. It is most active at dusk and at night. It sometimes hides by freezing in position with the head pointed upward. It has a broad, black whisker, which no other heron or bittern has. In flight, it has blackish flight feathers. It seldom calls when flushed, and it does not flock. It makes a hollow croaking or pumping sound on the breeding grounds (Robbins *et al.* 1983).

Least bittern (*Ixobrychus exilis*)

This bittern breeds in most of the eastern United States. It is common but very shy and usually remains hidden in tall freshwater grasses and sedges. This is the smallest heron and a weak flier. It will run and climb rather than fly; it seldom flies more than

30 m. Both sexes have large wing patches of buff and chestnut. A rare dark phase of this bird is a rich chestnut. It hides by freezing. The call is about four identical soft "coos" (Robbins *et al.* 1983).

Family Ciconiidae
Wood stork (*Mycteria americana*)

Wood storks, the only stork in North America, breed in the southeastern United States. It is locally common in southern swamps, marshes, and ponds. It has a long, thick bill and a dark, unfeathered head. Immature birds have a lighter colored head and neck and a yellow bill. It flies with its neck and legs extended, often soaring. Wood storks have slow, powerful, and loud wingbeats. It feeds on fish, reptiles, and amphibians. It nests in colonies in trees. The call is humming notes (Robbins *et al.* 1983).

Order Gruiformes

Cranes and their allies are a diversified group. All are wading birds with long legs. The average clutch for rails and gallinules is six to fifteen eggs (Robbins *et al.* 1983).

Family Rallidae
Black rail (*Laterallus jamaicensis*)

This rail winters along the Gulf Coast but also breed in most of the eastern United States. This is the smallest of the rails. Rails are wading birds with long legs. It is rather common, usually found in salt marshes among cordgrasses and rarely in freshwater marshes. It can be identified by its tiny size, the black coloration, the black bill, and the white spots on its back. The call is a "kickee-doo," heard late at night (Robbins *et al.* 1983).

Clapper rail (*Rallus longirostris*)

Clapper rails are large marsh birds that walk more often than they fly. They average 30 cm in length and have a wingspan of about 51 cm (USGS 2000). Weights range between 160 and 400 g, males being about 20% heavier than females (USGS 2001b).

Both sexes are similar in appearance, having gray-brown plumage, short necks, and long curved bills. The bills of males are colored pink to bright orange on the sides and bottom (USGS 2000). The rail's underside has white stripes. The extremely short tail is cocked upward. As in other rails, the body is compressed laterally. The voice is a series of loud unmusical ticks.

The eastern breeding range of clapper rails extends from New Hampshire south to the Florida Keys and Carribean Islands, as well as along the Gulf Coast as far as Texas. In the west, their breeding range extends from San Francisco to Mexico. Southern populations are year-round inhabitants and northern populations migrate to southern regions to overwinter (USGS 2001b).

Clapper rails are solitary ground nesters. They prefer saltwater, brackish, and freshwater marshes, and mangrove swamps (USGS 2001b). Clapper rails are opportunistic and generalist feeders. However, when possible they will primarily feed on aquatic invertebrates, as well as fish and plant matter. Examples of prey include small crabs, crustaceans, slugs, insects, small mammals and birds, small fish, and eggs. Clapper rails forage by either probing the sediment in shallow water, or by gleaning surface water (USGS 2000; USGS 2001b).

King rail (*Rallus elegans*)

King rails breed in the eastern half of the United States and live year-round along the Atlantic and Gulf Coasts. This large, rust-colored rail is usually found in freshwater marshes, though it is occasionally found in brackish marshes. It is browner than the clapper rail, with stronger bars on the belly. The chicks are black with a white bill. The call is similar to that of the clapper rail, but it is shorter and more musical and resonant (Robbins *et al.* 1983),

Common moorhen (*Gallinula chloropus*)

Common moorhens live year-round along the Atlantic and Gulf Coasts and breed in the eastern half of the United States. It is common in freshwater marshes and along the edges of lakes. It resembles a duck, but it has a bright, red frontal plate on the head and a yellow-tipped, chicken-like bill. The flanks are edged with white, and the

wings are entirely dark. It feeds along the edge of open water and seeks cover in dense vegetation when disturbed. It swims well and walks on lily pads. The call is a hen-like cluck (Robbins *et al.* 1983).

Purple gallinule (*Porphyrula martinica*)

Purple gallinules breed in the southeastern United States and are permanent residents along the coasts, from Central Florida to Texas. It is less common than the common moorhen, which it closely resembles in its habits. The adult is unmistakable, with a green back and purple head and underparts. This white plate on the front of the head is characteristic of this species. Immature birds are browner and paler. The calls are similar to the common moorhen's (Robbins *et al.* 1983).

Order Charadriiformes

Shorebirds, gulls, and allies form a diverse group of wading and swimming birds. Most are white, gray, or brown, with long, pointed wings and long legs or webbed feet. The sexes are similar. Most feed along shores, a few inland. They feed on small invertebrates. Two to four eggs are laid. Many are highly migratory (Robbins *et al.* 1983).

Family Charadriidae
Wilson's plover (*Charadrius wilsonia*)

Wilson's plovers are permanent residents of the southern Atlantic and Gulf Coasts of the United States. It is uncommon and local. This plover prefers sandy beaches and mudflats. It has a long, broad, dark eye stripe, heavy black bill, and wide, dark neck band. The feet are dull pink. The female is much paler than the male. The call is a "wheet" (Robbins *et al.* 1983).

Killdeer (*Charadrius vociferus*)

Killdeers are found across most of North America. They breed from Alaska to Newfoundland and southward, and winter as far north as British Columbia, Utah, the Ohio Valley, and Massachusetts. They can be found year-round in the Calcasieu

region (McLaren/Hart-Chemrisk 1998). Killdeers are very easy to identify. They have two black bands that circle their chest and one that wraps around their head. Their caps are brown, wings are black, and breast and belly are white. They have a long, brown tail that extends past their wing tips as well as long flesh-colored legs. Killdeers usually measure 23 to 27 cm in length with a 17.5 cm wingspan and weight of 100 g (USGS 2000; University of Michigan 2001; eNature Field Guide 2001).

Killdeers are found in open areas such as plowed fields, golf courses, and short-grass prairies. Their diet primarily consists of aquatic and terrestrial insects. Nesting killdeers have an interesting way of distracting predators. They will act as if they are mortally wounded and lure predators away from the nest. This is achieved by dragging themselves away from the nest, making them look as though they are broken, or hopping around on one foot (USGS 2000; eNature Field Guide 2001; Environment Canada 2000). The call is a "killdeer."

American woodcock (*Scolopax minor*)

Woodcocks winter in the southeastern United States but breed throughout the eastern half of the United States. Year-round populations also occur in the southeastern United States with the exception of peninsular Florida and right along the Gulf Coast. This bird is rather common but nocturnal. It lives in moist woodlands, swamps, and thickets. It is stocky and has short legs, a short neck, and a very long bill. It will allow a close approach and will then explode with whistling wings. The call is a nasal "peent" (Robbins *et al.* 1983).

Family Laridae
Laughing gull (*Larus atricilla*)

Laughing gulls are the largest and darkest of the black-headed gulls. These are medium-sized gulls with long wings and bills. During the summer their heads are black, their back and wings are grey, and the trailing edge of their wings is white. Laughing gulls range in length from 38 to 43 cm and have an average weight of 325 g (USGS 2001b). Laughing gulls are permanent residents of the Gulf Coasts and southern Atlantic Coasts of the United States. This species breeds along eastern coasts from Connecticut to the Gulf of Mexico. During winter months, laughing gulls

nest in areas south of Virginia (USGS 2001b). Laughing gulls are a summer resident in the Calcasieu Estuary (McLaren/Hart-Chemrisk 1998).

Laughing gulls are colonial breeders, nesting with other gulls and terns. They primarily inhabit areas along coasts in either bays, salt marshes, or estuaries. Nesting may also occur in agricultural and industrial areas. Laughing gulls display high nest site fidelity (USGS 2001b).

Laughing gulls are carnivorus scavengers, eating aquatic invertebrates, fish, and carrion. They forage either by using aerial site, diving from the surface, or by skimming the waters surface. Their diet may include small fish, insects, garbage, sewage, and waste from fishing boats (USGS 2000, USGS 2001b). The call is a variety of low chuckles.

Sandwich tern (*Sterna sandvicensis*)

Sandwich terns breed along the southern Louisiana coast and are uncommon. They are found on sandy beaches. No other North American tern has a black bill tipped with yellow. It has a long, slender bill, black legs, and a slight crest. The forehead of immature birds is mostly black. Adults have white foreheads and crowns during the winter. It fishes offshore. The call is a loud, grating "kirri" (Robbins *et al.* 1983).

Gull-billed tern (*Sterna nilotica*)

These pigeon-sized birds are usually found in coastal marshes and on sandy beaches. They breed along the east coast of North America from Long Island to the Gulf of Mexico. They also breed locally in Salton Sea, California (eNature Field Guide 2001). It is the whitest of the North American terns and is larger-bodied than the common tern. It has a short, thick, black, gull-like bill and broad, very white wings. The tail is less forked than in most terns, and the legs are long and black. The flight is more gull-like than that of other terns. It rarely dives but hawks for insects over marshes. It has a characteristic nasal 2- or 3- syllable call (Robbins *et al.* 1983).

Royal tern (*Sterna maxima*)

In North America, royal terns breed along the east coast from Maryland to Texas. They overwinter from North Carolina, the Gulf Coast, and southern California southward (eNature Field Guide 2001) of the Gulf Coast, most of the Atlantic Coast, and the southern Pacific Coast of the United States. This large tern is quite common but is strictly limited to saltwater. It has a crest in all plumages and can be identified at a distance from the smaller terns by the thick, orange bill and slower wingbeat. It has a white forehead, light wing tips, short legs, and a deeply-forked tail. It feeds almost entirely on fish which it catches from inlets or offshore. The call is a "chirrip" (Robbins *et al.* 1983).

Caspian tern (*Sterna caspia*)

Caspian terns are the largest tern. They measure between 48-58 cm in length and have wingspans of about 135 cm. The sexes are similar in appearance. They have a predominantly white to grey body with a black cap and a red bill. North American colonies of Caspian terns can be found as far north as the Mackenzie Valley, the Great Lakes, and Newfoundland. They also range south to the Gulf Coast and Baja California. During the winter, northern populations move south of North Carolina and northern California to their wintering grounds. Caspian terns are usually solitary birds, sometimes coming together in small feeding colonies. Nests are either a small depression in the ground or dead grasses shaped into a bowl (eNature Field Guide 2001; USGS 2000).

Caspian terns forage on sandy and pebbly beaches along lakes, rivers and sea coasts. They almost exclusively eat fish, but also forage on aquatic invertebrates. It is also interesting to note that Caspian terns are more aggressive than most terns, often eating the young and eggs of other terns (eNature Field Guide 2001; USGS 2000). The call is a very loud, harsh "kraaa."

Black skimmer (*Rynchops niger*)

Black skimmers live year-round along the Gulf Coast and the central and southern Atlantic Coasts of the United States. The large, red bill of black skimmers is very distinctive. This is the only bird on which the lower part of the bill is longer than the upper. They fly low over the water, with the lower bill cutting the surface. Upon contact with food, they quickly close their bill. Adults have black backs and sides with white bellies. The call is a loud, low-pitched, resonant "auw"(Robbins *et al.* 1983).

Order Columbiformes

Pigeons and doves are small-headed, short-legged, swift-flying birds with permanent wings and fanned or tapered tails. Females are duller than males. All species coo and bob their heads when walking. They eat grains, small seeds, acorns, and fruits. Nests are generally in trees. There are usually two, white eggs (Robbins *et al.* 1983).

Family Columbidae
Rock dove (*Columba fasciata*)

This common, introduced pigeon occurs throughout the United States. It has a white rump and a dark, terminal tailband. It glides with wings raised at an angle, and the wing tips collide on takeoff. It nests on buildings (Robbins *et al.* 1983).

Mourning dove (*Zenaida macroura*)

This is the most common dove in suburbs and farmlands. It is found year-round in most of the United States. It has a slim body and a long, tapered tail. The flight is swift and direct, without coasting. The whistling wings are characteristic. It feeds in flocks but nests singly. The call is an "ooah-oo-oo-oo" (Robbins *et al.* 1983).

Common ground-dove (*Columbina passerina*)

These doves are resident in the extreme southeastern United States as well as eastern and western Mexico. They are common in brush and farmlands. Their wings flash a bright rufous in flight. On the ground, they look like a miniature mourning dove. Their usually nest on the ground. Their call is a series of identical, low, soft whistles (Robbins *et al.* 1983).

Order Cuculiformes

These slender birds have round wings, curved upper mandibles, and long, graduated tails with short outer tail feathers. They are sluggish birds of forest and brush and eat hairy caterpillars. The sexes are similar. They lay between two and twelve eggs (Robbins *et al.* 1983).

Family Cuculidae
Yellow-billed cuckoo (*Coccyzus americanus*)

These cuckoos breed in most of the United States. They have large, white spots on the black undertail surface, a bright rufous flash when the wings are open, and the lower mandible is yellow. They are found in woods and brush, especially during outbreaks of tent caterpillars, on which they feed. Their song is guttural and toneless (Robbins *et al.* 1983).

Order Strigiformes

Owls are large-headed, short-necked birds of prey. They are mostly nocturnal and can be best seen and heard at dusk. Their large eyes are fixed in their sockets, so they move their entire head to shift their gaze. The flat, round facial disk conceals the large, internal ear flaps. They fly silently hunting for rodents and other mammals. The females are larger than the males. Most small owls and some large owls are cavity nesters and lay between two and eight eggs (Robbins *et al.* 1983).

Family Strigidae
Eastern screech owl (*Otus asio*)

The eastern screech owl is a permanent resident of the eastern United States. It is a common, small, eared owl found in towns, orchards, and small woodlots. Its plumage is bright rusty or gray. It nests in cavities. The song is a quavering whistle (Robbins *et al.* 1983).

Great horned owl (*Bubo virginianus*)

Great horned owls are found year-round in North America. This owl is common and has large ear tufts. It is white with shades of brown and black. The white belly has vertical dark barks (Robbins *et al.* 1983).

Barred owl (*Strix varia*)

Barred owls are permanent residents of the eastern half of the United States and southern Canada. This owl is common in southern swamps, and low, wet woods (eNature Field Guide 2001). It has dark eyes and no ear tufts. It has white bars on brown above and brown bars on white below. In flight, the barred owl resembles the great horned owl. It usually nests in cavities. Barred owls typically hoot about 8 times in a series (Robbins *et al.* 1983).

Family Tytonidae
Barn owl (*Tyto alba*)

Barn owls are permanent residents of most of the United States. This large, light-colored owl is known by its heart-shaped face, small dark eyes, and long legs. Its belly is white and plain or very lightly marked. It is strictly nocturnal and hunts rats and mice in farmyards, marshes, and fields. It has a peculiar habit of lowering its head and moving it back and forth. It nests in barns, tree cavities, and abandoned buildings. It does not hoot but has a soft ascending wheezy cry (Robbins *et al.* 1983).

Order Caprimulgiformes

Goatsuckers are nocturnal insect eaters with large, flat head, small bills, enormous mouths, and distinctive white patches in the wings or tail. Many are named for their call. They usually lay two eggs on the ground (Robbins *et al.* 1983).

Family Caprimulgidae
Chuck-will's-widow (*Caprimulgus carolinensis*)

This goatsucker breeds in the southeastern United States and is common in pine woods. It has no white in the wings, a buff-colored body, and is fairly large. Males have a white throat band, while the female's is buffy. Its call sounds like its name (Robbins *et al.* 1983).

Common nighthawk (*Chordeiles minor*)

Common nighthawks breed throughout the United States and most of Canada. They differ from goatsuckers in their long, pointed wings, slightly forked tails, and white wing patches. They become active before dark, flying above treetops and houses. Nighthawks sit lengthwise on limbs and diagonally on wires. The call is a nasal "peent" (Robbins *et al.* 1983).

Order Apodiformes
Family Apodidae

Swifts feed almost exclusively on flying insects caught on the wing with their wide mouths. They fly continuously, except in heavy rain. Their wings are built for speed and are long, stiff, slender, and slightly decurved. They appear to beat their wings alternately. The sexes are similar. Swifts nest on cliffs, in chimneys, and in hollow trees. The clutch ranges from three to six white eggs (Robbins *et al.* 1983).

Chimney swift (*Chaetura pelagica*)

Chimney swifts breed in the eastern two-thirds of the United States. These solid, dark brown birds are usually seen in flocks. The tail is stiff, slightly rounded, and never

forked or fanned. When chimney swifts are overhead, a noisy chatter of chirping notes is heard. They roost by the hundreds in tall chimneys during migration and form a huge funnel formation at dusk. Their call consists of short, rapid chips (Robbins *et al.* 1983).

Family Trochilidae

Hummingbirds are the smallest North American birds. They all have long, slender bills adapted for reaching deep into tubular flowers. Their wings beat so fast that they produce a humming sound. All species feed while hovering and can fly backward. They migrate during the day, flying low. They lay two small, white eggs (Robbins *et al.* 1983).

Ruby-throated hummingbird (*Archilochus colubris*)

This hummingbird breeds throughout the eastern half of the United States and southern Canada. The adult male has a bright red throat. These tiny birds are green above and white below. They are often detected by the rapid, squeaky chipping made in flight or by the hum of the wings. They are found near tubular flowers (Robbins *et al.* 1983).

Order Piciformes

Woodpeckers have a strong bill that is sharply pointed for chipping and digging into trees for wood-boring insects. The stiff tail is used as a prop. Most species drum on limbs, poles, or something similar. They have undulating flight, with the wings folded against the body after each series of flaps. Their nest is in a cavity chiseled deep into a large tree branch or trunk. Woodpeckers lay between four and eight white eggs (Robbins *et al.* 1983).

Family Picidae
Northern flicker (*Colaoptes auratus*)

The yellow-shafted race of the northern flicker permanently resides in the eastern half of the United States. It is common in open country near large trees. Flickers are about the size of a jay with a brown back, no white on the wings, and a black breast crescent. It has a white rump and yellow under the wings and tail. It is often seen on the ground eating ants and displaying. The call is a loud repeated "flick" or "flicker" (Robbins *et al.* 1983).

