NC STATE UNIVERSITY
COLLEGE OF VETERINARY MEDICINE

Fish Health Management

June 21 – 23, 2005

NC State College of Veterinary Medicine
Raleigh, NC

Course Schedule

Thursday, July 21, 2005		
Time	Talk	Page
8:30 – 9:15am	Continental Breakfast/Registration	
9:15 – 9:30am	Introductions, Course Overview *Dr. Greg Lewbart*	
9:30 – 11:30am	Fish Ecology & Physiology *Dr. Cliff Swanson*	7
11:30am – 12:30pm	Lunch	
12:30 – 2:00pm	Clinical Anatomy *Drs. Greg Lewbart & Craig Harms*	17
2:00 – 2:45pm	Nutrition *Mr. George Blasiola*	39
2:45 – 3:00pm	Break	
3:00 – 4:00pm	Immunology *Dr. Ed Noga*	Notes not provided
4:00 – 5:30pm	Freshwater & Marine Water Quality & Aquarium/Pond Systems *Dr. Jack Gratzek & Mr. George Blasiola*	49
5:30 – 5:45pm	Break	
5:45 – 7:15pm	Freshwater & Marine Water Quality & Aquarium/Pond Systems, continued *Dr. Jack Gratzek & Mr. George Blasiola*	61
7:15 – 8:00pm	Social Mixer with heavy Hors d'oeuvres	

Friday, July 22, 2005		
7:45 – 8:15am	Continental Breakfast	
8:15 – 9:15am	Reef Systems *Mr. Wade Lehmann*	63
9:15 – 9:30am	Break	
9:30 – 10:30am	Biosecurity *Dr. Jay Levine*	65
10:30 – 11:30am	Public Aquarium Systems *Mr. Stuart May*	83
11:30am – 12:30pm	Lunch	
12:30 – 1:30pm	Diagnostic Techniques *Dr. Craig Harms*	85
1:30 – 2:15pm	Clinical Pathology *Dr. Craig Harms*	93
2:15 – 2:30pm	Break	
2:30 – 5:00pm	Laboratory: Anatomy, Surgical Demonstration, Sample Collection, & Necropsy Techniques *Faculty*	
5:00 – 6:00pm	Fungal Diseases *Dr. Ed Noga*	Notes not provided
6:00 – 9:00pm	Dinner at NC Museum of Natural Sciences	
Saturday, July 23, 2005		
7:45 – 8:15am	Continental Breakfast	
8:15 – 9:15am	Environmental Diseases *Dr. Craig Harms*	97
9:15 – 10:30am	Viral & Parasitic Diseases *Dr. Greg Lewbart*	103
10:30 – 10:45am	Break	
10:45am – 12:15pm	Bacterial Diseases *Dr. Greg Lewbart*	111
12:15 – 1:00pm	Lunch	
1:00 – 3:00pm	Infectious Disease Laboratory *Faculty*	
3:00 – 3:15pm	Break	
3:15 – 4:00pm	Therapeutics *Dr Greg Lewbart*	117
4:00 – 5:00pm	Clinical Cases *Faculty*	
5:00 – 6:00pm	Course Summary/Questions/ Comments/Evaluations/Certificates	

Fish Health Management

Participant List

Jerome Albright
Priority One Services
6600 Fleet Drive
Alexandria, VA 22310
jerome@priorityoneservices.com

Geni Barnes
Virginia Living Museum
308 Mary Bierbauer Way
Yorktown, VA 23693
geni@cox.net

Heather Bates
NC Aquarium at
Roanoke Island
8 Croatan Court
Manteo, NC 27954
heather.bates@ncmail.net

Steve Buchholz
Apet
515 Dublin
Mundelein, IL 60060
sbuchholz@apetinc.com

L. Adam Church
NC Museum of Natural Science
1021 Avent Hill Apt. B3
Raleigh, NC 27606
church_a@hotmail.com

Jason Crichton
South Carolina Aquarium
100 Aquarium Wharf Road
PO Box 130001
Charleston, SC 29412
jcrichton@scaquarium.org

Dr. Thomas Edling
PETCO Animal Supplies
9125 Rehco Road
San Diego, CA 92121
thomase@petco.com

Dr. Karen J. Fischer - SA Emeriti
I-20 Animal Medical Center
5820 West I-20
Arlington, TX 76016
karen.fischer@comcast.net

Kate Freeman
NC State College of Veterinary
Medicine
1519 Hanover
Raleigh, NC 27608
ksfreema@ncsu.edu

Dr. Rae Gandolf
Pittsburgh Zoo & PPG Aquarium
One Wild Place
Pittsburgh, PA 15206
rgandolf@pittsburghzoo.org

Dr. Kari Granier
USDA-APHIS-VS
129 Colonial Circle NW
Cartersville, GA 30120
kari.l.granier@aphis.usda.gov

Amanda Grant - Pathology CVM '07
University of Illinois
406 Park Circle
St. Joseph, IL 61873
algrant@uiuc.edu

Dr. Lori J. Heintzelman - DVM
PO Box 2049
Lebanon, VA 24266
tirlith@hotmail.com

Jim Irving - Cold Fish Koi
Whetstone Creek Koi
1028 NC 704
Madison, NC 27025
support@whetstonecreekkoi.com

Robert B. Irving, Jr.
Irving Fish Hatchery
663 NC 704
Madison, NC 27025
irving_fish_hatchery@yahoo.com

Jack Jewell
Shark Reef at Mandalay Bay
3950 Las Vegas Blvd South
Las Vegas, NV 89119
jjewell@mrgmail.com

Philip Jull
Old Marple Veterinary Clinic
10 South Kirklyn
Upper Darby, PA 19082
pjull@hotmail.com

Rachel Kalisperis
South Carolina Aquarium
100 Aquarium Wharf Road
PO Box 130001
Charleston, SC 29412
rkalisperis@scaquarium.org

Melanie Landry
NC State College of Veterinary
Medicine
1021 Avent Hill Apt B3
Raleigh, NC 27606
mmlandry@ncsu.edu

Allen Lee - Koi Hobby
900 Club Drive
Monroe, NC 28112
allen.lee@siemens.com

Raymond Massey - Aquatics Supervisor
New Year Tech at NIEHS
111 TW Alexander Dr
MC:C0-01
RTP, NC 27709
massey2@niehs.nih.gov

Amanda Grant

Todd Matejovich - Research supply
Priority One Services
6600 Fleet Drive
Alexandria, VA 22310
todd@priorityoneservices.com

Dr. Brigitte Mercier
SPSNQ – Jardin Zoologique &
Parc Aquarium du Québec
9530, rue de la Faune
Charlesbourg, QC G1G 5H9
Canada
brigitte.mercier@spsnq.qc.ca

Kevin Mathis
NC Aquarium at Fort Fisher
900 Loggerhead Road
Kure Beach, NC 28449
kevin.mathis@ncmail.net

Ryan McAlarney
NC Aquarium at Fort Fisher
900 Loggerhead Road
Kure Beach, NC 28449
ryan.mcalarney@ncmail.net

Mitchell A. Morton
Foster Lake & Pond
183 Denmoor Court
Garner, NC 27529
mitch@fosterlake.com

Laura Muha
133 West Millstream Road
Cream Ridge, NJ 08514
lmuha@aol.com

Mary Orr -
Tetra
3001 Commerce Street
Blacksburg, VA 24060
mary.orr@tetra.net

Dr. Richard O. Overton -
USDA
190 Miles Road
Metter, GA 30439
notrevorich_2000@yahoo.com

Dr. James W. Provo
USDA-APHIS-VS
4629 Long Valley Road
Greensboro, NC 27410

Britton N. Purtee
NC Aquarium at
Roanoke Island
PO Box 2606
Manteo, NC 27954
wolf_obsidian@hotmail.com

Lory Scott - Curator
Virginia Living Museum
3510 Fontwell Court
Suffolk, VA 23435
lory.scott@valivingmuseum.org

Maria Serrano
NC State College of Veterinary
Medicine
8904 Turner Drive
Apex, NC 27539
meserran@ncsu.edu

Brian Shannon - Plymouth
NC State University
207 Research Station Road
Plymouth, NC 27962
dbshanno@unity.ncsu.edu

Dr. Julie Sheldon - '03
Oklahoma State University
College of Veterinary Medicine
1807 North Hartford Apt 914
Stillwater, OK 74075
juliesheldon@aol.com

Laura Shewmon
1315 Pineview Drive
Raleigh, NC 27606
lshewmo@unity.ncsu.edu

Dr. Raghu Shivappa - Researcher
NC State College of Veterinary
Medicine
4700 Hillsborough Street
Raleigh, NC 27606
raghunath_shivappa@ncsu.edu

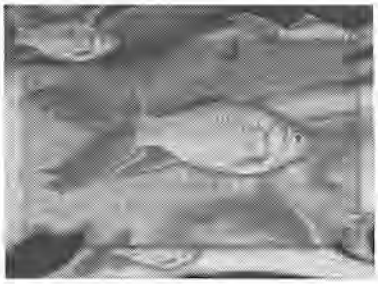
Dr. Rosemary B. Sifford
USDA-APHIS-VS
930 Main Campus Dr Ste 200
Raleigh, NC 27606
rosemary.sifford@aphis.usda.gov

Jeff Smith
NC Museum of Natural Science
5740 Poolside Drive
Raleigh, NC 27612
jeffrey.smith@ncmail.net

Jessica Sprague - '07
University of Illinois – Urbana-
Champaign
209 South Dodson Drive
Urbana, IL 61802
jlyons2@uiuc.edu

FISH ECOLOGY & PHYSIOLOGY

Cliff Swanson, DVM, MS, DACVA
NC State College of Veterinary Medicine
4700 Hillsborough Street
Raleigh, NC 27606



What is the Physiological Prime Directive?

* Survive to reproduce!

Physiological "Big Picture" Issues Supporting the Prime Directive

* Nutrition
* Locomotion
* Protection
  * Physical
  * Immunity
* Sensory perceptions including recognition of conspecific individuals

Homeostasis

- Control processes maintain an internal equilibrium enabling other physiological systems to function properly
  - Endocrine and autonomic nervous system
- Adaptive mechanisms alter physiological processes in the face of STRESS
  - External
  - Internal

Homeostatic Issues

- Oxygenation
- Body temperature
- Water, solute, pH balance
- Immunity

What is a Fish? (I)

- Pointy-headed definition - A fish is a poikilothermic, aquatic vertebrate, having gills for respiratory and ionic exchange with the ambient water, a variety of appendages developed as fins, and a body usually covered with scales.

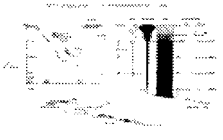
What is a Fish? (II)

- Bottom line definition - A fish is an aquatic vertebrate whose reproductive mission in life is supported by behaviors and cellular processes that accomplish homeostatic requirements identical to those of terrestrial species...except a fish does it in water!
- Survivorship success story
  - Part of Earth's biomass for over 400 million years
  - Live in the largest and most diverse habitat on the planet

Habitat Diversity

- 25,000 total species
- 41% freshwater species
  - 0.0093% of all water on Earth contains almost 1/2 the fishes
- 58% seawater species
  - 45% live above 200 m
  - Deep sea is largest volume habitat on Earth
    - 90% of water covers depth >1000 m
- 1% migrate between

Seawater Habitats



- Zones by depth
  - Littoral < 200 m
    - Most abundant, esp. tropical
  - Mesopelagic 200-1000 m
  - Bathypelagic 1000-4000 m
    - Immense volume
    - Permanently dark
    - Constant 2-5 degrees C
  - Abyssal 4000-6000 m
  - Hadal > 6000 m

Challenges of the Aquatic Environment

- Ambient temperature conducted to cells
  - Affects rates of metabolic reactions
- Low oxygen tension
  - Water contains about 1% oxygen by volume compared to 21% in air (95% reduction)
- Frictional and inertial drag
  - Water ~800x more dense, and 50x more viscous than air

Challenges of the Aquatic Environment

- Osmolarity
  - Seawater hypertonic to tissues - species dehydrate to environment
    - Drink water and excrete Na^+ across gills
  - Tissues hypertonic to freshwater - species must continuously excrete water
- pH
  - Gills are site of hydrogen ion balance and are in direct contact with water where CO_2 tension is low
- Low visibility

Contribution of Ventilation to Acid-Base Balance and Blood Oxygenation

* Ventilation is the mechanical process of moving the O_2-containing medium into contact with the tissue surface where O_2 and CO_2 are exchanged
* O_2 fuels oxidative metabolism
* CO_2 elimination is one of the primary determinants of H^+ balance
* pCO_2 drives ventilation in mammals

O_2 Acquisition and CO_2 Elimination by Fishes

* Venilation driven by pO_2, not pCO_2 in fishes
* Water is a huge sink for CO_2 which freely diffuses down its partial pressure gradient across the gills
  * CO_2 elimination independent of ventilatory effort
* Very low O_2 partial pressure in water is a physical constraint on O_2 extraction by blood
* Fishes must ventilate a large volume of water in order to bring enough O_2 to the gill surface to support metabolic needs

Hypoxemia

* Hypoxemia refers to low O_2 tension in the blood (O_2 in solution)
* Hypoxemia becomes a critical, life threatening event when the pO_2 falls below the threshold required to fully load hemoglobin (Hb) in red blood cells (RBC)

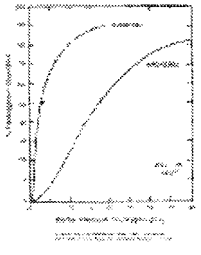
Hemoglobin

- Iron-containing protein in RBC
- Carries O_2 to tissues, and CO_2 and H^+ to gills
- Each Hb molecule can carry up to 4 molecules of O_2, but no more than that
- O_2 loading and unloading characteristics are affected by many other factors including pH, temperature, and nucleoside triphosphate concentration

Oxygen Transport by Hb

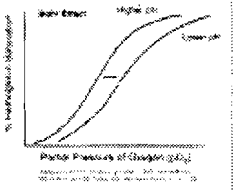
- Hb represents 99% of the total O_2 carrying capacity of blood
- Oxygen-Hb dissociation curve is sigmoidal because of cooperativity in O_2 loading
  - Binding of each successive O_2 molecule facilitates next until all 4 binding sites are occupied (Hb becomes saturated)
- Hb loading with O_2 has a very high physiological priority
- Adaptive mechanisms to acute stress protect this function

Normal Hb Dynamics in Fishes



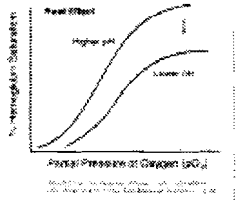
- O_2 transfers to blood from water via ventilation across gills
- Hb loads as a function of the O_2 partial pressure
- Species differences
  - Less active species inhabiting low O_2 habitats have higher affinity Hb

Normal Hb Dynamics At Tissues



- Blood delivers O_2 to tissues where Hb unloads because of increased CO_2 and H^+ concentrations that reduce Hb affinity for O_2 (Bohr effect)
- Hb picks up CO_2 and H^+

Normal Hb Dynamics At Gills

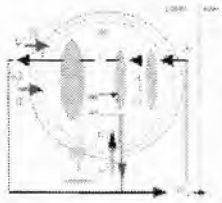


- CO_2 lost from Hb into water down concentration gradient
- RBC becomes less acidic and Hb affinity for O_2 increases (Haldane effect)
- Hb loads O_2 again

Control of Oxygen Transport by Hb

- Metabolic demands for O_2 supply and CO_2 removal vary with activity level of fish
- Environmental O_2 tensions and H^+ concentrations can vary
- The integrated Stress Response mediated by circulating epinephrine released from the head kidney includes modulation of the internal RBC H^+ concentration in a range that optimizes Hb-O_2 loading even in the face of systemic acidosis

Na+-H+ Exchange Pump of Teleost RBC



- Epi release evoked by a variety of stimuli including acidemia, hypoxemia, hypercarbia
- Epi stimulates a ß-adrenergic receptor on the RBC membrane
- ß-receptor turns on the RBC Na+/H+ antiporter
- Reduced RBC H+ favors O_2 loading

FISH ANATOMY
(adapted from Fish Medicine, first edition, WB Saunders)
© M.K. Stoskopf 2004

In fish medicine, as in any other clinical specialty, a basic knowledge of anatomy is important. Unfortunately, the structural diversity of fishes and the lack of basic anatomic information on even relatively common fish species makes obtaining this knowledge difficult. This chapter focuses on clinically important aspects of fish anatomy and is intended to serve as a reference for the clinician on specific organs in a variety of fishes. Some clinically important aspects of vascular anatomy and organology are also presented. The information presented in this chapter by no means represents the limits of diversity found in fishes, but within the confines of a single chapter it should go a long way toward preparing the clinician for the surprises in store.

Nearly everyone's concept of a typical fish is what is known as a fusiform fish. These fishes include most of the sportfish and commercially important species, such as trout, salmon, and bass. The reason to cover this type of fish in some detail is that the majority of fish seen in clinical practice fall into this category. It is also the type of fish we know the most about. In particular, these rainbow trout is perhaps the most extensively studied species of fish. Of course, there actually is no single, "typical" fusiform fish, and each species has certain adaptations that allow it to succeed in its particular environmental niche. While exploring the anatomy of the trout, some common variations in other fusiform fish will be noted.

THE RAINBOW TROUT, A FUSIFORM TELEOST

EXTERNAL ANATOMY

In describing the gross anatomy of a fish, most anatomists divide it into three body regions, the head, the trunk, and the tail. The head extends to the caudal margin of the gill covering or operculum. The trunk continues from the head to the most caudal point of the peritoneal cavity which in many fusiform fish is indicated externally by the urogenital and anal openings. The muscular tail begins at this point and extends caudally. The term, fusiform, refers to the overall shape where the head is smaller than the trunk with a taper from the mouth to a point behind the pelvic fins that is usually the deepest (dorsal to ventral) point of the fish. From this point the body tapers again to the end of the caudal peduncle.

SCALES

A major difference between the rainbow trout and the majority of other fusiform fish is the relatively light scale pattern of the trout. Scales are translucent dermal origin plates that project into the epidermis forming a protective environmental barrier for the fish. Trout have oval shaped cycloid scales with smooth borders that are considered more primitive than the ridged ctenoid scales found on many fusiform fish.

Scale types have some clinical significance dictating handling and treatment procedures. Lightly scaled fish like the trout, and particularly scaleless fish such as the catfish, can be much more sensitive to drugs and toxins in the water. This affects some

treatment protocols. Very fine scaled fish like the herring, jacks, and tuna are easily damaged, even in simple procedures, such as capture and transfer. These fish often require anesthesia for examinations and simple procedures, including injections. On the other extreme, large scaled fish, such as sturgeon, bonefish, and tarpon, can be very difficult to inject with remote injection devices like pole syringes, and large scales may complicate surgical efforts.

FINS

The fins of fish are often used as clinical landmarks or to locate lesions so that they can be monitored over time. A basic knowledge of fin nomenclature and how it varies on different types of fish is useful. The trout has a fairly complete compliment of fins. The paired pectoral fins originate high on the sides of the trunk just behind the gill openings. The musculoskeletal girdle for these fins is attached to the skull. The other paired set of fins on the trout are the pelvic fins that are set close together on the ventral surface, cranial to the urogenital and anal openings. The trout also has the four unpaired fins found in most fusiform teleosts. The cranial or first dorsal fin begins further caudally on the dorsal surface of the trout than in other fusiform fish like perch, bass, swordtails, and guppies. The caudal or second dorsal fin of the trout is located behind the first dorsal and is sometimes known as the "adipose" fin. The anal fin is found just caudal to the genital and anal openings of the fish on the ventral surface. The caudal fin is the terminal fin and is commonly referred to as the "tail."

INTERNAL ANATOMY

MUSCULOSKELETAL SYSTEM

For a clinician, the musculoskeletal anatomy of a fish is perhaps most easily accessible through radiography. Teleost fish have axial and appendicular skeletons of true bone with the same basic components of mammalian skeletons. Although skeletal patterns vary considerably, the clinician evaluating a case can usually rely on the conservation of symmetry and functional design. For additional insight into the structure of the fish skeleton, study the radiographic plates in the chapter on clinical procedures.

One of the most clinically important aspects of the muscular system is demonstrated in the trout, but is more consequential in many marine pelagic fish. This is the difference between dark and light muscle. The dark or red muscle in the trout and most other fusiform fish is located just beneath the skin. In the trout this is a middle band of muscle, but in tuna and other fish it can be much more extensive. The dark muscle of fish is thought to be involved in sustained swimming activity. It has a higher lipid content and is histologically distinct from the white muscle. The clinical importance of the location of dark muscle on a fish is that there are distinct differences in drug kinetics when injections are made into dark muscle. Anesthetics, for example, tend to distribute much differently when injections are made into the dark muscle, compared with injections into white muscle. This aspect of anatomy and pharmacology warrants considerable study with other drugs, but should be kept in mind when a clinician is selecting injection sites and angles in larger fish.

THE CARDIOVASCULAR SYSTEM

The circulatory system of fish is relatively simple. Poorly oxygenated blood circulates from the cranially placed heart to the gills where it is oxygenated by a countercurrent system in the gill lamellae. It then continues through the dorsal aorta to the arteries supplying the various organs and tissues and then returns to the heart via the venous system. The heart of any teleost, including trout, is considered a two chambered heart, although four distinct regions are readily distinguishable. The thin walled sinus venosus receives venous blood from hepatic veins and the common cardinal veins and empties into the somewhat thicker atrium through paired sino-atrial valves. The atrium, usually considered the first chamber of the fish heart, transfers blood to the muscular ventricle through the nonmuscular atrioventricular valve. The fourth component of the trout heart is the bulbus arteriosus that contains no cardiac muscle, but consists entirely of elastic connective tissue and smooth muscle. The bulbus anteriosus receives blood from the ventricle through semilunar ventriculobulbar valves and via its elastic recoil, pushes it through the ventral aorta to the gills. In lungfish and coelacanth, the atrium and ventricle are partially divided into two symmetric chambers and are much like amphibian hearts.

The physiology resulting from the vascular patterns in the tail and flank regions of fishes are clinically important. A renal portal system can have major effects on the pharmacokinetics of drugs administered intramuscularly or at various intravenous sites. Anatomists have identified eight anatomical patterns of blood flow in the caudal region of various fishes. These can be summarized into four clinically significant physiological groups.

Most fishes have a venous return system in which the majority of the blood returning from the tail and sides of the body passes through the kidney before going to the heart. The caudal vein divides into right and left renal portal veins that pass along the dorsal surface of each respective kidney sending branches into the renal parenchyma. Segmental veins from the flank also empty into the renal portal veins and subsequently the kidney. Blood is carried away from the kidney by the caudal cardinal veins. This complete renal portal system occurs in pike, all of the sharks, skates and rays, the lungfishes, ciscos, whitefish, burbot, tiger barbs, sculpins, porcupine fish, balloonfish, mullets, sea robins, cusk-eels, rockfishes, ocean perch, red fish, cowcods, chilipeppers, goatfishes, mackerels, polypterids, and sole.

Other fishes, particularly the cyprinids and anguillaform eels, have systems in which only some blood passes through the renal portal system, and a portion passes directly to the hepatic portal system. In cyprinids, this occurs through a communication between the caudal vein and the caudal intestinal vein. In eels, special venous arcades occur between the renal portal veins and the intestinal vasculature. With this system, some blood from the tail goes to the renal portal system, and some goes to the hepatic portal system.

The second most common caudal venous pattern in fishes is found in tench, cod, grenadiers, eelpouts, ide, dace, rudd, and some members of the cyprinid family. In this system, some blood from the caudal vein is taken directly to the heart, completely bypassing all portal systems. In the tench, only blood from the ventral portion of the tail goes to the kidneys. In the other fishes, the caudal vein branches into a left renal portal vein that goes to the left kidney and a right branch that goes directly to the heart.

In the fourth pattern of caudal venous return, all blood from the caudal vein is taken directly to the heart, and the kidneys are supplied only from segmental veins from the flank musculature. This pattern is found in the salmonids, the perch, including yellow perch, the tunas, lumpfish, and lumpsuckers. In these fish, no blood from the tail is filtered through a portal system before being recirculated by the heart.

Fish have a well developed lymphatic system (Harder, 1975) consisting of lymph vessels that start as blind ducts near the submucosa of the gastrointestinal tract, the spleen, liver, and other parenchymal organs. These vessels join to form larger ducts that parallel the venous system and eventually empty into it. Fish do not have lymph nodes; however, lymphoid tissue is found several places. It is probably best termed lymphomyeloid tissue since, it is multipotential. In lampreys and sharks, as well as many other fish, this tissue is found in the cranial kidney. In bony fish it can be located in the mesentery.

THE RESPIRATORY SYSTEM

The pharynx of trout and most teleosts can be divided into three cavities, the buccal cavity and two opercular cavities. The entrances of the opercular cavities from the buccal cavity are guarded by mandibular and maxillary valves. This differs from the sharks (elasmobranchs), which have a single orobranchial cavity and five or more parabranchial cavities on each side of the fish. The presence or absence of the mandibular and maxillary valves is important clinically when it is desirable to ventilate a fish artificially. Many teleost fish must have their mouth moved in order to open the valves and allow water to ram over the gills.

Trout have four gill arches with holobranchs and one with a hemibranch called the pseudobranch. These arches are supported by cartilaginous rods extending from the floor to the roof of the opercular cavities of the pharynx. The cranial aspects of the gill arches are modified into gill rakers, which help prevent food and debris from reaching the respiratory components of the gills. The structure of the gill rakers is different enough between species to have been employed as a taxonomic tool. Gill raker structure seems to parallel the feeding habits of the species.

The arrangement and form of the actual gills on the posterior aspects of the gill arches have also been used as taxonomic features even to the level of differentiating individual species of trout. Each gill arch has two rows of gill filaments. Each row is called a hemibranch, and a holobranch. Each hemibranch consists of a row of gill filaments that stand up like teeth on a comb. Abductor and adductor muscles allow the fish to spread and collect hemibranchs on an individual gill arch.

Gill filaments, often referred to as primary lamellae or lamellae in older literature, are formed on the caudal edges of the gill arches. Each gill filament has secondary lamellae (respiratory platelets) that run perpendicular to the axis of the primary lamella. In some fishes, there is only one row of secondary lamellae on each side of the filament, while in others there are two rows on each side. New secondary lamellae are formed at the tip of the gill filaments in bony fish, and the oldest secondary lamellae are, therefore, located at the base of the gill filament. In sharks, new secondary lamellae are developed in a growth zone at the base of the gill filament, and the secondary lamellae at the tip of the gill filament are the oldest. The gills are supplied by a dual arterial system with one

artery providing for oxygenation of blood by the gill and the other supplying the arch and lamellae (Laurent and Dunel, 1976).