Pileated woodpecker (*Dryocopus pileatus*)

Pileated woodpeckers are residents of the eastern United States and most of Canada. They are a wary bird of extensive deciduous or mixed forests, including swamps. Its back is solid black, and it has a conspicuous red crest. It has white under the wings as well as on the neck, and is a strong flyer. Its drumming is distinctive: loud, slow, and softer at the end. Their distinctive call is in a series (Robbins *et al.* 1983).

Red-bellied woodpecker (*Melanerpes carolinus*)

The red-bellied woodpecker occurs year-round through the eastern United States. This bird is common in southeastern woodlands. The mature bird has a black and white laddered back, a red cap and hind neck, while immature birds have brown heads. It has a rattle and a low, short, hoarse call (Robbins *et al.* 1983).

Red-cockaded woodpecker (*Picoides borealis*)

This rare woodpecker occurs throughout the southeastern United States wherever there is suitable old-growth, longleaf pine woods. It is a ladder-backed woodpecker with a solid black nap and cap and large, white cheek patches. Males have a small patch of red behind their eyes (eNature Field Guide 2001). Nest cavities in large longleaf pines are recognized by oozing gum. Red-cockaded woodpeckers are listed as endangered by the state of Louisiana and the United States (Robbins *et al.* 1983).

Red-headed woodpecker (*Melanerpes erythrocephalus*)

Red-headed woodpeckers live year-round in the eastern United States. They prefer open deciduous woods. The adult head is entirely red, and it has a large, white wing area. It has a white belly. Its call is a raucous "kwrrk" (Robbins *et al.* 1983).

Hairy woodpecker (*Picoides villosus*)

Hairy woodpeckers are permanent residents throughout most of the United States and Canada. This bird is common in mature deciduous or mixed forests. This medium-sized woodpecker has a long bill and a vertical, white stripe down its back. The male has a red patch on the back of its head. It has a rattle, and the call is a loud "peek" (Robbins *et al.* 1983).

Downy woodpecker (*Picoides pubescens*)

Downy woodpeckers look like miniature hairy woodpeckers. They are small with short, slender bills. They live year-round in most of North America. The outer tail feathers are barred. The call is a soft "pik," and the rattle descends in pick toward the end (Robbins *et al.* 1983).

18.2.2 Mammals Resident to the Calcasieu Estuary Ecosystem

Order Didelphimorphia
Family Didelphidae
Virginia opossum (*Didelphis virginiana*)

This marsupial is commonly called the opossum or simply possum. The opossum is common throughout Louisiana, occurring in nearly all wooded areas and in the coastal marshes. It ranges throughout most of the eastern United States and extreme southern Canada to Costa Rica. It is not common north of Illinois and Pennsylvania or west of central Kansas and has been introduced into the Pacific coast states. Opossums generally prefer open deciduous forest near permanent water, but they also live in

heavy forest, marshes, wood lots, shrub thickets, forest edge habitats, and agricultural areas (Lowery 1974; Choate *et al.* 1994).

Adults are about as large as a domestic cat but have shorter legs and a heavier body. The fur is long and dense and is interspersed with exceptionally long white guard hairs. Males are slightly larger than females. Females have an external, fur-lined abdominal pouch for carrying the young. Their general color is grayish white above with the fore and hind quarters darker and the belly lighter. The head is conical with a pointed snout. The tail is white and scantily haired except at the base. The front feet have five claw-bearing toes, while the hind feet have four claw-bearing toes and a large first toe that is opposable or thumb-like and clawless. Opossums have more than 50 teeth, more than any other Louisiana land mammal. Their life span rarely exceeds two years. It has a distinctive ambling gait and often hangs from a tree by its tail. It is known for its behavior of playing dead. Dogs, foxes, coyotes, bobcats, and great horned owls will occasionally include opossums in their diets, their biggest predator are cars (Lowery 1974; Choate *et al.* 1994).

One of the reasons for the opossum's success is that it eats almost anything, including insects, fruit, berries, vegetables, birds, eggs, and anything else it can scavenge. Its habits are almost entirely nocturnal. It spends the daylight hours "holed up" and emerges at nightfall to begin foraging for food. Mating occurs in late January or early February. Pregnancy lasts only around 13 days. The young get to the pouch, which contains 13 teats, on their own. The nest is usually in a hollow tree, fallen log, or other secluded place. Weaning takes place when the young are between 75-80 days old. They remain with their mother, riding on her back or in the pouch, for three to four months. A second litter may be produced in early summer (Lowery 1974; Choate *et al.* 1994).

Order Insectivora
Family Soricidae
Southern short-tailed shrew (*Blarina carolinensis*)

The short-tailed shrew occurs throughout the wooded portions of the state, except for the coastal parishes. It is most common in wooded areas but it can also be found in

brushy thickets adjacent to forests. It ranges from Florida westward to Texas and north to southern Illinois and Virginia (Lowery 1974; Choate *et al.* 1994).

This small mammal is dark slate gray (sometimes brownish) above and below. The underparts are only very slightly, if at all, lighter than the upper. The short tail, long snout, tiny eyes, and ears concealed in hair distinguish the short-tailed shrew from any mouse. Males are only slightly larger than females. They are high strung and nervous, and few survive past their first year. The shrew is active year-round. The nest is usually under a tree or fallen stump, usually 30 to 38 cm below the earth's surface, and the gestation period is 21-22 days. The average number of young is 6-7 (Lowery 1974; Choate *et al.* 1994).

The short-tailed shrew will attack and kill prey much larger and more powerful than itself and has no problem with cannibalism. It eats mainly insects, such as ants, ground beetles, caterpillars, or earthworms, some vegetation, and amphibians but will consume any available vertebrate. It has a ravenous appetite and has been known to consume its weight in food every 24 h. The toxic effects of shrew bites are well known. Few animals feed on shrews except for owls. Cats, and sometimes dogs, will capture them but seldom eat them (Lowery 1974; Choate *et al.* 1994).

Least shrew (*Cryptotis parva*)

The least shrew is widespread throughout Louisiana, except in the southeastern part of the state, wherever there are grassy fields, stands of broom sedge (*Andropogon*), fencerows, hedgerows, roadsides, railroad rights-of-way, or thickets along the edges of woodlands. The species lives in the eastern United States, westward to eastern Colorado, and southward through eastern and central Mexico to eastern Panama (Lowery 1974; Choate *et al.* 1994).

This species can be confused only with the short-tailed shrew, but it is much smaller. It weighs about 4.5 g, and its body is not much larger than some bumblebees. Its upper parts are generally brown (less slaty gray), while its underparts are markedly paler. The fur is fine and short but not as velvety as that of the short-tailed shrew. Like other shrews, the sexes are difficult to distinguish (Lowery 1974; Choate *et al.* 1994).

In Louisiana, the shrew reproduces from early spring to mid-fall, and females bear several litters each year. The gestation period is thought to be 21-23 days but may be less, and the average number of young is five. Weaning occurs after three weeks. The nests are in subterranean excavations. Its food includes beetles, bugs, crickets, grasshoppers, earthworms, millipedes, sowbugs, spiders, slugs, snails, salamanders, small frogs, and some plant material. It also enters beehives feeding on larvae and pupae. The remains of this shrew are frequently found in owl pellets. Hawks, snakes, and spotted skunks also prey on shrews (Lowery 1974; Choate *et al.* 1994).

Family Talpidae
Eastern mole (*Scalopus aquaticus*)

The eastern mole is found throughout the uplands of Louisiana. It does not occur regularly in coastal situations, even on higher ground. Grassy prairies, meadows, pastures, gardens, cemeteries, lawns, golf courses, and wooded areas with loose, moist, sandy or light loam soils are all preferred habitats. The mole occurs in the eastern half of the United States. The northern extent of their range is from southeastern South Dakota, central Minnesota, the southern peninsula of Michigan, eastern Pennsylvania, and southern New England (Lowery 1974; Choate *et al.* 1994).

Moles are about the size of small rats but are very distinctive. They have special adaptations for digging and spend about 99% of the time underground in their tunnels. The mole is slate gray with an extremely short tail. They have velvety fur, short front legs, webbed feet, a conical head, a bare nose, no external ears, and very small eyes. They nest in one of the deep tunnels, and the gestation period is about five weeks. Only one litter is produced per year, and the number of young per litter is usually four. Moles reach sexual maturity after about one year and can live up to three years. Moles eat any kind of insect they encounter, as well as larvae, large quantities of earthworms, sowbugs, millipedes, centipedes, and slugs. Occasionally, they eat frogs, small mammals, and plant material. They have relatively few predators, but foxes, skunks, snakes, and owls may occasionally prey on them. Shrews will eat their own babies (Lowery 1974; Choate *et al.* 1994).

Order Chiroptera
Suborder Microchiroptera

Two families of this suborder of bats occur in Louisiana. All bats found in Louisiana are almost exclusively insect eaters.

Family Vespertilionidae

Bats in this family mate in the late summer and fall months. The sperm is stored in the female until the following spring, when ovulation takes place (Lowery 1974; Choate *et al.* 1994).

Southeastern myotis (*Myotis austroriparius*)

This species occurs in the southeastern United States, ranging westward to northeastern Texas and adjacent Oklahoma and northward to the southern parts of Indiana and Illinois. It occurs throughout the state of Louisiana except for the extreme southern and southeast portions of the state. This medium-sized bat, with woolly fir, is gray to brown above, but varies from bright orange-brown and tan to whitish below. The feet have long hair extending beyond the toes. Females are more brightly colored than males (Lowery 1974; Choate *et al.* 1994).

These bats are active year round in Louisiana. They may go into torpor for a few days when the daily temperatures drop below 4.4 Celsius. They use caves, mines and other man-made structures, as well as hollow trees. Maternity colonies are formed in early spring. They leave their roosts early in the evening and proceed to water to drink before foraging. They fly low when feeding. Most breeding takes place during the fall, followed by delayed fertilization. This species bears twins as a rule. Predators include opossums, snakes, and raccoons (Lowery 1974; Choate *et al.* 1994).

Red bat (*Lasiurus borealis*)

The red bat occurs throughout Louisiana except for the extreme southeastern part of the state. It is found throughout the eastern United States and southeastern Canada, south to eastern Mexico. The red bat is migratory in the northern part of its ranges.

Some of them may use Louisiana as an overwintering ground. This medium-sized bat has relatively long, pointed wings. Females average slightly larger than males. The upper parts of the male are brick red, while the females are purplish red. Each shoulder has a yellow-white patch, and they have short, round ears (Lowery 1974; Choate *et al.* 1994).

Red bats are solitary and roost in trees during the day, frequently in Spanish moss. They begin pursuing insects, usually in clearings at heights of 3 to 9 m, shortly after sunset and are often active up to midnight. A bat may return night after night to the same clearing and often searches for food around street lights (Lowery 1974; Choate *et al.* 1994).

Little or nothing is known about the breeding behavior of red bats in Louisiana. Young are born from May to late June, after a gestation period of 80-90 days. They have from one to four young. The mother will carry her small young with her when feeding; they are left behind when they are larger. Since it roosts in open places, it can be affected by severe weather, and it is preyed on by some birds (Lowery 1974; Choate *et al.* 1994).

Seminole bat (*Lasiurus seminolus*)

The Seminole bat occurs in the southeastern United States, except in southern Florida, and along the Atlantic Coast. This species is similar to the red bat except that it is a deep mahogany, with a light frosting above. There is no color difference between the sexes. The Seminole bat inhabits wooded areas, in both deciduous and coniferous trees. It is a solitary bat and roosts in clumps of Spanish moss. The Seminole bat give birth to as many as four young. They are subjected to predation by birds, as well as losses resulting from adverse weather because of their exposed roosting (Lowery 1974; Choate *et al.* 1994).

Hoary bat (*Lasiurus cinereus*)

The hoary bat ranges over most of North America and Mexico, except in the extreme northern part of the continent and southern Florida. It occurs throughout Louisiana except for the southern and southeastern portions of the state. In many parts of its

range, the bat seems to be associated with coniferous forests. It spends the day roosting in trees or shrubs, usually 3 to 5 m above the ground. This bat is easily recognized by its large size (it is the largest bat in Louisiana). It is dark brown or umber with the hairs on the dorsum and belly strongly tipped with white. The face and chin are yellow, and it has short, round ears. Females are slightly larger. The hoary bat is highly migratory. They are strong fliers and normally appear well after sunset. They fly with the tail and interfemoral membrane curved forward under the body, in contrast to the red and Seminole bats which fly with their tails extended except when using the membranes to assist in the capture of an insect. Females bear two young in late spring that are capable of flight after four to five weeks (Lowery 1974; Choate *et al.* 1994).

Northern yellow bat (*Lasiurus intermedius*)

The northern yellow bat occurs along the Atlantic coastal plain, west along the Gulf Coast, along the lowlands of eastern and southern Mexico, across and Yucatan Peninsula, and Cuba. This is the second largest bat in Louisiana and is fairly common in the southern part of the state. It typically inhabits coniferous or deciduous forests near permanent water. It is entirely yellow (sometimes grayish or brownish) with long wings and short ears. Females are larger than males and have two to four offspring. All young are on the wing by the end of June or early July. Yellow bats leave their solitary roosts in the leaves of trees and in Spanish moss well before dark, often as early as sunset. They are strong, direct fliers and are usually recognized by their large size and strong wing beats (Lowery 1974; Choate *et al.* 1994).

Evening bat (*Nycticeius humeralis*)

The evening bat is one of the most common bats in Louisiana and is found in the eastern half of the United States. Its northern range extension is from northeastern Nebraska, northern Illinois, southern Michigan, and central Pennsylvania. This bat is small to medium sized with short, narrow wings. It is dark brown above and yellowish brown below; individuals will occasionally be grayish. The ears are small, thick, and rounded at the tips. Both sexes are similar, and immatures are characterized by their blacker and more scanty pelage. The evening bat roosts communally in hollow trees, under loose slabs of bark, and in attics and abandoned

houses, barns, outbuildings, warehouses, cisterns, and other similar sites. This species does not begin to fly until late twilight. At first it flies above the trees and descends lower as darkness falls. The young, usually two but sometimes only one, are born in late May and June (Lowery 1974; Choate *et al.* 1994).

Rafinesque's big-eared bat (*Plecotus rafinesquii*)

In Louisiana, the Rafinesque's big-eared bat is fairly widespread. It occurs in the southeastern United States, with the exception of southern Florida. This species cannot be confused with any other bats that occur in Louisiana. It has immense ears, more than 2.5 cm in length, and prominent lumps on its nose. It is dark brown above and grayish white below. The sexes are similar, and the young are nearly black. Favorite roosting sites of this colonial species include hollow trees, attics, lofts of barn and other outbuildings, open cisterns, culverts, and old, dilapidated, and abandoned houses in rural areas. This bat is hardly ever seen on the wing since it emerges only after dark. Nursery colonies, consisting of a dozen or more females, are found in spring. In Louisiana, the young are apparently born in May and early June. Females normally bear only one offspring at a time (Lowery 1974; Choate *et al.* 1994).

Family Molossidae
Brazilian free-tailed bat (*Tadarida brasiliensis*)

The Brazilian free-tailed bat ranges across the entire southern part of the United States through Mexico, the West Indies, and Central America, as well as over most of South America. In Louisiana, the bat is virtually statewide (except in the extreme southern part of the state) and is probably one of the most common bats in the state. Most of the Brazilian free-tailed bats in Louisiana migrate to the south in winter. This bat is brownish black above and grayish brown below. It has velvety fur, triangular leathery ears, a black muzzle with vertically arranged wrinkles on the upper lip, a long tail, long hairs on its toes, and narrow wings. Males may be larger, and young do not differ from the adults except for their smaller size and shorter ears. Free-tailed bats prefer the attics of old buildings for their large colonies but are not adverse to roosting in hollow trees, where they sometimes share quarters with the evening bat. There is a mass exodus of bats at nightfall. Mating is in the spring, in a span of a few days,

and it is followed immediately by ovulation and fertilization. Nearly all the young, a single baby for each female, are born within a period of two weeks. Females will accept and nurse any baby that attaches itself to one of her nipples (unlike bats in the family described above). Free-tailed bats in the attic of a building probably have few enemies. Rat snakes and corn snakes are excellent climbers and sometimes find their way into other daytime retreats to prey upon them. In roosts in hollow trees, the bats may be captured by owls and raccoons (Lowery 1974; Choate *et al.* 1994).

Order Edentata
Family Dasypodidae
Nine-banded armadillo (*Dasypus novemcinctus*)

The nine-banded armadillo now occurs throughout the state of Louisiana; it first appeared in 1917. It is now found throughout the southeastern United States, over most of Mexico, and through Central America to southern South America. For the past 100 years or so, it has been expanding its range from southern Texas. No other mammal, except maybe the opossum, is more frequently found dead on highways. The armadillo is about the size of an opossum but heavier. Most of the animal is encased in a bony carapace with bands of triangular "scutes". The head is covered with a plate of scutes, and the tail is encased in a series of bony rings. The ears are thick, leathery, and hairless, and the feet and legs are covered with scutes (Lowery 1974; Choate *et al.* 1994).

Males are slightly larger than females, and sexual maturity is attained at one year of age. Armadillos eat mostly insects, including beetles and their larvae, millipedes, centipedes, and many others, plant food, snails, slugs, earthworms, amphibians, and reptiles. They are predominantly nocturnal but are also frequently seen feeding during the day. The armadillo digs burrows and roots around for food. They cannot stay in these burrows for extended periods of time, something they are forced to do during cold weather. This factor limits the northward range of these armadillos. Breeding occurs in July or August, but implantation is delayed until November. After a 120-day gestation period, four well-developed, identical young are born. The life span of armadillos is about four years. Bobcats, wolves, coyotes, and dogs prey on the armadillo (Lowery 1974; Choate *et al.* 1994).

Order Lagomorpha
Family Leporidae
Eastern cottontail (*Sylvilagus floridanus*)

The eastern cottontail ranges widely over most of Louisiana, except in sections of the coastal marshes. It occurs in the eastern two-thirds of the United States, extreme southern Canada, southern and western Mexico, and parts of Central America. This species is a typical rabbit, with long ears, large hind legs and feet, soft fur, and a short, fluffy tail that is white beneath. The upper parts vary from grayish brown to reddish brown. The nape is rust colored, and the face and flanks are gray. The tops of the feet and belly are whitish, and a cream-colored ring surrounds the eye. Females average slightly larger than males. The young resemble the adults, but the fur is softer and more woolly (Lowery 1974; Choate *et al.* 1994).

The eastern cottontail is most frequently found in forest edge habitats, open areas, pastures, and grassy areas adjacent to croplands. Rabbits eat a variety of grasses, depending on the time of year, such as rye grass, vetch, chufa, oat, soy beans and various truck crops. The nest is a saucer-like depression, filled with grass and hair, that is usually in a dense stand of grass, often between a stand of taller vegetation such as a hedgerow or briar thicket. The breeding season extends throughout the year, although the main period is from February to mid-October. The litter size averages 3.7 in Louisiana. The gestation period is 28 to 32 days, and a female may produce as many as seven litters per year. Young rabbits are weaned and on their own after three to four weeks. Since the female usually mates immediately after birth, she is about ready to deliver another litter (Lowery 1974; Choate *et al.* 1994).

Few eastern cottontails live a long time. They have many predators including owls, foxes, coyotes, bobcats, mink, weasels, striped skunks, crows, dogs, cats, and snakes. Cottontails are also hunted, and the mowing, plowing, and burning of fields takes a heavy toll on the young (Lowery 1974; Choate *et al.* 1994).

Swamp rabbit (*Sylvilagus aquaticus*)

The swamp rabbit occurs in parts of Kansas, Missouri, Illinois, Indiana, Oklahoma, Texas, Arkansas, Louisiana, Mississippi, Alabama, Georgia, and South Carolina

(Lowery 1974; Choate *et al.* 1994). It occurs throughout the state in Louisiana. The swamp rabbit resembles the eastern cottontail but is larger and darker. It is much browner than the cottontail, with black on the back. The eye ring is pinkish cinnamon instead of cream. The sexes are similar. They measure between 45 and 55 cm in length with a 6.7 to 7.1 cm long tail and weigh between 1.6 and 2.7 kg (eNature Field Guide 2001).

Swamp rabbits occur mainly in wooded areas and in the coastal marshes. It is generally replaced by the cottontail in dry, upland cultivated areas and pastures, and it replaces the cottontail in hardwood bottomland swamps and coastal marshes. Swamp rabbits are numerous where canal banks, wooded ridges, and other areas provide dry cover, runways, and resting places but are perfectly at home in dense marshes. They will often hide in the water or swim to avoid predators. Swamp rabbits eat emergent aquatic vegetation and succulent herbaceous vegetation. They feed mostly at night but can be found feeding during the daylight hours, especially after rain (Lowery 1974; Choate *et al.* 1994).

The young are born year-round. The greatest reproductive activity is from late January to the end of September, with a peak between February and May when an abundance of green vegetation is available. The number of young varies from one to six, and the gestation period is 39-40 days. About four or five litters may be produced annually. The nest is a slight depression of earth filled with grasses mixed with rabbit hair. Swamp rabbits have the same predators and pressures as eastern cottontails. Flooding of their habitat by excessive rains (and hurricanes) can also take a heavy toll on these rabbits. The average adult life span is 1.8 years (Lowery 1974; Choate *et al.* 1994).

Order Rodentia
Family Sciuridae
Eastern gray squirrel (*Sciurus carolinensis*)

Grays squirrels are found in the eastern half of North American, including southern Canada. In Louisiana, they are found wherever there are trees except for the coastal marshes. With their long, bushy tails, gray squirrels are gray or grayish brown (sometimes with a yellowish tint) above and white or grayish white below. Both

sexes are similar in size and color. The gray squirrel is readily distinguished from the fox squirrel by its smaller size, overall grayish coloration, and white underparts (Lowery 1974; Choate *et al.* 1994).

Gray squirrels build two types of nests. One is a den in a tree cavity; the other type of nest is made of leaves and twigs that is a waterproof hollow ball or dome placed in the fork of a tree or intertwined among the terminal parts of the branches. The hollow ball nests at the end of branches are most often used for escape and resting, although they are sometimes used for reproduction. Gray squirrels, which have a nervous disposition and are noisy, are active at all hours but are most active in the mornings and late afternoons. They are well known for burying large quantities of nuts and acorns when they are plentiful. In addition to feeding on mast, they also feed on seeds, fruits, buds, flowers, leaves, inner bark, and some insects, bird eggs, and young birds (Lowery 1974; Choate *et al.* 1994).

Breeding activity occurs from late December to late February and again from late May to the middle of August. The gestation period averages 44 days. Therefore, the first litter is born between mid-February and mid-April, and a second litter may appear anytime from June through September. The number of young in a litter is variable but averages close to three. Young squirrels are on their own after three to four months. The female will often leave the den to the young and seek out a new site for her second litter. Gray squirrels become sexually mature after one year. Gray squirrels are usually protected by good cover; however, they are preyed upon by bobcats, foxes, weasels, snakes, hawks, and owls. Squirrels are also hunted (Lowery 1974; Choate *et al.* 1994).

Fox squirrel (*Sciurus niger*)

Fox squirrels are found in the eastern half of the United States. In Louisiana, they are virtually statewide, with the exception of in the coastal marshes and on some coastal islands. These large squirrels are rust colored or black and have long, bushy tails, but this species varies drastically, in color, geographically. Their habitat preference is for rather open situations in hardwood forests or in tracts of mixed hardwoods and pines. They can, however, be found in deep swamps in stands of cypress, tupelo, bitter pecan, and other hardwoods. Where fox and gray squirrels occur together and often

compete for the same habitat, the gray squirrel often appears to be the more successful, replacing its larger relative (Lowery 1974; Choate *et al.* 1994).

The breeding biology of the fox squirrel is much like that of the gray squirrel. In Louisiana, there are two major periods, one in spring and one in summer, when young are born, although pregnant females can be found all year. Breeding begins in late December/early January and declines to a low by late February. In May and June, it reaches another peak, which tapers off in July. A female is capable of producing two litters a year after she is two years old. The gestation period is 45 days, and the average number of young to a litter is three, ranging from one to six. The young are under parental care for about three months (Lowery 1974; Choate *et al.* 1994).

Fox squirrels are not early risers, although they are active in the morning (Lowery 1974; Choate *et al.* 1994). They also are active around midday (except during the midsummer heat) and then again in late afternoon. Practically any kind of vegetable food, whether under the soil or in a tree, is eaten by fox squirrels. They also eat bird eggs and larvae, pupae, adult insects, winged maple seeds, tulip poplar fruit, ripening corn, buds, and different berries when in season. Southeastern populations are more partial to pine cones and fungi. In these locations eastern fox squirrels play an important role in forest management. They eat fungi which is later deposited into the soil in squirrel excrement. Once in the soil, the fungi aids in the germination and growth of trees (eNature Field Guide 2001). Young forest and areas where lumbering has been intense is marginal squirrel habitat. Middle-aged to old forests produce the best mast and, therefore, are prime squirrel habitat. Some predation of the fox squirrel does occur, mostly by hawks.

Southern flying squirrel (*Glaucomys volans*)

Southern flying squirrels are found in the eastern United States, except in extreme southern Florida. They occur in forested areas throughout the state of Louisiana. The flying squirrel is characterized by the fold of skin that extends along the sides of the body from the wrists of the forelegs to the ankles of the hind legs. The body and tail are flat, and it has dense, soft, plain brown fur. The sides and folds of skin are dark brown, and the underparts are white. Flying squirrels have large eyes (Lowery 1974; Choate *et al.* 1994).

This squirrel, which is much more gregarious than its tree squirrel relatives, is seldom seen because of its nocturnal habits. Flying squirrels do not actually fly but glide from tree to tree. Food of the flying squirrel consists mainly of vegetable substances, especially seeds, fruit, berries, acorns, nuts, corn, grain, insects, bird eggs, and small vertebrates. Theirs nests are most frequently in tree cavities, but they occasionally use outside nests made of leaves and twigs. Mating takes place twice a year with litters being produced in spring and again in early fall. Gestations is 39-40 days, and the number of young varies from two to four. The young are able to care for themselves after six weeks. The flying squirrels nocturnal lifestyle and arboreal habitat help to protect it from predation. However, domestic cats and owls will catch a few (Lowery 1974; Choate *et al.* 1994).

Family Geomyidae
Baird's pocket gopher (*Geomys bursarius*)

Baird's pocket gophers occur chiefly in the Great Plains, from western Wyoming and southern and eastern South Dakota north through eastern North Dakota to extreme southern Manitoba, central northern Minnesota, and western Wisconsin south to northeastern Indiana, southeastern Missouri, eastern Arkansas, north-central and western Louisiana, central-southern Texas, and eastern New Mexico. For their predominantly subterranean existence, pocket gophers are among the most specialized of all rodents. They possess massive shoulders and arms, as well as heavily clawed forefeet that enable them to dig underground burrows. This medium-sized gopher varies from light brown to black, with some white spotting around the face. They have large cheek pouches, short hair, tiny eyes, and inconspicuous ears. This genus is easily distinguished by the two longitudinal grooves on each of the upper incisors, which, along with the two lower incisors, lie outside the mouth (Lowery 1974; Choate *et al.* 1994).

Where it occurs, the mounds of dirt that it brings to the surface from its underground excavations dot the landscape. The critical factor governing the distribution of this species in Louisiana is the availability of suitable soils. They can dig their tunnels only in sandy loam; therefore, they are not found in river bottoms, where the soils are heavy and the groundwater is high. During most of the year, a pocket gopher lives a

solitary and highly territorial existence. During breeding season, males will extend their tunnels in search of a receptive female (Lowery 1974; Choate *et al.* 1994).

The gestation period is around 40 to 50 days, and two litters may be produced annually. Young are found from May to December, and the average litter size is three to four. They begin dispersing from parental burrows at two months of age, establishing their own burrows and becoming sexually mature at about three months of age. A strong intolerance exists between immatures and adults and between the immatures themselves. The pocket gopher feeds on a wide variety of roots, tubers, other vegetative matter, and some insects. It obtains most of these items by extending its tunnels, but harvesting operations are sometimes carried out aboveground. Pocket gophers are not often captured by predators because of their underground habits, except occasionally by snakes, weasels, skunks, bobcats, coyotes, hawks, and owls (Lowery 1974; Choate *et al.* 1994).

Family Castoridae
Beaver (*Castor canadensis*)

The beaver ranges over most of North America, with the exception of Mexico, the desert southwest, and most of Florida. It was virtually extirpated or its numbers greatly reduced in many parts of its range because of its great demand in the fur trade in the early history of North America; however, it is making a comeback in many areas. Beavers are the largest rodent in North America. It has a broad, flat, black tail and black feet. The loud slap of their tail on the surface of the water to signal danger is well known. The fur consists of a dense, gray underfur that is overlaid dorsally with long, coarse, shiny guard hairs that give the coat a rich, glossy brown color. The sexes are similar in size and color (Lowery 1974; Choate *et al.* 1994).

Beavers are hard workers. They fell trees of considerable diameter, cut them into sections that they can manipulate, and drag or float them to their dam. The dams that beavers construct in Louisiana are less massive than those built in other areas because the waterways are usually slow-moving, sluggish streams or bayous due to the state's flatness. They build lodges in either open water or on stream banks. Lodge entrances are located beneath the waters surface. Loblolly pine, sweetgum, silverbell, sweetbay, and ironwood are among the woody plants most frequently utilized for dams, lodges,

and food, but they will use many other species. Beavers eat the bark of wood plants but will also consume large quantities of oak mast, aquatic vegetation, and the stems of many plants such as stinkweed, giant ragweed, water hyacinth, cross vine, corn, and switch cane (Lowery 1974; Choate *et al.* 1994).

Beavers are generally considered to be monogamous and to mate for life. However, they are occasionally polygamous. A beaver family usually contains one adult male, one adult female, a one-year old litter, and the succeeding litter. The beaver begins to breed in the second year, and the gestation period is about 120 days. The number of young in a litter is usually two to four. Birth occurs in April or May, and the young nurse for about six weeks. The beaver has few enemies. However, alligators and large carnivores probably account for the loss of a few young (Lowery 1974; Choate *et al.* 1994).

Family Cricetidae
Marsh rice rat (*Oryzomys palustris*)

The marsh rice rat ranges throughout the state of Louisiana and is most abundant in the southern half of the state. It occurs in the southeastern United States and most of Central America. This species is a small rat with a scantily haired tail that is equal in length to the head and body combined. The upper parts are dark gray, mixed with black, and the underparts are grayish white. The tail is brown above and white below, and the feet are white. It has fairly large eyes and prominent ears. The sexes are alike in size and color (Lowery 1974; Choate *et al.* 1994).

Its habitat is always rather wet, marshy places such as canal banks, grassy ditches, the edges of swamps, lakes, ponds, bayous, and streams, and fields with wet soil. The marsh rice rat can be found in marshes not subject to high tides and flooding. It is a good swimmer and diver, and is seldom, if ever, found in dry fields or well-drained woodlands far from water. Rice rats eat mostly seeds and the succulent parts of various available plants, but also eat insects, snails, crustaceans, bird eggs, and baby birds. Rice rats are heavily preyed upon by all hawks and owls and by water snakes (Lowery 1974; Choate *et al.* 1994).

The nest, which is composed of finely shredded dry grasses and sedges, is usually placed either in a slight depression on the ground or above the ground in wet areas in a tangle of vegetation. Rice rates are highly fecund and can produce as many as seven litters per year. However, since the life span of a female is less than one year, the average number of litters is probably no more than five or six. The gestation period is 25 days, and the number of young varies from two to five but is density dependent. Females usually mate within 10 hours of giving birth. Weaning occurs after 11-13 days, and the young are sexually mature after about 50 days (Lowery 1974; Choate *et al.* 1994).

Eastern harvest mouse (*Reithrodontomys humulis*)

The eastern harvest mouse occurs in the southeastern United States. In Louisiana, it occurs throughout the state except for the southern coastal plain (Lowery 1974; Choate *et al.* 1994). This small mouse is brown or black above with ash-colored underparts and has a small tail. The sexes are alike. They measure between 11 and 15 cm in body length and have tails between 4 and 7 cm. Eastern harvest mice range in weight from 10 to 15 g (eNature Field Guide 2001).

The harvest mouse lives in abandoned fields, weed-filled ditches, in briar thickets, and under tangles of honeysuckle and is nocturnal. The food of this species consists almost entirely of weed seeds, grain, green vegetation, and occasionally insects. Harvest mice are preyed upon by hawks and owls, as well as other predators (Lowery 1974; Choate *et al.* 1994).

The nest is either on the ground or above the ground in a clump of grass. Breeding occurs throughout the year with most births taking place between March and November. The gestation period is 21-22 days, litters often contain two or three young. Weaning takes place after two to four weeks, and mice are sexually mature after 12 weeks (Lowery 1974; Choate *et al.* 1994).

Fulvous harvest mouse (*Reithrodontomys fulvescens*)

This species occurs from southeastern Kansas and southern Missouri, south through western Arkansas, southwestern Mississippi, and throughout Louisiana. It is also found throughout most of Oklahoma and Texas, southeastern Arizona, and southward over most of Mexico to Nicaragua. Fulvous harvest mice are golden brown above, sometimes with black down the center of the back. The sides of the face and body are distinctive in that they are tawny or orange. The underparts are of grayish white, and it has a long tail. Both sexes are similar (Lowery 1974; Choate *et al.* 1994).

Fulvous harvest mice are one of the most common small mammals in Louisiana. They occur in uncultivated fields, briar thickets on the borders of woodlands, dense tangles of low vegetation adjacent to fencerows, and similar habitat. They are also common on canal banks and other high ground in coastal areas (Lowery 1974; Choate *et al.* 1994).

Harvest mice reach sexual maturity in two months or less and breed throughout the year. Nests are built 0.3 to 1 m above the ground and are made of finely shreds grasses and sedges. Gestation averages 22-23 days, and two to four young are normally born. An individual that survives for as long as a year has lived a long life. This mouse is seldom active during the day. Its diet consists almost entirely of weed seeds and small invertebrates but occasionally eats green vegetable matter. Fulvous harvest mice are eaten by hawks, owls, and snakes (Lowery 1974; Choate *et al.* 1994).

Cotton mouse (*Peromyscus gossypinus*)

The cotton mouse occurs in the southeastern United States. In Louisiana, it is extremely common and occurs wherever there are large woods or forest. It lives in dense underbrush along edges of streams, in bottomland hardwood forests, hammocks, and swamps, in margins of cleared fields, in old fields, and some upland forested areas. The cotton mouse swims well, and they may prefer wet areas. The cotton mouse has large, black eyes, large ears, white underparts and feet, a bicolored tail, and yellowish brown upper parts. The sexes are similar, and immatures are gray above (Lowery 1974; Choate *et al.* 1994).

Cotton mice are nocturnal and eat insects, spiders, slugs, snails, seeds, and fungus spores. Cotton mice build their nests under logs, in tree holes, and sometimes in buildings. The nest is lined with finely shredded, soft materials. Breeding occurs year round, but most young are born in the late fall and early winter. Young females begin to produce litters when only five to six weeks old. A litter usually consists of four, and the gestation period is about 21 days. Few mice live as long as a year, and they have many predators (Lowery 1974; Choate *et al.* 1994).

White-footed mouse (*Peromyscus leucopus*)

The white-footed mouse occurs in the eastern United States, south to the Yucatan Peninsula. In Louisiana, it occurs statewide with the exception of the extreme southern part of the state. This medium-sized mouse has a short tail and white feet. It is distinctly bi-colored and is grayish brown to reddish brown above and white below (Lowery 1974; Choate *et al.* 1994).

This mouse is common in woodlands in fallen logs, brush piles, and rocks, and in shrubs along fence rows and streams. They are nocturnal and active throughout the year. They construct nests of dry plant material beneath logs, stones, piles of brush and refuse, underground burrows, abandoned bird and squirrel nests, and in hollow trees. They eat seeds, nuts, fruits, other plant materials, insects, and other small invertebrates. Females produce several litters per year. Gestation lasts 22-23 days, and the average litter size is three or four. White-footed mice are significant food items for owls, foxes, bobcats, cats, and snakes (Lowery 1974; Choate *et al.* 1994).

Hispid cotton rat (*Sigmodon hispidus*)

The hispid cotton rat occurs across the southern United States and over most of Mexico and Central America. In Louisiana, it occurs statewide and is one of the most common mammals. This is a medium-sized, robust rat; the tail makes up about 40% of its total length. The coarse fur of its upper parts is black and tipped with yellow or tan. The sides are more uniformly tan, and the underparts are grayish white. The sexes are similar, and immatures are full grown after five months (Lowery 1974; Choate *et al.* 1994).