Most teleost fish, including the trout, have a pseudobranch. The pseudobranch arises embryologically from the caudal hemibranch of the mandibular gill arch and is supplied with oxygenated blood, and, therefore, has no respiratory function (Harder, 1975). Thus the name pseudobranch. As early as 1839, two morphological types of pseudobranchs were recognized, those with distinguishable lamellae in contact with the water and those that were covered and had no close water contact (Muller, 1839). Free pseudobranchs are essentially identical to normal gills. They are true hemibranchs that may be shorter than a normal gill hemibranch. This kind is found in blennys, herrings, and flounders. Covered pseudobranchs have some connective tissue overlaying them. This kind of pseudobranch is found in individual species from a number of orders of fish. Embedded pseudobranchs are covered by the mucous membrane of the opercular cavity and are separated from the opercular cavity by folds of tissue. These may be located in the tissues at the base of the skull rather than on the operculum. Covered pseudobranchs have been graduated into four types based on their degree of isolation from water, but the clinical usefulness of this classification is not clear (Leiner, 1938; Bertin, 1958).

The pseudobranch is distinct, from the carotid labyrinth of some air breathing fish. In the trout, the pseudobranch is located on the underside of each operculum. In a striped bass, on the other hand, it is attached to the hyoid arch and resembles a gill. The pseudobranch lies deep in the roof of the opercular cavity in carps.

The function of the pseudobranch is unknown. Efferent blood from the organ passes to the brain and the eyes, and a direct vascular connection exists to the choroid of the eye in salmonids (Yasutake and Wales, 1983). A variety of functions have been proposed based upon the histological structure of the organ, including a primary osmoregulatory function. Connections to the vascular rete of the swim bladder in some species have also led to the postulation that it may be involved in filling the swim bladder (Lagler et al., 1977).

DIGESTIVE SYSTEM

The rainbow trout is a carnivorous fish and, therefore, has a relatively short and simple digestive tract. Teeth are present, but not particularly prominent. Like most fish, the mouth is devoid of distinct consolidated salivary glands (Anderson and Mitchum, 1974), although, considerable mucus is produced by buccal glands. The short esophagus enters a J-shaped stomach that has both cardiac and pyloric regions that can be distinguished on the mucosal surface, but not the serosal surface. The intestines have been assigned a variety of names by various investigators, but we will adopt the terms small and large intestine. Alternate terms that may be encountered in the literature include intestine and rectum (Burnstock, 1959; Bullock, 1963), ascending and descending intestines (Weinreb and Bilstad, 1955), and anterior and posterior intestine.

There is no real problem in identifying the structures as bowel, but some fish, such as the goldfish and carp, do not have a defined stomach. This occurs in species from a number of orders. If fish do have a stomach, it is either straight, as is found in the pike, U- or J-shaped, as in the sharks and most fish with stomachs, or it has a gastric diverticulum that pouches off of the U-shaped stomach, as in the freshwater angelfish. The demarcation between the stomach and small intestine may be completely lacking or

the pylorus can be dramatically marked by numerous pyloric diverticulae. There may or may not be a demarcation between the small bowel and large intestine. The entire bowel can be no more complicated than a straight tube or it can be looped in various numbers of coils. The variations are many, but they all end at the anus.

In the trout, as the small intestine originates from the pylorus, pyloric diverticulae arise. These blind sacs are encased in adipose tissue that encompasses the diffuse pancreas of the trout that can be identified histologically. No discrete pancreas will be found on gross examination, and the multiple small ducts from the pancreatic tissue emptying into the small intestine are difficult, if not impossible, to appreciate grossly. In other fish, the pancreas is also diffusely distributed, usually along the portal veins as they emerge from the intestinal serosa. The exocrine tissue is more diffusely distributed than the endocrine tissues, which are usually located near the liver or embedded within hepatic tissue. Islets are not numerous. The pancreas is present as a discrete organ only in the sharks, lungfish, and some catfish.

The exocrine ducts of the pancreas in fishes develop from three anlage, with usually only one persisting late in development. However, all three can be found in many salmonids. In the majority of fish species, the single persistent duct develops from the right ventral anlage and is designated in some literature as "Wirsung's duct." In skates, the persistent duct develops from the dorsal anlage and is called "SANTORINI'S DUCT."

The liver of the trout is not distinctly divided into lobes. It is located in the cranial coelomic cavity originating just caudal to the transverse septum that separates the peritoneal and pericardial cavities. It is located predominately on the left side of the abdomen and is supplied by a hepatic portal system. The liver is connected to a prominent gallbladder via an hepatic duct. The gallbladder in turn empties into the pyloric end of the stomach through a bile duct.

The livers of various fish can be quite pigmented. This is due to the presence of so-called melano-macrophage centers, which are sometimes considered indicative of inflammatory response. The opposite extreme is a light, almost white appearance of the liver, particularly in captive-reared animals fed prepared diets. This condition can be seen occasionally in wild fish, but is prevalent in captive-reared fish.

The swim bladder, (gas bladder, air bladder, pneumatic tract), is actually derived from the digestive system. In some fish (physostomes), including trout, the swim bladder maintains an active communication with the esophagus through the pneumatic duct. In late embryonic and early post-embryonic development the swim bladder forms as a diverticulum of the esophagus. Young trout can gulp air from the surface to fill this pouch. Food can accidentally enter the swim bladder in very young trout, where it won't be digested, but will instead cause future swim bladder infections (Ross et al. 1975).

The swim bladder is a retroperitoneal organ. Its course of development varies in different fish species, resulting in a variety of final adult forms (Fig. 9). The simplest form of the swim bladder itself is found in the sturgeons and salmonids, including trout. In these fish, the swim bladder is a wide, short duct with only one chamber. In the carp group, the swim bladder is divided into cranial and caudal compartments with a connecting isthmus. In the codfishes the swim bladder is divided into three chambers.

In trout, the opening to the esophagus is eventually closed by a sphincter muscle, which controls the pneumatic duct allowing rapid gas uptake or dispersal. The pneumatic

duct in salmonids and catfish arises from the right side of the esophagus. In the carp, the duct develops from the left side of the esophagus. In contrast, the "lungs" of lungfish and polypterus originate from the ventral side of the foregut.

Other fishes that do not maintain a connection between the digestive system and the swim bladder (pysoclistous fish) develop one or more prominent rete mirabile associated with the bladder. These complex counter current capillary systems are often found in the cranio wall of the bladder and may be found in the cranial wall of each chamber in multichambered species (Jasinski and Kilarski, 1971; Grizzle and Rogers, 1976). The rete are thought to enhance the availability of oxygen, and to a lesser extent, other gases to the epithelium of the swim bladder for concentration in the bladder itself. The rete mirabile of the swim bladder may be associated with the structurally similar choroid body of the eye, which is present in some, but not all, fishes (Lagler et al. 1977). In addition to the rete mirabile, many, but not all, fish with prominent cranio capillary retes also have an anteriodorsal capillary network called the "oval." The oval is derived from the prevesicle of the embryonic pneumatic duct and is thought to function in the removal of gases from the swim bladder. Some fish, including the carp, catfish, and many eels have both a pneumatic duct and a rete mirabile associated with their swim bladders.

In some fish, such as in the butterflyfish, the swim bladder is highly complicated and penetrates into the centra and both transverse processes of many vertebrae, creating pneumaticized vertebrae (Schwartz, 1969). The butterflyfish floats on the surface and gulps air frequently and its swim bladder may participate in respiration.

The swim bladder is missing entirely in many pelagic and deep sea fish. The organ can be located in embryos of mackerels, tunas, and flounder, but flounder lose their swim bladders when they begin to live on the bottom, and adult mackerel and tunas do not have a swim bladder. On the other hand, over half of the benthic fish have very large swim bladders. Some of these deep sea species have their swim bladders filled with lipids rather than gas.

The actual function of the swim bladder remains somewhat speculative. It is certainly involved in the maintenance of hydrostatic equilibrium. In some fish it may be involved in respiration, sound production, and possibly perception of pressure fluctuations, including sound waves (Reyer, 1977; Ross, 1979).

HEMATOPOIETIC ORGANS

Fish do not have bone marrow to serve as a hemopoietic organ. Their erythrocyte production is accomplished by the same tissues that produce their lymphoid response, lymphomyeloid tissue, located diffusely in the mesentery. The primary site of hematopoiesis in the trout is the cranial kidney (Yasutake and Wales, 1983). The gross anatomy of the kidney is discussed below under the urogenital system. The spleen of the trout is adjacent to the stomach; it is also an accessory hematopoietic organ.

Much of the lymphomyeloid tissue of the fish is found in the spleen. The spleen of the trout is adjacent to the greater curvature of the stomach. In other fish, it is often found near the lesser curvature of the stomach or in stomachless fish near that part of the intestine that passes where the stomach would be. The position of the spleen is similar to what would be expected in mammalian anatomy, with some exceptions. The lampreys do not have a spleen, but instead have a fat-body or sheath. In the lungfish, the spleen and pancreas are together, but well delineated. The capsule of the spleen often contains

considerable pancreatic tissue in other teleosts. Fish spleens can be pigmented. Pigment nodules result in a spotted spleen appearance in some fishes.

The trout, like most fish, has a distinct thymus that is a highly vascular, paired, bilateral organ found under the dorsal edge of the operculum just dorsal to the gill arches. In contrast to mammals, the fish thymus is difficult to locate in young fish, but can be seen grossly in fish older than five months (Yaustake and Wales, 1983). In some fish the thymus is found within the cranial kidney. In air breathing catfish, the thymus is caudal to the accessory respiratory organ, much more caudal than in most fishes. In the sharks, the thymus is fairly distinct and is located on the dorsal inner edges of the gill slits.

THE ENDOCRINE SYSTEM

The pituitary of the trout is located ventral to the diencephalon much as in a mammal. Compared to other fish, the pituitary of the trout is foreshortened craniocaudally and deep dorsoventrally. Otherwise, it is unremarkable.

The thyroid gland of the trout is diffuse, a common situation in many species of fish. It cannot be identified as a discrete organ. Follicles are found throughout the course of the ventral aorta and the branchial arteries. Follicles can even be found in the retro-orbital tissues.

Fish do not have discrete adrenal organs. The two types of tissue are distributed in the kidneys and along larger blood vessels. The component found in the kidney is sometimes called the "interrenal organ." In trout, this organ is located in the lymphoid portion of the cranial kidney. In most fish it is found in a layer around the posterior cardinal vein and its tributaries.

The second component of fish adrenal tissue, the suprarenal organ, is homologous to the adrenal medulla. It develops out of the nerve tissue of the sympathetic ganglia and remains in close contact. These are sometimes called the "paraganglia." Suprarenal tissue may contain a large number of neurons, along with chromaffin cells. In sharks and rays, the suprarenal organs are along the aorta and form larger complexes referred to as "corpora axillaria." This is the origin of the term "suprarenal organ" since, in sharks the corpora axillaria is found on the dorsal side of the caudal kidney. In bony fish the "suprarenal organ" is almost always located in the region of the cranial kidney.

The kidney also contains the corpuscles of Stannius. These are only found in bony fish, not in the sharks. They are piles of cells arranged in alveoli, usually located dorsal and in the midline of the caudal kidney where the left and right kidneys come together. Sometimes they are located on the ventral side of the kidney. Most fish have two corpuscles of Stannius. Some catfish have three to seven scattered along the ventral surface of the caudal third of the kidney. Trout and other salmonids have four to 14 of these structures. Removal of these organs affects electrolyte balance. The corpuscles become very active when fish have external damage that breaches the epithelial osmotic barrier. The corpuscles are considered endocrine organs since they do not have ducts.

The ultimobranchial bodies are also called the "postbranchial" or "suprapericardial" bodies. They are located near the gill-slits. In mammals, the ultimobranchial bodies are fused into the thyroid. In fish, they produce calcitonin. They are NOT anlage of parathyroid glands. Fish do not have discrete parathyroid organs.

In trout and salmon, the ultimobranchial bodies are bilateral, located ventral to the esophagus, near the sinus venosus. In the rays, the ultimobranchial bodies are also paired,

but found in the caudal wall of the pericardial cavity. In sharks, they are found only on the left side of the body on the dorsal wall of the pericardial cavity, between the elements of the last gill arch.

UROGENITAL

Kidneys in fishes vary considerably. Classically, they are described as they occur in the trout, as a fused, single, long, thin organ lying retroperitoneally just ventral to the spinal column occupying the dorsal wall of the coelomic cavity along its entire length. This is an oversimplification. The pronephros and the cranial portion of the mesonephros, often designated as the cranial anterior or head kidney can be topologically distinct from the caudal portion of the mesonephros designated as the caudal posterior or tail kidney. Even when the organs are continuous, the cranial portions of the kidneys usually remain separate, but the caudal portions are most often fused to some degree. Numerous authors have had the chance to classify fish kidneys into various groups and subgroups, none of which are particularly relevant in diagnostic efforts. What is important is to be sure to sample both the cranial and the caudal kidney when performing a postmortem examination. Not only is the gross morphology quite variable, but the histology can be startling, particularly in species that have completely aglomerular kidneys.

The genital organs of the male trout have been well studied (Robertson 1958, Henderson 1962). The elongate testis bound in a thick white capsule of connective tissue is supported by a distinct mesorchium arising from the swim bladder peritoneum. The testis consists of elongate blind, branching seminiferous tubules that produce spermatozoa that are discharged into a short deferent duct at the posterior pole, which carries the sperm to the genital pore. The reproductive and urinary systems are completely separate in the male trout, as well as in the female trout (Henderson 1962).

The ovaries of the trout are not as well studied as the male gonads, but are very similar to those in other fish species (Yamamoto et al. 1965). Trout ovaries are long, paired organs that develop a beaded texture and enlarge significantly as oocytes mature during spawning season. They are supported by a mesovarium, and their vascular supply enters along the dorsomedial aspect of the gonad. The tunic of the ovary is very thin and transparent compared to the tunic of the testis. After ovulation, a number of oocytes remain behind and are eventually resorbed. These have been called "transitory corpora lutea" and "corpora atretica," but all evidence suggests that these are merely unovulated oocytes (Dodd 1960).

The trout develops oviducts from a modified Mullerian duct during embryonic development, but these nearly disappear in the adult trout. Ova are shed into the coelomic cavity before being passed through the remaining caudal portions of the oviducts at the genital pore.

SENSORY

The eyes of the trout are somewhat similar to those of other fish. They are bilaterally placed, in bony sockets of the skull, where they are independently moveable by three paired extrinsic oculomotor muscles. Although most fish are described as having no eyelids, trout have a single fatty membrane around the eye. The presence of a modified eyelid is common in fish. The sclera of the cranially compressed globe of the trout eye is supported by an annular ring of cartilage continuous with nasal and temporal

plates of true bone. The trout cornea is elliptical and considerably thinner at the nasally located center, a common finding in rapid swimming fish (Prince 1956). An annualar ligament connects the cornea to the outer rim of the iris (Prince, 1956). This ligament, along with the protrusion of the spherical lens through the pupil, appears to severely limit iris movement (Ali 1959). The pupil in most boney fishes is circular with some elongation in the direction required for acquiring food. In the trout the pupil elongation is ventromedial (Anderson and Mitchum 1974). Some fishes that inhabit deep water where light levels are extremely low, technically have no pupil since their entire iris is missing (Harder 1975).

To accommodate, the inelastic lens of the trout must be moved toward the retina by a special retractor lentis muscle (Pumphrey 1961). This is the common mode of accommodation in fish eyes. Fundoscopic examination of the trout reveals a lack of a fovea for binocular vision. This would be expected considering the lateral placement of the eyes on the head of trout.

Olfaction in the trout is centered in the nasal passages where water is circulated between the pairs of nasal openings as the fish swims (Watling and Hillemann 1964). Likewise, taste organs in trout are centered in the buccal cavity and esophagus. Some similar sensory organs are found on the surface of the head, but not to the extent found in species like the catfish.

The lateral line system of the trout consists of bilateral supraorbital, suborbital, mandibular, and temporal canals on the head. These are continuous with the lateral line runs the length of the body at about halfway between the dorsal and ventral limits of the trunk (Weber and Schiewe 1976).

The pineal organ lies between the midbrain and dorsal forebrain in most species of fish including the trout. It is often considered a sensory organ. In young fish it is visible through the cartilage of the cranium. The pineal is thought to be sensitive to light and may have an important secretory function for trout. A complete understanding of the function of this tubular organ that is connected to the third ventricle remains elusive (Dodt 1963; Falcon and Meissi 1981).

Contrary to popular belief, fish do have ears. The ear of the trout consists of semicircular canals and three endolymph filled ampullae, the utriculus, the sacculus, and the lagena. Each of these ampullae contains a calcareous otolith. The bilateral endolymph filled labyrinths of fish are encased in the bone and cartilage of the posterior part of the cranium. They are thought to maintain muscle tone, balance, and perception of gravity, as well as serve in the reception of sound (Lowenstein 1971).

CHANNEL CATFISH

The external appearance of the channel catfish varies depending on the age, sex and geographical location of the fish. Generally the fish is slender with a slightly depressed head that is approximately one-fifth its total length. The skin is devoid of scales. Eight barbels are present around the subterminal mouth, four dorsal, and four ventral. The maxillary barbels are the largest, located on the upper lip. Much smaller nasal barbels are found on the cranial margins of caudal nares. The two pairs of barbels on the lower lip are the larger outer mandibulary barbels and the smaller central mental barbels.

Experienced practitioners can usually determine the gender of a mature channel catfish on the basis of the wider flat head and pigmented under body and jaw of the males. Females have a fuller body, and lighter color during spawning. Mature males also have a distinct urogenital papilla. The female genital opening is separate from the urinary opening and has small flaps over it.

The fins of the catfish are relatively unremarkable. There are paired pectorals which each have one hard ray with a serrated posterior edge and nine soft rays. The dorsal fin begins about one third of the body length from the rostrum and has one serrated spine and six soft rays. The second dorsal fin is an adipose fin with no rays, being supported only by fibrous connective tissue. The paired pelvic fins each have eight soft rays, and are found immediately cranial to the anal and urogenital openings near the halfway point of the body. The anal fin is caudal to the anus and is a taxonomic feature in catfish, with each species having different numbers of rays. The caudal fin of the catfish is deeply forked.

The lateral line apparatus of the channel catfish is readily visualized. On the body it runs from the base of the caudal fin to the head along the midbody. Near the head it arches dorsad. Branches extend from the main lateral line to the adipose fin. On the head, branches form the supraorbital canal cranial and medial to the cranial nares, proceeding caudad above the eye. An infraorbital canal is present caudal and lateral to the cranial nares running below the eye. Just caudal to eye, the infraorbital canal turns dorsad and joins the supraorbital canal to form the postocular commissure, which proceeds caudally. A preoperculomandibular canal starts on the lower jaw running caudally from just cranial and lateral to the mandibular barbel. This canal joins the postocular commissure and helps form the cephalic lateralis, which connects with the lateral line of the body just behind the post-temporal bone. Small pit organs (ampullary organs) are also abundant on head, and less so on the body. They form a depression or pit in dermis that is readily visible to the naked eye. The function of these pit organs is not known, although there is some thought that they might be electroreceptors.

INTERNAL TOPOLOGY

The most remarkable aspect of the catfish skeleton is the absence of several bones in the skull, but this is more of taxonomic interest than clinical. Catfish have four pairs of gill arches which are covered by opercula. Gill filaments are present on each arch along with two rows of gill rakers. Catfish have an oral valve consisting of flaps of skin, which jut caudally into the mouth from the upper and lower jaws to prevent respired water from exiting through the mouth.

The channel catfish is omnivorous (Jearld 1970; Cross 1967) and has an alimentary canal that is from one to three times the length of its body (Gammon 1970), and a U-shaped stomach with discernable fundic and pyloric regions. There are no pyloric cecae (Gammon 1970). An intestinal sphincter, visible only on opening the gut, separates the large and small intestine.

The liver of the channel catfish is relatively small. It can be yellowish brown to dark red and is not divided into distinct lobes. The liver extends only a few millimeters caudal to the ventral pectoral girdle. The caudal edge ends at the level of the caudal edge of the base of the pectoral fin on the right side. It extends a few millimeters further caudal

on the left. The gallbladder is on the right caudal aspect of the liver, and a common bile duct enters the cranial small intestine just caudal to the liver.

The pancreas in catfish is scattered as white nodules, sometimes referred to as "Brockman bodies," in the mesentery around the bile duct, the hepatic portal vein, and between the spleen and liver. There are multiple pancreatic ducts that run parallel to the common bile duct, but dump separately into the small intestine. The spleen is located on the left side of the catfish, above the stomach. It is usually translucent and sits just cranial to the gonads.

The swim bladder of the channel catfish is retroperitoneal, sitting between the completely divided cranial and caudal kidneys. It is semirigid, opaque white, and has three chambers, one cranial and two caudal. The transverse septum, which divides the cranial chamber from the caudal pair, is incomplete. The longitudinally separated caudal chambers both communicate freely with the cranial chamber. The channel catfish is physostomous with a pneumatic duct between the midventral aspect of the cranial swim bladder chamber and the dorsolateral right side of the esophagus very close to the stomach. The cranial chamber also comes in direct contact with the skin, forming lateral cutaneous areas.

Although the kidneys are fused laterally, the cranial and caudal kidneys of the catfish are completely separated by the interposing swim bladder. The cranial kidney is composed of endocrine and hemopoietic tissue and is substantial. It generally extends caudad to just behind the gills. The caudal kidney has lateral margins, which can wrap around the posterior swim bladder chambers. The caudal kidney extends cranially to the middle of the dorsal fin and has two Stannius corpuscles located laterally. Two ureters (opistonephric or wolffian ducts) go through the length of the caudal kidney and open at the urogenital pore into the urinary bladder. The urinary bladder is in the caudal body cavity just ventral to the caudal kidney.

The gonads of the catfish are immediately ventral to and attached by mesenteries arising from the peritoneum covering the caudal kidney and swim bladder. Short ducts extend from the caudal ends of the gonads to the genital pore. The testes are lobate with numerous finger-like projections. There are distinct differences in the gross appearance of the cranial and caudal regions of the testes. The cranial portion is composed of the seminiferous tubules. The paired ovaries are tubular and enclosed in a tunica albuginea.

GOLDFISH

The external appearance of goldfish is extremely varied. Like the various breeds of dogs, goldfish have been bred into a myriad of forms and shapes at the whim of goldfish "fanciers." Some of these forms are discussed in the taxonomy chapter of the special medicine section on goldfish, koi, and carp. The more extreme forms have their own particular problems based upon the deformations of their anatomy. For example, fish with protruding eyes are prone to ocular trauma and infections. Rotund breeds like lion heads and orandas are difficult to evaluate for ascites or abdominal distension, an important sign of viral and bacterial diseases affecting goldfish. Similarly, breeds with peculiar scale formations like the pearl-scale breeds, can be falsely diagnosed as suffering from external parasitism, or pathological abdominal distension.

Goldfish, like other cyprinids, have no teeth in their jaws. They do generally have three rows of pharyngeal teeth on the lower pharyngeal bones. These pharyngeal teeth are

true teeth and are used to crush food against a masticating or carp stone on the basioccipital bone. This carp stone is also called the pharyngeal process. It surrounds several major vessels to the head and is covered with a horny epithelium.

Goldfish have the classic four gill arches. The greatest number of gill filaments is on the first gill arch, as is typical of most fish. The pseudobranch of the goldfish lies deep in the roof of the opercular cavity. The heart of the goldfish lies under and just caudal to the gills. A distinct transverse septum separates the pericardial cavity from the abdominal cavity.

The abdominal cavity of goldfish is dominated by a long complex gastrointestinal system compatible with their primarily herbivorous gustatory preference. The esophagus appears to enter the coiling intestines directly. There is no distinct stomach or even a dilatation of the gastrointestinal system, nor are there any pyloric cecae. Usually three coils of intestine nestle a prominent spleen in the center of the intestinal coil.

The swim bladder of goldfish is two chambered with an isthmus and distinct cranial and caudal compartments. The two chambers cannot be closed off from each other unless the isthmus is damaged; however, each chamber has its own rete in its cranial wall. The size of the chambers can be nearly equal or quite disparate in the heavy-bodied breeds of goldfish. These fish are prone to swim bladder disorders which affect their ability to maintain proper equilibrium in the water. The two chambers of the swim bladder are nearly equal volume in Koi and traditional forms of goldfish.

The cranial and caudal kidney of the goldfish are distinctly separated. The cranial kidney is retroperitoneal, just in front of the cranial chamber of the swim bladder. The caudal kidney lies between the cranial and caudal swim bladder chambers. The gonads of the goldfish lie just ventral to the swim bladder. They change size dramatically with the reproductive cycle. The testes fuse posteriorly to form a Y-shaped organ.

The goldfish liver has two main lobes that unite cranially, dorsal to the esophagus. A deep indentation in the cranial dorsal surfaces of the liver accommodates the cranial swim bladder chamber. The gallbladder is surrounded by the right liver lobe. The left lobe of the goldfish liver is occasionally referred to as the splenic lobe since it protects the spleen in a caudal ventral impression. There are usually two additional small accessory liver lobes in goldfish, one just caudal to each of the two main lobes. The left accessory caudal lobe of the liver can be rudimentary (Webber 1827). Similar liver structure is found in Koi, Ide, and Pomfret.

The dorsal mesentery of the goldfish forms a sagittal septum that separates the two main liver lobes, forming a sagittal septum, which contains the hepatic vasculature. Occasionally the liver will be completely divided by this septum, forming two distinct livers that are never joined cranially during embryonic development. The hepatic septum contains three hepatic veins, one arising from the left lobe and two from the right liver lobe, which carry blood from the liver to the heart.

STURGEON

The sturgeon is individual, large, shield-like scales give it a primitive, armored appearance. The heterocercal tail fin, where the backbone runs along the dorsal edge of the caudal fin, gives the tail a shape reminiscent of the sharks. The sturgeons are obviously designed for bottom feeding, with a prominent ventral mouth and barbels common in other bottom dwellers. Sturgeon can grow to immense size and are extremely

important economically for their production of caviar. The species found in the United States are relatively uncommon and are the focus of considerable effort to restock them.

The operculum of the sturgeon does not completely cover the gills. Four gill arches have two hemibranchs each. The heart is located in a heavily protected pericardial cavity that is separated from the abdomen by a thick connective tissue septum.

While young sturgeons have teeth, adults are toothless. Their digestive tract is relatively short. The strong muscular esophagus is lined with longitudinal folds and delivers food to a U-shaped stomach. The opening of the pneumatic duct into the stomach is prominent. The pyloric region as a distinctive structure, that consists of a complex network of pyloric cecae which open into each other forming a sponge-like structure, which obscures much of the stomach from view. The small intestine is very short and almost a saccule which runs cranial from the pylorus to the intestinal valve demarcating the beginning of the spiral intestine. The spiral intestine is similar to the spiral valve found in sharks and rays. It begins at the most cranial reach of the gastrointestinal tract and runs caudally to the anus.

The right lobe of the liver is much more extensive than the left, extending caudally to approximately the level of the caudal tip of the pectoral fin. The left lobe of the liver is much smaller than the right and only extends to the level of the caudal margin of the origin of the pectoral fin. The gallbladder is visible in the cranial ventral portion of the right hepatic lobe. The spleen is long and has multiple lobes. It lies on the left side of the abdomen next to the stomach and is rather large compared to the spleens of other fish. The pancreas of the sturgeon is found as dispersed bits of tissue throughout the abdomen.

The swim bladder of the sturgeon is a simple single ellipsoid chamber with a prominent pneumatic duct that opens into the stomach. The swim bladder is displaced to the right side of the abdomen and is not located along midline as would be expected. The wall of the swim bladder is opaque and relatively thick.