Hispid cotton rats inhabit edges of woodlands, roadsides, along railroad grades, uncultivated fields, grassy ditches, thickets, tangles of open vegetation, and pine forests with dense stands of broom sedge. It occurs on the high ground of marshes such as canal banks. Cotton rats subsist mainly on vegetable matter, but also eat insects, crayfish, bird eggs, baby birds, and other animal life. Unlike most rodents, this rat is active day and night. It is heavily preyed upon by hawks, owls, and other predators. The average life span of the cotton rat is about six months (Lowery 1974; Choate *et al.* 1994).

Cotton rats are highly fecund. They breed year round, and litters are born in rapid succession. Nests are crudely constructed of grasses and stem fibers and are placed in cup-shaped depressions on the ground, under boards, logs, or rocks, or in clumps of broom sedge. The gestation period is around 27 days, and the average litter is five to seven. Young are weaned after five to seven days, grow rapidly, and are well developed. Sexually maturity is reached as early as 40 days (Lowery 1974; Choate *et al.* 1994).

Eastern wood rat (*Neotoma floridana*)

The eastern wood rat occurs in most of the eastern half of the United States. Wood rats occur throughout the state of Louisiana, except in the extreme southeast portion of the state. Eastern wood rats have dense and soft fur, very large ears, and large and bulging eyes. They are blackish brown above, yellow on the sides, and gray on the cheeks. The underparts are white and the tail is distinctly black above and white below. The white feet are comparatively small for this fairly large rat. The sexes are similar, and young are much grayer than adults. Lactating females possess two pairs of teats in the groin region similar to the udders of a cow. Adults of both sexes have scent glands in the center of the belly that exude a greasy, yellow secretion (Lowery 1974; Choate *et al.* 1994).

Eastern wood rats are common in hardwood bottomland forests and brush-bordering streams, roads, and fields, in rocky upland outcrops, but are rare, or absent, in dry, wooded uplands where pine is predominant. They occur in the coastal marshes on tree islands. Wood rats eat mostly vegetable matter such as buds, seeds, nuts, roots, tubers, succulent herbs, grasses, and berries. They rely heavily on oak mast in the

bottomland swamps, occasionally eating snails and insects. Even though wood rats are almost exclusively nocturnal, they have many predators such as owls, coyotes, foxes, weasels, skunks, raccoons, and large snakes (Lowery 1974; Choate *et al.* 1994).

Wood rats are known for their two types of large nests. One is a huge pile of sticks on the ground at the base of a hollow tree or hollow log. The second type of nest is built above the ground, anchored to the limbs of a small sapling or in a tangle of vines. Which type of nest is built may be related to the degree of forest flooding. They are nicknamed "pack rat" since almost any kind of bright, shiny object can be found in wood rat nests. Eastern wood rats are not nearly as prolific as other rats and mice. They usually reach sexual maturity after seven to eight months. Only two, or possibly three, litters are produced annually, and the usual litter consists of two to four young. The gestation period is about 35 days, and weaning takes place after about 20 days. Wood rats may live two to three years (Lowery 1974; Choate *et al.* 1994).

Muskrat (*Ondatra zibethicus*)

Muskrats occur in Canada, Alaska, and most of the continental United States. In Louisiana, they occur throughout the state and are especially numerous in the coastal marshes. They range inland where rice is grown and are found along bayous and lakes throughout most of southern Louisiana. Muskrats are one of the main staples of the Louisiana fur trade. This fairly large rodent has small eyes and ears. The incisors are outside of the mouth, allowing it to gnaw underwater. The rear feet are webbed with fringes of stiff hairs, and the scaly tail is vertically flattened. The upper fur is dense with guard hairs and is a rich dark brown. The underparts are gray to white. Males are slightly larger than females (Lowery 1974; Choate *et al.* 1994).

Muskrats live in saline, brackish, and freshwater marshes, ponds, sloughs, lakes, ditches, streams, and rivers. They live in family groups and build large houses out of vegetation, in wet areas, using burrows where possible. They reproduce year round. The adult female is capable of producing seven to eight litters per year. The gestation period is 25-30 days, and the average number of young is around six or seven. Muskrats become sexually mature after about two months. Yearly populations of muskrats fluctuate considerably. Common muskrats begin to forage in the middle of the afternoon. They are opportunistic and eat mainly aquatic vegetation but also

consume turtles, crabs, crayfish, mussels, and small fish. Muskrats are eaten by many predators (Lowery 1974; Choate *et al.* 1994).

Family Capromyidae
Nutria (*Myocastor coypus*)

The nutria was introduced into Louisiana in 1938, when 20 animals arrived from Argentina. They multiplied rapidly and escaped into the wild. Because of the rumor that they ate undesirable aquatic vegetation and because their fur brought a good price, people all over the state began to purchase nutria and release them. Nutria occur normally in temperate South America. They have been introduced widely in the United States and are found throughout the state of Louisiana. They have done great damage to marshes, dikes, levees, and crops and have caused the decline of the muskrat populations (Lowery 1974; Choate *et al.* 1994).

The nutria is a large robust rat, almost equal in size to a beaver. It has huge front teeth and small ears. The fur is dense, soft, and slate overlaid with long, glossy dark brown or yellowish guard hairs. The side of the face and body are often yellow, and the chin and the opt of the muzzle are white. The tail is cylindrical, and the feet are black. Nutria are excellent swimmers, and the hind feet are webbed (Lowery 1974; Choate *et al.* 1994).

Nutria prefer freshwater marshes and normally eat about 1.1 to 1.6 kg of aquatic vegetation per day. They usually eat species that man does not want controlled, passing up the more undesirable species. They are most active at night, consuming food both on land and while floating in the water. Nutria dig burrows in canal banks or levees or take over old burrows. Similar to most rodents, the nutria is quite prolific. The number of young in a litter averages 4.5, and the gestation period is about 130 days. Weaning occurs after five to seven weeks. Sexual maturity is reached after four to eight months. The alligator is the nutria's main predator, but the young are captured by many predators that live in the swamps and marshes (Lowery 1974; Choate *et al.* 1994).

Order Carnivora
Family Canidae
Coyote (*Canis latrans*)

The coyote ranges widely in North America and occurs throughout the state of Louisiana. This dog-like canid resembles a small German shepherd. The color of the coyote is variable. Typically, the underparts are light gray to pale yellow with black guard hairs. The muzzle, back of the head, base of the ears, and nape are strongly tinged with yellow or buff. The area of white around the lips of the coyote is narrow and sharply demarcated. The upperside of the tail, like the back, is white and yellow below, with a black tip. The throat, belly, and inside of the legs are white to pale yellow, and the outside of the hind legs is reddish. The tail of the coyote is usually held low while running (Lowery 1974; Choate *et al.* 1994).

Males average slightly larger than females. The vocalizations of the coyote consist of barks, yelps, and yaps in a series that often starts out as a solo performance but becomes a chorus as other members of the pack join in. The coyote inhabits grasslands, brushy areas, forest edges, and both upland and bottomland forests. They live singly, in pairs, or small packs. The dens are in gullys, under roots or overhanging banks, and in places studded with thickets and dense cover. Coyotes breed in late winter/early spring. The gestation period lasts for about 63 days, and the young usually number from five to seven. Both parents care for the young. After 8 to 12 weeks, the young begin to learn to hunt for themselves. Coyotes eat rabbits, rats, mice, and other wild mammals, birds, carrion, insects, and plants. Man is the coyote's only serious predator (Lowery 1974; Choate *et al.* 1994).

Red fox (*Vulpes vulpes*)

The range of the red fox extends over most of North America to the southern United States. It is absent from the extreme southeastern United States, much of the Great Plains, the Pacific coast, the Great Basin, and southern Texas. In Louisiana, it occurs statewide except in the extreme southern portions of the state. The red fox can be found in mixed oak-pine wooded uplands interspersed with farms and pastures or in the vicinity of cane fields in bottomlands. This small dog-like canid is predominantly reddish-yellow in color. The tail is reddish-yellow, except the end is black, with a

white tip. The nose pad, ears, legs, and feet are black, while the cheeks, throat, and belly are white. It has long, erect ears, and a long, pointed nose (Lowery 1974; Choate *et al.* 1994).

The red fox is principally nocturnal, but it is active during the day, especially at twilight. It eats mainly small mammals such as rabbits, rats, mice, birds and bird eggs, carrion, insects, fruits, and plant materials. Males are slightly larger than females. The voice is a short yelp terminating in a throaty sound or a yapping scream. Red foxes excavate their dens in banks or gullies or take over dens of another animal. Red foxes are monogamous and mate for life. Breeding takes place in late fall or early winter and usually four to five young are born after a gestation period of 53 days. Both parents help with the raising of the kits. Weaning takes place when the young foxes are about two month of age, and they leave the den when about six months old. They become sexually mature after about a year. Humans and their dogs are predators of the red fox. However, some kits may be lost to owls and other predators (Lowery 1974; Choate *et al.* 1994).

Gray fox (*Urocyon cinereoargenteus*)

The gray fox ranges throughout Louisiana except in the extreme southern part of the state. This species occurs in the United States, except for the northwest portion of the country, and ranges throughout Mexico, most of Central America, and into northwestern South America. The gray fox is easily distinguished by its gray coloration, smaller size, and black-tipped tail. The back of the ears, sides of the neck, band across the lower throat, the legs, and the sides are tawny or reddish brown, while the belly, the borders of the upper jaw, and the throat are white (Lowery 1974; Choate *et al.* 1994). Both sexes of the gray fox are similar. Kits are black at an early stage but soon begin to show the patterns of the adults.

Gray foxes prefer mixed pine-oak woodlands bordering on pastures and fields with weed patches. It is not often seen because of its crepuscular and nocturnal habits. The fox spends its days in a dense thicket, in a hollow tree or log, or in an underground den. Gray foxes regularly climb trees. Their food consists of rats, mice, rabbits, adult and larval insects, birds and their eggs, and plant material such as berries, fruits, corn, and acorns (Lowery 1974; Choate *et al.* 1994).

Mating of gray foxes occurs in late winter. After a gestation period of 53 to 63 days, usually three to five young are born. Both parents participate in the raising of the young. The pups begin to hunt for themselves by the time they are three months old, but often remain together as a family unit until the following fall. They begin to breed the next year. The average life span is four to five years. Other than humans and their dogs, the gray fox has few enemies. Young foxes are preyed upon by bobcats, coyotes, and owls (Lowery 1974; Choate *et al.* 1994).

Family Ursidae
Black bear (*Ursus americanus luteolus*)

Black bears were formerly widespread in North America. In many areas, the species has been extirpated or its numbers have been vastly reduced. In Louisiana, remnant populations still exist. Black bears are listed as a threatened species by the state of Louisiana and the United States. The few bears that remain in Louisiana today are confined to the large expanses of hardwood swamps. Black bears are huge, bulky animals with long, dense, glossy black hair. The tail is very short and inconspicuous. It has a blunt face, small eyes, and a yellowish brown muzzle. Males average somewhat larger than females, and the normal life span is fifteen years (Lowery 1974; Choate *et al.* 1994).

During the day, bears bed down in dense thickets, canebreaks, and rock or brush shelters. They are normally nocturnal but sometimes are active during the day. Bears den in a variety of situations, including road culverts, hollow logs, and tree cavities. They are excellent climbers and often have their dens in cavities well above the ground. Bears sleep for extended periods in their dens. In Louisiana, they may leave their dens for short periods during favorable weather in midwinter (Lowery 1974; Choate *et al.* 1994).

Mating occurs from May to July. The gestation period for black bears is 100 to 210 days, and young are often born while the female is in her den for the winter. Usually two cubs are born, and they remain with the mother through the following fall, sometimes longer. The female first gives birth at three years of age and breeds in alternate years thereafter. Bears are omnivorus and as much as 95% of their diet consists of vegetable matter. They occasionally eat mice and squirrels, but oak mast,

field corn, muscadines, blackberries, and honey are consumed in large quantities. Their only predators are humans and their dogs (Lowery 1974; Choate *et al.* 1994).

Family Mustelidae
Long-tailed weasel (*Mustela frenata*)

The long-tailed weasel occurs from southern Canada to Peru, with the exception of a small area in the southwestern United States and northwestern Mexico. It is found throughout the state of Louisiana, except in the extreme south. This small, long-bodied animal with short legs and a long tail can hardly be confused with any other mammal. It is uniformly brown above, with yellow or white below. The fur is fine and short, and one quarter or one third of the well-furred tail is black. Both sexes are similar in appearance (Lowery 1974; Choate *et al.* 1994).

Weasels live in forest edge habitats as well as in brushlands, woodlands, fencerows, and sometimes agricultural and urban areas. Weasels are aggressive, swift, and nimble and forage at all hours. Their main food is rats, mice, voles, and rabbits, but they occasionally capture small, ground-feeding birds, amphibians, reptiles, and invertebrates. They can climb and swim, although not expert at either. They run with an arched back. Weasels breed in the summer, and a single litter, averaging five to eight, is born the following spring, usually in April, after a gestation period ranging from 205 to 337 days (implantation is delayed). The den of a weasel is usually in a rotten log, hollow stump, or a hole in the ground. The nest chamber is floored with grasses and mouse or shrew fur. Females reach sexual maturity after about four months, while males reach maturity after about a year. Predators of the weasel include owls, snakes, foxes, dogs, and coyotes (Lowery 1974; Choate *et al.* 1994).

Eastern spotted skunk (*Spilogale putorius*)

Eastern spotted skunks are found throughout most of the western United States, the southeastern United States, and most of Mexico and Central America. In Louisiana, they are found throughout the state, except in the southeastern part of the state. This small skunk, about the size of a squirrel, has glossy jet black fir and a bushy tail. It has four pairs of long, symmetrically arranged white streaks with white spots on the

head, chin, back, rump, and sides of the basal portion of the tail. Females are smaller than males (Lowery 1974; Choate *et al.* 1994).

These skunks live in a variety of habitats, including pastures, woodlands, forest edge, croplands, fencelines, hedgerows, and farmyards. When a spotted skunk is confronted by an enemy, it performs numerous displays. If that doesn't work, it emits a fine vapor, that is strongly-scented and pungent, from its anal glands. Spotted skunks are mostly nocturnal but may be seen at twilight. Insects, such as grasshoppers, crickets, and ground beetles, constitute a large part of the skunk's diet, but it also eats rats, mice, reptiles, salamanders, frogs, crayfish, small birds, bird eggs, fruit, and corn. It makes its home in natural cavities and crevices, hollow logs, old armadillo holes, and shallow depressions under tree roots, as well as under old buildings, sheds, piles of lumber, and other similar places. Spotted skunks breed during late winter, and the young are born in the spring after a gestation period of about 60 days. The litter usually contains four to five young, and weaning occurs at around 54 days. A second litter may be produced in late summer. Females begin breeding at about 9-10 months of age. Predators of the spotted skunk include dogs, owls, foxes, coyotes, and bobcats (Lowery 1974; Choate *et al.* 1994).

Striped skunk (*Mephitis mephitis*)

Striped skunks occur widely throughout southern Canada and the United States south to northern Mexico but are locally absent in some places, such as the deserts of the southwestern United States. In Louisiana, they are widespread but absent from the extreme southeast part of the state. Striped skunks are about the size of a domestic cat. They have triangular-shaped heads, short legs, and long and bushy tails. A narrow white stripe starts on the forehead and extends to the shoulders where it divides into longitudinal stripes. The amount of white on striped skunks is extremely variable. The striking feature, as with all skunks, is the presence of two anal glands that produce an oily, smelly substance (Lowery 1974; Choate *et al.* 1994).

These skunks prefer open habitats or forest edge. Wherever striped skunks are found, their presence is made known by their scent. They are frequently seen dead on the road. Striped skunks are not quick to use their scent; they usually do so after much provocation and numerous displays. Striped skunks use old dens of foxes, armadillos,

and other mammals, or refuse dumps, stumps, or beneath old buildings for their homes. Skunks mate in late winter and after a gestation period of up to 75 days, five to seven kits are born. Weaning occurs after eight to ten weeks, and the offspring stay with their mother until fall. Striped skunks eat large quantities of insects, along with many rats and mice. They also eat frogs, salamanders, crayfish, bees, bird eggs, and small amounts of plant material. Owls, coyotes, bobcats, foxes, and dogs are known to prey on striped skunks (Lowery 1974; Choate *et al.* 1994).

Family Felidae
Bobcat (*Lynx rufus*)

Bobcats occur in southern Canada, the entire United States, and most of Mexico. They range throughout the state of Louisiana, except in the extreme southern portion. Bobcats are medium-sized cats, larger than domestic cats. They are long-legged and have a ruff of fur on their short faces that appear as sideburns. Their general color is yellowish-brown or reddish-brown streaked and spotted with black. The underparts are white with black spots and bars. The tail is reddish-brown above and white below with dark bars and a white tip. Males are larger than females. Kittens are heavily mottled (Lowery 1974; Choate *et al.* 1994).

The bobcat seems to prefer areas with dense understory vegetation, including heavily wooded uplands and bottomland forests, brushy areas, swamps, and partly open farmlands. Rabbits are the most significant component of a bobcat's diet; however, they also eat squirrels, rats, mice, small birds, fawns, livestock, and carrion. It hunts at all hours and either sneaks up or lies in wait for its prey. Breeding takes place in mid-winter and after a gestation period of about 62 days, two to three kittens are born. The den is usually under an uprooted tree but may be in a cane thicket. The kittens stay with their mother until the following spring (Lowery 1974; Choate *et al.* 1994).

Order Artiodactyla
Family Cervidae
White-tailed deer (*Odocoileus virginianus*)

White-tailed deer are found throughout Louisiana where there is suitable habitat (Lowery 1974; Choate *et al.* 1994). They occur throughout southern Canada and the

United States, except in some of the western states, Mexico, and Central America into South America. This deer has long legs, hoofed toes, a naked nose pad, and a moderately short tail that is white beneath. The upper parts are reddish in summer and grayish brown in winter, and the underparts are white. Males have antlers during part of the year and usually weigh between 68 and 141 kg. However, some males can reach 200 kg. Females are smaller than males and weigh between 41 and 96 kg. Body size ranges from 68 to 114 cm in height, 1.88 to 2.13 m in length, with a tail from 15 to 33 cm (Environment Canada 2000; eNature Field Guide 2001). The fawn is reddish-brown above with white spots. When a white-tailed deer is alarmed, it will snort, stomp on the ground, and/or raise its tail, revealing its bright white marker. This acts as a warning signal to other deer. The white tail also helps fawns follow their mothers as they flee the disturbance (eNature Field Guide 2001).

White-tailed deer are browsers and grazers and are very adaptive. The food plants utilized by deer in Louisiana vary widely according to the particular habitat and what is available. Important plants eaten include mulberry, trumpet creeper, titi, strawberry bush, yellow jasmine, holly, willow, honeysuckle, dewberry, and greenbrier. Oak mast and the wild pecan are particularly important for deer. Over the course of a day white-tailed deer will consume between 2.25 and 4 kg of food. They will also drink water from a variety of sources such as rain, snow, dew, or other water source. White-tailed deer have a four-part stomach which enables them to consume foods that other animals cannot (eNature Field Guide 2001).

Breeding of white-tailed deer takes place from October to January. After a gestation period ranging from 195 to 212 days, twins or a single fawn is born. Sexual maturity is reached after about 1.5 years. White-tailed deer are usually nocturnal, bedding down in protected areas as morning approaches. They are excellent runners and can reach speeds around 58 km/hr. They are also capable of jumping 2.6 m vertically and 9 m horizontally (eNature Field Guide 2001). Few deer live longer than ten years (Lowery 1974; Choate *et al.* 1994).

Order Sirenia
Family Trichechidae
West Indian manatee (*Trichechus manatus*)

The West Indian manatee occurs along the coast and in coastal rivers of the southeastern United States from North Carolina southward to southern Florida and westward in the Gulf of Mexico to southern Texas and Veracruz, and through most of the West Indies and the Caribbean waters of Central America to northern South America. The manatee is only a casual occurrence along the central northern Gulf Coast. The manatee is listed as endangered both by the state of Louisiana and the United States. This aquatic species is unique among mammals of North America. Boat propellers and, more recently, diseases and cold weather have killed large numbers of manatees. It has a round body with a small head, no visible neck, forelimbs modified as flippers, no hind limb, spatulate tail, overhanging lips with bristles, absence of hair except for a few stiff bristles, and lack of external ears. It is dull gray to black in color. Adults measure 2.4 to 4.3 m in length and weigh from 136 to 544 kg. Males average somewhat larger than females. The gestation period is about 180 days, and young (usually one) at birth weigh 18 to 27 kg and measure slightly over 1 m. Females begin to breed at the age of three or four years. This gentle giant is a strict vegetarian and consumes large amounts of aquatic vegetation (Lowery 1974; Choate *et al.* 1994).

Order Cetacea
Family Delphinidae
Common dolphin (*Delphinus delphis*)

Like other dolphins, common dolphins are very social and playful animals. They are rarely found alone and school in groups from a few dozen to several thousand (University of Guelph 2000). Common dolphins are wide-ranging. They can be found in the Mediterranean, Black, and Red Seas, the Atlantic and Pacific oceans, as well as the Gulf of Mexico. They are small dolphins weighing between 100 and 136 kg and measuring 1.5 to 2.4 m in length. Females are usually smaller than males. Common dolphins have black to grey backs and creamy-white bellies. They have long pointed beaks containing between 40 and 61 teeth per side. Their teeth are specially designed to hold onto slippery fish. Common dolphins have a

distinguishable hourglass pattern that runs along their side (University of Michigan 2001; University of Guelph 2000).

Common dolphins breed in the spring and fall. The gestation period lasts for 10 to 12 months. Females usually give birth to one baby; however, twin and triplet births have been seen. Babies come out tail first, measuring about 1 m in length and weighing between 11 and 16 kg. Sexual maturity is reached between the ages of 12 and 15 years. The total life span of common dolphins is believed to be from 35 to 40 years (University of Michigan 2001).

Common dolphins can be found in coastal waters as well as deeper waters further out to sea. They will often move with schools of prey fish. Examples of prey include herring, pilchard, anchovies, nocturnal hake, sardines, small bonito, and sauries. They also forage on squid and octopus. A common dolphin can consume up to 9 kg of fish per day (University of Michigan 2001). Common dolphins usually feed in groups. They will circle prey and take turns charging into the middle of the school, swallowing their catch whole. Another foraging technique involves chasing prey to the surface, bumping the fish out of the water, and catching it in midair. Common dolphins have few predators, though they do have to watch out for sharks and killer whales (University of Michigan 2001).

Atlantic spotted dolphin (*Tenella frontalis*)

Atlantic spotted dolphins are born gray and become more spotted as they age. This spotting aids in the determination of age and the individual identification of the dolphin. This dolphin measures 2.3 m. Atlantic spotted dolphins feed on squid and fish and commonly occur over the continental shelf in the Gulf of Mexico (MMS 2001).

Pantropical spotted dolphin (*Tenella attenuata*)

These slender dolphins are born gray, spotting as they age. Their lips are a bright white. This species feeds on fish and squid. This spotted dolphin can reach a length of 2.6 m. Pantropical spotted dolphins are the most abundant dolphin in the Gulf of

Mexico. They are commonly found along the continental slope in the north-central and western Gulf of Mexico (MMS 2001).

18.2.3 Amphibians Resident to the Calcasieu Estuary Ecosystem

Order Caudata
Family Ambystomatidae
Spotted salamander (*Ambystoma maculatum*)

Spotted salamanders are found throughout Louisiana in pine-hardwood forests. This is a stout and sluggish salamander that is bluish-black, black, or brownish-black with rounded yellow or orange spots in two rows from the head to the end of the tail. Its belly is dark gray, and it averages 1.5 to 1.75 cm in size (Dundee and Rossman 1996; Conant and Collins 1998).

This is a secretive species, largely subterranean, and its presence is often undetected except during the brief breeding season. It appears to prefer soils with some clay content and a preponderance of hardwood trees. It hides in rodent burrows, in logs, or beneath logs and litter. In wet stream bottomlands or woodlands where the water table is high, it may be found anytime. Spotted salamanders eat worms, arthropods, snails, and slugs (Dundee and Rossman 1996; Conant and Collins 1998).

Large numbers of them can be found crossing roads on rainy nights during the breeding season, which occurs from late December to late February. Several hundred animals may use a single, temporary pool for breeding. Up to 300 eggs are laid in clusters, attached to submerged sticks or leaves. The eggs hatch within a few weeks, and larvae grow to a length of 4.0 to 7.5 cm before transforming between late March and June (Dundee and Rossman 1996; Conant and Collins 1998).

Marbled salamander (*Ambystoma opacum*)

Marbled salamanders occur throughout Louisiana north of the coastal marshes. This stocky, sluggish salamander, ranging in length from 10.2 to 12.7 cm, is bluish-black with white or silvery crossbands on the back and tail. This common, but secretive,

species is most often encountered in bottomlands or in wet woodlands under logs, boards, and debris. It can be found under logs any time of the year (Dundee and Rossman 1996; Conant and Collins 1998).

Marbled salamanders eat millipedes, centipedes, spiders, insects, and snails. This salamander breeds during the fall (October through January) and lays its eggs on land in protected, moist depressions, usually under logs. The eggs are guarded by the female and hatch when rain floods the depressions. Larvae are dark brown or black and transform in the early spring (Dundee and Rossman 1996; Conant and Collins 1998).

Small-mouthed salamander (*Ambystoma texanum*)

This medium-sized salamander (14 cm) is relatively slender with a narrow head and small mouth. It varies in color from dark slate to bluish-black or brown, usually with lichen-like markings more numerous and lighter on the lower sides. This species occurs throughout Louisiana except in the coast marshes (Dundee and Rossman 1996; Conant and Collins 1998).

This species occurs in many habitats but is most prevalent in wet woodlands and bottomlands where hardwoods are common. It uses all sorts of ponds and ditches as breeding sites. It can be encountered under logs, when the water table is high, and in crayfish burrows. Eggs, laid singly or in clusters of two or three, are deposited on sticks, vegetation, and under leaves on the bottom of the pool. Breeding occurs from December to February, and the larvae transform in March and April (Dundee and Rossman 1996; Conant and Collins 1998).

Family Amphiumidae
Three-toed amphiuma (*Amphiuma tridactylum*)

This amphiuma occurs statewide in all sorts of freshwater habitats. It is a large, eel-like salamander that reaches 107 cm in length. It has two pairs of tiny nonfunctional legs, each with three toes. It is dark brown to black above, its belly is light gray, and there is a dark patch on the throat. Eggs are laid from April through early September and are attended to by the female. The incubation period may be five months. There

is some evidence that this species may be ovoviviparous. Nests are in damp places, and larvae lose their gills immediately after hatching. They eat crayfish, earthworms, fish, skinks, snails, and insects (Dundee and Rossman 1996; Conant and Collins 1998).

Family Plethodontidae
Dwarf salamander (*Eurycea quadridigitata*)

This salamander is found throughout Louisiana, except in the coastal marshes. It is a small (rarely more than 8 cm in length), terrestrial salamander that is yellowish brown to brown with a dark dorsolateral stripe on each side. It sometimes has black specks on the back, and the underside is yellow (Dundee and Rossman 1996; Conant and Collins 1998).

This is an abundant species that occupies spring seeps, swampy ground, damp hardwood and pine forests, pine margins, and stream bottomlands. Dwarf salamanders presumably eat small invertebrates. It is usually found hidden beneath cover objects, such as logs and boards, but also crosses roads on rainy winter nights. Breeding occurs from October through February. White eggs are laid singly or in small groups and are attached to leaves and twigs. Transformation of the larvae usually takes place from mid-April to late summer (Dundee and Rossman 1996; Conant and Collins 1998).

Family Salamandridae
Eastern newt (*Notophthalmus viridescens*)

Newts occur statewide, except for the coastal salt marshes. There are two forms of this animal: the eft, which is an immature phase found in terrestrial situations, and the adult newt, a secondarily aquatic form that develops after the eft stage or that may develop directly from larvae without entering the eft stage. The eft stage is dull olive to red with scattered black specks and is up to 8 cm in length. The adult is up to 12 cm in length. It has a compressed tail and fins and is brown to olive with a black-speckled yellow belly (Dundee and Rossman 1996; Conant and Collins 1998).

Efts are most commonly found in woodlands under logs, boards, and debris. The aquatic phases are found in ditches, woodland ponds, swamps, and sluggish streams. They eat all sorts of invertebrates, especially arthropods and snails, and may feed on salamander eggs on occasion. Eggs are laid, singly, and attached to vegetation, from January to June (Dundee and Rossman 1996; Conant and Collins 1998).

Family Sirenidae
Lesser siren (*Siren intermedia nettingi*)

Sirens are found statewide in freshwater habitats. This unusual, neotenic salamander rarely exceeds 3 cm in length in Louisiana. It has an eel-like body, no hind limbs, four toes on each foreleg, and bushy gills. Its color ranges from grayish-blue to olive, brown, or black with scattered dark dots (Dundee and Rossman 1996; Conant and Collins 1998).

This species' major habitat is ditches and ponds, but it also utilizes small and large streams, swamps, woodland pools, and sloughs. They are known to occur in brackish water. Sirens are secretive and usually burrow into the mud and debris of the bottom. If the habitat dries up, this salamander can burrow into the bottom and form a cocoon to protect it from desiccation. Sirens eat snails, crustaceans, and insects. Not much is known about the reproduction of this species, but eggs probably hatch in mid-winter in Louisiana (Dundee and Rossman 1996; Conant and Collins 1998).

Order Anura
Family Bufonidae
Gulf Coast toad (*Bufo valliceps*)

The Gulf Coast toad is found along the Gulf Coasts of Texas and Louisiana. It mainly occurs in south and southeastern Louisiana. Some of these toads may also be found in Arkansas and Mississippi (USGS 2001a). This is an easily recognized large toad, attaining a maximum length of nearly 13 cm in Louisiana (Dundee and Rossman 1996; Conant and Collins 1998). Males are considerably smaller than females. Toads have dry, warty skin, and they hop. This toad has sharply-ridged, well-developed cranial crests, a conspicuous yellowish cream mid-dorsal stripe, and a broad yellowish cream dorsolateral stripe on each side. The color is variable; some are black and

orange, while others are predominantly white and yellow. This toad appears to predominate in agricultural and wet hardwood areas. They have been found in salt marshes and on the coastal prairie. During the winter they can be found under logs in hardwood forests.

Vocalization, a short trill lasting 2-6 seconds, has been heard from April through the middle of September (Dundee and Rossman 1996; Conant and Collins 1998). Eggs are laid in the usual long gelatinous string characteristic of toads, often in shallow pools. Tadpoles are dark with light dorsal saddles. This toad probably breeds in brackish waters in Louisiana. Larval development takes place in 20-30 days, and the tadpoles may reach a length of 2.5 cm. Insects are common prey for these toads (USGS 2001a; eNature Field Guide 2001).

Order Anura
Family Bufonidae
Woodhouse's toad (*Bufo woodhousei*)

This toad is moderately-sized (5 to 8 cm) with low, narrow, and sharply-ridged cranial crests. It has a light mid-dorsal stripe and is yellowish to gray with dark brown or black dorsal spots (each with three or more warts). This toad is highly variable since it hybridizes with Gulf Coast toads and southern toads (*Bufo terrestris*). These toads are found throughout Louisiana and have been found on high ground in the coastal marshes and in salt marshes (Dundee and Rossman 1996; Conant and Collins 1998).

Woodhouse's toad's call is a nasal "w-a-a-h" lasting 1-2.5 seconds. They breed from March through early July. The eggs are usually arranged in single, sometimes double, lines. The dark tadpoles have a darkly-pigmented throat patch. The larval period may last as long as 60 days. This toad normally breeds in temporary pools but may also use quiet pools in streams (Dundee and Rossman 1996; Conant and Collins 1998).

Family Hylidae
Northern cricket frog (*Acris crepitans*)

This frog is found throughout the state of Louisiana, most often along the margins of freshwater lakes and streams. A race of this species has been observed in pools in

coastal marshes. This small, agile, ground-dwelling treefrog reaches about 3.5 cm in length. The back has elongate dark spots on a gray to green or rust background, sometimes with a prominent green, yellow, or rusty-orange mid-dorsal stripe. It has numerous vertical dark and light bars on the snout, a dark triangle between the eyes, a white belly, and an irregularly edged dark stripe on the back of the thigh (Dundee and Rossman 1996; Conant and Collins 1998).

These treefrogs typically remain on the ground, except during heavy rains, and rarely stray from water. They make huge leaps when disturbed, plunging into the water and hiding submerged among vegetation. They are active all year as long as the weather is mild. Northern cricket frogs eat tiny arthropods, especially insects and spiders, as well as some snails, worms, crustaceans, and other invertebrates (Dundee and Rossman 1996; Conant and Collins 1998).

Northern cricket frogs breed from March through September, may call in any month, and are more prone to call during the day than any other frog in Louisiana; however, its choruses are greatest at night. The call is a repeated "gick" and sounds like two pebbles being rubbed together. Breeding takes place in a pond or pool. During breeding, males fertilize the eggs as they are released one at a time by the female. The developmental period is 40-90 days. Tadpoles have a black-tipped tail. Growth is rapid, and adult size may be reached two months after metamorphosis (Dundee and Rossman 1996; Conant and Collins 1998; University of Michigan 2001).

Green treefrog (*Hyla cinerea*)

Green treefrogs are found south of Delaware into Florida and the Florida Keys. From there they range west into Texas, north through central Arkansas, west to Tennessee and Illinois. Green treefrogs range throughout Louisiana wherever freshwater breeding sites are available. This medium-sized treefrog reaches a length of 6.5 cm. It is usually light to dark green with some scattered golden flecks and a well-defined, sharp-edged white or yellow stripe extending from the upper lip almost to the groin. The posterior edge of the lower hind limb and foot also has a light stripe (Dundee and Rossman 1996; Conant and Collins 1998). Like other treefrogs, green treefrogs have large toe pads and long legs.

These treefrogs are most abundant in forested areas or around water courses, ponds, and lake margins (Dundee and Rossman 1996; Conant and Collins 1998). They can be found on leaves, in crevices, under the eaves of buildings, or on almost any vertical surface. They hide under bark and logs in cool weather. Green treefrogs often congregate in large groups of several hundred frogs (University of Michigan 2001; eNature Field Guide 2001). Their food primarily consists of arthropods, such as snails, beetles, and spiders. These frogs are eaten by large aquatic birds, ribbon snakes, garter snakes, water snakes, raccoons, and fish.

Green treefrogs call from April to September. Males usually call from perches above the water. Their call is a monotonous "quonk, quonk, quonk" and sounds like the dull ringing of a cowbell. Breeding sites range from temporary ditches to lake, slough, and lagoon margins. They have been reported to breed in brackish marshes and shallow pools in coastal marshes. The eggs are laid in surface masses attached to floating vegetation. Tadpoles have mottled tail fins, a light stripe from eye to snout, and sometimes two light body blotches. Transformation has been observed from July through late October (Dundee and Rossman 1996; Conant and Collins 1998).

Squirrel treefrog (*Hyla squirella*)

Squirrel treefrogs are found in the southern United States, from Virginia to the Florida Keys, west to central Texas, and as far north as Mississippi. Squirrel treefrogs are abundant in southern Louisiana lowlands and flatlands where suitable temporary freshwater pools may form (Dundee and Rossman 1996; Conant and Collins 1998). They also occur in hardwood stream valleys in scattered locations in central and northern Louisiana. This small treefrog (about 4 cm) varies from green to brown, often with irregularly distributed small, dark spots on the back, a poorly-defined wavy or ragged-edged light stripe from the upper lip to the anterior part of the side. It can be confused with small green treefrogs. Males and females are very similar.

Squirrel treefrogs are nocturnal, feeding on insects during the night and hiding or sleeping under cover during the day (University of Michigan 2001; eNature Field Guide 2001). These treefrogs will gather around lights where they feed voraciously on insects (Dundee and Rossman 1996; Conant and Collins 1998). They prefer shallow, weedy, temporary pools for breeding sites and have been know to breed in

brackish marshes. Rain will stimulate the males to call. They will call during the day before rains. Breeding choruses have been heard from March to November. The call is a raucous "waak, waak", usually from a low perch on the bank in emergent vegetation. Around 1,000 eggs are enclosed in two jelly envelopes and deposited on the bottom of the breeding pool. The tadpoles are greenish with a pigmented throat and mottled fins. The larval period lasts at least 40-50 days.