The gonads and kidneys of the sturgeon are in intimate contact. The testes run from the pericardial region to near the anus. Cranially, they are attached directly to the peritoneum and caudally they are suspended by a mesorchium. Sperm pass through the kidneys to reach the primary urinary ducts. Male sturgeon also have a patent oviduct, which appears structurally identical to that seen in females. The function of this duct in males is not known. The ovaries are also connected to the kidneys but the connection is reduced and apparently nonfunctional. Ova are released into the body cavity and taken up by the oviducts (mullerian ducts), which carry them to the urogenital opening that is just caudal to the anal opening.

The eyes of the sturgeon have two crescent shaped bony lamellae, one ventral and one dorsal, which are located between the sclera and the cornea to lend support to each globe. These lamellae are distinct from the subconjunctival ossicles that are also present.

FRESHWATER ANGELFISH

The freshwater angelfish are very popular aquarium fish. These cichlids have a very striking appearance, being laterally flattened, presenting a large lateral body surface. Their anatomy is not that well studied. Externally their most striking feature is their modified body form and fins.

The operculum and gills of the freshwater angelfish are similar to other cichlids. The heart of the freshwater angelfish is hidden under the cranial ventral tip of the liver. It

lies in a triangular compartment pericardial cavity, bounded cranially by the bony limit of the gill chamber.

The abdomen of most freshwater angelfishes is filled with considerable fat. The liver is the most prominent organ in the abdomen. There are two major lobes, one on each side of the abdomen, which join cranially. The cranial ventral portion of the liver lies over the cardiac atrium. The stomach is moderately indistinct and usually hidden under the liver. It is U-shaped often with a gastric caecum. The intestines are relatively long, arranged in coils in the lower abdomen. The spleen lies at the ventral caudal tip of the liver in the mid abdomen. It is usually round or bean shaped.

The swim bladder of the freshwater angelfish is thin walled and transparent. It is a single chamber cranially, which is split into two fingers caudally, lying deep in the tail musculature. The two fingers of the caudal swim bladder are separated by three to four ventral vertebral processes. The kidneys of the freshwater angelfish are retroperitoneal and are not separated into distinct cranial and caudal organs, however, the cranial kidney fills the space between the cranial aspect of the swim bladder and the skull and is a prominent organ. The kidneys run the entire length of the abdomen and are fused, lying just below the spinal column.

FLOUNDER

The external anatomy of an adult flounder is a dramatic departure from the anatomy of other fishes. The flounder lives its life on its side. During early development, the right eye migrates clear to the left side of the flounder's head and the animal assumes a position with it's right side down and left side up. The pectoral fins remain on each side of the animal, positioned high at the level of the middle of the operculum. Flounder have strong teeth in both jaws and a distinct fleshy, tongue that is continuous with the gill isthmus and has a spade-like distal tip. There are four pairs of gill arches covered completely by the operculae. The cloaca opens on the ventral side (right) of the fish under the anal fin.

As might be expected, the abdominal contents of the flounder are asymmetrical. The liver is essentially a single lobe that occupies most of the left or upper side of the fish. On the right side or down side of the fish, only two small portions of the liver are visible, protruding around the gallbladder. A short, thick walled esophagus empties directly into the stomach which is approximately twice the length of the esophagus. The stomach has a small pyloric pouch. Three large, thick, pyloric cecae, about half as long as the stomach, branch off of the small intestine. Two branch off on opposite sides of the small intestine, about 1.5 cm after the pylorus. The third cecum branches off the intestine another 1.5 cm after the first two cecae. The intestinal tract is short, approximately the same total length as the stomach, with the rectum emptying into the cranial aspect of the cloaca.

The bean shaped spleen of the flounder is found in the lesser curvature of the stomach. It is generally half the size of the pyloric cecae. Paired gonads are nestled into a pocket in the tail musculature. The oviducts are prominent and end in an opening in the caudal aspect of the cloaca. The cranial and caudal kidney are continuous, located retroperitoneally under the vertebral bodies. There is no swim bladder, as might be expected in a bottom dwelling species.

The heart is located approximately one third of the body width dorsal to the base of the anal fin. It is most superficial on the upper or left side of the flounder. There is a well developed bulbus arteriosus, which receives the blood from the ventricle and sends it to the gills.

Locating the brain of a flounder can be a challenge. The eyes are used as landmarks. In the flounder, the brain is located approximately 1.5 eye diameters caudal to the right or upper eye. It is interesting that the optic nerves of the flounder do not appear to cross.

MARINE ANGELFISH

The tropical marine angelfish are another group of laterally compressed fish with distinct body form. They are quite colorful and have a modified fin structure with a single dorsal fin running along the majority of the dorsal surface and a single ventral fin rather than separate pelvic and anal fins. Externally the lateral line is prominent. In the blue angelfish the lateral line begins just behind the dorsal commissure of the opercular arches. From there it follows the line of the back, running about one fifth of the total body depth below the dorsal limit of the body, to the base of the caudal peduncle. The lateral line of the grey angelfish begins about 1 cm above the dorsal commissure of the operculum and parallels the dorsal ridge of the body about one quarter of the body width down. The lateral line continues to the caudal peduncle where it runs in a straight line, slightly above horizontal midline, to the beginning of the caudal fin. There are half a dozen sensory pits around the eye and the top of the mouth in the grey angelfish. The pupil of the grey angel fish is asymmetric, and elliptical, with the narrow radius of the ellipse toward the nasal canthus.

The operculum of marine angelfish can have prominent spines, usually on the ventral caudal aspect. Some marine angelfish will also have bony spines on their chin. Additional defensive spines occur on the ventral body, caudal to the anus at the start of the ventral fin.

Under the operculum there are four pairs of complete gill arches with two hemibranchs each. The heart of the marine angelfishes lies in a well defined pericardial cavity nearly obscured by the gills above the bony ventral notch formed by the cleithrum and coracoid bones. In a full grown specimen the heart is a full 4 cm deep from the ventral surface of the fish. The heart is a typical fish heart with a very large and prominent thin walled atrium.

In the abdomen of the marine angelfishes, the liver is usually nearly symmetrical, but extends slightly further caudal on the left side. There is a single ventral lobe which cradles the stomach and joins the rest of the liver with a broad isthmus. The gallbladder is prominent and lies dorsal to the stomach, often, extending caudally to lie between the origins of the gonads.

The esophagus is extremely large, approaching 3 cm in diameter in larger specimens, with distinct corrugations in the serosal surface. The esophagus empties into a large distinct C-shaped stomach which occupies much of the abdominal cavity. The pylorus of the stomach lies adjacent to the cardia. A distinct cardiac and fundic region of the stomach can be identified grossly. In the French and grey angelfishes there are 18 small thin pyloric cecae radiating from the beginning of the small intestine. The blue

angelfish has between 20 and 24 pyloric cecae. The small intestine is long, up to 75 cm, and is over five times as long as the colon it empties into.

The pancreas is diffuse and not identifiable as a discrete organ. The very flat spleen of the marine angelfish is technically on the right side of the fish, but lies nestled in the fold where the esophagus enters the stomach, nearly on midline. Its location is similar to the location of the spleen in the lesser curvature of the stomach in mammals. The spleen is usually darkly pigmented by melanocytes in its capsule. It is well protected from accidental damage from intraperitoneal injection by the stomach and bowel.

The swim bladder of the marine tropical angelfish is thick and fibrous with a thin endothelial lining. It is a single chamber cranially, which is split into two fingers caudally, lying deep in the tail musculature. The two fingers of the caudal swim bladder are separated by six prominent ventral vertebral spines in the French and grey angelfish and by three enlarged ventral processes in the blue angelfish.

The cranial kidney of marine angelfishes is very prominent and well separated from the caudal kidney. The large cranial kidney lies dorsal to and directly behind the gill chamber as a single mass. It extends caudally the width of one or two vertebrae and is nearly spherical. The caudal kidney, on the other hand, is extremely thin and very minor, appearing simply as blotches in the ventral view of the vertebrae. Ureters from the caudal kidney enter a very thin walled urinary bladder, which lies in the caudal ventral quadrant of the abdomen behind the bowel. The urethra is extremely short, running from the ventral aspect of the urinary bladder to open into the cloaca, just caudal to the rectal opening. The gonads run ventrad from the origin to the gallbladder to the level of the cloaca, forming a Y-shaped organ. The sexual ducts empty into the cloaca.

The brain of marine angelfish can be difficult to locate because of the disc like shape of the head and body. The best method for locating the brain is to use the eye as a landmark. The brain lies on midline, just at the dorsal limit of the globe or the orbit if the eye has been removed. The optic nerve moves nearly directly medial to cross just under the cranial portion of the brain.

SEA HORSE

The seahorse is often mistaken for an invertebrate, probably because of its hard plate-like exterior. It is a bony or teleost fish, however, and possesses a backbone. The hard outer skin of the seahorse complicates clinical procedures and simple injections. The sea horse swims in a vertical head up position using its pectoral fins to stabilize itself. These fins are located high on its body, essentially on its head, where many people expect to find ears. The seahorse propels itself through the water with the help of the single dorsal fin. There is a small pelvic fin, which is of no value in locomotion.

Seahorses are sexually dimorphic and their gender can easily be determined by examining their genital openings near the pelvic fin. Females have a small inverted crescent shaped anus just cranial to a very small punctate reproductive opening. Both openings are located cranial to the pelvic fin. The male seahorse has a similar crescent shaped anus above the pelvic fin, but has an elongated genital opening below the pelvic fin rather than above it. This is the opening to the pouch.

The gills of the seahorse are relatively unaccessible because the operculum has a nearly complete membranous attachment to the body. The modified tail is suitable for grasping and holding to anchor the seahorse in position.

The internal anatomy of the seahorse is nearly as unusual as the external anatomy. An esophagus leads to a stomach, which is merely a dorsal pouch, at the crook of the seahorse's neck. The pylorus is inapparent or missing, making the stomach very difficult to appreciate. The intestines of the male seahorse are nearly 50 percent longer than those of the female. There is a gallbladder, a discrete organ at the most cranial portion of the liver, just below the crook of the neck. The liver is most evident on the right side of the animal where it takes a bend caudally about half way down the abdomen.

The kidneys are paired and retroperitoneal with no separation between the cranial and caudal portions. The gonads can be quite variable. The testes often appear as thin white lines running on the ventral surface of the kidneys. The ovaries are low, paired orange structures.

The heart of the seahorse is a dark triangular organ in the apex of the neck, just behind the gills. It surrounds the esophagus with three major vessels on each side, one coursing cranially to the gills, two running caudally, bringing return blood from the liver and kidneys. The gills of the seahorse are quite striking structures, quite distinct from those of other fishes. Seahorse gills are hemispherical organs with many individual gill projections giving the organ the appearance of a small chrysanthemum. They lie in a closed gill chamber, and gill biopsy is not practical.

BROWN SHARK

The brown shark is an elasmobranch, a member of the chondroichthys. Like all other sharks, skates, and rays, its entire skeleton consists of cartilage rather than bone, although the jaws are heavily mineralized. Externally, the brown shark covered by small placoid scales which give the skin a rough sandpaper texture. The placoid scale is derived from the same embryonic progenitor as the teeth of sharks. These scales, which are histologically identical to the shark's teeth, are also found in the mouth and throat, extending down the pharynx to the level of the caudal gill chamber. The teeth of sharks are replaced in sets as they are used and broken or lost.

The fins of the brown shark are distinctive, being considerably more substantial than the transparent membranes supported by thin rays and spines found in most bony fish. Nevertheless, the same fin nomenclature is applied. The majority of shark species have very streamlined fusiform body conformations. They may or may not have a complete compliment of fins. The variation seen most frequently is the absence of a dorsal fin. The tough fibroelastic properties of shark skin allow it to hold sutures very well, but at the same time, rapidly dulls surgical needles. The lateral line of most sharks is quite prominent.

Like most sharks, the brown shark has five pairs of individual gill slits. The gills are located within the gill slits which can be closed off by highly developed gill septae. The thymus is fairly distinct, located in the upper inner edges of the gill slits. The ultimobranchial bodies are only found on the left side of the body, on the dorsal wall of the pericardium between the elements of the last gill arch rather than on both sides (Harder 1975).

Internally, the clinically significant comparative anatomy points for a shark are its large liver, the presence of a spiral valve, the absence of a swim bladder, and the presence of a rectal gland. The liver of the brown shark is extremely large and may make up nearly 25 percent of the animal's body weight. It has two nearly equal sized lobes which run

nearly the entire length of the abdomen. The gallbladder is in the cranial part of the left liver lobe. Not all sharks have gallbladders.

The esophagus of the brown shark empties through a well developed esophogastric sphincter into a J-shaped stomach, which has a distinct pylorus. Many sharks do not have the esophogastric sphincter of the brown shark, which probably inhibits regurgitation. The small intestine of the brown shark is relatively short, emptying through an intestinal valve into the spiral colon, sometimes called the spiral valve. The spiral colon provides a greatly increased surface area for nutrient absorption in the otherwise short digestive tract of the shark.

The rectal gland is found dorsal to the colon, opening into that part of the intestine just before the cloaca. The gland is suspended by a mesentery of the colon. It has also been called the cecal gland, the anal gland, the supra-anal gland, or the digitiform appendix. It is a thick walled tube with a narrow central lumen. The glandular tissue of the tube gives rise to many tubules that are arranged running radially to the lumen (Harder 1975). Blood from the rectal gland is fed into the hepatic portal system. It is thought that this gland has a major role in sodium chloride excretion and sodium balance.

The pancreas is a discrete organ in the brown shark. It wraps around the small intestine near the pylorus. The spleen of the brown shark is elongated and essentially bilobed. Sharks frequently have accessory spleens which are not the result of any traumatic event. The kidneys of the shark are retroperitoneal and the left and right kidney are completely separate. The cranial edge of the kidney is usually just below the base of the front edge of the first dorsal fin. Directly underneath the kidney are the gonads. In the male shark, the cranial portion of the kidney serves as an epididymis. Just caudal to this portion of the kidney is a region that produces liquid to accompany the sperm as it runs caudally through the seminiferous tubules and the sperm duct. This is sometimes called Leydig's gland. Two masses of lymphomyeloid tissue, the epigonal organs, run along the dorsal abdomen nearly obscuring the kidneys and testis in the male or the kidneys, uterus, oviduct, and ovary of the female shark.

In sharks and rays, the suprarenal organs are found along the aorta forming large complexes referred to as "corpora axillaria." This is the source of the term "suprarenal organ." In sharks the corpora axillaria is found on the dorsal side of the caudal kidney.

The brown shark has eyelids and a nictitans. The wall of the eye is strengthened by a cartilage layer, which is thick at the posterior part of the globe and thins as it nears the limbus of the cornea. There is a tapetum lucidum.

THE SOUTHERN STING RAY

The rays are also elasmobranchs, or cartilaginous fish, but they have a very distinct body form, quite different from the sharks. Rays and skates are flattened dorsoventrally, maintaining normal dorsal ventral orientation in their environment, as opposed to the laterally flattened flounder, which lay on their side. The rays are covered by placoid scales much like those found covering the sharks. The mouth of a ray is oriented downward to facilitate bottom feeding. The mouth is a crushing type of mouth, with broad crushing plates suitable for diets based primarily upon mollusks and crustaceans.

An important adaptation in the skates and rays to bottom feeding is the spiracle system, which allows water for respiration to be taken in on the dorsal side of the fish,

moved past the parabranchial gill cavities, and expelled underneath through the five pairs of ventral gill slits. This minimizes the intake of silt and bottom debris, but also complicates gill biopsy. The spiracle is a remnant of the gill slit between the first and second gill arches. The true spiracle has a short hemibranch that is the same as the pseudobranch in bony fish (Harder 1975).

The thyroid gland of the skates and rays is a disc shaped compact organ located in the ventral throat region similar to that found in sharks. It is not diffusely distributed like in the bony fishes. The ultimobranchial bodies are paired and found caudal to the pericardium.

The internal organs of the rays are similar to those of the shark. When the abdomen is opened, essentially all abdominal organs are obscured by the massive liver. Only the caudal aspect of the stomach and the spiral colon peak out from under the large liver. The digestive system consists of a short esophagus with an esophageal valve just above a series of transverse rugae in the cranial stomach. There is a transition zone, and then the rest of the true stomach is lined with deep longitudinal rugae. The stomach ends at a pylorus that dumps into a very short, saccule-like intestine that leads to the spiral valve. The spiral valve empties into the short rectum, which in turn empties into the cloaca. The ray has a true cloaca.

The spleen is on the left side of the abdomen, well covered by the large liver. The pancreatic duct of the ray develops from the dorsal anlage just as it does in sharks and skates (Harder 1975). This duct is commonly called "Santorini's duct." The kidneys run retroperitoneally along the spine as in other fish. In the rays, as in the sharks, the suprarenal organs are found along the aorta and form larger complexes referred to as "corpora axillaria." The interrenal glands are paired and enclosed in a connective tissue capsule between the caudal ends of the kidneys. There is no swim bladder. The gonads are located ventral to the kidneys.

Male rays have copulatory organs that are modifications of the medial edges of the pelvic fins. These are called "claspers." They have a dorsal longitudinal groove. During copulation the distal end of the clasper is inserted into the oviducts opening the female's cloaca. Rays also have a clasper gland beneath the ventral skin of the claspers that produces a white viscous secretion that coagulates in water and may serve to seal off the clasper groove and help in semen transport (Lamarca, 1963).

Rays and skates have a rectal gland similar to the one in sharks. The gland is a thick walled tube with a central lumen. The glandular tissue is divided into lobes and the ducts run parallel to the length of the organ rather than radially as they do in the sharks. The gland receives blood from a branch of the caudal mesenteric artery, which divides to form a vascular network. There is apparently no countercurrent principle involved in the relationship of the ducts and the vascular supply (Harder, 1975). Blood from the rectal gland goes into the hepatic portal system. In some of the rays, the rectal gland has an S-shape rather than the usual finger shape.

Skates have a paired bilateral organ of unknown function. It is found lateral to the vas deferens of the testicle and opens near the genital opening into the cloaca. It can be quite large and consists of many folds of epithelium. It secretes a clear liquid that is essentially free of protein and complex polysaccharide with a pH of 9.2 and a carbon dioxide content of 210 mmoles/liter (Harder 1975). This gland is called the alkaline gland or sometimes Marshall's gland.

Rays have no eyelids, but instead are able to pull their eyes back into their head for protection. There is a tapetum lucidum choriodeale between the lamina vasculosa and the lamina choriocapillaris which is different from the usual teleost eye (Harder 1975).

Literature Cited

Ali, M.A. (1959) The ocular structure, retinomotor and behavioral response of juvenile Pacific salmon. Can. J. Zool. 37:965-996.

Anderson, B.G., Mitchum, D.L. (1974) Atlas of trout histology. Wyo. 'GameFish Dep., Bull. 13. Pp. 1-110.

Bertin, L. (1958) Organes de la respiration aquatique. In: Trait'e dezoologie (P.P. Grass'e, editor) Vol. 13 pp 1303-1341. Masson, Paris.

Bullock, W.L. (1963) Intestinal histology of some salmonid fishes withparticular reference to the histopatho logy of acanthocephalan infections. J.Morphol. 112:23-44.

Burnstock, G. (1959) The morphology of the gut of the brown trout, Salmotrutta. Q. J. Microsc. Sci. 100:199-220.

Cross, F.B. (1967) Handbook of Fishes of Kansas. Univ. Kan. Mus. Nat.Hist., Misc. Publ. 21. 156 p.

Dodd, J.M. (1960) Gonadal and gonadotrophic hormones in lower vertebrates.In: Marshall's Physiology of Reproduction (parkes, A.S., editor) 3rd ed., Voll, Part 2, Chap. 11 pp. 417-582. Longmans, Green, New York.

Dodt, E. (1963) Photosensitivity of the pineal organ in the teleost, Salmoirideus (Gibbons). Experientia 19:642-643.

Falcon, J. and H. Meissi. (1981) The photosensory function of the pinealorgan of the pike (Esox lucius L.). Correlation between structure andfunction. J. Comp. Physiol. 144 Part A:127-138.

Gammon, R.L. (1970) The Gross and Microanatomy of the Digestive Tract andPancreas of the Channel Catfish, Ictalurus punctatus. M.S. Thesis. KansasState Univ., Manhattan, Kansas. 52 p.

Grizzle, J. M., Rogers, W.A. (1976) Anatomy and histology of the channelcatfish. Auburn University Agricultural Experiment Station, Auburn, Alabama.94 pp.

Groman, D.B. (1982) Histology of the Stripped Bass. American FisheriesSociety Monograph No. 3. Bethesda, Maryland.

Harder, W. (1975) Anatomy of Fishes. E. SchweizerbartschVerlagsbuchhandlung, Stuttgart, Germany.

Henderson, N.E. (1962) The annual cycle in the testes of the eastern brooktrout, Salvelinus fontinalis (Mitchill). Can. J. Zool. 40:631-641.

Hoar, W.S. (1955) Reproduction in teleost fish in The ComparativePhysiology of Reproduction and the Effects of Sex Hormones in Vertebrates(Chester Jones and P. Eckstein editors) Mem. Soc. Endocrin., No. 4:5-24.Cambridge Univ. Press, London.

Hughes, G.M. (1984) General Anatomy of the Gills in Fish Physiology Vol. XA(W.S. Hoar and D.J. Randall, editors). Academic Press, New York, pp 1-72.

Jearld, A. Jr. (1970) Fecundity, Food Habits, Age and Growth, Length-weightRelationships and Condition of Channel Catfish, Ictalurus punctatus(Rafinesque), in a 3300 Acre Turbid Oklahoma Reservoir. M.S. Thesis, OklahomaState Univ., Stillwater, Oklahoma. 78 p.

Lagler, K.F., Bardach, J.E., Miller, R.R., and Passino, D.R.M. (1977)Ichthyology, 2nd ed. John Wiley & Sons, Inc., New York. 545 pp.

LaMarca, M.J. (1963) The functional anatomy of the clasper and clasper glandof the yellow sting ray, Urolophus jamaicensis (Cuvier). J. Morph. (Philad.),114:303-323.

Laurent, P. (1984) Gill Internal Morphology in Fish Physiology Vol. XA (W.S.Hoar and D.J. Randall, editors). Academic Press, New York, pp 73-184.

Laurent, P. and Dunel, S. (1978) Hisophysiologie-relations anatomiques desionocytes (cellules a chlorue) avec le compartiment veineux branchial:d'efinition de deux types d"epith'elium de la branchie des poissons. Comptesrendus Academic de Sciences Paris, S'erie D:1447-1450.

Leiner, M. (1938) Die Physiologie der Fischatmung. Akad. Verlagsges.Leipzig.

Lowenstein, O. (1971) The labyrinth. in Fish Physiology Vol 5 (W.S. Hoarand D.J. Randall, eds.) Academic Press, New York, pp 207-240.

McLean, J.R. and Nilsson, S. (1981) A histochemical study of the gas glandinnervation in the Atlantic cod, Gadus morhua. Acta Zoologica 62:187-194.

Muller, J. (1839) Vergleichende Anatomie der Myxinoiden. III. Uber dasGefassys tem. Abh. Akad. Wiss., Berlin pp 175-303.

Prince, J.H. (1956) Comparative Anatomy of the Eye. Charles Thomas Pub.Springfield, Ill. p 355.

Pumphrey, R.J. (1961) Concerning vision in The Cell and the Organism(Ramsay, J. A. and V.V. Wigglesworth editors) Cambridge Univ. Press, New York.Chapter 13, pp 193-208.

Reyer, H.U. (1977) The role of the swim-bladder in vertical movement offishes (Carassius auratus, Salmo gairdneri, and Tilabia mariae). Biology ofBehavior 2:109-128.

Robertson, O.H. (1958) Accelerated development of testis after unilateralgonadectomy with observations on normal testis of rainbow trout. US FishWild. Serv. Fish. Bull. 58:9-30.

Ross, L.G. (1979) The haemodynamics of gas resorption from the physoclistswimbladder: the structure and morphometrics of the oval in Pollachius virens.J. Fish Biology 14:261-266.

Ross, A., Yasutake, J.W.T. and Leak S. (1975) Phoma herbarum, a fungal plantsaprpophyte as a fish pathogen. J. Fish. Res. Board Can. 32:1648-1652.

Schwartz, E. (1969) Luftatmung bei Pantodon buchholzi und ihre Beziehung zurKiemenatmung. Z. vergl. Physiol. 65:324-339.

Walls, G.L. (1942) The Vertebrate Eye and Its Adaptive Radiation. HafnerPub Co. New York (Reprinted 1963) Chapter 14, pp 563-591.

Watling, H. and Hillemann H.H. (1964) The development of the olfactoryapparatus of the grayling Thymallus arcticus J. Fish. Res. Board Can.21:373-396.

Weber, D. D. and Schiewe, M.H. (1976) Morphology and function of the lateralline of juvenile steelhead trout in reltion to gas-bubble disease. J. FishBiol. 9:217-233.

Weber, E.H. (1827) Uber die Leber von Cyprinus carpio, die zugleich dieStelle des Pancreas zu vertreten scheint. Arch. Anat. Physiol. 2:294-299.

Weinreb, E. L. and Bilstad, N.M. (1955) Histology of the digestive tract andadjacent structures of the rainbow trout, Salmo gairdneri irideus. Copeia1955:194-204.

Yamamoto, K., Oota, I., Takano, K. and Ishikawa T. (1965) Studies on thematuring process of the rainbow trout, Salmo gairdnerii irideus - I.Maturation of the ovary of a one-year old fish. Bull. Jpn. Soc. Sci. Fish.31:123-132.

Yatsutake, W.T. and Wales, J.H. (1983) Microscopic Anatomy of Salmonids: AnAtlas. USDA Fish and Wildlife Service, Resource Publication 150, Washington,DC.

FISH NUTRITION: AN OVERVIEW OF DIETARY REQUIREMENTS

George Blasiola, Aquatic Biologist B.S, M.S

Introduction: Maintaining good fish health depends on the quantity and quality of nutrients received in the daily diet. Although nutritionally related diseases are uncommon in nature, fish maintained in captive situations are prone to nutritionally related diseases. The requirements for some nutrients also increase during episodes of stress including disease. In order to ensure normal growth, good structural tissue and organ integrity and function, normal reproduction and resistance to infection fish must receive nutritionally balanced diets.

To minimize nutritional problems, diets must be formulated from high quality and varied components that provide balanced and essential nutrients in the correct amounts. The diet should provide the basic macronutrients protein, fat, carbohydrate, fiber and vitamins and minerals. The majority of the research on the nutritional requirements of fish has been conducted on economical species such as salmon, trout, catfish, carp, and mahi-mahi with limited research on marine and freshwater aquarium fish. However, with the expanded interest of maintaining fish for both for aesthetic enjoyment as well as scientific research we are accumulating more information on the nutritional requirements of some aquarium species.

A majority of freshwater aquarium fish are captive bred and easily adapt to a wide range of foods. On the other end of the spectrum, less than 50 species of marine fish are captive bred. This present the problem of providing a diet that replicates the natural diet for many marine fish. Basic diets should be based as much as possible on the types of foods that fish feed on in the natural habitat, especially important with wild caught marine fish.

Lecture Outline:

Introductory Remarks

- Dietary Requirements

Feeding habits of Fish in Nature

- Extreme diversity of food habits
- Linked to structure and form
- Food habits can change from juvenile to adult as well as from season to season.
- Evolution of new species presents the problem of finding a new food source.

Anatomical Differences and Digestion

- Mouthpart specialization
- Presence or absence of teeth; some species have pharyngeal teeth
- Variable digestive tract length depending on food habits
- Presence of pyloric cecae in some species
- Inner surface of intestine of some species have folds or projections to increase food absorption

Natural Food Habits and Behaviors of Fish: Classification

- Carnivorous
- Herbivorous
- Omnivorous

- Surface
- Midwater
- Bottom

Considerations in Feeding Aquarium Fish

- Wild caught vs domestic bred? If wild, is it a specialist feeder?
- Providing food that will be acceptable in size, nutritional quality and proper sensory ingredients to elicit a feeding response.
- Providing a diet that is similar in nutritional composition to those foods found in the natural habitat.
- Consideration of nutritional requirements of juvenile vs adults

Feeding Behaviors: Freshwater and Marine Fish

- Surface Feeders
- Midwater Feeders
- Bottom Feeders

Priority Energy Requirements of Fish

- Daily Activities
- Maintenance
- Growth
- Reproduction

Factors Affecting the Metabolic Rates of Fish

- Water Temperature
- Species
- Age
- Physical Condition
- Feeding Habit

Major Categories of Aquarium Foods

- Live
- Frozen
- Freeze-Dried
- Prepared Dry

Prepared Dry Food Categories

- Flake (drum dried, extruded)
- Steam Pelleted
- Extruded
- High Moisture

Potential Problems Associated with Feeding Aquarium Fish

- Frequency
- Amount
- Overfeeding
- Underfeeding
- Inappropriate Particle Size
- Acceptability (palatability)

Nutrient Requirements of Fish

- Protein
- Carbohydrates
- Lipids
- Fiber
- Vitamins
- Minerals

Proteins

- Composed of amino acids
- Essential and Non-Essential
- Animal proteins more complete than plant
- Amino acid profile of diet is critical

Carbohydrates

- Sugars, starches and cellulose
- Converted to simple sugars by fish
- Not all fish utilize carbohydrates well
- Spares protein

Lipids

- Fats, phospholipids, waxes and steroids
- Spares protein in diets
- Essential for normal growth and health
- Specific requirements known for certain species

Essential Fatty Acids for Marine Fish

- Marine fish require n-3 HUFA'S
- Of the various fatty acids two are especially critical for normal growth: DHA and EPA
- Docosahexaenoic Acid (DHA) and Eicosapentaenoic Acid (EPA)

Food Enrichment Techniques

- Supplying of DHA and EPA critical for larvae survival
- Live foods such as Artemia and Rotifers can be enriched with HUFA's by feeding foods containing these fatty acids
- Commercial products available
- Live food can be fed first with live microalgae such as Chaetoceros or freeze-dried algae such as Schizochytrium.

Fiber

- Indigestible portion of diets
- No specific requirements known

Vitamins

- Catalysts for biochemical transformations
- Not an energy source
- Fat soluble (A,D,E,K)
- Water soluble (B complex,C)

Vitamin C (Ascorbic acid)

- Must be supplied in diet
- Antioxidant
- Required for various functions
- Amount required related to stress
- Essential for wound repair
- Critical role in disease resistance
- Needed for normal bone repair

Minerals

- Fish absorb some minerals from water
- Calcium, Phosphorous, Magnesium

Nutritional Deficiencies

- Macronutrient
- Micronutrient
- Malnutrition
- Toxins in Diet

Signs of Nutritional Deficiencies

- Low disease resistance
- Abnormal Color
- Reduced growth
- Abnormal behavioral signs
- Lowered fecundity
- Low survival rate of offspring

Macronutrient Imbalances in Fish Diets

- Protein: Inadequate quantity and quality of amino acids
- Carbohydrates: Excess
- Fiber: Excess prevents nutrient uptake
- Fat: Excess, deficiency or improper type
- Minerals: Deficiency

Malnutrition

- Partial or Total Nutrient Deprivation
- Difficult to Reverse in later stages
- Can be caused by parasitism
- Serious with juvenile fishes
- Signs include weight loss and sunken abdomen

Toxins

- Absorbed from the food
- Herbicides, pesticides, rancid fats
- Antinutrients (thiaminase)
- Mycotoxins

Formulating Fish Foods

- Commercial Foods and Lab/Home Formulations
- Ingredient Components

Formula Development Calculations

- Basic Calculations for Fish Food Formulas
- Expanded Formula Calculations
- Adding Medications

Basic Gelatinized Formula

- Criteria for Ingredient Selection
- Preparation

Pigments

- Carotenoids
- Xanthophylls
- Pterins

Spirulina (*Spirulina platensis)*

- Microscopic algae rich in protein, vitamins, essential amino acids, minerals, essential fatty acids and pigments.
- Extensive research indicates it can increase growth rate, fertility, survival rate and coloration when fed to fish.
- High xanthophylls concentration of 5,800 mg/kg

Stomach Analysis

- Basic research methodology for determining what fish feed on in the natural habit.
- Interpretation of contents can be difficult, as some items in the gut could have been accidentally ingested.
- The analysis allows comparison of gut contents of various fish at different stages of life cycle and different seasons.

Basic Guidelines for Feeding Aquarium Fish

- Match the food type as closely as possible with diet of fish in the natural habitat
- Consider the life cycle stage of the fish, change diet as required
- Supplement with a variety of foods
- Avoid maintaining fish that have highly specialized feeding requirements
- If using prepared foods read label carefully. Avoid aquarium "junk foods"
- Store foods properly to minimize nutrient degradation
- Use live foods with caution

Selected References

Barclay, W. and S.Zeller. 1996. Nutritional enhancement of n-3 and n-6 fatty acids in rotifers and *Artemia* nauplii by feeding spray-dried *Schizochytrium* sp. Journal of the World Aquaculture Society 27(3): 314-322.

Boonyaratpalin, M. and N. Unprasert. 1989. Effects of pigments from different sources on colour changes and growth of Red *Oreochromis niloticus.* Aquaculture 79: 375-380.

Halver, J. 1989. Fish Nutrition. Academic Press, New York

Hanley, F. 1987. The digestibility of foodstuffs and the effects of feeding selectivity on digestibility determinations in Tilapia, Oreochromis niloticus (L), Aquaculture 66:163-179.

Henson, R.H. 1990. Spirulina algae improves Japanese Fish Feeds. Aquaculture Magazine 16(6): 38-43.

Mahajan, C.L. and N.K. Agrawal. 1979. Vitamin C deficiency in Channa punctatus Bloch. Jouranl of Fish Biology 15:613-622

Randall, J.E. 1967. Food habits of reef fishes of the West Indies in Proceedings of the International Conference on Tropical Oceanography. pp. 665-847 University of Miami, Coral Gables, FL

Spotte, S. 1992. Captive Seawater Fishes: Science and Technology. John Wiley and Sons, New York

© 2005 All rights reserved .George Blasiola

FRESHWATER AQUATIC DIET FORMULATION **BASIC GELATINIZED FOOD BASE** **WORKSHEET**

Formula Reference based on: Gratzek, p 51, Fish Health Mgmt Course Notes UGA

Batch Amount Target (gm) 142
Batch Amount Actual (gm): 42 (Dry)
FORMULA: Initial
Date: 7/31/2003

Ingredient	Amount Added	Protein %	Amount	Fat %	Amount	Fiber %	Amount	Ash %	Amount	Carbohydrate %
Flake Food	20	45	9	5	1	8	1.6	5	1	
Canned Sardines, O	5	16.3	0.815	12.5	0.625	0	0	4	0.2	
Cod Liver Oil	3	0	0	100	3	0	0	0	0	
Corn Oil	1	0	0	100	1	0	0	0	0	
Gelatin	10	6	0.6	0	0	0	0	2	0.2	
Plankton,F.D	2.5	60	1.5	10	0.25	19	0	5	0.125	
Water (gm)	100									
Vitamin C	0.5									
Vitamin E (10 units)										
Ingredient Total (gm)	142		11.915		5.875		1.6		1.525	
Proximate Analysis (%) :		**28.36905**		**13.9881**		**3.809524**		**3.630952**		**42.20238095**
As Fed:		**8.24**		**4.2**		**1.14**		**1**		**15.42**

Preparation:
Add 50 ml of water to a bowl or beaker. Mix in the corn oil and codliver oil.
Add vitamin C and E. Macerate sardines with a knife or in a blender or food processor.
Add 10 gms of gelatin to 50 ml of water, allow to siften, heat until dissolved. Allow to cool to slightly.
Add to other ingredients and mix well. Note: can all be mixed in food processor or blender.
Pour into small packages, lay flat in refrigerator to set, freeze.

FRESHWATER QUALITY AND AQUARIUM SYSTEMS

John B. Gratzek DVM, PhD

Health Information about Water
(A report from the Athens Clark County Public Utilities Dept, July 1, 2002)

"The sources of drinking water (both tap and bottled water) include rivers, lakes, streams, ponds, reservoirs, springs and wells. As water travels over the surface of the land or through the ground, it dissolves naturally occurring minerals and, in some cases, radioactive material, and can pick up substances resulting from the presence of animals or from human activity".

Contaminants that may be present in source water include the following:

Microbial contaminants, such as viruses and bacteria may come from sewage treatment plants, septic systems, agricultural livestock operation and wildlife.

Inorganic contaminants such as salts and metals, which can be naturally occurring or result from urban storm runoff, industrial or domestic wastewater discharges, oil and gas production, mining or farming.

Pesticide and herbicides, which may come from a variety of sources such as agriculture, urban storm water runoff and residual uses.

Organic and chemical contaminants, including synthetic and volatile organic chemicals, which are by-products of industrial processes and petroleum production, and can also come from gas stations, urban storm water run off and septic systems.

Radioactive contaminants, which can be naturally occurring or be the result of oil and gas production and mining activities.

In order to ensure that tap water is safe to drink, the EPA prescribes regulations that limit the amount of certain contaminants in water provided by public water systems. Food and Drug Administration regulations establish limits for contaminants in bottled water which must provide the same protection for public health.

The message is... drinking water is generally a good source of water used for aquaria.

Aquarium systems can be simple (inexpensive) or complex:

FLOW THROUGH SYSTEMS

One of the simplest examples of keeping fish is that found in trout production areas of the Northwest as exemplified by trout raceways in Idaho's snake valley. The source of water is springs from the surrounding mountains with the optimum water temperature at about 58 F. Because of the steady flow of water waste products are minimized in this raceway. EPA regulations mandate that but wastewater from

raceways be diverted into settling ponds to minimize waste products from contaminating local rivers.

Experimental flow-trough systems shave been devised using heated dechlorinated municipal water. A unit at the Veterinary College at the University of Georgia was devised Research flow through system used for experiments on channel catfish. City tap water had to be dechlorinated heated and provided with uniform water flow. The system eliminated differences between aquaria. Such flow-trough systems engender high costs for water, heating and dechlorination.

Central Filtration Systems:

Central filtration systems are flow through systems which recycle aquarium water after adequate filtration of fish wastes and destruction of pathogenic organisms for fish. These systems are commonly used in public aquaria, wholesale fish establishments, pet stores, and aquatic research centers.

The advantages of such a system include:

- Uniform water quality
- Ease of water quality control
- Labor and time saving
- disease prevention

The components include:

- An efficient mechanical filtration apparatus not unlike those used for swimming pools
- Ample substrate for biological filtration. Substrates can be a gravel bed, foam (as in mattress foam cushions), or a variety of plastic surfaces. Circular wheel-type discs (wet-dry filter) constructed from plastic roofing have been used with good success. Their rotation in water provides a self cleaning function. Commercial filtration products including bio-wheels, and wet-dry trickle systems can be found in the internet under **AQUATIC ECOSYSTEMS**
- An ultraviolet light sterilization system with enough power to kill protozoan parasites is required. This power is described as KWS/cm^3 (kilowatt seconds per square centimeter). The minimum power to inactivate Ichthyophthirius multifillis intermediates. By experimentation the minimum wattage was 91,000 KWS/cm^3 . Because ultraviolet lamps lose power with time, a common practice is to double the wattage and to change bulbs after several months of usage. Bacteria and viruses are readily killed at these wattage levels.

FILTRATION BASICS

Filtration of aquarium water –independent of the size of the system- consists of three components:

- **Mechanical filtration** is the simple entrapment of solid wastes, which results in the clarification of water. The filter matrix can be sand, pea gravel, synthetic beads, foam (sponge-like) or floss. The slides illustrate the many filter types available including a. undergravel plates, a simple box filter, a foam filter, an electric powered filter and a canister filter shown supplying water to a planted aquarium. With time any mechanical filtration system will be converted to a biological filter.
- **Biological Filtration** is the oxidation of ammonia to nitrite and to nitrates by Nitrosomonas sp. and Nitrobacter sp. respectively. These bacteria are aerobic and autotrophic – which means that they require ammonia and nitrite for their multiplication. The mineralization of feces, and food is effected by a host of invertebrates which are found in an active filter system

  Filter facts:

  1. Clean the filters, the bacteria are "sessile" –they are firmly attached to the substrate by a mucopolysaccharide. Never use cleaning fluids except water. Bacteria attached to the filter substrate will remain viable for many months if kept wet.
  2. Dead areas in a filter bed become anaerobic leading to the production of hydrogen sulfide (H_2S), which is very toxic. Deep cleaning of undergravel filters is easily done using distended –end siphon tubes. There may be an inconsequential loss of bacteria during cleaning.

- **Chemical Filtration:** Consists of the addition of a specific substance to a filtration system to get a desired effect:
  1. **Activated carbon:** Because of its high adsorptive capacity a good grade of activated carbon will adsorb a variety of organic compounds including color producing substances, and a wide variety of therapeutic chemicals including antibiotics. Activated carbon must be replaced periodically. It has been used to terminate a treatment including antibiotics, and paraciticides. Filters containing activated carbon should be disconnected during a treatment regime.
  2. **Zeolites:** A variety of clays will adsorb ammonia. When used in an aquarium these clays become saturated with ammonia and provide an excellent substrate for nitrifying bacteria.. The clays can be regenerated with a 3% salt solution. In the author's opinion, the best use of clays in a freshwater system is to provide a buffering action in soft water areas. Brand names products like "Ammosorb" are available. Kitty Litter is also an ammonia adsorber.
  3. **Ultraviolet lights:** Must be "in line" with the filter system. Used in large facilities such as hatcheries or pet stores. With enough wattage an ultraviolet light system will kill viruses, bacteria and many protozoans. UV systems are used to control algae growth in Koi ponds.

4. **Protein skimmers** (Foam fractionators): The foaming of a fluid results in bubbles, which disrupt the chemical bonds of protein molecules. If this process is confined to a small area, the proteins are deposited on the sides of a bubble chamber as denatured proteins. Protein skimmers are used primarily in salt water systems.

Chemical Changes in a newly established aquarium:

The **Conditioning Period: (Run-in period)**

When a freshwater or saltwater aquarium is initially set up with new filters and gravel the addition of fish initiates dramatic changes in the water quality. New filters and gravel lack the nitifying bacteria, which oxidize ammonia to nitrites and nitrites to nitrates. In a microbiologically naïve system ammonia will initially appear followed by an elevation in nitrite and nitrate levels. Deaths of fish will be seen during the ammonia and nitrite peaks. Fish will appear lethargic and on postmortem examination the blood will be brown in color indicating a methemoglobinemia. This condition with a positive nitrite test is ample evidence that nitrites are responsible for the deaths. Some fish such as the *Serpae tetras* are not affected by nitrites possibly because of a high level of liver methemoglobin reductase.

How to Avoid/Reduce the Length and Intensity of the Conditioning Period:

1. Add a few fish at intervals
   - Use hardy fish: goldfish, *Serpae tetras*
   - Test waters frequently and change as required. If a nitrite level is 2 ppm, a 50 % water change will only reduce nitrites by 50 %. Complete water changes may have to be made. Refill with tap water with the temperature adjusted within 3 degress of the aquarium water. Dechlorinate immediately after filling.
2. Conditioning without use of fish
   - Seed Aquarium with conditioned gravel and/or water or use a conditioned foam filter. Rinse the gravel to be added with water. This will tend to remove parasites or their intermediates..
   - Add 20 mg/liter (pinch) of ammonium chloride; urine can be used (teaspoon?)
   - Test water periodically and add fish when ammonia and nitrite levels disappear
   - Continue testing water –keep records

3. What about:
   - Ammonia absorbers: These are complex clays which will absorb ammonia from fresh water systems. They will reduce measurable ammonia but nitrites still appear. They are not effective in salt-water aquaria. In fact, a salt water bath is used to remove absorbed ammonia from absorbers used in freshwater systems
   - Bacterial cultures: As discussed above, transfer of gravel from a conditioned aquarium to a newly established aquarium is a standard

practice. Use as much gravel as practical but rinse with water to remove parasites or their eggs. A conditioned foam filter is an excellent and easy way to seed a new aquarium with bacteria.

- Other: Purified bacterial cultures and "enzymes" have been sold for conditioning aquaria. Their efficacy has not been proven

The Established Aquarium

The Established Aquarium

Characteristics:

- Absence of ammonia, and nitrites
- Nitrate levels increase
- pH levels may fall in water with low alkalinity (bicarbonates)
  > pH under ~ 5.0 will result in inhibition of nitrification
- Organic substances will increase
  > Aquarium water may develop a yellow hue which can be removed by activated carbon.
  > Algae / diatoms will accumulate
  > Immunological suppressors may increase

Avoid problems by good husbandry:

- Test water regularly
- Change water and clean filters and gravel beds at regular intervals. Amount: 25% to 90%
- Buffer water (adjust pH) if required
- Replace activated carbon
- Clean algae from glass
- Feed high quality food.

Water Testing:

Who offers these services:

- Pet stores generally will offer a free testing service for customers. The tests that are offered usually include pH, ammonia, and nitrites.
- Agricultural Extension Departments may offer testing services.
- Selected veterinary clinics (Tell them to bring their dogs in for a checkup)

Types of tests:
The types of testing equipment can vary from the very expensive such as specific ion probes or individual colorimetric tests where a color is developed and compared with a color scale. Freshwater aquarists generally require kits testing for ammonia, nitrites and pH levels. Other kits, which may be required, are available from pet store outlets. Electronic testing devices can be purchased from:

LaMotte Company,
P.O. Box 329
802 Washington Ave.
Chestertown, Md. 21620
Phone: 800- 344-3100

Specific Water Quality Problems:

Chlorine:

Municipal water supplies are chlorinated. The amount of chlorine added will vary depending on the season. Additional chlorine may be added after a rain where bacteria may have been translocated from an agricultural area to an adjacent river that supplies the local city water.

Chlorine can be removed by:

- Addition of sodium thiosulfate: Best method
  Available is pet stores .
  Make a 13% solution of sodium thiosulfate; add 2-3 drops per gallon of water
  Photographic "Hypo" used as a fixative in photography is 5 % sodium thiosulfate
- Letting water stand –diffusion. Takes too long
- Aerating water aggressively
- Passing chlorinated water through activated carbon

Is dechlorination always required after a water change?

No. Chlorine will react with a wide variety of organic substances including organic material in aquarium water. The presence of chlorine cannot be detected after fresh chlorinated tap water is added to an aquarium. Obviously, the amount of water changed is critical. Any water changes over 25% should include dechlorination after the fresh water is added. Sodium thiosulfate should be added into the aquarium immediately after a water change.

Professionals should always recommend the addition of sodium thiosulfate to inactive chlorine

Chloramines

In some areas of the country water sources contain significant levels of humic acids. When chlorine reacts with these organics trihalomethanes are formed which at 4 ppb (parts per billion) may be carcinogenic. To circumvent this problem, certain waterworks in the USA combine ammonia and chlorine to form **chloramines .**

Chloramines do not form trihalomethanes but are antibacterial and toxic to fish.

The addition of sodium thiosulfate (see above) will inactivate the chlorine leaving an ammonia ion.

Removal of Chloramines

- Add sodium thiosulfate (see above).
- What about the residual ammonia?

  * Rapidly oxidized in a well-conditioned aquarium

  * pH control: If pH is low predominant ions will be NH_4 (non-toxic ammonium) rather than NH_3 (toxic ammonia). Lowering of pH will solve the problem
  * In new setups (no nitrifying bacteria) consider use of an ammonia adsorbant

Are chloramines really a problem? Many municipal water systems have been using chloramines for years during periods of rain. The public was unaware of this. Reports from fish hobbyists suggested that no problem was noted. Routine use of sodium thiosulfate apparently was the solution.

Hardness and Alkalinity:

Hardness refers to the amount of minerals in water. Depending on the soil type that rain water percolates through, the water could contain more or less minerals. **Cations** (positively charged ions) such as calcium and magnesium are prominent in water however other elements can be present depending on the area. These include Iron, Zinc, Boron, Silicon and others. Test kits express hardness as ppm of calcium carbonate.

Anions are negatively charged and include bicarbonates, carbonates and hydroxides. The term "total alkalinity" measures the amount of these ions. Test kits measure total alkalinity as ppm equivalents of carbonates. Results of a total alkalinity are reported as ppm calcium carbonate.

Hardness Scale for

Description	Parts per million ($CaC0_3$)
Soft	0-75 ppm
Mod. hard	75-150 ppm
Hard	150-300 ppm
Very hard	300 + ppm

Significance of hardness, alkalinity levels and pH of water.

1. **Hard water** implies a high alkalinity and good buffering capacity. In hard water areas aquarium water is well buffered and pH levels will be stabilized. The pH in some hard water areas of the county may be at 7.8 or higher. The biological significance of a very high pH is that any ammonia will be in the NH_3 form which is toxic
2. **Soft water** implies a low alkalinity and poor buffering capacity. In an aquarium system where hydrogen ions are being generated by nitrification, the pH will naturally be lowered and may drop two pH units (logs). A drop from pH 7 to pH 5 represents a 100-fold increase in hydrogen ion concentration. Nitrification will be affected at that level and ammonia will appear. Fish will be stressed.

Acceptable pH limits:

Freshwater fish: 6.5 to 7.8 --- 7.0 is optimal
Saltwater fish: 7.8 to 8.3

How to increase pH in soft water systems:

1. Add Calcium carbonate: Dolomitic limestone or oyster shell can be placed in a filter canister. Use of a filter bag is suggested since the material may have to be eventually replaced. Oyster shell is inexpensive and available at feed stores. Dolomitic limestone or "crushed coral" – used as substrate in marine systems -- is available at pet outlets. Monitor pH with a test kit!
2. Pre set powdered or liquid buffers are available at pet outlets.

How to adjust pH of Hard Water:

In certain parts of the country water may be very hard with a pH over 7.5. Lowering pH in hard water is difficult using pH adjusters. The hardness and alkalinity of hard water must first be reduced before pH adjusters will be effective.

- Distilled water: Distillation of water removes both cations and anions from water. Deionization of water also removes cations and ions Cannisters with mixed resins are available.
- In recent years a process referred to as **Reverse Osmosis** has been developed: The process involves passing water through a semi permeable membrane which traps the ions. Water deionized by reverse osmosis (RO water) is routinely sold at upscale pet stores. Regardless of the method of deionization one must determine the proportion of tap water and deionized water to get the desired pH. Consider using 4 parts of deionized water to 1 part of tap water. Use small test proportions to determine hardness and alkalinity levels with hardness and alkalinity test kits. Adjust proportions as necessary. Fortunately, salt mixtures are sold for reconstituting deionized water.

- Home water softeners: Home softening units remove calcium and magnesium ions. Alkalinity, consisting of carbonates, silicates, etc remains unchanged! Since the anions are responsible to high pH of water, the high pH problems will persist However, the water is great for laundry uses.!

Ammonia problems and solutions:

Toxicity of ammonia in water shifts with pH changes:

Ammonia (NH_3) ion is toxic to fish and is prominent in higher pH water. It the pH of the water is decreased towards neutrality (ph 7) or lower, the non-toxic ammonium ion (NH_4) predominates. This shift can be used to avoid toxicity. While the physiologic effects of ammonia are not fully understood, **acute** ammonia toxicity, by its effect on gill cells, results in disturbances of respiration and osmoregulation. Low levels of ammonia (**chronic toxicity**) produce gill cell hypertrophy, leading to hyperplasia and eventually necrosis. The end physiological effect is a loss of gas exchange, excretion, and osmoregulation. Fish do not eat well and appear lethargic.

Solving Ammonia problems :

- Test water for ammonia and pH
- Determine cause:
  1. Is filter system biologically active? (Conditioned)
  2. Overfeeding results in elevated ammonia levels.
  3. Too many fish for the filtration system
  4. Chemical inactivation effect on filters such as methylene blue qnd Erythromycin
- Take appropriate corrective action:
  1. Change water immediately
  2. Remove some fish if appropriate, reduce amount of feed if necessary
  3. Reduce pH
  4. Ammonia adsorbers
  5. Augment biological filtration by addition of conditioned filters of any kind. Easily moved foam filters are especially useful for this purpose.

DEPENDING ON THE SITUATION USE ANY OR ALL OF THE ABOVE

Nitrite Problems

- Nitrites are a product of nitrification. In a microbiologically conditioned filtration system, nitrites are rapidly oxidized to nitrates. Nitrites at taken in by gill cells and combine with iron in the cytochrome oxidation system to form methemoglobin which cannot carry oxygen. Fish die of anoxia.
- The presence of nitrites in an aquarium should be expected during the conditioning period. A diagnosis of nitrite toxicity can be confirmed by water testing,
- Susceptible fish are lethargic, show no interest in food and may have flared gills. Dark blood noted during a necropsy procedure indicates a methhemoglobinemia.

- In a mixed population of fish, the owner will report that some fish are living while others have died. Many fish are not susceptible to the effects of nitrites. Resistant fish include goldfish, and Serpae tetras among others. Many species of catfish appear to be susceptible.
- Low levels of nitrite may be tolerated. The effects of low levels are not well documented.
- **Solutions:**
  1. Change water –but expect nitrites to be produced until enough nitrifying bacteria (Nitrobacteria) have been established in the filter bed. Water may have to be changed daily. Fish could be transferred to a "conditioned" aquarium until the primary aquarium is cycled.
  2. The addition of salt to water has been shown to block the uptake of nitrites in some fish. Salt has been shown to block the uptake of nitrites in highly susceptible channel catfish at 20 ppm. In practical terms, approximately .75 grams (one eight of a teaspoon) per 10 gallons of water is an acceptable dosage. Iodized salt will not cause toxicity problems. Salt, along with a sodium thiosulfate, should be added after a near complete change of water. **Note**: Aquarists for 60 years have lauded the effects of salt on the general health of fish. Salt can have both a nitrite sparing effect as well as inhibiting some parasites

Nitrates

Nitrates are the end product of nitrification. In nature nitrates are utilized by plants as part of the nitrogen cycle. In aquaria nitrates stimulate plant and algae growth and they are not toxic for fish. Aquarists are aware that aquatic plants will flourish in a well lighted aquarium which has been conditioned.