Gray treefrog (*Hyla chrysoscelis-versicolor* Complex)

Gray treefrogs are found throughout Louisiana. The taxonomy of this species is unresolved. These moderately large treefrogs (6 cm) have granular, warty skin. The back is various shades of gray to brown to green, with large, irregular, dark blotches forming elongated projections. The rear of the thighs is yellow or yellowish-orange with dark markings, and there is a light spot under the eye (Dundee and Rossman 1996; Conant and Collins 1998).

The gray treefrog is a forest dweller, but is frequently encountered in pastures and open areas. Gray treefrogs eat mainly insects. They can be found on vegetation and under cover in the winter. They are extremely arboreal, and males can be heard calling high in the trees. Males have been heard from mid-March through September. The call is a harsh or melodious trill. Breeding sites include semipermanent farm ponds, woodland pools, and especially ditches. Up to 2,000 eggs are laid in small packets, either free-floating or attached to vegetation. The tadpoles have large, dark blotches on long, red-finned tails. The larval period is from 45 to 65 days (Dundee and Rossman 1996; Conant and Collins 1998).

Spring peeper (*Hyla crucifer*)

Spring peepers occur throughout Louisiana except in coastal marshes. This small treefrog (3.5 cm in length) is brown to brownish-olive with a dark transverse mark between the eyes and a large, x-shaped mark on the back. The spring peeper is a woodland species. It does not ascend into vegetation much higher than low shrubs. Spring peepers eat mostly arthropods (Dundee and Rossman 1996; Conant and Collins 1998).

Spring peepers have been heard calling in Louisiana from November through May. Most of the calling is at night. The call is a clear, single-note whistle that is repeated. Eggs have been observed from January through April. Up to 1,000 eggs are deposited singly and attached to vegetation in temporary or semipermanent pools. The tadpoles are not easily recognized and transform into tiny frogs 0.9 to 1.4 cm in length. At least 45 days are required for transformation (Dundee and Rossman 1996; Conant and Collins 1998).

Striped chorus frog (*Pseudacris triseriata*)

These chorus frogs occur throughout the state of Louisiana. They have been found in cypress forest-bottomland hardwood habitats in the coastal marshes. This small, slender frog (up to 3.5 cm in length) has a variable dorsal pattern, usually consisting of three longitudinal dark stripes, streaks and/or spots on a gray to brown color. A dark triangular mark is usually present between the eyes, the upper lip has a light stripe, and a dark stripe passes from the snout, through the eye, and along the side to the groin (Dundee and Rossman 1996; Conant and Collins 1998).

This ground-dwelling frog occurs in all kinds of habitats from cultivated fields to forests but is especially abundant in forested areas. This species is also sometimes found in low, weedy vegetation or under groundcover. It eats small insects and invertebrates. Striped chorus frogs are winter-breeders. Calls are heard from late October through early April. The call, which is a grating chirp like running your thumb along the teeth of a stiff comb, is usually heard at night; however, rain can stimulate calling during the day. Breeding sites are shallow, temporary pools, ditches, and flooded field where emergent vegetation or a grassy margin is present. The males usually call from the vegetation. Females lay 500-1,500 eggs in clusters attached to vegetation. The tadpoles have dark dots on the back, a bicolored, striped tail, and freckled tail fins (Dundee and Rossman 1996; Conant and Collins 1998).

Family Ranidae
Bullfrog (*Rana catesbiana*)

Bullfrogs occur throughout Louisiana wherever permanent freshwater habitat exists. This is the largest frog in the United States (maximum length 20 cm). The back is

green or brown, with scattered black dots, mottling, or a maze of curving dark marks. Its belly is yellow, often heavily mottled. Dorsolateral ridges are absent from its trunk, it has a large eardrum, and the toes of its hind feet are extensively webbed (with longest toes projecting beyond web; Dundee and Rossman 1996; Conant and Collins 1998).

Bullfrogs can be found around almost any lake, pond, slough, permanent ditch, sluggish stream, freshwater marsh, or swamp, virtually anywhere that freshwater provides a year-round refuge. Bullfrogs are highly aquatic, and often yelp as they leap into the water when disturbed. They frequent the water's edge and lie dormant under water during cold weather. They eat anything that will fit into their mouth; including insects, crayfish, fish, frogs, salamanders, snakes, birds, mice, bats, and others (Dundee and Rossman 1996; Conant and Collins 1998).

In Louisiana, breeding begins in March and extends throughout the summer. Bullfrogs often call during the day, usually from a bank or on top of floating vegetation in their respective territories. Their call is unmistakable and sounds like a resonant "brrr woooom." The egg mass of up to 20,000 eggs is an enormous floating film deposited among water plants and shrubs in the water. In Louisiana, the larval period may be up to five or six months. The large tadpoles are green with black dots; the belly is white or yellow and mottled, and dots are on the fins (Dundee and Rossman 1996; Conant and Collins 1998).

Green frog (*Rana clamitans*)

Green frogs are found throughout Louisiana wherever suitable freshwater pools are available. This frog is plain brown to dull bronze with indistinct, irregularly-placed, dark spots on its back, with a white belly. It has extensively webbed toes and dorsolateral ridges on its back (Dundee and Rossman 1996; Conant and Collins 1998).

This species is the most frequently encountered ranid frog in Louisiana. It is prevalent in swamps, marsh margins, and forested areas, where it prefers pools but also occurs in streams. When disturbed, it squawks as it jumps into the water. It eats gastropods and arthropods. Water snakes and ribbon snakes often prey on green frogs (Dundee and Rossman 1996; Conant and Collins 1998).

The call of the green frog sounds like a plucked banjo. It can be heard day and night from mid-March to September. Males are usually well-spaced in quiet, shaded pools. Breeding sites may be permanent or temporary pools. The 1,000 to 3,000 eggs are deposited as a surface film in open water or attached to vegetation. Tadpoles have an olive green back with dark spots. The tail is long and mottled or black, and the fins are low. The larval period is estimated to be 70-85 days. They may overwinter as a result of late breeding activity (Dundee and Rossman 1996; Conant and Collins 1998).

Southern leopard frog (*Rana sphenocephala*)

Southern leopard frogs are found from southern New York State to Florida, in the west from eastern Texas north to eastern Kansas, in northern Missouri, southern Illinois and Indiana, and in the south of Kentucky and Tennessee (USGS 2001a). Southern leopard frogs are widely-distributed throughout Louisiana near any temporary or permanent freshwater (Dundee and Rossman 1996; Conant and Collins 1998). This gray, tan, or green medium-sized frog (13 cm) has rounded, elongated, irregularly-placed dark brown or black spots on its back. The toes of the hind feet are webbed for more than half their length, and the eardrum has a spot in the center.

This frog frequents virtually every water body, although it tends to be displaced in wet woodlands and swamps by the green frog. In more exposed situations, leopard frogs are most likely to be encountered around permanent pools. They appear to tolerate some degree of brackishness. They feed mainly on insects, earthworms, and other invertebrates. Predators, such as birds, snakes, and raccoons, feed extensively on leopard frogs (Dundee and Rossman 1996; Conant and Collins 1998).

Leopard frogs call (day and night) and lay eggs throughout the year, but the major breeding period is from December through February. The call is varied and may be a series of clucks, a low guttural trill, or a low-pitched chuckling. The flat egg mass (1,000-1,500 eggs) is either attached to vegetation or lies on the bottom in shallow water. The tadpoles are olive to yellowish with dark spots, and the larval period ranges from 50 to 75 days (Dundee and Rossman 1996; Conant and Collins 1998).

Family Microhylidae
Eastern narrow-mouthed toad (*Gastrophryne carolinensis*)

These interesting toads range through the southeast of North America and into the Florida Keys. They are also found west to east Texas and Oklahoma (University of Michigan 2001). Narrow-mouthed toads occur throughout the state of Louisiana (Dundee and Rossman 1996; Conant and Collins 1998). This squat, short-limbed frog reaches an adult size of 2.5 to 3.8 cm. It has a pointed head, a transverse fold of skin behind its eyes, and its toes are not webbed. The transverse fold of skin can be flipped forward over their eyes, acting like a windshield wiper to rid their bare eyes of insects. Its color is gray, brown, or reddish-tan, with a wide, dark area that tapers toward the head. Males are distinguished by their dark throats. Their backs and underbellies are covered with light and dark mottlings. They are a smooth-skinned toads, which is rare.

Forested areas are the best place to find narrow-mouthed toads, especially in hardwood floodplains. They will also use meadows if suitable ground cover exists. They have been found on beaches and under boards in salt marshes. These toads are also excellent at burrowing and can disappear under the ground in a matter of minutes. They primarily breed in temporary pools, but will occasionally breed in salt water. These small toads are gregarious and secretive during the day. They eat all sorts of arthropods and a few snails, but ants, termites, and beetles are the major food items. They have irritating skin secretions (Dundee and Rossman 1996; Conant and Collins 1998).

Narrow-mouthed toads breed from late April to September. Males have been heard calling from late March through early October, usually at night but sometimes during the day. Males prefer to call from clumps of grass in the water. The call is a low "b-zzzz" for a second or so and sounds like a bleating lamb. Up to 1,000 eggs are laid in masses that form a surface film. Hatching occurs quickly (within a few days), and the tadpoles resemble wide, miniature frying pans. The larval period lasts from 20-70 days (Dundee and Rossman 1996; Conant and Collins 1998).

18.2.4 Reptiles Resident to the Calcasieu Estuary Ecosystem

Order Crocodylia
Family Alligatoridae
American alligator (*Alligator mississippiensis*)

American alligators are the largest reptiles found in North America. This very large lizard-like reptile has tough leathery skin overlying bony plates on its back and tail (Dundee and Rossman 1996; Conant and Collins 1998). Females are smaller than males, measuring between 1.8 and 2.4 m with males reaching lengths up to 5 m. They have a long, broadly rounded snout and protruding eyes and nostrils. The back is uniformly dark in adults, but it is black with yellow crossbands in juveniles. They have short, stout legs and a large tail that accounts for about half of their body length. American alligators occur throughout Louisiana where there is suitable habitat.

Alligators occur in many bodies of still or slow-moving water such as lakes, bayous, swamps, canals, and ditches. However, the species is most abundant in the extensive coastal marshes of Louisiana. They enter salt marshes on occasion, but do not nest there. In a swamp or marsh or along the banks of streams or lakes, they will dig large holes that are connected to large underground dens to use during drought or as a winter retreat. Alligators are voracious predators and will eat any animal they can capture and crush with their powerful jaws (Dundee and Rossman 1996; Conant and Collins 1998).

Female alligators do not breed until they are nearly 10 years old and at least 1.8 m long (Dundee and Rossman 1996; Conant and Collins 1998). Courtship and mating occur in April and May, and nest building and egg laying takes place from late May to early July. During the breeding season, males are territorial and bellow loudly. In southwestern Louisiana, the female constructs the nest mound from marsh vegetation she strips from an area near the mound. She lays from 2 to 58 eggs and guards the nest during the incubation period (ranging from 62-65 days). The incubation temperature determines the sex of the embryos. When the young alligators are ready to hatch, they begin to call. She helps them hatch and get to the water. Newborns are only 15 to 20 cm long when they hatch. Mothers are very protective of their newborns, guarding them from predators such as racoons, birds, and other predators.

Despite this protection, 88% of newborns will fall victim to raccoons or birds. Hatchlings usually stay with their mother until the following spring. Growth is rapid, about a foot per year. In terms of range, female alligators occupy smaller areas than males. Males will occupy areas greater than two square miles. Both sexes increase their range during courtship and breeding (University of Michigan 2001).

Order Testudines
Family Chelydridae
Common snapping turtle (*Chelydra serpentina serpentina*)

Snapping turtles are found throughout the state of Louisiana in freshwater and the coastal marshes. This large, aquatic turtle reaches almost 50 cm in carapace length. Adults weigh between 4.5 and 16 kg and can weigh up to 34 kg. It has a large head, long neck and tail (2/3 to 1 3/4 the length of the carapace), and its eyes are visible from above. The carapace is dark with three longitudinal rows of keels (Dundee and Rossman 1996; Conant and Collins 1998).

This turtle may be found in almost any kind of freshwater situation, particularly permanent ponds, lakes, and streams. It also enters brackish waters in marsh areas. Although highly aquatic, snapping turtles can be found on land, such as when crossing roads. They are active at all hours. Snapping turtles rarely bask out of water. If they do, it is usually on a mud flat. Snapping turtles have powerful jaws and eat sponges, worms, mollusks, insects, crustaceans, fishes, amphibians, reptiles, birds, mammals, carrion, and aquatic vegetation. Snapping turtles fall prey to alligators, and the young are eaten by birds, mammals, snakes, and fish (Dundee and Rossman 1996; Conant and Collins 1998).

Nesting occurs from May through September, and 20 to 30 tough-shelled, spherical, white eggs are laid. The nest is on land as much as several hundred meters from the water. Incubation time varies between 55 to 125 days (Dundee and Rossman 1996; Conant and Collins 1998).

Alligator snapping turtle (*Macroclemys temminckii*)

Alligator snapping turtles occur statewide in Louisiana. This is one of the world's largest freshwater turtles, reaching a carapace length of at least 80 cm and a weight of over 137 kg. It has a huge head with a strongly hooked beak and a long tail. Its eyes are not evident from above. It has a very rough, dark brown carapace with three rows of keels, but it also has prominent dorsal keels and an extra row of scutes on each side of the carapace (Dundee and Rossman 1996; Conant and Collins 1998).

These huge turtles are most commonly found in large rivers, canals, lakes, and oxbows, but they also enter swamps near rivers. In the coastal marsh, they are most evident in the freshwater lakes and bayous. They have a pink process on the floor of their mouths that resembles a wriggling worm. They lie still at the bottom of a lake or river with their mouth open facing upstream and use this lure to catch fish. Nesting takes place from April to early June, and about 25 eggs are laid. The eggs are spherical, tough, and white. The hatchlings are almost black in color (Dundee and Rossman 1996; Conant and Collins 1998).

Family Kinosternidae
Common musk turtle (*Sternotherus odoratus*)

Musk turtles are found throughout Louisiana except for the brackish areas of the coastal marsh. This turtle attains a maximum length of 14 cm. The carapace is olive brown to black, often with dark spots or streaks. The carapace is smooth in adults but has a prominent mid-dorsal keel and smaller keels on the sides in juveniles. The toes are webbed, the head has two white or yellow stripes on each side, and barbels are present on the chin and throat (Dundee and Rossman 1996; Conant and Collins 1998).

The musk turtle occurs in a variety of fresh waters. The criteria are that the water be permanent, currents are absent or slow, and the bottom is soft. It prowls on the bottom for long periods of time and only occasionally climbs out of water to bask. It exudes foul-smelling musk from its glands.

Musk turtles are omnivores. Brittle-shelled eggs are laid from March through July. The clutch averages one to four eggs and they are deposited in dug nests, in the open,

under debris, or in decaying logs. Incubation is 60 to 85 days (Dundee and Rossman 1996; Conant and Collins 1998).

Mississippi mud turtle (*Kinosternon subrubrum hippocrepis*)

Mud turtles occur throughout Louisiana. This small, aquatic turtle does not exceed 12.4 cm in carapace length. It has two light lines on each side of the head. The carapace is smooth in adults but has two weak lateral keels and a mid-dorsal keel in young. The carapace is olive to black, and the plastron is yellowish brown to dark brown in adults and red in hatchlings (Dundee and Rossman 1996; Conant and Collins 1998).

Mud turtles are associated with shallow waters of ditches, ponds, swamps, bayous, and other bodies of water where the bottom is soft and there is little water movement. They are particularly abundant in the coastal marsh, often frequenting distinctly brackish water. Mud turtles rarely bask. They burrow in mud or into rotting logs to estivate or hibernate. When mud turtles are disturbed, they produce a foul-smelling musk. Mud turtles are omnivorus and eat snails and frogs. The reproductive season is long, ranging from December through July. Clutches average two to four eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Family Emydidae
Three-toed box turtle (*Terrapene carolina triunguis*)

This land turtle reaches a carapace length of 22 cm. It occurs throughout the state of Louisiana, in high areas of coastal marshes. The carapace is either high domed and plain brown or has a variable pattern of orange or yellow spots, blotches, or radiating lines. It has three toes on the hind foot. The plain plastron has hinges so the entire body and legs can be enclosed within the shell (Dundee and Rossman 1996; Conant and Collins 1998).

Box turtles primarily inhabit open woodlands, but also turn up in fields wherever forests are nearby. They may be found under logs during cool weather. During the winter, box turtles hibernate in burrows or under deep leaf litter. They may lie in shallow water during hot weather. Juveniles are rarely seen, probably because they

uses burrows and forage under leaf litter (Dundee and Rossman 1996; Conant and Collins 1998). Box turtles have small, well-defined home ranges.

Box turtles are omnivorus, eating plants, fungi, smaller animals, insects, and carrion. They mate in the spring, and nesting occurs from May through October. Eggs are deposited in excavations 8 to 10 cm deep and then covered. They can live a long time, up to 100 years (Dundee and Rossman 1996; Conant and Collins 1998).

Ornate box turtle (*Terrapene ornata*)

These turtles range from the southern regions of South Dakota, Iowa, and east Illinois, south to Louisiana and Texas, and west to southwest Arizona (eNature Field Guide 2001). Within the state of Louisiana, ornate box turtles are classified as critically imperilled (McLaren/Hart-Chemrisk 1998). These turtles occur mainly in southwestern Louisiana along the coastal prairies but may be found almost anywhere in the state (Dundee and Rossman 1996; Conant and Collins 1998).

This small turtle reaches a carapace length of just over 15 cm. The carapace is flattened on top and is brown or black with a bold pattern of yellow lines, often with a mid-dorsal yellow stripe. The toes are slightly webbed, and the hind foot has four toes. The plastron has a strong pattern of yellow lines on a dark color and is hinged so that the entire body can be enclosed within the shell. Males have either red or orange eyes and a larger tail than females. The eyes of female ornate box turtles are brown (eNature Field Guide 2001; WDNR 1999).

Ornate box turtles inhabit prairies and savannahs. During hot weather, they gather in large numbers in shallow ponds. They burrow to hibernate. They eat mainly insects and earthworms but also consume vegetation. They will also eat carrion when it is available. Berries are eaten during the appropriate seasons, as are dandelions. Ornate box turtles usually fulfill their water needs from the plant and animal material they eat, but will drink water directly if necessary (eNature Field Guide 2001; WDNR 1999).