The term **"balanced aquarium"** has been used to describe an aquarium system where nitrates and carbon dioxide (from fish) provide nutrients for plants and, in turn, the plants provide oxygen for the fish. Such natural balanced aquaria were not equipped with power filters. Good lighting is essential. Generally, the amount of fish in a naturally balanced aquarium is minimized.

In filtered aquaria the periodic removal of nitrates is beneficial for controlling algae.

Hydrogen Sulfide (H_2S)

Hydrogen sulfide is a gas that has the odor of rotten eggs. It is very toxic; it arrests cellular respiration by bonding to iron in mitochrondrial cytochromes, thereby arresting aerobic metabolism leading to suffocation and sudden death.

Hydrogen sulfide is a product of anaerobic decomposition and collects as a gas in anaerobic pockets in aquarium gravel or pond muck. If such anaerobic pockets in aquarium or fishponds are disturbed, the gas is released and within a very short time (minutes) fish begin to "roll over" and die.

Avoidance and Treatment:

Filters should be kept clean – detritus from gravel beds should be removed regularly by agitating the gravel bed. Since anaerobic pockets are found deep in a gravel bed, a distended end siphon tube is best used for this purpose since gravel will not be sucked into the siphon tube. Such systems are available at pet store

outlets. Filters in Koi ponds should be cleaned regularly as should the detritus from the bottom of the pond. Filters of Koi pons should be run continuously. Turing off a filter of a Koi pond at night to save on electricity should not be done. Hydrogen sulfide can form rapidly and will be released the filter are activated in the morning. Note smell of rotten eggs!
Fish can be saved if the problem is **recognized immediately.** Fish should be removed to fresh water immediately. They will not eat for several days and show little or no movement during their recovery. Since the gills have been damaged, aeration should be maximized during convalescence.

Carbon Dioxide

Carbon dioxide is an end product of nitrification, respiration of the fish and presents no problem in well-aerated aquaria.
Carbon dioxide is a problem in the shipment of fish in oxygenated plastic bags. An interesting phenomenon is seen when fish shipped from long distances arrive "dead". When the fish are transferred to well aerated water, they will regain consciousness from the apparent carbon dioxide narcosis.
Fish farmers are aware of the optimal packing densities for shipping various varieties of fish.

Use of carbon dioxide gas for plant growth : Plants require carbon dioxide for growth. Diffusion units are available from retail outlets. Home made carbon dioxide units have been popularized by Mark Ching. He suggests using a yeast culture to generate carbon dioxide gas. See Freshwater and Marine Aquarium August 2002, pp182-186, or refer to http://thekrib.com for do-it-yourself systems.

Contact Mr. Ching at mark12085@aol.com.

Oxygen:

- Well water is usually low in oxygen: If well water is used for aquaria, increase oxygen by surface agitation of water
- Decreases of oxygen:
  1. Small Koi ponds without agitation and insufficient cleaning.
  2. Excessive loading of an aquatic system
     Aquarists have suggested "One inch of fish per gallon of water". This is very conservative. **I have 15 Koi in an 800-gallon pond.** (Better to be conservative and not push the envelope)
- Addition of formaldehyde(a paraciticide) will decrease oxygen. Agitate water if formaldehyde is used.
- Elevated water temperatures will decrease the oxygen in water. Agitation is important.

Water Temperature :

- Fish species will tolerate variations in temperature in nature. Extreme variations are an exception.
  1. Goldfish and Koi will live in near freezing water but will not eat and should not be fed at temperatures below 55 F. Consider 75 F to 80 F as an acceptable range for most tropical fish.
- Sub optimal temperatures will affect:
  1. Appetite, metabolism and growth
  2. Activity of fish
  3. Breeding behavior
  4. Immune response and healing

- Seasonal water changes in ponds:
  Not infrequently Koi will develop ulcers with the onset of warmer water temperatures. It is thought that such fish are carriers of an organism during the winter months (possibly a virus and/or a bacterium (Aeromonas sp.). With the change of seasons, the pathogens respond relative quickly to an elevated environmental temperature while the immunological system of the fish responds much slower. This lagging effect provides a window of susceptibility for Koi during the spring turnover. The solution is to keep the water heated throughout the winter season. A thorough cleaning of pond in the fall of the year is an additional precaution to avoid spring diseases in fish.

Plants and water quality:

- Live plants in an aquarium utilize nitrates, provide shade, escape areas as well as the "natural look" for a beautiful aquarium
- Conditions for optimal growth:
  1. Avoid deep aquariums to optimize lighting
  2. Aquarium should be conditioned
  3. Water pH 6.5 –7.5
  4. Light: At least 2 watts of broad spectrum per gallon of water
  5. Light period 12 hours is suggested. Plants will vary in their tolerance
  6. Healthy plants: See the following sites for pictures and tips on care. **tropica aquarium plants.com** and **wet plants.com**, These sites provide tips on grooming and planting
  7. Gravel depth should be 4-5 inches deep.
  8. Fertilizer: Water in a conditioned aquarium provides nitrates. Addition of chelated iron and supplemental carbon dioxide is suggested
  9. Filter: Surface water movement should be minimized to prevent the escape of available carbon dioxide.
  10. Snails: Remove snail "pods" from leaves of plant prior to placement in the aquarium
      Place plant in alum bath/ one tablespoon per gallon.
      If snails are present in the aquarium, small Koi will find them tasty and eliminate the problem.

MARINE AQUARIUM SYSTEMS: ENVIRONMENTAL FACTORS AND COMPARISON OF FRESHWATER AND MARINE AQUARIA

George Blasiola, Aquatic Biologist B.S, M.S

Introduction: Marine fish and other aquatic organisms are greatly influenced by the chemical, biological and physical changes in the aquatic medium in which they inhabit. Aquatic environments depending on whether they are freshwater or marine are subject to various environmental fluctuations. In comparison to many freshwater habitats, marine systems such as coral reefs tend to be remarkable stable with very little fluctuation.

In captive situations, marine systems are subject to environmental deterioration if specific parameters are not monitored. As with freshwater systems, ammonia, nitrite, nitrate, pH and temperature need to be monitored. However, in marine systems it is important to measure other parameters such salinity and calcium. Maintaining a marine system requires an understanding of some of the differences between freshwater and marine systems as it relates to water quality and other environmental factors.

Lecture Outline:

Environmental Factors Affecting Marine Systems: Biological, Chemical, Physical

- Biological
- Chemical
- Physical

Biological Factors

- Space Requirements
- Population Density
- Pathogens

Chemical Factors

- Water Chemistry
- Pollutants
- Diet
- Metabolic Wastes
- Chemical Treatments

Physical Factors

- Temperature
- Sound
- Dissolved Gases
- Light
- Handling and Transport

Characteristics of Traditional Marine Aquariums

- Primary Filter is Undergravel
- Fish are the predominate fauna
- Absence of Macroalgae
- Use of decorative coral skeletons
- Basic Water Testing ammonia, nitrite, nitrate, pH, salinity
- Primary purpose of light is aesthetics

Characteristics of Reef Type Aquariums

- Use of trickle filters, live rock, algal turf filtration
- Invertebrates predominate
- Macroalgae often present
- High population of live corals
- Reliance on basic water testing and specialized monitoring of calcium, phosphate, strontium and others.
- Lighting source critical in health of corals and macroalgae.

Comparison of Marine with Freshwater Aquaria

- Tank Size
- Filtration
- Substrate
- Filter Media
- Lighting
- Aeration
- Specific Gravity
- Ammonia
- Ammonium
- Nitrate
- Run In Period
- Water Changes and Amounts
- Substrate Cleaning
- Fish Density

© 2005 All rights reserved .George Blasiola

PROPER SETUP AND MAINTENANCE OF MARINE REEF AQUARIUMS

Daniel W Lehmann, BS

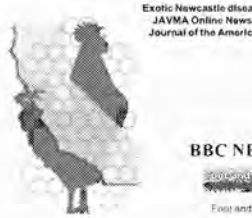
A. Parameters of Concern
    a. Nitrogen
    b. Phosphate
    c. Salinity
    d. Alkalinity
    e. pH
    f. Temperature
B. Additives
    a. Calcium & Alkalinity
        i. 2 parts
        ii. reactors
    b. Other
        i. Strontium
        ii. Iodine
    c. Food
C. Basic Components of a Successful System
    a. Lighting
        i. Metal halide
        ii. Fluorescent
            1. t5
            2. VHO
            3. PC
    b. Sumps and Pumps
    c. Circulation
        i. Requirements
        ii. Devices
    d. Filtration
    e. Live Rock/Live Sand
D. Advanced Components
E. Cycling
F. Medications and Treatments
    a. Marine Ich
    b. Red Bugs
    c. Isopods
    d. Brown Slime/coral diseases
G. References

Biosecurity to Prevent the Introduction of Diseases into aquaculture farms

Jay F. Levine DVM MPH
Aquatic Animal Epidemiology
And Conservation Genomics Laboratory

Overview

- Introduction to biosecurity
- Biosecurity considerations
- Biosecurity in practice
- NSCU aquatic pathogen facility



Exotic Newcastle disease strikes game birds in California
JAVMA Online News
Journal of the American Veterinary Medical Association

BBC NEWS

Foot and mouth Thursday, 23 January, 2003, 11:07 GMT

Sheep Foot and mouth rules set to be eased

The Straits Times INTERACTIVE

MAY 8, 2004 SAT

US to check its role after China SARS outbreak

August 1, 2003
Maine officials battle new outbreak of ISA

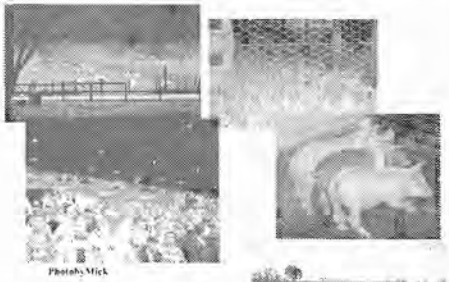
Infectious Salmon Anemia, Maine,
USA March 23, 2001:
The first identification of infectious salmon anemia (ISA) in the US has been made in an aquaculture pen near Treat Island in Cobscook Bay, Maine.

Spring viremia of carp control program launched

May 15, 2003

NEWS RELEASE Wisconsin Department of Natural Resources
To be released in the Sept. 10, 2002 DNR News CONTACT:
Sue Marcquenski, Marty Engel,Dr. Myron Kebus, DATCP

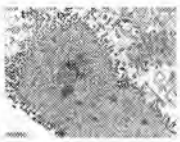
SUBJECT: Exotic fish virus suspected of killing carp
MADISON , Wis. – An exotic fish virus, spring viremia of carp, is suspected of killing more than 10 tons of carp last spring and the previous fall in Cedar Lake in St. Croix and Polk counties in northwestern Wisconsin.



High Density
Large susceptible populations

The list of potential pathogens is lengthy

- Bacterial, parasitic, viral, fungal
- Endemic:
  - koi herpes virus
  - spring viremia of carp virus?
  - other fish rhabdoviruses
- OIE listed Pathogens
  - Epizootic hematopoietic necrosis
  - Infectious hematopoietic necrosis
  - *Oncorhynchus masou* virus disease
  - Viral hemorrhagic septicemia
  - Spring viremia of carp virus



- Bivalves

Infection with:

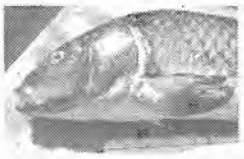
- *Marteilia refringens*
- *Marteilia sydneyi*

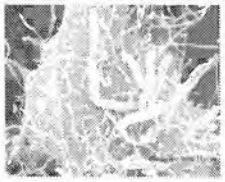
- *Mikrocytos mackini*
- *Bonamia ostreae*
- *Bonamia exitiosus*

Crustaceans

- **Taura syndrome**
- **White spot disease**
- **Yellowhead disease**
- **Tetrahedral baculovirosis**
- **Spherical baculovirosis**
- **Infectious haematopoietic necrosis**
- **Crayfish plague (*Aphanomyces astaci*)**
- **Spawner-Virus disease**

One bug, One disease

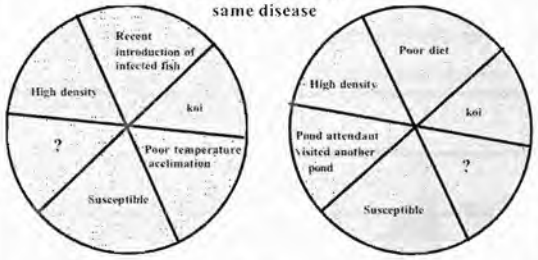




Aeromonas hydrophilia

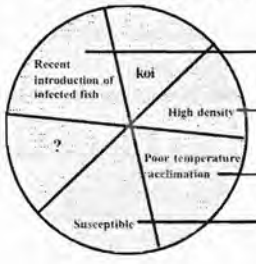
An Epidemiologic Approach:

Different pathways
same disease



Agents Hosts Environment

A biologic risk based approach
If you know the risk factors you can prevent introduction without knowing the pathogen



Active surveillance

Optimize density

Careful temperature acclimation

Vaccination

How do we effectively minimize disease-related problems?

Optimize the rearing/holding environment

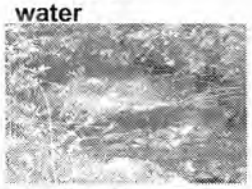
Conditions suitable for species

- Coldwater species vs. Warm water species
- Just survival vs. preferred conditions

Careful Site Preparation

- Pond, pen, or tank location:
  - access, utilities, wildlife, shading, security
- Pond, pen, or tank characteristics that meet species needs

Source of water



Environmental parameters

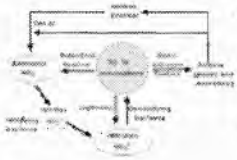
- Dissolved oxygen
- Temperature
- Hardness
- Alkalinity
- pH
- Turbidity

A few basics: understanding of nitrogen cycle

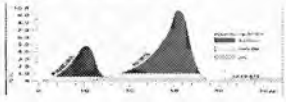
Ammonia, nitrite, nitrate

Ammonia and nitrosomonas

Nitrite [NO2] and nitrobacter



Atmospheric deposition



Movement of fish

- Time
- Temperature
- Dissolved oxygen

Diet

- Appropriate for species
- Quantity
- Quality
- Delivery: floating etc.
- Frequency
- Time of day
- Temperature
- Overfeeding
  - on vacation

Filtration

- Open system vs. Closed system
- Particulate removal
- Nitrogenous wastes
- Clarity

Density

- Appropriate number of fish for volume of water
- Fish size, environmental conditions, species behavior
- Account for fish growth

Taken together

Ist step

Reducing Risk By Implementing Best Management Practices

- **Commercial aquaculture**
  - growth/productivity/profitability
  - product safety and quality
  - environmental practices
  - sustain health
- **Ornamental ponds**
  - growth
  - sustain health
  - sustain investment

When your making money who cares?

- Catfish industry
- Bait fish industry
- Shrimp industry

Most aquaculture industries have ignored biosecurity

- High density = disease problems
  - white spot
- Farms abandoned
- Environmental problems
  - Salt water incursion
  - Damage to mangroves
  - Nutrient loading
  - Lose of subsistence rice farming

Risk-based biosecurity

Don't have to know the disease to prevent the problem

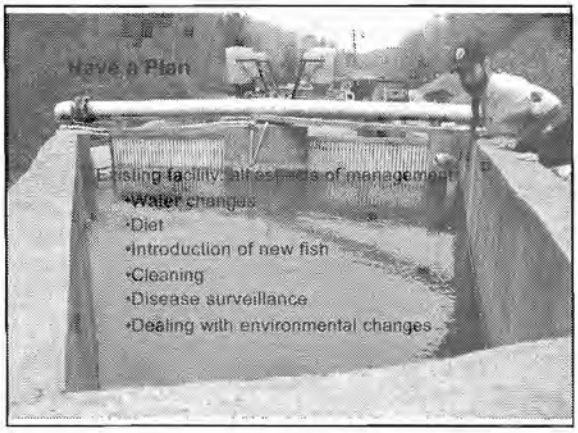
- Introduction of a pathogen
  - Purchase of infected fish
  - Immediate introduction into established pond
  - No acclimation, and a large temperature change at introduction
- Eliminate Risk factors
  - Careful selection and history of fish before purchase
  - Quarantine of new arrivals
  - Careful acclimation

Where to begin to improve biosecurity

- Risk assessment - Step by step review
- Critical control points

Whether you have a large farm or a single small pond : If its worth doing: Have a Plan

- New pond
  - Work through construction step by step
  - List the steps for fish introduction, and maintenance

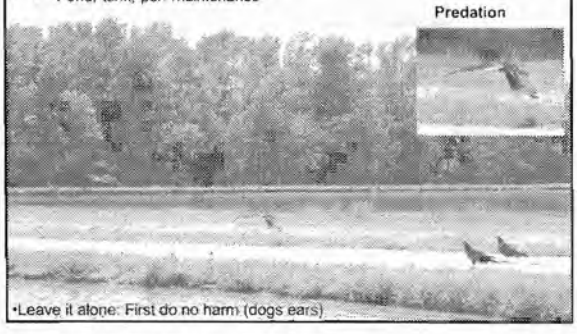


What are the two most important things affecting the success of a biosecurity plan?

A plan is of no value if you don't use it

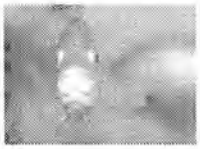
Farm Maintenance Issues

- Stock processing : All in All out vs. Continual restocking
- Handling of stock
- Pond, tank, pen maintenance



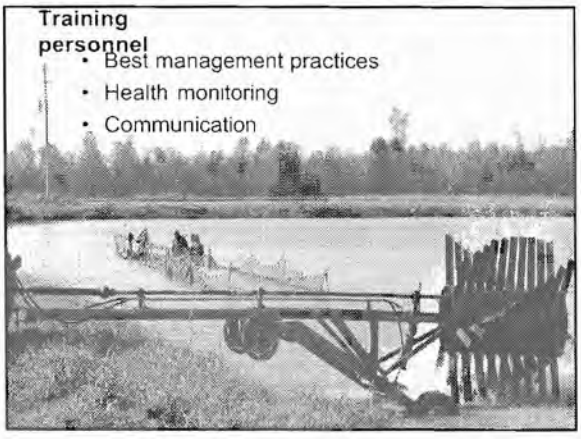
Quarantine

- Fish Abstinence: Don't introduce new fish from unknown or questionable sources
- But if you must: Have a quarantine tank, or pond
- Get a fish health examination from your veterinarian



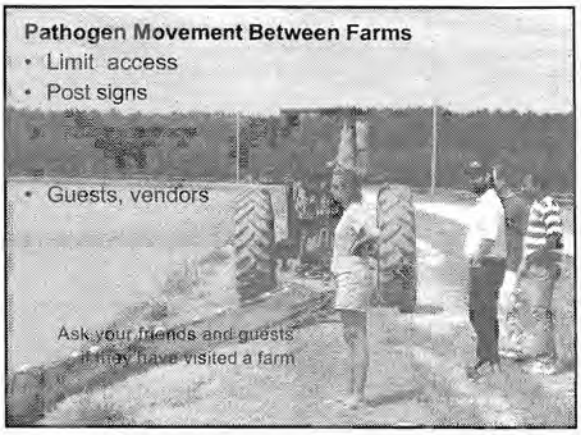
Training personnel

- Best management practices
- Health monitoring
- Communication



Pathogen Movement Between Farms

- Limit access
- Post signs
- Guests, vendors



Surface water sources

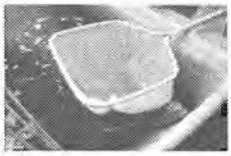
Water treatment

Cleaning and Disinfection

- Cleaning
- Proper storage
- Proper application procedures
  - Dilution
  - pH
  - Quartenary ammonia - cotton mops
- Spectrum: specific disinfectants for
  - specific agents
  - Bleach (viruses)

Equipment

- Dipping nets in disinfectant and rinsing before use
- Surface disinfection of equipment



Foot baths: Do they work?

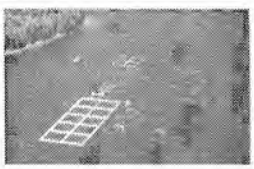
Foot-covers: slick surfaces

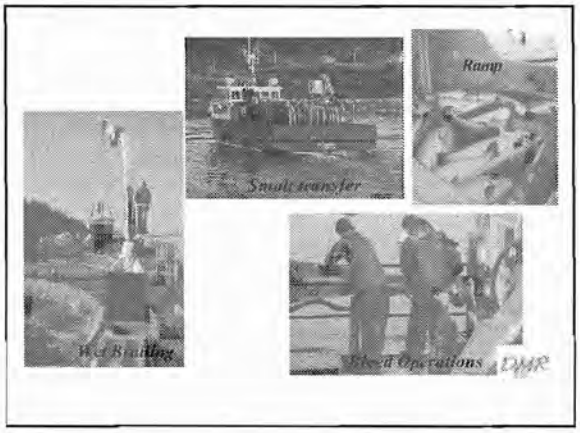
- Gira et al. J Clin Microbiol. 2004 42(4):1813-7. Furunculosis due to *Mycobacterium mageritense* associated with footbaths at a nail salon.
- Langsrud et al. J Appl Microbiol. 2003 95(1):186-95 Characterization of *Serratia marcescens* surviving in disinfecting footbaths.

Between farm, pond, and net

- In addition to the fish : nets, tanks, bags, vendors, veterinarians

Salmon industry
- Whale boats and salmon
- Biosecurity during transport





Active disease surveillance

- Observation
  - Food consumption
  - Other behavior
- Physical examination
- Physical signs
  - Lesions
  - Changes in coloration
- Veterinary examination
  - Complete blood count
  - Chemistry panel
  - Serology, PCR

Improving herd immunity: Vaccination

Few vaccines for fish viruses: none commercially available in the US

Antimicrobials

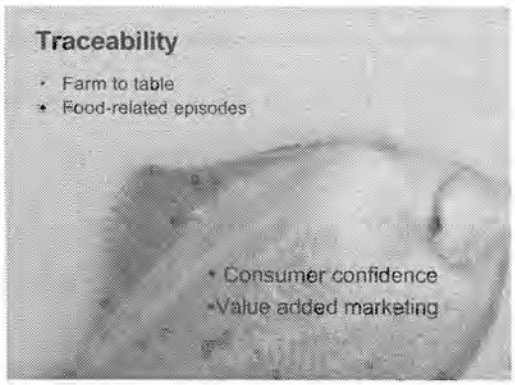
- Limited availability
  - oxytetracylcine (Terramycin)
    - withdrawal times
      - catfish 21 days
      - salmonids 21 days
  - sulfadimethoxine (Romet)
    - withdrawel times
      - catfish 3 days
      - salmonids 42 days
- Do they work?
  - anti-microbial resistance
- Industry image
  - Consumer confidence
  - e.g. salmon industry

Food security = food defense

- Product monitoring from farm to retail
  - Maintaining cold chain
  - Maintaining security

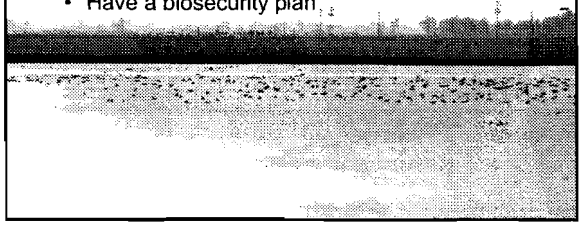
Traceability

- Farm to table
- Food-related episodes



Summary

- Epidemiologic view of disease
- You don't need a specific diagnosis to prevent disease problems
- Biosecurity practices can prevent pathogen introduction
- Have a biosecurity plan



AQUARIUM LIFE SUPPORT SYSTEMS

Stuart E. May
Aquarist NAIB (Construction & Opening)
Husbandry Curator
NC Aquarium at Pine Knoll Shores

LSS Simplified

- Most large Public Aquariums operate on a closed system.
- Water is circulated thru filters and tanks of animals.
- The water is cleaned and reused.
- Occasionally a portion of the re-circulated water is dumped and replaced

Typical Holding System LSS

- Will operate systems from a few hundred gallons to thousands of gallons
- Will operate salt and fresh water.(Plus or Minus Components)

Pump

- Several different types centrifugal pumps can be used for aquarium operation. (Submersible, Non-submersible, Magnetic and Shaft Drive)
- Impeller uses spinning force to pressurize water as it moves thru pump.

Surface Skimmers

- Remove water from tanks by gravity (Standpipe)
- Avoids suction capture of animals on pump intakes
- Maintains constant water level
- Prevents water loss during pump or power failure.

Mechanical Filters

- Collect and concentrate particulates
- Usually measured micron sizes.
- Usually need to be stopped and cleaned
- Occasionally require major overhaul.

Biological Filters

- Enhance growth of beneficial bacteria by increasing surface area and gas exchange.
- Can be found in some fashion on all wetted surfaces
- Can operate as trickle or fully submerged.

Protein Skimmers

- Usually designed for salt water systems.
- Sometimes called Foam Fractionators
- Removes waste before acted on by Bio-filter
- Basic styles include: Downdraft, Venturi, and Air-driven
- Combination chemical & mechanical filter
- Uses surface tension of micro-bubbles to filter systems.

Chemical Filters

- Usually built in side stream or tangent off main filtration path.
- Can be operated as a trickle filter or fluidized
- Adsorbs DOC, some metals, and other chemicals
- Activated carbon can be recharged but is usually thrown away

LSS at its Best

- Delivers clear clean water to animals
- Water maintains high concentration of animals (Biomass)

DIAGNOSTIC TECHNIQUES, ANESTHESIA AND SURGERY

Craig Harms, DVM, PhD, DACZM
North Carolina State University
College of Veterinary Medicine
Department of Clinical Sciences
Center for Marine Sciences and Technology
303 College Circle
Morehead City, NC 28557

(excerpted from Harms CA. Fish. In: Fowler ME & Miller RE (eds). Zoo and Wild Animal Medicine V. Philadelphia, Saunders, 2003)

Many cases may be resolved on the basis of the history, water quality testing, physical exam, and a few simple biopsy procedures. Fish may be seen on a field service or in clinic basis. If seen in clinic, most cases can be handled on an out-patient basis, eliminating the need for fish holding facilities. Determine early on if euthanasia of some fish for diagnostic purposes is an option or not. For example, a tropical fish breeder who has lost 25 of 200 fish and needs to sell 100 to break even for that batch may be willing to sacrifice 5 - 10 fish to reach a diagnosis, while the owner of a prize koi wants you to save that fish. If fish are submitted to the clinic, request that the owner bring affected fish, and a separate water sample for water quality testing. Fish should ideally be transported in sufficient water (about 1 L/cm fish) with an equal volume of air or oxygen (or use portable aerators for long transport) in an aquarium bag, packed in a styrofoam cooler to moderate temperature fluctuations. Be prepared to supply aeration immediately upon arrival, as many fish will not be transported optimally.

Points to determine in the history include experience of the client, how long the aquarium has been set up, what other fish are present and if any others are affected, how long the fish has been present in the aquarium, recent additions and losses, use of quarantine, frequency of feeding and content of diet, prior treatments, type of filtration, size of aquarium, aquarium maintenance, a description of the problem, and results of any water quality monitoring performed by the owner.

Water quality testing should be performed as soon as possible on submitted samples, as some parameters are very labile. Temperature and dissolved oxygen are generally only reliably measured on site. Essential measurements include ammonia, nitrite and pH. Depending on the system, hardness or alkalinity (e.g., African cichlids) and salinity (e.g., marine tank) may also be considered essential. Other parameters which may be useful in some cases include chloride, chlorine, copper, and nitrates. Timing of sampling is important for parameters which fluctuate, and special sampling and storage procedures are required for some substances.

In the physical exam, note behavioral abnormalities (e.g., increased respiration rate, piping, flashing, whirling), external lesions (e.g., ulcers, frayed fins, tumors), coloration (e.g., generalized or focal darkening, reddened fins), contour (e.g. abdominal swelling, spinal deformities), and eye lesions.

Wet mounts of a skin scraping, fin clip and gill biopsy are some of the most useful diagnostic procedures available. They are used primarily to diagnose ectoparasite infections, but in some cases are also useful for bacterial infections (particularly columnaris) and environmental gill damage. Motion is critical to the identification of many protozoan parasites, which makes wet mounts superior, in many cases, to stained cytology or histology. Anesthetizing the fish facilitates obtaining biopsy specimens. Prepare slides with a drop of the fish's water prior to initiating biopsies. Skin scrapings should be performed with just enough pressure to obtain a few scales along with mucus and epithelial cells, using either a cover slip or blunted scalpel blade. Fin clips can be obtained with a pair of fine scissors. Gill biopsies are obtained by deflecting the operculum and cutting only the tips of a few lamellae. The samples are placed immediately on the previously prepared slides and cover-slipped for microscopic examination. These procedures are usually extremely safe, although in a severely compromised fish the stress of the procedure plus some blood loss from the gill biopsy could precipitate death of the fish.

Venipuncture in fish less than 7.5 cm is impractical on a survival basis, but fairly simple on larger fish. The caudal vein is the usual target, and can be approached ventrally or laterally. Whole blood or buffy coat wet mounts can be used to diagnose hematozoan parasites (e.g., trypanosomes) or an overwhelming septicemia. Interpretation of fish hematology and serum chemistries is not as advanced as in other areas of veterinary medicine, but are becoming more routine and can yield useful information, especially when serial samples are obtained.

Fecal samples are usually overlooked in fish medicine, but they are possible to obtain. Fresh samples are essential for meaningful interpretation since feces are rapidly colonized by free-living aquatic microbes, and break up quickly. Fish often provide a fecal sample out of water under the influence of anesthetic.

Bacterial cultures may be obtained ante-mortem from skin and gill lesions, although interpretation may be complicated by opportunistic aquatic bacteria. Non-lethal kidney aspirate techniques have been described, but in most cases culture of internal organs is done post-mortem. Kidney, spleen and visible lesions should be cultured from post-mortem cases. Special culture techniques are required for some fish pathogens, some relatively simple (e.g., room temperature or cooler incubation, added salt for some marine pathogens), some more complex.

Radiographic and ultrasonographic imaging are increasingly being used in fish clinical medicine. Fish can be removed from the water with or in some cases without anesthesia for the brief period required for obtaining radiographs. Plain radiography works well for evaluating bone and swim bladder, making it useful for cases of trauma and buoyancy disorders. Because fish normally have little fat in the coelomic cavity and no gas in the gastrointestinal tract, contrast is generally insufficient to evaluate viscera. Positive contrast studies can be used to delineate the gastrointestinal tract. Fish are well suited for ultrasonographic imaging as they may be left submerged with the water acting as a coupling agent. Ultrasonography is useful for imaging soft-tissues, complementing plain radiography. CT and MRI image acquisition times are becoming more rapid, making these studies in fish more feasible.

Full necropsies are sometimes required to reach a diagnosis. Because dead fish autolyze so rapidly, euthanizing moribund fish is most likely to yield useful results. A fish found dead, and dead for an unknown period of time, is rarely worth necropsying (though exceptions occur). The fish may be euthanized with an anesthetic overdose followed by cervical transection. Necropsy procedures are a personal preference, but a systematic approach is important. Skin scrapes, fin and gill clips should be obtained if not already done. Kidney cultures may be obtained via either a dorsal or ventral approach. Most pathologists excise the left lateral body wall to expose the viscera. Tissue sampling for histopathology, virus isolation, toxicology, electron microscopy, etc., is identical to that for other animals. Very small fish are not amenable to routine necropsy procedures, but may be preserved whole (puncture coelomic cavity for good fixative penetration) and examined in sagittal or longitudinal sections on a single slide. Squash preparations of organs are useful for obtaining rapid diagnosis of granulomas, parasitic cysts, and intestinal parasites.

Anesthesia

Anesthesia facilitates examination, transport, and obtaining diagnostic samples; it is required for surgery; and, when necessary, can be overdosed for euthanasia. Physical and chemical parameters of the anesthesia water should closely match those of the aquarium or pond water of the fish to be anesthetized. Because anesthetic agents tend to cause respiratory depression, adequate aeration is mandatory. Since some anesthetic agents markedly affect pH, addition of buffers may be required to avoid inducing acidosis. Water temperature has a direct effect on metabolic rate, and therefore affects the rates of induction and recovery, with higher temperatures speeding both.

Food should be withheld for at least one feeding cycle prior to anesthesia. Although aspiration pneumonia is not a hazard in fish, regurgitation can clog gill rakers and foul the water. For water-borne anesthesia, water and containers for induction, maintenance, and recovery should all be prepared ahead of time.

Anesthetic stages are gauged by activity, reactivity to stimuli, equilibrium, muscle tone, respiratory rate, and heart rate. Broad stages include sedation, narcosis or loss of equilibrium, and anesthesia, and may be further subdivided into light and deep planes.

Many compounds have been employed as fish anesthetics. Only the more commonly used and/or readily available are included here.

Tricaine methanesulfonate (MS-222, Finquel®) is the most widely used fish anesthetic, and is the only one approved in the United States for use in food fish, with a 21 day withdrawal period. Administered as a water-borne solution, tricaine is absorbed across the gill epithelium and is cleared primarily through the gills. Tricaine is conveniently administered as a pre-mixed stock solution of 10 g/L (10,000 ppm or mg/L, or 10 mg/ml). The stock solution is unstable in light and should be kept in a dark container. Tricaine solutions are acidic and should be buffered prior to administration to fish. Saturating with sodium bicarbonate buffers the stock solution between pH 6.0 and 7.5. Although generally considered a safe anesthetic, tricaine margins of safety are narrower for young fish in warm, soft water, and there is variation across species. Recovery from short procedures is rapid (less than 10 minutes if properly dosed), with prolonged recoveries (up to 6 hrs) from longer procedures. Tricaine is dosed at 100 - 200 mg/L for rapid induction, 50 - 150 mg/L for maintenance, and 15 - 50 mg/L for sedation.

Eugenol (clove oil, 4-allyl-2-methoxyphenol) is an immersion anesthetic alternative to tricaine.[23] Clove oil (90 - 95% eugenol) is available over-the-counter from pharmacies, and 100% eugenol is available from chemical supply companies. Though classified "generally regarded as safe" by the FDA as a food additive, it does not have approved status as a fish anesthetic at this time. Eugenol is incompletely soluble in water, particularly at cold temperatures. A 1:10 mixture of eugenol into 95% ethanol yields a 100 mg/ml stock solution. Final concentrations of 40 - 120 mg/L may be used for anesthetizing fish (the contribution of ethanol to the anesthetic effect is nil at these concentrations). Lower doses of eugenol have equivalent effects on induction, recovery and blood gases as higher doses of tricaine. Compared with tricaine, eugenol is less expensive and less bulky as a stock solution, but it has shorter history of use than tricaine. Anecdotal reports indicate that the margin of safety may be lower in some species. In addition to its anesthetic properties, eugenol may have some beneficial antimicrobial effects.

Metomidate is a non-barbiturate imidazole hypnotic that has been used as a water-borne anesthetic agent in fish. It is available under the trade name Marinil® in Canada, but has not received approval in the US. It is readily water soluble and should be stored in tight light-protected containers. Some fish anesthetized with metomidate turn very dark transiently. It is not believed have analgesic properties, and muscle fasciculations occur at low doses. It therefore may not be appropriate as a single agent for surgery, although it is useful for sedation and tranquilization. Gouramis are believed to be very sensitive to metomidate. Dosages are 5 - 10 mg/L (up to 30 mg/L for some species) for anesthesia, 2.5 - 5 mg/L for heavy sedation, 0.5 - 1 mg/L for light sedation, and 0.06 - 0.2 mg/L for transport sedation.

Ketamine is a familiar injectable short-acting dissociative anesthetic. It may be used alone or in combination with an alpha-2 agonist, which has the advantage of partial reversibility. Teleost fish are refractory to the effects of both ketamine and alpha-2 agonists, making these agents more appropriate as an aid to restraint rather than as a substitute for water-borne anesthesia. Elasmobranchs are more susceptible than teleosts. Published dosages for teleosts are 66 - 88 mg/kg IM ketamine alone or 1 - 2 mg/kg ketamine plus 50 - 100 ? g/kg medetomidine IM reversed with 200 ? g/kg atipamezole IM; for elasmobranchs 12 - 20mg/kg ketamine plus 6 mg/kg xylazine IM.

Propofol has been used successfully in bamboo sharks as a 2.5 mg/kg IV bolus administered in the caudal vein. Righting reflex was lost within 5 minutes and returned in 60 - 200 minutes.

Lidocaine has been used as an immersion anesthetic with variable results, but may have application as a local anesthetic in combination with immobilizing agents. Care must be taken not to overdose small patients with local injections (< 1 - 2 mg/kg total dose).

Butorphanol may be used at 0.4 mg/kg IM once for post-operative analgesia. Treated fish tend to be more active, swim higher in the water column and return to feeding more quickly following surgery than untreated fish. Analgesics and analgesia have not been extensively studied in fish.

Anesthesia may be delivered by any of the usual routes of administration: orally (PO), parenterally (IV, IM, intracoelomic) or inhalation (topical to gills, bath, water-borne), although oral administration is rarely practiced due to difficulties in precise

dosing and uncertainties in rate and degree of absorption.

Parenteral administration is usually IM or intracoelomic. The parenteral mode is best suited for larger fish, for which injection site trauma is less of a hazard, and for larger volume aquariums, where adding water-borne anesthetics to the entire tank would be impractical, and confinement and capture of the fish in a smaller volume of water would be problematic. Injections may be made by hand syringe, pole syringe, or dart.

Water-borne anesthesia is the most widely used route of anesthetic administration for fish. For bath treatment the drug can be brought to the desired concentration in water containing the fish, or the fish can be placed in an induction tank containing anesthetic water. For short procedures lasting less than 5 minutes, this may be all that is necessary. The fish may be removed from the water for the necessary manipulations, and either returned to the anesthesia water to extend the procedure, or moved to recovery water when the task is completed.

For longer out-of-water procedures water must be delivered in continuous flow to the gills, and the entire surface of the fish must be kept moist. Anesthesia water delivery to the gills can be achieved by a non-recirculating or recirculating system. A simple non-recirculating design uses empty IV fluid bags as reservoirs and a drip set for delivery, with flow rate regulated by the drip set clamp. This system works well for small fish. A recirculating system is well suited to large fish, where conservation of anesthesia and water are a concern. Numerous recirculating systems have been described,but the basic idea is that anesthesia water from a reservoir is delivered to the gills and is collected in a sump and returned repeatedly to the fish. The simplest recirculating system has the operator returning the sump water to the reservoir by hand, or switching the position of the sump and reservoir tanks. Less labor intensive designs use pumps to return water to the fish. In both recirculating and non-recirculating systems, flow should be normograde (in the oral cavity and out the opercular opening) to achieve optimal gas and anesthesia exchange.

Adjusting anesthetic depth may be challenging. One simple method is to prepare 60 ml syringes containing anesthesia-free water or more concentrated anesthesia solution. These solutions can be delivered to the gills directly as needed, based on the patient's condition.

A variety of parameters may be monitored during anesthesia induction and maintenance. Respiration (opercular movement) is probably the most important reference of anesthetic depth. Others include loss of equilibrium (during induction), jaw tone (which may be present in the absence of opercular movement), pupil size, color of fin margins, and response to tactile or surgical stimuli. Although pulse is not readily palpable in fish, heart rate can be monitored by ECG (using subcutaneous leads), cardiac ultrasonography, or Doppler blood flow probes. Blood gases can be measured in patients which are sufficiently large. Pulse oximetry is not consistently successful in fish.

If opercular motions cease, or other signs indicate that depth of anesthesia is too great, anesthesia-free water should be directed across the gills. The recovery tank should be well aerated and free of anesthetic agents. When returning a fish to the recovery tank, the fish can be faced into the water flow from the filter/aerator. Larger fish can be held with the mouth open and pulled forward, ramming water across the gills. Monitor jaw tone and opercular movement. Jaw tone and even biting can precede return of opercular movement. Once the fish begins breathing on its own, it is best left alone. Continue to

monitor respiration, motion, and equilibrium until the fish is fully recovered, and periodically thereafter.

It is sometimes difficult to determine immediately whether or not a fish has expired. Do not give up on resuscitation attempts prematurely, as respiratory arrest can precede cardiac arrest by an extended period of time. Electrocardiography or ultrasonography are useful in detecting heart beats in very bradycardic patients.

Euthanasia may be accomplished by an overdose of any of the anesthetic agents discussed. Larger fish which can not easily be transferred to a bath treatment may have the anesthesia solution poured directly over the gills. To be certain the euthanasia is complete, once the fish is deeply anesthetized, cranial concussion, spinal transection, or exsanguination can be performed.

Surgery

Once safe out-of-water anesthesia can be performed, surgery is a straight-forward undertaking. Indications for fish surgery include surgical sexing and other laparoscopic procedures, coelomic exploratory, external and internal mass removal, gastrointestinal foreign body, swim bladder repair, catheter implantation and other research applications. Positioning can be accomplished using open-cell foam blocks custom fit to the patient. The foam block restrains the fish in the desired position (particularly important for laterally compressed fish positioned in dorsal recumbency), prevents slipping, maintains moisture of skin in contact with the foam, and allows anesthesia water exiting the opercula to percolate through to be collected for recirculation. Exercise caution in the position of the eyes. Standard skin disinfecting protocols are extremely irritating to fish skin, and damage to the mucous layer far outweighs any benefit of a transient reduction in microbial flora. Rinsing or gentle swabbing with sterile saline, confined to the incision site, suffices to prevent complications as long as sterile technique is followed. A single pre-operative prophylactic antibiotic dose may be employed. Heavy scales can impede incisions, and a single row may need to be removed along the incision line. Scales should be removed individually by forceps with gentle caudal traction to limit epithelial damage. The surgical field may be established with a clear plastic drape held in place with a thin line of Vaseline, or, in larger fish, by sewing the drape to the edge of the incision. The plastic drape also helps retain moisture around the patient. An abundance of freely mobile skin is rarely found in fish, so large skin defects such as those resulting from external mass removal are difficult to close and often must be managed for second intention healing. The coelomic cavity is normally approached via a ventral midline incision, with latero-dorsal extensions to facilitate exposure to the swim bladder. Self-retaining retractors also improve exposure to the viscera. The coelomic cavity of koi, and perhaps other species, naturally contains light adhesions which must be broken down to access and visualize the various organs. Microsurgical techniques are required for many species of small ornamental fish. A wide range of suture materials have been used successfully in fish. Because incisions are exposed to the aqueous environment, monofilament sutures have advantages in reducing the potential for bacteria entering the incision site via capillary action as with multifilament sutures. Synthetic absorbable suture may not be readily absorbed in fish. The monofilament polyglyconate elicits a lesser inflammatory reaction in fish skin than the multifilament polyglactin 910. A single layer closure is adequate for small fish, but if a 2 layer closure is feasible, it is should be

performed. Some advocate the use of surgical glue as a final sealant and for added security, but may be problematic in that severe dermatologic reactions have rarely been associated with surgical glue in certain species of fish, particularly marine angelfish. When used alone surgical glue is associated with a higher incidence of dehiscence than suture closure in fish, and when used in combination with suture it creates added drag on the suture line once elevated from the skin by underlying mucus production. Application of a thin layer of povidone iodine ointment to the closed incision seems to reduce incidence of post-operative superficial fungal infections. Salt for a few days postoperatively at 1 - 3 g/L for freshwater fish can reduce the osmotic stress induced by anesthesia and surgical defects of the epithelium. As in any species, a good plane of nutrition is essential for wound healing.

CLINICAL PATHOLOGY

Craig Harms, DVM, PhD, DACZM
North Carolina State University
College of Veterinary Medicine
Department of Clinical Sciences
Center for Marine Sciences and Technology
303 College Circle
Morehead City, NC 28557

Clinical pathology, cortisol and weight values 2 weeks before and 48 hours after surgery in 30 koi carp. Due to non-normal distribution of some data (Shapiro-Wilk test), all summary statistics are reported as median, lower and upper quartiles (25th and 75th percentiles). Pre- and post-surgical values are compared by Wilcoxon matched pairs signed ranks test. Following application of the sequential Bonferoni correction, only the change in LD is no longer statistically significant. Creatinine was invariably below instrument range (0.1 mg/dL) and is not included in the table. (From Harms CA, Lewbart GA, Swanson CR, Kishimori JM, Boylan SM. 2005. Behavioral and clinical pathology changes in koi carp (Cyprinus carpio) subjected to anesthesia and surgery with and without intra-operative analgesics. Comp Med 55: 223-228.

	Pre-surgery median	Pre-surgery quartiles	48 h post-surgery median	48 h post-surgery quartiles	p value
PCV (%)	36	32, 38	25	21, 28	< 0.0005
TS (g/dl)	2.2	1.9, 2.4	1.8	1.3, 2.1	< 0.0005
Glucose (mg/dl)	36	28, 43	82	62, 100	< 0.0005
Calcium (mg/dl)	8.4	8.2, 8.9	8.4	7.9, 8.8	ns
Phosphorus (mg/dl)	6.4	5.6, 6.6	5.3	4.7, 5.7	< 0.0005
TP (g/dl)	2	1.7, 2.2	1.9	1.5, 2.1	< 0.0005
Albumin (g/dl)	0.9	0.75, 0.95	0.8	0.7, 0.9	< 0.0005
Globulin (g/dl)	1.1	1.0, 1.3	1.1	0.8, 1.2	< 0.0005
A/G	0.74	0.65, 0.88	0.75	0.70, 0.84	ns
AST (U/liter)	103	74, 138	600	423, 750	< 0.0005
CK (U/liter)	3387	2628, 5676	5658	5532, 5750	0.003
LD (U/liter)	132	93, 432	416	257, 649	0.046
Na (mmol/liter)	134	132, 137	134	131, 136	ns
K (mmol/liter)	3.4	3.0, 3.7	3	2.7, 3.3	0.001
Cl (mmol/liter)	115	113, 116	111	107, 113	< 0.0005
Bicarbonate (mmol/liter)	6	5, 7.5	8	8, 9	< 0.0005
Anion gap (mmol/liter)	17.4	14.9, 18.5	17.5	16.9, 19.6	ns
Cortisol (µg/dl)	23.7	16.3, 35.4	22.5	11.4, 29.9	ns
Weight (g)	123	77, 160	114	76, 152	< 0.0005

PCV = packed cell volume, TS = total solids, TP = total protein, A/G = albumin/globulin ratio, AST = aspartate aminotranferase, CK = creatine kinase, LD = lactate dehydrogenase, Na = sodium, K = potassium, Cl = chloride, ns = not statistically significant

Descriptive statistics for blood chemistry parameters analyzed for 28 dasyatid stingrays collected in coastal waters of South Carolina, Georgia, and northern Florida. Most values for bicarbonate, albumin and lactate dehydrogenase fell below instrument measurement ranges (5 mmol/L, 1 g/dl, and 5 U/L, respectively), and were not included. Cain DK, Harms CA, Segars A. 2004. Plasma biochemistry reference values of wild-caught southern stingrays (Dasyatis americana). J Zoo Wildl Med 35: 471-476.

Parameter	**Median**	**10th percentile**	**90th percentile**
PCV L/L (%)	0.22 (22)	0.15 (15)	0.25 (25)
Total Solids g/L (g/dl)	56 (5.6)	42 (4.2)	60 (6.0)
Total Protein g/L (g/dl)	26 (2.6)	16 (1.6)	32 (3.2)
Sodium mmol/L	315	295.9	326.2
Potassium mmol/L	5.0	3.2	6.4
Chloride mmol/L	342	301	362
Calcium mmol/L (mg/dl)	4.12 (16.5)	3.01 (12.06)	4.82 (19.3)
Phosphorus mmol/L (mg/dl)	1.5 (4.7)	0.98 (3.0)	2.1 (6.4)
Urea Nitrogen mmol/L (mg/dl)	444 (1243)	423:0 (1184.9)	461.6 (1293.1)
Glucose mmol/L (mg/dl)	1.69 (30.5)	0.94 (16.9)	2.35 (42.4)
Aspartate aminotransferase U/L	14.5	3.6	61.2
Creatine kinase U/L	80.5	11.7	296.5
Lactate mmol/L	3.1	<2.0	6.2
Osmolality mOsm	1065	1007.6	1144.3

Bonnethead shark (*Sphyrna tiburo*) PCV and plasma biochemistry reference values.* (From Harms C, Ross T, Segars A. 2002. Plasma biochemistry reference values of wild bonnethead sharks, Sphyrna tiburo. Vet Clin Path 31: 111-115.)

Parameter	n	Median	Lower Quartile	Upper Quartile	Minimum-Maximum
PCV (%) [L/L]	21	24 [24]	22 [22]	26 [26]	17-28 [17-28]
Total solids (g/dl) [g/L]	20	6.3 [63]	6.0 [60]	6.8 [68]	5.8-7.5 [58-75]
Total protein (g/dl) [g/L]	24	2.9 [29]	2.7 [27]	3.4 [34]	2.2-4.3 [22-43]
Albumin (g/dl) [g/L]†	24	0.4 [4]	0.4 [4]	0.4 [4]	0.3-0.5 [3-5]
Globulins (g/dl) [g/L]	24	2.6 [26]	2.3 [23]	3.0 [30]	1.9-3.8 [19 - 38]
A/G ratio	24	0.14	0.13	0.17	0.12 - 0.19
Sodium (mmol/L)	24	282	279	285	273 - 292
Potassium (mmol/L)	24	7.3	6.4	7.9	5.7 - 9.2
Chloride (mmol/L)	24	290	285	296	277 - 304
Bicarbonate (mmol/L)†	24	3	2	4	0 - 5
Anion gap (mmol/L)	24	-5.8	-7.6	0.1	-15.7 - 7.5
Calcium (mg/dl) [mmol/L]	24	16.8 [4.2]	16.2 [4.0]	17.4 [4.3]	15.8 - 18.2 [3.9 - 4.5]
Phosphorus (mg/dl) [mmol/L]	24	8.8 [2.8]	7.5 [2.4]	10.0 [3.2]	5.9 - 12.7 [1.9 - 4.1]
Urea nitrogen (mg/dl) [mmol/L]	24	1004 [358]	986 [352]	1028 [367]	944 - 1068 [337 - 381]
Glucose (mg/dl) [mmol/L]	24	184 [10.2]	165 [9.2]	191 [10.6]	155 - 218 [8.6 - 12.1]
Aspartate aminotransferase (U/L)	24	42	33	66	15 - 132
Lactate dehydrogenase (U/L)	24	<5	<5	<5	<5 - 11
Creatine kinase (U/L)	24	82	47	233	18 - 725
Osmolality (mOsm/kg)	24	1094	1078	1111	1056 - 1139

*Standard units [SI units]

†Includes values below the reliable limit of detection of the analyzer.

ENVIRONMENTAL DISEASES

Craig Harms, DVM, PhD, DACZM
North Carolina State University
College of Veterinary Medicine
Department of Clinical Sciences
Center for Marine Sciences and Technology
303 College Circle
Morehead City, NC 28557

(excerpted from Harms CA. Fish. In: Fowler ME & Miller RE (eds). Zoo and Wild Animal Medicine V. Philadelphia, Saunders, 2003)

Water quality problems are the major cause of captive fish morbidity and mortality. An understanding of the major water quality parameters and how they interact is essential for diagnosing and treating fish disease.

Dissolved oxygen (DO) should be maintained above 5 ppm (mg/L), with 6 - 10 ppm ideal. DO lower than 5 ppm can be tolerated for varying periods of time by some species, but at the cost of opportunistic infections and poor growth. Cold-water fish generally require higher DO than warm-water fish. DO is reduced with increasing temperature and salinity. In ponds, the balance of photosynthesis and respiration is the major determinant of DO, with peak concentrations at the end of the day, and lowest levels near dawn. Causes of and contributors to environmental hypoxia include power outages interrupting air pump operations, high fish biomass, and increased oxygen demand due to decay of a high organic load (e.g. algal death, excess feed, and dead fish themselves). Signs of hypoxia include aggregation of fish around oxygen sources, including at the air-water interface ("piping"), increased respiratory rate, survival of only air-breathing fish, and a characteristic (but not pathognomonic) agonal response of gaping mouth and flared opercula. DO is very labile and should be measured on site for meaningful results.

Optimal pH range varies by species. Many tropical freshwater fish come from poorly buffered waters and do best in slightly acid conditions (pH 6.5 - 6.8), except for African rift lake fish which do best in hard alkaline water (pH 7.6 - 8.0). Marine waters are more stable and alkaline, so marine fish should require a higher range (pH 8.0 - 8.5). Fish can adapt, up to a point, to gradual changes in pH. Metabolic activity of fish and nitrifying bacteria produce acids, resulting in a downward trend in aquarium pH, which must be countered by periodic partial water changes. The actual pH of the water is somewhat less important than how it relates to ammonia. At low pH, ammonia exists as the less toxic ammonium ion (NH_4^+) which is unable to pass through gill membranes; at high pH it is found as the more toxic uncharged NH_3 which readily passes through gill membranes (see below).

Alkalinity in water chemistry terms is a measure of the capacity of water to neutralize a strong acid. It is an aggregate measure of buffering capacity, primarily dependent on the anions bicarbonate and carbonate, and is expressed as calcium carbonate equivalents. It should not be confused with the term alkaline, which refers to a pH > 7. Hardness is the sum of divalent cation concentrations, almost exclusively

calcium and magnesium, and may also be expressed in terms of calcium carbonate equivalents. Hardness is also expressed unequivocally as mg/L calcium (mg/L Ca x 2.5 = $CaCO_3$ equivalents), or in the German scale of degrees hardness (°dH; $CaCO_3$ equivalents in mg/L = °dH x 17.9). Hardness and alkalinity are usually closely related, because alkalinity of most natural waters derives from carbonate salts of calcium and magnesium. The two parameters can vary independently, however. For instance, addition of sodium bicarbonate would raise alkalinity and not hardness, and addition of calcium chloride would raise hardness and not alkalinity.

Salinity is the salt content of water expressed as g/L or parts per thousand (ppt). Natural water ranges from 0 to 40 ppt, with freshwater being < 0.5 ppt and sea water from 33 - 37 ppt. (For reference, 0.9% physiologic saline is 9 ppt.) Salinity can be estimated from refractive index (temperature calibrated refractometers; convenient but expensive) or specific gravity (hydrometers; inexpensive but less convenient).