These turtles mate in the spring and nest from early May to mid-July. Two to eight eggs are laid in shallow cavities and covered with dirt. The eggs hatch after about

two months of incubation. Juvenile turtles are rarely encountered. These turtles may live as long as 50 years (Dundee and Rossman 1996; Conant and Collins 1998).

Mississippi diamondback terrapin (*Malaclemys terrapin pileata*)

This terrapin is found in the saline portions of the coastal marsh of Louisiana and reaches a carapace length of 24 cm. Diamondback terrapins have a dark brown or black carapace that has strongly developed concentric grooves, ridges on each scute, and a prominent tuberculate mid-dorsal keel. The plastron is yellow, and the head and legs are gray with dark spots. The toes are webbed, and the head is often very broad (Dundee and Rossman 1996; Conant and Collins 1998).

This is a turtle of brackish waters. Within the coastal marsh, it prefers the more open channels. They may bask on mud or float in channels. Terrapins spend the night and hibernate buried in mud. They have salt glands to excrete excess salt. Terrapins eat snails, clams, crabs, and vegetation.

In Louisiana, the terrapin grows quickly. Females are mature in six years and lay many clutches of 4 to 12 eggs per year, from April through July. Nests are made in sandy areas above the high tide water level (Dundee and Rossman 1996; Conant and Collins 1998).

Red-eared slider (*Trachemys scripta elegans*)

These turtles get their name from the red-orange spot behind their eyes and the speed with which they retreat when they feel threatened. They are found from Indiana to New Mexico and from Texas through to the Gulf of Mexico. Sliders occur in virtually all freshwater habitats throughout the state (Dundee and Rossman 1996; Conant and Collins 1998). They also enter brackish waters and coastal marsh ponds. These turtles have a broad, red stripe behind the eye, sometimes widened to form a reddish spot. Their toes are webbed and their chin is rounded. The carapace is rough with narrow, curving ridges. It is green with fine, yellow lines in juveniles. Adult carapaces are olive or brown with less obvious yellow lines. The plastron is yellow with paired, dark blotches. Old males are completely dark. Red-eared sliders are sexually dimorphic. Males are smaller than females, but have a larger tail and longer

front nails. In terms of size, red-eared sliders range from 13 to 20 cm (University of Michigan 2001; TPW 1996).

These turtles are very abundant in all sorts of pond, lake, and ditch habitats, and only occasionally inhabit areas with moving waters (Dundee and Rossman 1996; Conant and Collins 1998). They prefer muddy bottoms and large amounts of aquatic vegetation. They typically feed in the early morning. The diet of young red-eared sliders is about 70% meat and 30% plant matter. As adults this changes to 10% plant matter and 90% meat (University of Michigan 2001). Foraging takes place under water and includes the consumption of crayfish, aquatic insects, snails, and fish, as well as plants like arrowheads, water lilies, hyacinths, and duck weed. These turtles crowd onto projecting logs on sunny days. Huge numbers of slider nests are dug up by skunks, raccoons, and snakes. Many predators eat young and adult turtles.

Mating occurs in spring and fall, and eggs are laid in Louisiana from late March to mid-July. The average clutch is seven eggs. Red-eared sliders produce up to 3 clutches a year. The elliptical, leathery, white eggs are placed in a small excavation above the water and then covered with dirt. The eggs hatch in 68 to 70 days. Sexual maturity is reached in two to five years, and these turtles may live for 50 to 75 years (Dundee and Rossman 1996; Conant and Collins 1998).

Missouri cooter (*Pseudemys floridana hoyi*)

Cooters are found throughout Louisiana in sluggish streams, ponds, lakes, and ditches. This large, freshwater terrapin reaches 30.5 cm in length. Its toes are webbed, its head has numerous yellow stripes on the top, bottom, and sides; the underside of the chin is flat. The carapace is rough with narrow, curving ridges and is brown with yellow lines. The plastron is yellow or orange-yellow. It eats aquatic vegetation, insects, sponges, and bryozoans. The cooter is fond of basking on logs. Little is known about the reproductive habits of this turtle (Dundee and Rossman 1996; Conant and Collins 1998).

Western chicken turtle (*Deirochelys reticularia miaria*)

Chicken turtles occur throughout the state, except in the coastal marshes. This aquatic turtle attains a maximum length of 25 cm. It has vertical yellow stripes on the posterior surface of the hind legs and a broad yellow stripe on the front of the foreleg. It has webbed feet and an extremely long neck. The carapace is usually dark olive with a pattern of lighter-colored lines and numerous ridges (Dundee and Rossman 1996; Conant and Collins 1998).

The chicken turtle is almost exclusively a resident of ditches, ponds, and lakes. It rarely occurs in streams. It wanders and frequently turns up in temporarily flooded woodland pools. Chicken turtles are omnivorus. Little is known about its reproduction in Louisiana (Dundee and Rossman 1996; Conant and Collins 1998).

Family Cheloniidae
Loggerhead (*Caretta caretta*)

Loggerhead sea turtles are listed as threatened by the state of Louisiana and the United States Loggerheads have a large, block-like head, a reddish-brown carapace, and a yellow plastron. Adults may weigh more than 227 kg. The carapace length of adults averages about 1.1 m. They generally inhabit warm water over the continental shelf of the Atlantic basin, ranging as far north as Canada and as far south as Argentina. (LDFW 2001)

Loggerheads regularly enter marshes, estuaries, and coastal rivers. Well-drained dunes with clean sand and scattered, grassy vegetation is ideal nesting habitat. In Louisiana, loggerheads have been found throughout the coastal region, but nesting has been recorded only from the Chandeleur Islands. Mating takes place in late March through early June. In Louisiana, nesting occurs between May and August, peaking in late June. Females lay an average of 2 clutches of 60-80 eggs per season, on 2- or 3-year intervals. Sexual maturity is reached between 16 to 40 years of age. Loggerhead sea turtles are omnivorus and feed on aquatic plants, crustaceans, mollusks, jellyfish, squid, sea urchins, and fish (LDFW 2001).

Kemp's ridley (*Lepidochelys kempii*)

Kemp's ridleys are the only sea turtle with an almost circular carapace, which may be dark gray, brown, black, or olive. Ridleys are the smallest sea turtles, weighing between 36 and 45 kg with a carapace length of 50-70 cm. These sea turtles are listed as endangered by the federal government and the state of Louisiana (LDFW 2001).

Kemp's ridley sea turtles are primarily restricted to the Gulf of Mexico, although juveniles may be carried by the Gulf Stream. Most of the nesting is restricted to one beach near Rancho Nuevo, Tamaulipas, Mexico. Although this sea turtle does not nest in Louisiana, the estuarine and off-shore water provide key feeding and developmental sites. They prefer warm bays and coastal waters, tidal rivers, estuaries, and seagrass beds. Deep water channels and estuaries provide ideal hibernation sites. Kemp's ridleys eat crabs, crustaceans, mollusks, fish, jellyfish, squid, and starfish (LDFW 2001).

Family Trionychidae
Pallid spiny softshell (*Apalone spinifera pallida*)

Softshells occur throughout Louisiana and have been recorded from brackish areas of the coastal marsh. This medium-sized to large, flattened, aquatic turtle reaches 54 cm in carapace length. The snout is tubular, the toes are webbed and clawed, the neck is long, and the body is covered with a soft, leathery shell. It is olive-gray to yellowish brown in color. Tiny projections on the upper surface of the carapace make it feel like sandpaper (Dundee and Rossman 1996; Conant and Collins 1998).

The spiny softshell is found in lakes, oxbows, lagoons, borrow pits, drainage ditches, and rivers. Nesting occurs from May through August. Females produce clutches of 4 to 32 eggs, probably several times per year. The eggs hatch from late August to October (Dundee and Rossman 1996; Conant and Collins 1998).

Order Squamata
Suborder Lacertilia
Family Polychridae
Green anole (*Anolis carolinensis*)

Green anoles occur throughout the state of Louisiana (Dundee and Rossman 1996; Conant and Collins 1998). These diurnal lizards are found in the Southeastern United States, from east Texas to southern Virginia. They are often called American chameleons because they can change color. Green anoles measure between 13 to 20 cm, males generally being larger than females. They most often appear bright green, but can also be grey or brown. Green anoles have long, skinny bodies and pointed heads. Males can be distinguished from females by their pink dewlap or throat fan, used for courtship and in defense of territory (University of Michigan 2001; eNature Field Guide 2001). In coastal marshes, they are found on high ground and levees.

Anoles occur in deep forest, but seem to prefer dense shrubs in more open areas. Large populations occur in urban areas. During the winter, they retreat under boards, beneath the bark of dead trees, in rotting logs, and at the base of palmettos. They feed on insects and other small arthropods. They typically eat flies, beetles, moths, and spiders. Prey is stalked and must move in order for the green anole to see it (University of Michigan 2001; eNature Field Guide 2001).

Breeding begins as early as February. Female produce a single egg approximately every two weeks throughout the breeding season and deposit them under leaves, logs, or other debris. Growth is rapid, and individuals reach sexual maturity in less than a year. They breed the first spring following hatching (Dundee and Rossman 1996; Conant and Collins 1998).

Family Teiidae
Six-lined racerunner (*Cnemidophorus sexlineatus*)

Racerunners range throughout Louisiana except for the Mississippi alluvial plain. In the western part of the state, they occur in the coastal marshes, in high areas. This large lizard (27cm) has a long, whip-like tail. The body is dark with six, narrow, light stripes, often with a green tint on the sides of the body (Dundee and Rossman 1996; Conant and Collins 1998).

This species prefers open, well-drained, sandy habitats. They burrow under bushes and in holes in cliff banks. Racerunners are the fastest-moving lizard in the state and prefer the sun. On overcast days, they hide under boards and logs. Arthropods are

the main food item for racerunners. Clutch size averages one to eight eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Family Scincidae
Ground skink (*Scincella lateralis*)

These diurnal lizards are wide-ranging in North America. They are found from New Jersey to Kansas through to the deep south and the Gulf of Mexico. In Louisiana, this skink occurs statewide (Dundee and Rossman 1996; Conant and Collins 1998). This shiny, brown lizard is small to medium-sized (14.5 cm) with a pair of dark dorsolateral stripes, a white or yellow belly, and smooth, flat scales. It is a common lizard and occurs anywhere that will support lizards, from deep forests to city backyards. They usually occur under logs, boards, or other debris but they prowl at the surface in dead leaves and clumps of grass. Ground skinks eat small insects, spiders, and earthworms. Eggs are laid from May through September, and the clutch size varies from one to five eggs. Parents provide no care for their young (eNature Field Guide 2001).

Five-lined skink (*Eumeces fasciatus*)

Five-lined skinks occur throughout Louisiana. They occur in coastal marshes in higher areas and on natural levees. These skinks are fairly large (up to 21 cm) and shiny with flat, smooth scales. During the breeding season, the head of large males is orange red. Small skinks are black with narrow white or yellow stripes and a blue tail. Large adults are brown with a gray tail and have dull stripes (Dundee and Rossman 1996; Conant and Collins 1998).

Five-lined skinks are found in pine and hardwood forests where habitats are usually damp. They are often found on the sides of living trees, on, in, and under logs, beneath bark on stumps and dead trees; under slabs and chips in sawdust piles; and beneath debris in trash piles. This species feeds on a wide variety of arthropods and small vertebrates. Eggs are laid from May through August. The female guards her clutch that averages seven to nine eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Broadhead skink (*Eumeces laticeps*)

Broadhead skinks are found throughout the state of Louisiana. They do not occur in the coastal marshes and are rare in the southwestern part of the state. Small skinks are black with narrow white or yellow stripes and a blue tail. Large adults have a brown body and tail and have stripes that are dull or absent. This lizard is large (up to 33 cm) and shiny, with smooth, flat scales, and males have an orange-red head during the breeding season (Dundee and Rossman 1996; Conant and Collins 1998).

This skink is the most arboreal in Louisiana and prefers hardwood trees. They often live in hollow limbs and other cavities. During the winter, they can be found under the bark of logs and standing dead trees. They eat almost anything that is small enough to capture, including other vertebrates as well as arthropods. Little reproductive data are available for Louisiana. Females often remain with their eggs; clutches range from 6 to 16 eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Family Anguidae
Western slender glass lizard (*Ophisaurus attenuatus attenuatus*)

Glass lizards are found throughout Louisiana. This is a very elongated (up to 1.1 m), legless lizard with a fold of skin along the side of the body and a pattern of narrow dark stripes extending onto the very long tail. It has external ear openings, movable eyelids, and a tail more than twice as long as the body (Dundee and Rossman 1996; Conant and Collins 1998).

Slender glass lizards are more abundant in the high areas of the coastal marsh in southwest Louisiana than in any other part of the state. They are found in thick, high grass adjacent to brush or moist bottomlands. They are found under logs in damp woods adjacent to marshes and in open pinewoods. They eat insects, spiders, snails, and eggs of other reptiles. Little reproductive data are available for this species in Louisiana (Dundee and Rossman 1996; Conant and Collins 1998).

Order Squamata
Suborder Serpentes
Family Colubridae
Mississippi green water snake (*Nerodia cyclopion*)

This water snake is found throughout most of the state of Louisiana. This heavy-bodied snake is olive-brown with an indistinct pattern of narrow dark crossbars, with the ones on the back alternating with those on the sides. The belly is dark brown marked with light spots or crescents. Green water snakes are seldom, if ever, found away from the immediate vicinity of water. They prefer still or very slow-moving water and may be extremely abundant under favorable conditions. They have been found in lakes, ponds, canals, ditches, bayous, rivers, swamps, marshes, rice fields, and flooded woods. They are primarily a diurnal basking snake during the cooler months and nocturnal and aquatic during the warmer months of the year. Mississippi green water snakes predominantly eat fish. Females ovulate from April through June and give birth to an average of 18.4 young from late July through September (Dundee and Rossman 1996; Conant and Collins 1998).

Diamondback water snake (*Nerodia rhombifer rhombifer*)

Diamondback water snakes are found throughout Louisiana, except in the extreme southeastern portion of the state. This heavy-bodied snake is tan to gray-brown with a pattern of dark brown to black chain-like markings. The belly is usually yellow (occasionally dusky brown) and marked with small crescents. It grows up to 1.6 m in length (Dundee and Rossman 1996; Conant and Collins 1998).

This snake lives in still or slow-moving waters, but it has also been found in fast-moving rivers, ponds, sloughs, and in driftwood and logjams. During the day, it can often be seen basking in overhanging branches of small trees. It will drop into the water if disturbed. It is mostly nocturnal from April through October. Diamondback water snakes feed almost exclusively on fish, but also eats frogs. The female ovulates from April through July and gives birth to broods ranging in size from 11 to 30 young from August through October (Dundee and Rossman 1996; Conant and Collins 1998).

Yellowbelly water snake (*Nerodia erythrogaster flavigaster*)

This water snake ranges throughout Louisiana. It is relatively heavy-bodied and moderately long (up to 1.6 m). This snake is gray, greenish-gray, or brown. It is usually plain but may have a light pattern of alternating dorsal and lateral blotches. The dorsal pattern is visible in juveniles. The belly is some shade of yellow with few dark markings (Dundee and Rossman 1996; Conant and Collins 1998).

Yellowbelly water snakes are found in ponds, sloughs, bayous, streams, rivers, lakes, swamps, drainage ditches, and rice fields. They are more likely to wander some distance from water than other species of water snakes. They very rarely bask and are primarily nocturnal. They mainly eat fish, but also prey on crayfish and frogs. Females ovulate during May and June and give birth in August and September. Broods range in size from 14 to 27 (Dundee and Rossman 1996; Conant and Collins 1998).

Blotched water snake (*Nerodia erythrogaster transversa*)

Blotched water snakes are found in extreme southwestern Louisiana. They are heavy-bodied and reach lengths of up to 1.6 m. These snakes vary in appearance and may be any shade of gray or brown. They have a pattern of alternating dorsal and lateral dark blotches. The blotched pattern may be very strong or light. The belly is virtually plain yellow (Dundee and Rossman 1996; Conant and Collins 1998).

These snakes are found wherever permanent or semi-permanent water occurs including ponds, sloughs, bayous, streams, rivers, lakes, swamps, drainage ditches, and rice fields. This water snake is primarily nocturnal and rarely basks. Their main food is fish, but they also eat crayfish and frogs. Females ovulate during May and June and give birth in August and September. The brood ranges in size from 14 to 27 (Dundee and Rossman 1996; Conant and Collins 1998).

Broad-banded water snake (*Nerodia fasciata confluens*)

Broad-banded water snakes are found throughout the state of Louisiana. They are long (up to 1.6 m) and heavy-bodied. They are tan or yellow with broad, dark bands. Some individuals may be almost black while others may be very light. The belly is light with large, squarish dark blotches. It is found in all aquatic situations and occurs to the very edge of salt or brackish water along the coast. This snake frequently basks. They eat a variety of fishes and frogs. Broad-banded water snakes ovulate in late spring/early summer and give birth (7 to 39 young) from July to September (Dundee and Rossman 1996; Conant and Collins 1998).

Gulf salt marsh snake (*Nerodia clarkii clarkii*)

Gulf salt marsh snakes are found in the coastal marshes of Louisiana. This snake is medium-sized (91 cm) and moderately heavy-bodied. It is dark with five light stripes. The belly is light with two rows of large dark spots. This snake is adapted for living in salt water. It obtains metabolic water from its food. It eats small fish and crabs. Brood size ranges from 2 to 44 (Dundee and Rossman 1996; Conant and Collins 1998).

Graham's crayfish snake (*Regina grahamii*)

Graham's crayfish snake occurs throughout Louisiana with the exception of portions of the western and southeastern portions of the state. This small-headed, brown snake has a dark-bordered cream or light gray stripe on the three lower dorsal scale rows and a cream or yellow belly, either unmarked or with a row of dark spots. It grows to 1.2 m (Dundee and Rossman 1996; Conant and Collins 1998).

This snake is found at the margins of ponds and streams, along sloughs and bayous, and in swamps, flooded rice fields, and ditches. It sometimes basks, but may be found under stones and debris, in holes in stream banks, and in crayfish burrows. It eats mostly crayfish, but other crustaceans, amphibians, and fish are also part of its diet. Females ovulate from April through July and give birth in late summer. The brood size ranges from 4 to 39 (Dundee and Rossman 1996; Conant and Collins 1998).