Nitrogen cycling is one of the most important factors in aquarium fish health. Ammonia is the primary nitrogenous waste product from fish, and is very toxic in its un-ionized form. Ammonia/ammonium ratios depend on pH, temperature, salinity and pressure. Total ammonia measurements are comprised of both ammonia and ammonium; the ammonia/ammonium ratio can be derived from standard tables supplied with test kits. Any ammonia detected in an established aquarium is an indicator of biological filtration deficiency (see below). Ammonia is extremely irritating to mucous membranes and causes gill damage that can interfere with oxygen exchange. Treatment is by partial water changes and possibly acidification of the water. Nitrifying bacteria convert highly toxic ammonia to the somewhat less toxic nitrite (NO_2^-) (Nitrosomonas sp. and others) and even less toxic nitrate (NO_3^-) (Nitrobacter sp. and others). Nitrite causes methemoglobinemia (brown blood disease), characterized by brown-colored gills, respiratory difficulty and occasionally a head up vertical position. Treatment is by addition of sodium chloride to the water and water changes. Chloride ions compete with nitrite for uptake by the gills. Nitrate is much less toxic than ammonia and nitrite, but can build up to subclinically harmful levels in the absence of periodic water changes. Nitrifying bacteria in the aquarium filters and on all surfaces are responsible for biological filtration. Under normal conditions it takes several weeks, adequate oxygen, and a source of ammonia for nitrifying bacteria to colonize and proliferate to functional levels in the tank. When an inexperienced aquarist overloads a new tank with fish, before biological filtration is adequately established, toxic ammonia concentrations result ("new tank syndrome"). Health of the biological filter must also be considered when medicating an aquarium with antimicrobial compounds, as iatrogenic ammonia spikes rather than the suspected microbial agent may kill the fish.

Chlorine and chloramines are added to municipal water supplies to make it safe for human consumption. Both are highly toxic to fish. Chlorine diffuses out of water if allowed to stand sufficiently long in a container with a large surface area, and diffuses more rapidly in aerated water. Instant removal of chlorine is accomplished with sodium thiosulfate at 7 g/1000 L (chlorine concentrations as high as 2.0 ppm), or with commercial products which usually contain sodium thiosulfate. Chloramines do not diffuse out of water, and chloramine-containing water must be filtered by activated carbon or treated or treated with a chemical neutralizer.

Copper sulfate may be added to pond water or municipal water to control algae, or gain access via copper pipes, and is also used to control fish parasites. Copper is highly toxic to invertebrates, and can be harmful to fish, particularly in soft fresh water with low DO.

There are 3 major functional types of filtration: biological, mechanical, and chemical, although single filters may combine more than 1 function. Biological filtration converts toxic ammonia to non-toxic nitrates (see above) and can take the form of undergravel filters, sponge filters or wet/dry filters. Mechanical filtration is designed to remove particulate matter, and includes submerged box filters, out-of-tank power filters, sand filters and diatom filters. Chemical filtration systems include ion-exchange resins, activated carbon, ozone, foam fractionation, reverse osmosis, and UV filtration to remove or modify chemical contaminants. Activated carbon is the chemical filtration most frequently employed in home aquaria. Activated carbon filtration must be disconnected when treating a tank with therapeutic agents, until you want to remove those agents.

Some blue-green algaes produce neuro- and hepatotoxins (e.g., microcystins) under the right conditions.

Temperature affects many water quality parameters (particularly DO), as well as fish metabolic rate and immune response, and protozoal parasite life cycles. Preferred optimum temperatures vary widely according to species. Temperate species tolerate greater fluctuations in temperature than tropical species, and a rapid drop in temperature is less detrimental than a rapid rise. Increasing water temperature is easily accomplished with submersible heaters; refrigeration systems are more expensive. A rule of thumb for temperature changes is 1 °C/h. Hyopthermia leads to inactivity, positioning on substrate, and anorexia. Hyperthermia increases oxygen demand just as DO is decreasing.

Fish are susceptible to sunburn, particularly if held in clear shallow water without shade in sunny conditions, or if forced to the surface by hypoxic conditions. Dorsal erythema may progress to ulceration.

Some species of fish may be destructive to substrate and tank appliances. Adequate shielding of susceptible equipment and provision of a diverse tank habitat can reduce the chance of potential hazards developing.

Electrical exposure from lightning or faulty wiring can fracture vertebrae, induce erratic swimming, or cause death. Lower level exposure to stray voltage may cause ill-thrift, elevated plasma cortisol and glucose concentrations, and altered plasma electrolyte balance.

Species Compatibility. Detrimental effects from tank-mate incompatibility may be obvious or subtle. Many variables such as stocking density, availability of food, water quality parameters and the availability of security areas (cover) all come into play. In certain circumstances the dominant individuals simply intimidate the submissive fishes, forcing them to hide and causing them to starve since they may not be getting any of the available food. A simple lack of gross lesions does not necessarily mean that there is not a compatibility problem.

Feeding

Adequate nutrition is an important component of preventive medicine. As in any species, diet offered may differ from diet consumed by the fish, so observing food intake is critical to any nutritional assessment. Fish are efficient at feed conversion, well over 50% for some species, which makes them attractive food-producing animals in aquaculture. Feed intake is energy limited. Digestible protein to digestible energy ratios for optimal growth range from 81 - 117 mg/kcal, substantially higher than for mammals because of the lower energy requirement for maintenance. Inadequate protein content in the diet may lead to excessive deposition of body fat. Some degree of hepatic lipidosis is common in captive fishes, and may reflect a fat storage function of this organ, though it may progress to a pathologic condition. Polyunsaturated fatty acids are required by fish, though the essential fatty acids differ between species. Protein and lipids are utilized well as energy sources by fish, with carbohydrate utilization varying considerably based on their natural dietary preferences. Carnivorous fish such as salmonids require a higher protein diet than omnivorous and herbivorous fish, while a high protein trout chow is less appropriate for koi and goldfish.

Some species of fishes readily eat almost any food offered, while others (especially certain marine reef fishes) have very specialized diets that may be exceedingly difficult to duplicate in captivity. When possible, a varied diet is recommended. Most of the commercially prepared flake and pelleted foods appear to be well balanced and palatable. Some piscivorous fishes will not consume prepared foods and must be fed whole fish, either live or frozen/thawed. Feeding live fishes has the obvious potential disadvantage of introducing potential pathogens into the aquarium. Some fishes including certain species of South American plecostomus catfishes seem to require driftwood in the aquarium upon which they graze and likely extract some nutrients. Other species enjoy fresh grated vegetables such as zucchini. Fish may consume from1 - 6% of their body weight daily, depending on food quality, water temperature, and metabolic activity (growth, reproduction). Required feeding frequency depends on the species, though smaller species do well with multiple feedings per day, and larger species may not require daily feedings. The adequacy of feeding quantity and frequency can best be gauged by visual assessment of lateral and head-on profiles and when possible by weighing the fish. Generally speaking, short term underfeeding is less detrimental than short term overfeeding due to secondary adverse water quality effects of overfeeding. The natural history and husbandry requirements of any captive fish should be thoroughly studied prior to acquisition to ensure an adequate diet can be procured.

Many nutritional disorders have been described in catfish and salmonids, but relatively little is known about formulating complete diets for the plethora of species kept in aquaria. The fact that so many species do well in captivity may be more a testament to their hardiness and adaptability than to our ability to provide an optimal diet. To avoid nutritional problems, be sure all the fish are eating and the food is fresh. Commercial food over 6 months old should not be used, and opened containers should optimally be refrigerated. Guidelines for storage and handling of fish used as food for piscivorous birds and mammals apply equally well to piscivorous fish.[1] Water soluble vitamins may leach rapidly from flake and pelleted food once added to water, so rapid consumption by the fish helps reduce the chances of deficiencies, and sinking feed delivers essential nutrients more efficiently to benthic feeders. Carotenoid pigments (e.g., astaxanthin,

castaxanthin, zeazanthin) are variably utilized by different species, but are important for maintaining desirable coloration of muscle (salmonids) and skin (goldfish, koi, etc.). The following condensed list contains nutritional disorders that have been described for aquaculture species (deficiencies except where noted):

Anemia: Vitamins C, B_{12}, E, K, folic acid, niacin, pantothenic acid, pyridoxine, iron, autoxidation of unsaturated fats (excess).

Scoliosis: Vitamin C, tryptophan.

Cataracts/(other ocular manifestations): riboflavin, tryptophan, methionine, manganese, Zn, (Vitamin A).

Pancreatic lesions: biotin, pantothenic acid.

IMPORTANT VIRAL AND PARASITIC DISEASES OF ORNAMENTAL FISH

Gregory Lewbart M.S., V.M.D., Dipl. ACZM
greg_lewbart@ncsu.edu

VIRAL DISEASES

Several viral diseases have been thoroughly described in ornamental fishes. The most commonly observed viral disease of tropical fish is called lymphocystis disease. This disease is caused by an iridovirus which infects connective tissue cells of the fish. The virus induces these cells to undergo extensive hypertrophy until the cells may actually be visible to the naked eye. Affected cells can increase a thousand fold in size. The disease appears to be more common in marine and brackish water fishes. Certain species of freshwater tropical fish like the green terror (*Aequidens rivulatus*) are prone to the disease. Members of the genera *Scatophagus*, *Monodactylus*, and *Changa* are all brackish water fishes that seem predisposed to lymphocystis disease. Stress is almost certainly a factor in this disease since outbreaks are frequently observed following capture and shipping of fishes. Gross lesions appear white and granular and usually are seen on the skin and fins. Occasionally, lesions will be seen in the mouth and on the gills. There is no proven chemotherapeutic treatment. Most cases are self-limiting if the fish is provided with proper water quality and nutrition. Surgery can be performed on affected fish by carefully scraping the hyperplastic fibroblasts clear of the fish with a sterile scalpel or scissors. This procedure should be performed quickly and the patient(s) should receive 5-10 days of topical antibiotic therapy following the surgery. A definitive diagnosis can be made by microscopically examining a scraping of the affected area. The enlarged connective tissue cells will appear circular and in clusters. These cells frequently emit a light orange hue under the microscope.

Several important viral diseases have been described in goldfish and koi. Spring Viremia of Carp (SVC) is a reportable disease caused by a rhabdovirus (*Rhabdovirus carpio*). Until this year (2002) the virus had not been found in North America and is considered a foreign animal disease (FAD) in this country. Mortality may reach 100% but is frequently much less. Younger fish are more susceptible than older fish and infected fish typically present with abdominal ascites and multiple organ hemorrhages. Transmission is horizontal and most cases occur in the spring or early summer when the water begins to warm. Diagnosis is usually made with viral isolation (from spleen and /or caudal kidney) and/or serum antibody titers. Koi or carp "pox" is a skin disease caused by herpes virus and not a pox virus. This rarely fatal disease is frequently self-limiting and may resolve when pond temperatures increase. Diagnosis is usually based on clinical presentation and histopathology. Koi herpes virus (KHV) is a recently described disease of koi that is frequently fatal. The virus causes a severe bronchitis and the affected fish literally die of hypoxia. Recent outbreaks of this disease have been reported in Israel, Japan, and the U.S.

PROTOZOAL DISEASES

The following list provides the correct spellings and a brief description of some important protozoal disease agents of tropical fish.

Brooklynella- The marine "cousin" of *Chilodonella*. Found on saltwater tropicals. Looks very similar to *Chilodonella*. This parasite is also easily treated with protozoicides.

Chilodonella- A ciliated protozoan which can cause high morbidity and mortality among freshwater tropical fishes at the wholesale and fish farming levels of the industry. Attacks skin and gills. Easily identified microscopically by its heart-shaped structure and slow circular motion when not crawling on the surface of the fish. Once diagnosed, this problem is easily treated with formaldehyde, malachite green or salt.

Cryptocaryon- The marine form of "Ich." Frequently referred to as "white spot disease." This large ciliate possesses both free-swimming and encysted stages. Many aquarists control its spread with the prophylactic use of copper sulfate in the water. A quarantine protocol for new fishes can greatly help reduce the spread of this disease.

Epistylis (Heteropolaria)- A stalked ciliate that is commonly found in freshwater containing a high organic load. Tends to colonize bottom dwelling fish such as the plecostomus catfish. Lesions appear pale and white in color and resemble a fungal disease. Microscopically, one sees a ciliated crown atop a long stalk that is prone to frequent contractions. Easily treated with formaldehyde however a clean well-filtered tank is the best solution to the problem. This disease is usually not fatal in itself but may open the fish up to secondary bacterial disease.

Henneguya- A sporozoan which presents in the form of small white cysts on the fins and gills of some fish. The cysts contain infective spores. Commonly seen on the dorsal fins of imported *Leporinus* species. Not harmful to the fish. Careful removal by scraping with a scalpel is the best treatment since the parasite is aesthetically undesirable.

Hexamita (Spironucleus)- These flagellated protozoa may cause severe gastrointestinal disease if present in large numbers. Normal inhabitant of fish digestive tract. As an ectoparasite it is believed to be involved with "Hole in the Head Disease" (Head and Lateral Line Erosion) common to Oscars and other cichlids. Treated effectively with metronidazole.

Ichthyobodo- Formerly (and still commonly) called Costia. A flagellated protozoal ectoparasite. A normal inhabitant of fish skin. Poor water quality and other stresses (especially crowding) may allow this normally mutualistic parasite to reproduce rapidly and overwhelm the host. It is responsive to treatment with formaldehyde and malachite green but tougher than most protozoa. Microscopically the protozoa are very small (5-10 microns), move rapidly, and are shaped like small sickles. They may be attached to host tissue or swimming free. Most common in freshwater species of fishes but has been reported from several marine fishes.

Ichthyophthirius- Known commonly simply as "Ich." The largest protozoal parasite of fish and one of the most commonly encountered. Trophozoites may reach 1.0 mm in diameter. Can affect skin, gills or both. Prevention is the best method of control although the parasite is susceptible to a variety of parasiticides including malachite green and formaldehyde.

Plistophora- A microsporidian sporozoan which is the causative agent for true "Neon Tetra Disease." The parasite is not specific to neon tetras and when present will attack the musculature of the affected fish. Infected muscle will contain numerous sporoblasts containing spores. Grossly infected muscle will appear white or pale. Certain bacterial skin diseases will produce similar gross lesions. Such sporozoan infections are usually unresponsive to treatment and diseased fish should be removed from the tank. High mortality is usually associated with this disease.

Tetrahymena- Commonly called "Guppy Killer Disease." A ciliated protozoan that can be free-living or parasitic. Common in crowded conditions and in water containing excessive organic debris. Unaffected by parasiticides because of its ability to burrow deeply into skin of host that ultimately protects parasites from chemotherapeutics. Best method of control is prevention through sound husbandry practices. These pear-shaped protozoa may be present in very large numbers when the infestation is severe.

Trichodina- A disc-shaped ciliate protozoan found on the skin and gills of many fish. Circular rows of denticles and a ciliary girdle give this parasite a unique radial symmetry. Probably not harmful when present in small numbers.

Uronema- The marine counterpart of *Tetrahymena*. This is also a significant disease-causing parasite. Tissue destruction by this protozoan will allow for secondary invasion by pathogenic bacteria.

TREMATODE DISEASES

Both monogenean and digenean trematodes parasitize tropical fishes. Monogenean parasites including *Dactylogyrus* and *Gyrodactylus* are ectoparasitic and can cause considerable damage to the host when present in high numbers. These parasites possess a multiple hooked attachment organ called an opisthaptor that disrupts the integrity of the host's skin and mucus membranes. These monogeneans can complete their entire life cycle on a single host and in some species the cycle may be as short as 60 hours if all environmental conditions are optimal. Crowding and other stress factors predispose tropical fish to monogenean trematode problems. These parasites are generally resistant to low doses of formaldehyde and even some organophosphates. Most freshwater monogeneans can be killed quickly with a 3 to 5 minute saltwater bath (30-35 parts per thousand). Glacial acetic acid or hydrogen peroxide dips will also kill these parasites. Dosage information is given in the provided articles and references. Praziquantel baths have also proved to be effective in killing some monogenean worms. While expensive,

this is a relatively safe treatment when used at a concentration of 10 parts per million for 3 to 6 hours.

The majority of digenean fluke problems appear to be primarily aesthetic in nature among tropical fish. Fish commonly serve as an intermediate host for these parasites that frequently have a complex life cycle. Invertebrates may be the first host and a bird or mammal the primary host. Encysted digeneans are commonly observed as metacercaria in the skin and underlying tissues of tropical fish. Occasionally these metacercaria are found in the coelomic cavity of tropical fish. Imported silver dollar fish species from South America are commonly infected with metacercaria belonging to the genus *Neascus*. Some fish may have only one or two metacercaria while others may harbor hundreds. This disease will not harm the fish and will not progress unless an appropriate primary host animal consumes the fish. Fish that are affected are sometimes said to have "Salt and Pepper" disease since the cysts become pigmented and the uplifted scales appear especially white or shiny. Another common digenean parasite is *Clinostomum* that is called the "Grub" by fish farmers in Florida. Excysted worms may be more than 5 millimeters long and are easily visible to the naked eye. If the metacercaria are not too numerous, they can be removed safely with a clean scalpel.

Occasionally, larvae belonging to the genus *Diplostomum* have been found associated with the lens in the eyes of tropical fish. In such cases the lens will become opaque and the fish may be blinded. There is no reported treatment for this disease.

CESTODE DISEASES

Tapeworms are found inhabiting the digestive tract of wild tropical fishes. Diagnosis can be made by fecal examination observing proglottids exiting the vent of a fish, or during necropsy. Recently, work has been published using praziquantel to treat infected fishes and it appears that certain tapeworms are susceptible to a dose as low as 2 parts per million in the water. Infected fish can be bathed in this solution for 3 hours with adequate aeration.
Tropical fish commonly act as an intermediate host in a cestode's life cycle and encysted tapeworm larvae called procercoids can be found in the coelomic cavity of tropical fishes.

NEMATODE DISEASES

Nematodes are common parasites of fish and can be especially abundant in wild species. In some cases the tropical fish is the definitive host and the nematodes will be found in the gastrointestinal tract. In other instances the fish is an intermediate host and the larval nematodes will be seen encysted beneath the skin, in the musculature or in the coelomic cavity. Medical treatment of the larval forms is very difficult because these nematodes are encysted and well protected. Some species of *Eustrongyloides* form large cysts just under the skin of tropical fish and can be removed surgically, especially if the fish is relatively large. As is the case with other encysted larval helminth parasites, the disease will usually not progress unless the fish is eaten by the definitive host.

Gastrointestinal nematodes can be observed on necropsy and ova are readily seen on examination of the feces. While the presence of these parasites may not cause a problem in nature, the stresses of captivity and shipping may exacerbate any parasitic problem. Nematodicides such as fenbendazole and piperazine may be incorporated into food in order to successfully treat these problems. The attached articles contain dosage and treatment information.

CRUSTACEAN DISEASES

There are several important crustacean parasites of tropical fish. *Laernea*, known commonly as "Anchor worm," is a modified copepod parasite that infects large scaled freshwater tropical and temperate species of fish. This parasite possesses a life cycle that includes microscopic pelagic larval stages that molt and grow several times before attacking the fish host. On the host the female anchor worm matures and produces two large egg sacs containing hundreds of *Laernea* eggs. This parasite is easily visible to the naked eye and may be more than 2 centimeters in length. They get their name from the attachment organ that is a highly modified structure that resembles the anchor on a ship. This structure is buried in the host's musculature and allows for the invasion of pathogenic bacteria. Plucking the parasites from the fish is warranted and usually results in inflamed areas that heal quickly. Organophosphates and glacial acetic acid dips are successful in treating the problem. The disease is especially common in imported and domestic goldfish.

The other major crustacean parasite is *Argulus*. This branchiuran crustacean is commonly called the "Fish Louse." Fish lice are flattened creatures with a very distinctive shape and appearance. They have a pair of eyespots and are about 5-10 millimeters in length. They move about the skin of a fish very effectively and camouflage themselves well on the host. They suck bodily fluids from the fish via a sharp stiletto that actually injects a small amount of toxin into the fish. These parasites are especially harmful to small fish. *Argulus* also possesses a life cycle with pelagic larval stages so the entire aquarium system may have to be treated with organophosphates to control the disease. Depending on temperature, the total life cycle takes between 6 and 20 weeks.

A less commonly seen group of crustacean parasites are the isopods. While most isopods are free living, members of the genus *Livoneca* can be parasitic. Terrestrial pill bugs commonly seen under rocks and logs are isopods and the aquatic parasitic forms resemble their land dwelling relatives. While *Argulus* is dorsoventrally compressed, isopods like *Livoneca* are laterally compressed and appear segmented.

FURTHER READING

The following list contains references that will aid the interested student or clinician in diagnosing and treating infectious disease problems of ornamental fish.

1. Brown L: Aquaculture for Veterinarians. Pergamon Press, Oxford, England, 1993, 447 pp.
2. Butcher RL: Manual of Ornamental Fish. British Small Animal Veterinary Association. Gloucestershire, England, 1992, 200 pp.
3. deGuzman E. and E.B. Shotts. Bacterial Culture and Evaluation of Diseases of Fish. Vet Clinics of North America Small Animal Pract., 1988, 18(2): 365-374.
4. Gratzek JB: Control and therapy of fish diseases. Adv Vet Sci Comp Med 27:297-324, 1983.
5. Gratzek JB: An overview of ornamental fish diseases and therapy. i Small Anim Pract 22:345-366, 1981.
6. Gratzek JB: Aquariology: the Science of fish Health Management, Tetra Press, 1992.
7. Herwig N: Handbook of Drugs and Chemicals Used in the Treatment of Fish Diseases. Springfield IL, Charles C Thomas, 272 pp., 1979.
8. Hoffman GL and FP Meyer: Parasites of Freshwater Fish. TFH Publications, Neptune City, NJ, 1974.
9. Johnson, EJ: Koi Health and Disease. Johnson Veterinary Services, 3805 Robinson Rd., Marietta, GA 30068, 1997, 141 pp.
10. Lewbart, GA: Self-Assessment Color Review of Ornamental Fish. Iowa State University Press, 1998, 192 pp.
11. Lewbart, GA: Emergency pet fish medicine. *In* Bonagura JD and Kirk RW (eds.): Current Veterinary Therapy XII: Small Animal Practice. Philadelphia, WB Saunders Co, 1995, 1369-1374.
12. Lewbart, G., Vaden, S., Deen, J., Whitt, D., Manaugh, C., Doi, A., Smith, T., and K., Flammer. Pharmacokinetics of enrofloxacin in the red pacu (*Colossoma brachypomum).* Journal of Veterinary Pharmacology and Therapeutics, 20:124-128, 1997.
13. Noga, EJ: Fish Disease: Diagnosis and Treatment. ISU Press, 2000, 367 pp.
14. Stoskopf, MK: Fish Medicine. Philadelphia: W.B. Saunders Company, 1993, 882 pp.
15. Tocidlowksi, ME, Lewbart, GA., and Stoskopf, MK. 1997. Hematologic study of red pacu (*Colossoma brachypomum*). Veterinary Clinical Pathology, 26(3):119-125.
16. Untergasser D: Handbook of Fish Diseases. TFH Publications, Neptune, NJ, pp.160, 1989.
17. The Veterinary Clinics of North America: Exotica Animal Practice. Various volumes, WB Saunders.
18. Inglis V, Roberts RJ, Bromage NR (eds): Bacterial Diseases of Fish, New York, NY, Halsted Press, 1993
19. Trust TJ, Bartlett KH: Occurrence of potential pathogens in water containing ornamental fishes. Applied Microbiology 28(1):35-40, 1974

20. Frerichs GN: Isolation and identification of fish bacterial pathogens, in Inglis V, Roberts RJ, Bromage NR (eds): Bacterial Diseases of Fish, New York, NY, Halsted Press, 1993, pp 257-283

Content of pages **111 – 115** excluded due to copyright restrictions. Blank pages 110 and 116 removed.

The citation for the excluded content is**: Lewbart GA. Bacterial Diseases of Pet Fish. Proceedings of the North American Veterinary Conference. 2001 15(PT. 1):715-18.**

This material is available in the collection of the NCSU Libraries.

For more information about access to the NCSU Libraries, visit www.lib.ncsu.edu.

PET FISH THERAPEUTICS

Gregory Lewbart M.S., V.M.D., Dipl. ACZM
greg_lewbart@ncsu.edu

Introduction:

Certainly one of the most commonly asked questions concerning the practice of pet fish veterinary medicine is, "what drug should I use and what's the dose?" An uncomplicated question with a complex and frequently ambiguous answer. The problem, of course, lies in the lack of sound pharmacokinetic data available and the overwhelming number of species involved. When environmental differences such as temperature, pH, and water hardness are tossed into the equation, selecting a drug and dosing regimen becomes even less objective.

The FDA is currently examining the wide availability of prescription drugs, especially antibiotics (for more information on the FDA and the use of drugs in aquaculture, consult the following Web Address: (www.fda.gov/cvm/). With the recent passing of the Minor Use and Minor Species (MUMS) Animal Health Act, we are likely to see some dramatic changes in the availability of drugs for use in fish (it is likely that these changes will mean more "approved" drugs). Such measures will necessitate sound pharmacokinetics, efficacy, and safety studies to support clinical use of antimicrobials and other chemotherapuetants in fishes. Relatively little research related to pharmacology has been reported in aquarium fishes. What little information exists is based on clinical efficacy and in-vitro trials using a number of different antimicrobials.

The purpose of these notes is to familiarize the reader with the most current pet fish pharmacokinetic information, provide a comprehensive list of drugs and related compounds, and discuss the variables that influence dosing regimens in pet fish.

Dosing Routes:

Due to their aquatic nature, generally small size, and frequently large numbers, a variety of atypical methods are utilized to deliver antibiotics to the ornamental fish. Standard parenteral methods can and commonly are used to dose aquarium fish with antibiotics, but the clinician must also be familiar with the terminology applied to water borne treatments. Table A contains these important definitions.

Drugs and Dosages:

The majority of the current information on chemotherapeutics used in aquarium fish has been extrapolated from the finfish aquaculture literature. There are a number of reasons for this, most of which revolve around funding for sound pharmacokinetic research. One review article summarized the entire body of literature on this subject (Stoffregen et. al., 1996). There are currently only two antibiotics approved for use in fish intended for human consumption (Romet-30® and Terramycin for Fish®). Most of the literature

dealing with antibiotic usage in aquarium fish is empirical and anecdotal. Several studies (Lewbart et. al, 1997; Doi et. al, 1998; Lewbart et al., 2005a; Lewbart et al., 2005b) report on the pharmacokinetics of antibiotics in the red pacu (*Piaractus brachypomum*) and koi (*Cyprinus carpio*), a tropical aquarium fish closely related to such important species as the tetras, hatchetfishes, silver dollars, and headstanders. Fortunately, the veterinarian treating aquarium fish can apply current extra label drug use regulations when selecting and initiating antibiotic therapy.

Routes of Antibiotic Administration for Ornamental Fish:

Bath- Usually refers to a treatment in which the drug is dissolved in the water in which the fish are swimming. The treatment usually lasts at least 15 minutes and less than 24 hours. Dosage is usually based on volume of water and not on fish biomass.