Gulf crayfish snake (*Regina rigida sinicola*)

The Gulf crayfish snake is found in most of Louisiana, except in the eastern portion of the state. This shiny, brown snake has a light stripe on the first dorsal scale row. It has a small head and a light belly with two rows of black spots. It grows to 80 cm in length (Dundee and Rossman 1996; Conant and Collins 1998).

This snake is highly aquatic. It stays hidden in vegetation, logs, boards, and other debris and is only found in the open at night or after heavy rains. Their main food is crayfish, but they also eat frogs, salamanders, and small fish. Females ovulate in May and June, and broods range in size from 6 to 14 (Dundee and Rossman 1996; Conant and Collins 1998).

Brown snake (*Storeria dekayi*)

Brown snakes range throughout Louisiana. One subspecies, the marsh brown snake (*Storeria dekayi limnetes*), occurs in the coastal salt marshes. A second subspecies, the Texas brown snake (*Storeria dekayi texana*) occurs in the western part of the state, while a third subspecies occurs in the eastern portion of the state. Because there is so much integration between the subspecies, they are not discussed separately. This snake is short (53 cm in length) and light-brown or gray-brown. It has a relatively broad, indistinct, pale vertebral stripe that is bordered or crossed by dark spots or a pair of light lateral stripes. The belly is white or pinkish (Dundee and Rossman 1996; Conant and Collins 1998).

Brown snakes are found around natural levees and muskrat houses in coastal marshes, in rocky ravines in northern hill country, and city backyards. They are also found in bogs, swamps, moist woods, and hillsides. Brown snakes are secretive during the day and can be found beneath logs, boards, rocks, or other debris, and they emerge at dusk. They eat earthworms, slugs, snails, soft-bodied insects, spiders, and small amphibians. Most births occur during June and August and broods range from 5 to 25 young (Dundee and Rossman 1996; Conant and Collins 1998).

Gulf Coast ribbon snake (*Thamnophis proximus orarius*)

The Gulf Coast ribbon snake is olive brown with a gold vertebral stripe and a yellowish lateral stripe on the third and fourth dorsal scale rows. The dark color does not extend onto the lateral margins of the belly scutes. It is slender and fairly long (up to 1.2 m). This snake occurs in and adjacent to the coastal marshes in the southern portion of Louisiana (Dundee and Rossman 1996; Conant and Collins 1998).

This snake is found in open areas or dense forests in or near ditches, ponds, lakes, streams, rice fields, and even flooded jeep trails. It has been observed swimming. Ribbon snakes eat fish and adult and larval amphibians. They do not eat earthworms or adult toads. Sexual maturity is reached after two years. Young are born from July through October, with the brood size ranging from 4 to 24 (Dundee and Rossman 1996; Conant and Collins 1998).

Eastern garter snake (*Thamnophis sirtalis sirtalis*)

Garter snakes are found throughout Louisiana except for portions of the western part of the state and the coastal marshes. They are brown, gray, or nearly black with a yellowish or red-orange vertebral stripe and a light yellow lateral stripe on the second and third dorsal scale rows (Dundee and Rossman 1996; Conant and Collins 1998).

These snakes occur in a wide variety of habitats such as meadows, marshes, woodlands, hillsides, along streams and drainage ditches, and in urban areas. Garter snakes are generalized feeders and eat earthworms, amphibians, small mammals, fish, leeches, snakes, crayfish, snails, slugs, birds, sowbugs, and insects. Nine to thirty-eight young are born in June and July (Dundee and Rossman 1996; Conant and Collins 1998).

Rough earth snake (*Virginia striatula*)

This brown snake is very short (32 cm) and has a pointed snout. Occasionally, there is an indistinct pale band across the back of the head. The belly is white or pink. Itranges throughout Louisiana except for the coastal marshes and extreme southeast portion of the state (Dundee and Rossman 1996; Conant and Collins 1998).

They are frequently found in stumps and under logs and loose bark in the forest, they occur in areas that have been disturbed by humans. They are found under boards, cardboard, or other debris and in plant litter. They feed mainly on earthworms. Mating occurs in March and April, and an average of five young are born from July through September (Dundee and Rossman 1996; Conant and Collins 1998).

Eastern hognose snake (*Heterodon platirhinos*)

Hognose snakes occur throughout Louisiana but are rare or absent from the extreme southeast part of the state. They are is heavy-bodied, moderately long (up to 1.2 m), and have an upturned snout. Their dorsal pattern is variable usually yellow, brown, gray, olive, orange, or red with large dark spots. Occasionally, it is solid black. The underside of the tail is usually lighter than the belly (Dundee and Rossman 1996; Conant and Collins 1998).

This snake occurs in a wide variety of habitats, but seems to be most abundant in dry, upland situations. Toads are the preferred food for the hognose snake, but it also eats frogs and salamanders. When disturbed, these snakes will flatten their necks and hiss or play dead. Hog nose snakes breed in the spring, and eggs are laid from June to October. The clutch size ranges from 9 to 25 eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Western mud snake (*Farancia abacura reinwardtii*)

Mud snakes are found throughout Louisiana. This glossy black snake has a bright red belly and is very long (up to 2.1 m). The red color extends onto the lower sides of the body as irregular bars. Mud snakes are rarely seen out of water, except when crossing roads during heavy spring rains. They are occasionally found in or under rotten logs

or in burrows along the banks of small streams. Adult mud snakes feed almost exclusively on amphiumas and sirens, large, eel-like salamanders. Mud snakes breed during the summer, and the eggs hatch in late summer or early fall. Clutch sizes range from 16 to 50 eggs, and the females usually remain coiled around the eggs during the incubation period (Dundee and Rossman 1996; Conant and Collins 1998).

Racer (*Coluber constrictor*)

Racers range throughout Louisiana. In coastal marshes, they are found on the high ground and natural levees. This snake is long (up to 2 m) and glossy. Its back is uniformly black, gray, brown, tan olive green, or blue (with white spots), and the belly is white, grayish white, light yellow, grayish blue, slate gray, or black. Young appear very different from adults and have a series of dark, mid-dorsal blotches on their gray backs. Five subspecies occur in Louisiana, three of them occur in the vicinity of the Calcasieu Estuary (Dundee and Rossman 1996; Conant and Collins 1998).

This common terrestrial snake is found in a wide variety of habitats. It is found in grassy uplands, wooded areas, brush piles, briar patches. They are good climbers and can be found in trees. Racers eat mice, rats, lizards, frogs, birds, and other vertebrates. Clutches of 6 to 18 granular, white eggs can be found from June through September in moist soil by decaying logs, in decaying wood pulp, in clumps of grass, and under boards. Hatching occurs from late July through late September (Dundee and Rossman 1996; Conant and Collins 1998).

Rough green snake (*Opheodrys aestivus*)

Rough green snakes occur throughout Louisiana. These snakes are extremely slender and green. Their bellies are white or yellow and they grow to 1.2 m in length. They are excellent climbers and are usually found on vines, in bushes and small trees; often near streams or lakes. They eat grasshoppers, katydids, crickets, larvae of moths and butterflies, and spiders. They frequently enter shallow bodies of water. Eggs have been found alongside decaying logs in June and July (Dundee and Rossman 1996; Conant and Collins 1998).

Texas rat snake (*Elaphe obsoleta lindheimerii*)

The Texas rat snake ranges from southern Oklahoma to Texas and Louisiana (University of Michigan 2001). Texas rat snakes occur throughout Louisiana but are replaced by another subspecies of rat snake in the northern part of the state (Dundee and Rossman 1996; Conant and Collins 1998). This blotched rat snake is very long (up to 2.6 m) and is variable in color and pattern. It is usually grayish brown or yellowish brown with large blotches that are brownish black or bluish black. It often has a black head, and the belly is mottled or checkered. Texas rat snakes are covered with keeled scales.

Rat snakes occur in a wide variety of habitats. They inhabit swamps, wooded areas, pastures, briar patches, cultivated fields, open sandy areas, trees and bushes, houses, and barns. They have been observed swimming in bayous and rivers. When cornered, they will often coil up and buzz their tail like a rattlesnake. They eat mice, rats, rabbits, birds, squirrels, lizards, and other vertebrates. Texas rat snakes kill their prey by constriction. The clutch size ranges from 6 to 44 eggs (Dundee and Rossman 1996; Conant and Collins 1998). Females lay 5 weeks after mating, and the eggs hatch 65 to 70 days later (University of Michigan 2001).

Speckled kingsnake (*Lampropeltis getula holbrooki*)

Speckled kingsnakes occur throughout Louisiana. They are found from southwestern Illinois to south Iowa, south to east Texas, and west to southwest Alabama. These snakes are often called “salt and pepper” snakes. They are shiny and black with a pattern of small, light-colored spots (usually one per scale). The dorsal spots fuse to form narrow crossbands in juveniles and some adults. The belly is usually light with black markings. This snake grows to 2.1 m (Dundee and Rossman 1996; Conant and Collins 1998).

These snakes are found in a wide variety of habitats, although they are probably most abundant in moist situations. They inhabit river swamps, coastal marshes, upland wooded areas, and stream valleys. They use logs, rocks, ledges, and thick clumps of vegetation as hiding places. Ridges or levees that border or extend into marshes or swamps support substantial populations. They have been observed swimming, and

their eggs have been found in muskrat houses. They eat mice, rats, snakes, including poisonous snakes, birds, turtle eggs, and lizards. Younger snakes forage mostly on lizards and other small snakes. Eggs are laid in July and August, and they hatch in late summer. Clutch size ranges from 5 to 17 eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Prairie kingsnake (*Lampropeltis calligaster calligaster*)

Prairie kingsnakes are found in the western half of Louisiana. This gray or tan snake has alternating dorsal and lateral series of black-bordered brownish or reddish blotches. In large adults, the pattern is often obscured by dark pigment. They are moderately long (1.4 m), and the belly usually has a checkerboard pattern. Kingsnakes are found in mixed pine-hardwood forests. They eat small rodents, snakes, lizards, and young birds. Eggs are laid in June and July and hatch in August and September. Clutches range from 5 to 17 eggs (Dundee and Rossman 1996; Conant and Collins 1998).

Family Elapidae
Texas coral snake (*Micrurus fulvius tener*)

This small-headed venomous snake had a pattern of alternating black, yellow, and red rings. The yellow rings are narrow and separate the wider red and black rings. The tip of the nose is black. This coral snake is found in the western half of Louisiana except for the coastal marshes. It grows to a length of 1.2 m (Dundee and Rossman 1996; Conant and Collins 1998).

Coral snakes are primarily inhabitants of forested areas. They have been found in open grasslands and swampy, wooded bottomlands and are often associated with logs. Coral snakes have a mild disposition. They do not strike but chew vigorously to inject their potent venom. They eat snakes and lizards. They deposit five to nine elongated eggs in June (Dundee and Rossman 1996; Conant and Collins 1998).

Family Viperidae
Southern copperhead (*Agkistrodon contortrix contortrix*)

Copperheads occur throughout Louisiana. This venomous, heavy-bodied, tan snake has a pattern of broad, reddish brown crossbands that are much narrower mid-dorsally than laterally. The belly is light brown marked with darker brown blotches. Juveniles have bright yellow tail tips. They have a narrow, dark line from the eye to the angle of the jaw, a deep pit in the side of the head between the eye and the nostril, and elliptical pupils. They grow to a length of 1.3 m (Dundee and Rossman 1996; Conant and Collins 1998).

These snakes are primarily found in lowlands, near swamps and cypress-bordered streams, but they also occur in hilly regions and wooded areas. Copperheads are mainly active at night. They eat mice, small birds, lizards, snakes, amphibians, and insects. Broods ranging in size from 5 to 10 are born from August through October (Dundee and Rossman 1996; Conant and Collins 1998).

Western cottonmouth (*Agkistrodon piscivorus leucostoma*)

Cottonmouths occur throughout Louisiana. This venomous snake is long (1.4 m) and heavy bodied. It is dark brown or black with a pattern of broad dark crossbands that usually disappear with age. The belly is brown and heavily blotched with black. Juveniles have a bright yellow tail tip. Cottonmouths have a broad, dark line from the eye to the angle of the jaw, a deep pit in the side of the head between the eye and the nostril, and elliptical pupils (Dundee and Rossman 1996; Conant and Collins 1998).

These snakes are found in virtually every permanent or temporary aquatic situation. In larger bodies of water, they are commonly seen basking on logs, brush piles, roots of fallen trees, or limbs overhanging the water. They are also found in bottomland forests and hillsides. When disturbed, they pull into a compact coil and hold their mouth open, revealing the cottony-white interior. They eat any vertebrate prey that they can swallow, but fish are the principal item in their diet. Between 2 and 20 young are born in August (Dundee and Rossman 1996; Conant and Collins 1998).

Western pygmy rattlesnake (*Sistrurus miliarius streckeri*)

Western pygmy rattlesnakes are found throughout Louisiana except in the northeast portion of the state. They are light in color, usually pale grayish brown with mid-dorsal dark spots that may either be highly irregular or form short bars. There are one or two conspicuous rows of dark spots on each side of the body. They often have a reddish dorsal stripe. These venomous snakes are between 38 and 51 cm long, with a tiny rattle and small tail. They are usually found near water, such as river floodplains, swamps, marshes, and wet prairies. They eat rodents, birds, frogs, and lizards (Dundee and Rossman 1996; Conant and Collins 1998).

18.3 References

Arnqvist, G. 1992. Brown pelican (*Pelicanus occidentalis*) foraging success related to age and height of dive. The Condor 94:521-522.

CDEP (Connecticut Department of Environmental Protection). 2000. Wildlife in Connecticut - Endangered and Threatened Species Series. http://www.dep.state.ct.us. Web page accessed on April 23, 2001.

Choate, J.R., J.K. Jones, Jr., and C. Jones. 1994. Handbook of Mammals of the South-Central States. Louisiana State University Press. Baton Rouge, Louisiana. 304 p.

Conant, R. and J.T. Collins. 1998. A field guide to reptiles and amphibians, Eastern and Central North America. Third Edition. Houghton Mifflin Company. New York, New York. 616 p.

Dundee, H.A. and D.A. Rossman. 1996. The Amphibians and Reptiles of Louisiana. Louisiana State University Press. Baton Rouge, Louisiana. 300 p.

Dunning, J.B., Jr. 1984. Body weights of 686 species of North American birds. Western Bird Banding Association Monograph No. 1. 38 p.

Ehrlich, P.R., D.S. Dobkin, and D. Wheye. 1988. The Birder's Handbook. Simon and Schuster. New York, New York. 785 p.

eNature Field Guide. 2001. http://enature.com. Web page accessed on April 27, 2001.

Environment Canada. 2000. Canadian Wildlife Service - Hinterland Who's Who Series. http://www.cws-scf.ec.gc.ca. Web page accessed on April 23, 2001.

Erwin, R.M. 1985. Foraging decisions, patch use and seasonality in egrets (aves: ciconiiformes). Ecology 66(3):837-844.

Hayman, P., J. Marchant, and T. Prater. 1986. Shorebirds: an identification guide to the waders of the world. Houghton Mifflin. Boston, Massachusetts. 412 p.

Knopf, A.A. 1977. The Audubon Society Field Guide to North American Birds. Eastern Region. Alfred A. Knopf, Inc. New York, New York.

LDFW (Louisiana Department of Fish and Wildlife). 2001. http://www.wlf.state.la.us. Web page accessed on May 2, 2001.

Lowery, G.H., Jr. 1974. The Mammals of Louisiana and Its Adjacent Waters. Published for the Louisiana Wildlife and Fisheries Commission. Louisiana State University Press. Kingsport Press, Inc. Kingsport, Tennessee. 565 p.

McLaren, Hart Environmental Engineering Corporation. 1996. Results of a two-year survey of nesting success of least terns (*Sterna antillarum*) at the PPG-Lake Charles Facility. Lake Charles, Louisiana.

McLaren/Hart-Chemrisk. 1998. Off-site ecological risk assessment for the PPG-Lake Charles Facility - Volume 1. Cleveland, Ohio.

MMS (Minerals Management Service). 2001. Cetaceans (Marine Mammals) - Whales and Dolphins in the Gulf of Mexico. http://www.gomr.mms.gov. Web page accessed on May 29, 2001.

NGS (National Geographic Society). 1983. Field Guide to the Birds of North America. National Geographic Book Services. Washington, District of Columbia.

NOAA (National Oceanic and Atmospheric Administration). 2001. Selective Photo Gallery of Otter Island Species. http://www.csc.noaa.gov. Web page accessed on April 23, 2001.

Robbins, C.S., B. Bruun, and H.S. Zim. 1983. A Guide to Field Identification, Birds of North America. Golden Press. New York, New York. 360 p.

TPW (Texas Parks & Wildlife). 1996. Nature Wildlife Fact Sheets. http://www.tpwd.state.tx.us. Web page accessed on May 2, 2001.

University of Guelph. 2000. Canada's Aquatic Environments. http://www.aquatic.uoguelph.ca. Web page accessed on May 4, 2001.

University of Michigan. 2001. Museum of Zoology: Animal Diversity Web. http://animaldiversity.ummz.umich.edu. Web page accessed on May 2, 2001.

USEPA (United States Environmental Protection Agency). 1993. Wildlife exposure factors handbook. Volume I of II. EPA/600/R-93/187a. Office of Research and Development. Washington, District of Columbia.

USEPA (United States Environmental Protection Agency). 1995. Trophic level and exposure analyses for selected piscivorus birds and mammals. Volume I. Analyses of Species in the Great Lakes Basins. Office of Science and Technology, Office of Water. Washington, District of Columbia.

USFWS (United States Fish and Wildlife Service). 2001. Endangered Species Program. http://endangered.fws.gov. Web page accessed on April 23, 2001.

USGS (United States Geological Survey). 2000. Patuxent Bird Identification InfoCenter. http://www.mbr.nbs.gov. Web page accessed on April 23, 2001.

USGS (United States Geological Survey). 2001a. Northern Prairie Wildlife Research Center. http://www.npwrc.usgs.gov. Web page accessed on May 2, 2001.

USGS (United States Geological Survey). 2001b. Biological and Ecotoxicological Characteristics of Terrestrial Vertebrate Species Residing in Estuaries. http://www.pif.nbs.gov. Web page accessed on April 23, 2001.

WDNR (Wisconsin Department of Natural Resources). 1999. Ornate Box Turtle Factsheet. http://www.dnr.state.wi.us. Web page accessed on August 24, 2001.