Dip- Refers to a treatment in which the fish is submerged in a particular solution for between 1 second and 15 minutes. Water volumes are usually smaller than those of bath treatments and drug concentrations are frequently higher.

Flush or Flow Through- Requires constant water flow. Most frequently used in raceways or narrow vats. Medicant is added to inflow area and fish are exposed to drug as it passes over them with the water current. Similar to dip procedure except fish may not have to be removed from their normal holding area.

Indefinite Bath- Medication is added to aquarium and usually there is no water change or immediate retreatment.

Injection- The antibiotic is given by injection with the aid of a hypodermic needle and syringe. Routes may be subcutaneous, intradermal, intramuscular, intravenous, and intraperitoneal (intracoelomic).

Oral- Medication is mixed with the food in order to treat the fish. Usually done by incorporating drug into a gelatinized food mixture. For larger fish patients, medication can be placed in a chunk of food and then fed or force-fed to the patient.

Topical- The antibiotic is applied directly to the lesion.

* Remember, before using any drug in the water, discontinue chemical (e.g. carbon) filtration during treatment as this will inactivate the medicant. Adequate aeration is also important during any water treatment.

**When antibiotics are used as bath treatments, ideally they should be used daily for 5-7 days. Water changes (at least 50%) should take place between treatments. This protocol is much easier to follow in a home or hospital aquarium than in a pet store or wholesale facility.

Formulary:

The following formulary is not meant to be a complete listing of all drugs available to treat fishes nor has the information provided been proven to be safe and effective on all species. The doses were compiled from several sources and my personal experience. The information herein is meant only to be a guideline and a quick reference of drugs and dosages for the treatment of pet fishes by a licensed veterinarian.

I. Anesthetic Agents

1. **Eugenol**. A practical alternative to MS-222 is clove oil (active ingredient eugenol) which is available at many pharmacies. Eugenol is not completely soluble in water and should be diluted 1:10 in 95% ethanol to yield a working stock solution of 100 mg/ml, since each ml of clove oil contains approximately 1 gram of drug. Concentrations of between 40 and 120 mg/L are effective in freshwater and marine species and results are comparable to MS-222, except that recovery may be prolonged.

2. **MS-222 (tricaine methanesulfonate).** This particular agent is purchased in a crystalline form and can be used to produce a working stock solution of 10 grams per liter in clean, dechlorinated water. By making dilutions from this stock solution, the clinician can accurately formulate safe and effective anesthetic solutions. Each liter of stock solution should be buffered with 3-5 grams of sodium bicarbonate (baking soda). The stock solution should be stored in a glass bottle away from light at room temperature. When prepared and stored in this manner, the stock solution is good for 30-60 days. Final concentrations of between 100 and 150 mg/liter will usually anesthetize most fish within a matter of three to five minutes.

II. Antibiotics

1. **Amikacin sulfate (Amiglyde-V®).** Can administer a loading dose of 5 mg/kg IM and then 2.5 mg/kg every 72 hours for 5 treatments total.

2. **Ceftazidime (Fortaz®).** This human cephalosporin drug has proved to provide good broad-spectrum activity with the advantages that its half-life is long and it kills *Pseudomonas*. Dose is 20 mg/kg IM or SC Q 72 hours.

3. **Enrofloxacin (Baytril®).** A quinolone compound that appears to be safe and effective in fishes. May be used IM, ICe and PO at a dose of 5-10 mg/kg/ Q 48 hours for 7-21 days. May be diluted with sterile saline since the concentrated form (2.27%) may be irritating. May also be used as a bath at a concentration of 2.5 mg/L for 5 hours Q 24 hours. Recent work here at the NCSU-CVM indicates 10 mg/kg ICe Q 5 days for koi at 20 degrees C.

4. **Florfenicol.** Florfenicol is a structural analogue of chloramphenicol similar to thiamphenicol, but with more activity against some organisms than chloramphenicol. It differs importantly from chloramphenicol in that it lacks the para-nitro group that is believed to be responsible for the problems of aplastic anemia. Based on salmonid literature, an estimated dose of 10-30 mg/kg administered IM every 24 hours would be required.

5. **Nitrofurazone.** Dose is 20 mg/L as a 5 hour bath. Repeat daily for 5-7 days. Several soluble forms exist.

6. **Oxytetracycline.** Doses are quite variable (10-70 mg/kg Q 24 hr) and can be given PO, IM, ICe or as an immersion. Consult a formulary for more information.

7. **Trimethoprim sulfadiazine (Tribrissen).** A good broad-spectrum antibiotic. Dose is 30 mg/kg given IM, PO, or IC Q 24 hours for 7-10 days. May also be used as a bath at a concentration of 20 mg/L for 5 hours Q 24 hours.

III. Parasiticides

1. **Diflubenzuron (Dimilin®).** Dose is 0.01 mg/L for 48 hours, 3 treatments, 7 days apart. Very effective but may kill desirable environmental invertebrates. Must have an EPA license to administer. Anecdotal reports exist where veterinarians are prescribing lufenuron (Program®) at a similar dose with good success.

2. **Fenbendazole (Panacur®).** A good parasiticide for intestinal nematodes. Can be given orally at a dose of 50-100 mg/kg and repeated in 2 weeks. Also can be used in a gel food at a concentration of 0.2% (200 mg/100 grams food) and fed for 3 days, repeated in 2 weeks.

3. **Formaldehyde.** Bath dose is 20-25mg/L for 12 to 24 hours followed by a 50% water change. A conventional dose which is commonly used is 1.0 ml of 100% formalin (37% formaldehyde)/ 10 gallons (38 L) of water. Encysted parasites like "Ich" and "Cryptocaryon" will require several treatments. Effective for some ectoparasites in koi as a 10-minute dip at a concentration of 100-150 mg/L (Noga, 2000). Monitor fish closely for signs of distress. Always change water between treatments.

4. **Levamisole phosphate (Ripercol®).** Dose is 2 mg/L for 12-24 hours as a bath treatment.

5. **Metronidazole (Flagyl®).** Normally used as a bath treatment at 10 mg/L Q 24 hours for 3 consecutive days with 25-50% water changes between treatments. Also can be used in a gel food at a concentration of 0.2% (200 mg/100 grams food) and fed for 10 days.

6. **Praziquantel (Droncit®).** Used to treat cestode and trematode parasites. Dose is 5-8 mg/kg IM, IC, or PO repeated in 2 weeks. Bath treatment is 5-10 mg/L for 3-6 hours, repeat in 7 days.

Trichlorfon. This organophosphate is effective in controlling crustacean ectoparasites at a dose of 0.5 mg/L. Commercially available compounds include Neguvon and Malthion. A BIOTEST IS STRONGLY RECOMMENED WHEN USING THESE COMPOUNDS. Treatment is usually repeated several times with 25-30 % water changes between treatments.

7. **Salt.** Safe for most species of ornamental fishes at a dose of 1 gram/L as an indefinite bath. Goldfish and koi will tolerate concentrations from 3-5 grams/L. Used as a general tonic for stress, and control of external parasites (2 grams/L will usually kill circulating protozoal parasites). Plants may be harmed at concentrations over 0.5 grams/L.

8. **Water.** Ectoparasitic protozoans must be treated with a dip or bath. Options: Many freshwater species will tolerate a 4-5 minute dip in full strength (30-35 g/liter) **seawater**. Marine fishes can be placed in a **freshwater** dip for 4-5 minutes. Aerate well and monitor very closely. Certain smaller fishes may not survive this treatment. If possible, test treatment on one fish first.

SUGGESTED READING:

Carpenter, J.W., Mashima, T.Y., and D.J. Rupiper. *Exotic Animal Formulary, 2nd Ed.* Saunders, Philadelphia, 2001.

Doi, A, Stoskopf, MK, and G.A. Lewbart. 1998. Pharmacokinetics of oxytetracycline in the red pacu (*Colossoma brachypomum*) following different routes of administration. Journal of Veterinary Pharmacology and Therapeutics, 21:364-368.

Gratzek JB: An overview of ornamental fish diseases and therapy. J Small Anim Pract 22:345-366, 1981.

Gratzek JB: Control and therapy of fish diseases. Adv Vet Sci Comp Med 27:297-324, 1983.

Herwig N: Handbook of Drugs and Chemicals Used in the Treatment of Fish Diseases. Charles C. Thomas, Springfield IL. 272 pp. 1979.

Lewbart G.A. 2005. Fish; Exotic Animal Formulary, J. Carpenter editor, Elsevier Publishing, pp. 5-29.

Lewbart G.A., Papich M.G., and D. Whitt-Smith. 2005. Pharmacokinetics of florfenicol in the red pacu (*Piaractus brachypomus*) after single dose intramuscular administration. Journal of Veterinary Pharmacology and Therapeutics, 28:317-319.

Lewbart G.A., Butkus D.A., Papich M., Coleman A.K., Krum H., and E.J Noga. A simple catheterization method for systemic administration of drugs to fish. Journal of the American Veterinary Medical Association 226(5):784-788.

Noga EJ: Fish Disease: Diagnosis and Treatment. Iowa State University Press, 367 pp., 2000.

Stoskopf MK: Fish Medicine. Saunders Co., Philadelphia, 882 pp., 1993.

(These notes are an updated version of a proceedings article from the 1999 ACVC Conference)

PET FISH CLINIC SUPPLIES

Gregory Lewbart M.S., V.M.D., Dipl. ACZM
greg_lewbart@ncsu.edu

INTRODUCTION:

The keeping of fish as pets is a hobby which has experienced increasing popularity over the past decade. In fact, more pet fish are kept as pets in the United States than any other single group of animals.1,2 In recent years and a growing number of veterinarians are gaining clinical experience with pet fish. 3,4 The same fundamental disciplines such as critical care, microbiology, parasitology, nutrition, pathology, and surgery that are applied to terrestrial animals can be applied to pet fish as well. The final ingredient to successful and rewarding pet fish practice is making your clinic "fish friendly." The following list of supplies, including key references, will provide the interested clinician with the tools necessary to work up that first fish case!

THE WATER TEST KIT:

An accurate and properly functioning test kit is probably the veterinary clinician's most valuable diagnostic tool. The basic pet fish diagnostic laboratory must be equipped to test for temperature, ammonia, nitrite, nitrate, pH, dissolved oxygen and total alkalinity. Test kits that measure copper and chlorine are also desirable. Heavy metal and miscellaneous toxin testing can be performed by most state diagnostic labs. Several companies manufacture test kits which accurately and inexpensively test the appropriate water quality parameters. Here is a list of suppliers of quality kits:

1. Hach Company, P.O. Box 389, Loveland, CO 80539, 1-800-227-4224 (www.hach.com)
2. Gilford Instrument Labs, Inc., Ovelin, OH 44074, 216-774-1041
3. La Motte Chemicals, P.O. Box 329, Chestertown, MD 21260, 301-778-3100
2. Orion Research, Inc., 840 Memorial Drive, Cambridge, MA 02139, 617-864 –5400 (www.orionres.com)

CLINICAL TEXTS:

Brown L: Aquaculture for Veterinarians. Pergamon Press, Oxford, England, 1993, 447 pp.

Butcher RL: Manual of Ornamental Fish. British Small Animal Veterinary Association. Gloucestershire, England, 1992, 200 pp.

Gratzek, JB: Aquariology: The Science of Fish Health Management. Morris Plains, NJ: Tetra Press, 1992, 330 pp.

Johnson, EJ: Koi Health and Disease. Johnson Veterinary Services, 3805 Robinson Rd., Marietta, GA 30068, 1997, 141 pp.

Lewbart, GA: Self-Assessment Color Review of Ornamental Fish. Iowa State University Press, 1998, 192 pp.

Noga, EJ: Fish Disease: Diagnosis and Treatment. Mosby, 1996, 340 pp.

Spotte S: Captive Seawater Fishes. New York, Lea and Febiger, 1992, 942 pp.

Stoskopf, MK: Fish Medicine. Philadelphia: W.B. Saunders Company, 1993, 882 pp.

Wildgoose, WH: BSAVA manual of Ornamental Fish (2nd ed.). British Small Animal Veterinary Association, Gloucester, England, 2001, 304 pp.

CLINIC SUPPLIES
EQUIPMENT:
Water test kit
Water sample bottles (plastic, 250 ml)
Refractometer
Rubberbands
Oxygen tank with regulator
Compound microscope
Assorted sizes of plastic fish bags
Assorted nets
Complete dissecting kit
Gram scale (to 1 kg)
Sterile surgical pack(s)
5 gallon bucket
Centrifuge
Assorted plastic totes/sweater boxes
Assorted glass aquaria
Sponge filters
Air pumps
Air tubing

DRUGS AND OTHER COMPOUNDS:
Amikacin
Atropine
Calcium gluconate
Ceftazidime
Commercial dechlorinator
Dexamethasone injectable
Emeraid II
Enrofloxacin 22.7 mg/ml injectable
Enrofloxacin 22.7 mg tablets

Epinephrine
Eugenol (clove oil) 1:9 with 95% ethanol (stock approx. 100 mg/ml)
Euthanasia solution
Fenbendazole
Furosemide
Heparin 1000 u
High energy/protein dog food (1 small can)
Lidocaine 2%
Lubricating jelly
Metronidazole
MS 222 (10 mg/ml buffered stock solution)
10% Neutral buffered formalin
Nexaband adhesive
Nitrofurazone
Nolvasan (dilute and nondilute)
PanalogR ointment
Povidone iodine ointment
Praziquantel tablets and injectable solution
Sea Salt (5 lbs)
Silver sulfadiazine cream
Sterile water
Trichlorfon 8%
Trimethoprim/Sulfamethoxazole tablets 960 mg

DIAGNOSTIC MATERIALS:
Glass slides
Coverslips
Assorted needles: 18g, 20g, 22g, 23g, 25g, 26g
Assorted syringes: 1 cc, 3 cc, 6 cc, 12 cc, 35 cc, 60.cc with lure or eccentric tip
Nonsterile 2X2 and 4X4 pads
Plastic pipettes
Microcentrifuge with putty
Microhematocrit tubes
2.5 ml blood collection tubes
- red top (clot tube)
- green (heparin)
- yellow (sodium chloride)

Microcontainer tubes (serum separator)
Bullet tubes (eppendorf) 1.5 ml
Culturettes
- minitip
- portacult
- regular
- ARD bottle

Assorted suture materials
Florescence strips

Assorted red rubber catheters 5 French-12 French
KimwipesR
Paper towels
Tongue depressors
Nonsterile gloves
Assorted scalpel blades

REFERENCES:

Lewbart GA. Pet fish medicine beckons to pioneer veterinarians. Veterinary Product News, March: 50-51, 1999.

Pet Industry Joint Advisory Council (PIJAC), PIJAC National Office Suite 400, 1220 19th Street, NW, Washington, DC 20036, 1997.

Purchase HG, Gloyd J, and Pitts JL: Opportunities for veterinarians in aquaculture. Journal of the American Veterinary Medical Association, 202(5):734-737, 1993.

Results of the AVMA survey of US pet-owning households on companion animal ownership. Journal of the American Veterinary Medical Association, 211(2):169-170, 1997.

Smith C: Pet fish medicine offers new challenges. Journal of the American Veterinary Medical Association, 205(9):1267-1271, 1994.

(These notes are an updated version of a 1999 ACVC Proceedings article)

Content of pages **127 – 133** excluded due to copyright restrictions. Blank page 134 removed.

The citation for the excluded content is: **Lewbart GA. Reproductive medicine in koi (Cyprinus carpio).Veterinary Clinics of North America Exotic Animal Practice. 2002 Sep;5(3):637-48, vii.**

This material is available in the collection of the NCSU Libraries.

For more information about access to the NCSU Libraries, visit www.lib.ncsu.edu.

KOI HERPES AND SPRING VIREMIA OF CARP

Gregory Lewbart M.S., V.M.D., Dipl. ACZM
greg_lewbart@ncsu.edu

(Draft of a 2005 North American Veterinary Conference Proceedings)

Introduction

Spring Viremia of Carp (SVC) is a reportable disease caused by a rhabdovirus (*Rhabdovirus carpio*) frequently referred to as Spring Viremia of Carp Virus (SVCV). Until recently (2002) SVCV had not been found in North America and is considered a foreign animal disease (FAD) in the United States. In the summer of 2002 the disease was identified in koi from a North Carolina fish farm; there have been subsequent confirmed cases in other states. Mortality may reach 100% but is frequently much less. Younger fish are more susceptible than older fish and infected fish typically present with abdominal ascites and multiple organ hemorrhages. Transmission is horizontal and most cases occur in the spring or early summer when the water begins to warm. Diagnosis is usually made with viral isolation (from spleen and/or caudal kidney) and/or serum antibody titers. Diagnosis should be confirmed with virus neutralization.[1] The disease is not restricted to koi and actually can affect several carp species and some other cyprinids. All suspect cases should be necropsied and the United States Department of Agriculture (USDA) contacted for proper routing of diagnostic samples. Confirmed cases must be reported to the USDA. A complete summary of the disease and diagnostic procedures can be found on the Office International des Epizooties (OIE) web site listed below. It is important to note that SVCV infected fish may also be suffering from a Gram-negative bacterial infection. Petty et al.[2] provide a detailed summary SVCV.

Koi herpes virus (KHV) is a recently described disease of koi that is frequently fatal. KHV is not a reportable disease. The virus causes a severe branchitis and the affected fish literally die of hypoxia. Since 1997, outbreaks of this disease have been reported in many European countries, Israel, Asia (including Japan), South Africa, and the U.S.[3-6] Two references provide excellent reviews of the subject.[6,7] There is a report with encouraging results that a live attenuated vaccine for this disease in koi may be practical.[8] There is also anecdotal evidence that koi "heated" to at least 30 degrees C (86 degrees F) may be resistant to an outbreak since the virus appears restricted to environmental temperatures of 18-26 degrees C (64.4-86 degrees F).[6] While this heat "therapy" may save the fish, it could produce sub-clinical carriers of the disease. Since the KHV genome has been determined, other vaccination options are being explored.[6] Gross clinical signs include enophthalmia, mottled gills, excessive mucus production, and dermatitis.[6,7] Mortality frequently exceeds 80% and is restricted to carp.[7] Diagnosis is made by virus isolation and confirmed by PCR of the viral isolates. Even without isolation of the virus PCR can be used to detect the DNA of virus in fresh, infected tissues.[5,9]

One of the best ways to prevent the transmission of these two viral diseases is through adequate quarantine and biosecurity protocols. All new koi should be quarantined for a minimum of 30 days before introduction to an established population. Biosecurity, in the form of good hygiene and proper disinfection procedures, will greatly reduce the spread of these and other infectious diseases of koi.

References:

1. Office international des epizooties (http://www.oie.int/), 2002.

2. Petty PD, Riggs AC, Klinger R, Yanong RPE, and R Francis-Floyd. Spring viremia of carp. University of Florida IFAS Extension (http://edis.ifas.ufl.edu), 2004 (VM-142).

3. Hedrick RP, Gilad O, Yun S, Spangenberg JV, Marty GD, Nordhausen RW, Kebus MJ, Bercovier H, and A Eldar. A herpesvirus associated with mass mortality of juvenile and adult koi, a strain of common carp. Journal of Aquatic Animal Health 2000;12:44-57.

4. Perelberg A, Smirnov M, Hutoran M, Diamant A, Bejerano Y and M Kotler. Epidemiological description of a new viral disease afflicting cultured *Cyprinus carpio* in Israel. The Israeli Journal of Aquaculture 2003;55(1:5-12.

5. Gilad O, Yun S, Andree KB, Adkison MA, Zlotkin A, Bercovier H, Eldar A, and RP Hedrick. Initial characteristics of koi herpesvirus and development of a polymerase chain reaction assay to detect the virus in koi, *Cyprinus carpio* koi. Dis Aquat Organ 2002;48(2):101-8.

6. Waltzek TB and RP Hedrick. Koi herpesvirus update 2004. California Veterinarian 2004; July-August issue.

7. Hartman KH, Yanong RPE, Petty D, Francis-Floyd R, and AC Riggs. Koi Herpes Virus (KHV) Disease. University of Florida IFAS Extension (http://edis.ifas.ufl.edu), 2004 (VM-149).

8. Ronen A, Perelberg A, Abramowitz J, Hutoran M, Tinman S, Bejerano I, Steinitz M, Kotler M.
Efficient vaccine against the virus causing a lethal disease in cultured *Cyprinus carpio*. Vaccine 2003;21(32):4677-84.

9. Gray WL, Mullis L, LaPatra SE, Groff JM, and A Goodwin. Detection of koi herpesvirus DNA in tissues of infected fish. J Fish Dis 2002;25:171-178.

STANDARD OF CARE: PET FISH

Gregory Lewbart M., VMD, Dipl. ACZM
greg_lewbart@ncsu.edu

M. Andrew Stamper DVM, DACZM
Disney's Living Seas
Orlando, FL USA

(Draft of a 2005 North American Veterinary Conference Proceedings)

Introduction

The keeping of fish as pets is a hobby with a long history. In recent years, particularly during the past decade, ornamental pond fish including koi and goldfish have become increasingly popular in various parts of the world. In fact, more pet fish are kept as pets in the United States than any other single group of animals including dogs, cats, small mammals, birds, and reptiles (AVMA, 1997; PIJAC, 1997). The hobby has also become more sophisticated in recent years and a growing number of veterinarians are gaining clinical experience with pet fish. Until the 1980's, except in rare instances, most of the medical care and husbandry practices were performed by the hobbyists themselves or with the help of the local pet store clerk or aquarium maintenance person. Many of these people are very knowledgeable and conscientious but there are no minimal training standards as there are in the veterinary profession. Additionally, we as veterinarians understand the principles of medicine, surgery, and animal husbandry. The same fundamental disciplines such as critical care, microbiology, parasitology, nutrition, pathology, and surgery that are applied to terrestrial animals can be applied to pet fish.

With regards to the Standard of Care (SOC) for this group of animals, koi, goldfish, and all commonly kept freshwater and marine bony fishes should be included. Sharks and other elasmobranchs, while occasionally kept in the home aquarium, are more frequently managed by aquarium and zoo veterinarians.

As our knowledge of fish diseases, therapeutics and water quality increase, more and more veterinarians will be qualified to responsibly work with these animals. Peer-reviewed articles on the clinical management of pet fish issues now appear in many veterinary journals. Nearly every major veterinary conference includes fish medicine in the program and several veterinary schools now offer continuing education courses on this subject. In addition, a growing number of textbooks and review articles contain valuable information on pet fish medicine. All veterinarians, even if they have never worked on a fish, have a broad understanding of disease processes, diagnostics, animal husbandry, and chemotherapeutics. The opportunity to apply this knowledge to a client's pet fish problem can be a rewarding experience. Currently, some clinicians report that 10% or more of their income is generated by fish patients.

Minimal Standards
Continuing Education (CE)
Veterinarians treating pet fish should maintain their state licensure(s) and attend at least one CE session focused on fish medicine every other year. It is recommended that fish veterinarians be an active member of the International Association for Aquatic Animal Medicine (IAAAM), American Association of Zoo Veterinarians (AAZV (or both), and the American Veterinary Medical Association (AVMA). The American Fisheries Society (AFS) is an excellent organization and dedicates resources to fish medicine and pathology.

Pet fish veterinarians are also encouraged to join local organizations, such as aquarium and garden pond clubs, and to attend or make presentations when possible.
A number of annual courses and programs (listed below) are available to the interested veterinarian and veterinary student, and while not mandatory, would be a good way to round out one's education.
-Regularly offered Continuing Education Programs:
Advanced Fish Medicine (University of Florida); every other year
*Aquavet I&II (University of Pennsylvania/Cornell University)
*Aquamed (Louisiana State University); every other year (even years; next in 2006)
Diseases of Warmwater Fishes (University of Florida); every other year
Fish Health Management Course (North Carolina State University)
-Annual Meetings of Note:
*AAZV
*Eastern Fish Health Workshop
*IAAAM
*NAVC
*WVC
-Internship Programs:
*New England Aquarium
*Mystic Marinelife Aquarium
*National Aquarium in Baltimore
*Florida Aquarium/UFL Tropical Aquaculture Laboratory
*Shedd Aquarium
Michigan State University
Mississippi State University (catfish medicine)
-Residency Programs:
North Carolina State University
University of Florida
*Not exclusively fish medicine

Staff training- Many veterinary meetings (NAVC, WVC, AVMA, etc.) regularly offer CE for veterinary technicians. In house training is recommended and can be used to supplement knowledge gained at CE meetings.

Recommended Library **(Note: Information is constantly changing and it is imperative to update your library with the current literature)**

Textbooks

-Brown L: Aquaculture for Veterinarians. Pergamon Press, Oxford, England, 1993, 447 pp.
-Carpenter, J et. al. Exotic Animal Formulary, Third Ed. Elsevier, Philadelphia, 2005.
- Ferguson HW. Systemic Pathology of Fish. Iowa State University Press, Ames, Iowa, 1989.
-Fowler ME, Miller RE (eds). *Zoo & Wild Animal Medicine: Current Therapy 4*. WB Saunders Co, Philadelphia, 1999.
-Gratzek, JB: Aquariology: The Science of Fish Health Management. Morris Plains, NJ: Tetra Press, 1992, 330 pp.
-Johnson, EJ: Koi Health and Disease. Johnson Veterinary Services, 3805 Robinson Rd., Marietta, GA 30068, 1997, 141 pp.
-Lewbart, GA: Self-Assessment Color Review of Ornamental Fish. Iowa State University Press, 1998, 192 pp.
-Noga, EJ: Fish Disease: Diagnosis and Treatment. Iowa State University Press, 2000, 340 pp.
-Roberts RJ. Fish Pathology. WB Saunders, Philadelphia, 1989.
-Saint-Erne, N. Advanced Koi Care, Erne Enterprises, Glendale, AZ, 2003, 194 pp.
-Spotte S: Captive Seawater Fishes. New York, Lea and Febiger, 1992, 942 pp.
-Stoskopf, MK: Fish Medicine. Philadelphia: W.B. Saunders Company, 1993, 882 pp.
- The Merck Veterinary Manual, Eighth Edition, Merck & Co., Inc., National Publishing Co., Philadelphia, PA, 1998.
-Treves-Brown KM. *Applied Fish Pharmacology*. Kluwer Academic Publishers, Dordrecht, The Netherlands, 2000.
-Wildgoose WH (ed): BSAVA Manual of Ornamental Fish, 2nd ed., Gloucester, England, 2001.

Journals

-Diseases of Aquatic Organisms
-ExoticDVM Magazine
-Journal of Aquatic Animal Health
-Journal of Fish Diseases
-Journal of the AVMA
-Journal of Zoo and Wildlife Medicine
-Seminars in Avian and Exotic Pet Medicine. Fudge, A., ed. Elsevier.
-Veterinary Clinics of North America: Exotic Animal Practice. Elsevier
-Veterinary Record

Hospital Physical Plant

Hospitalizing fish patients can be a challenge but will be necessary on occasion. A variety of aquarium support equipment and supplies including tanks, pumps, filters, nets, siphons, heaters, and water conditioners are required. In many situations the owner of the fish can provide some or all of the necessary materials, including conditioned biological

filter substrate, for the hospitalized patient. A room or portion of a room dedicated to fish life support is ideal. The majority of equipment and supplies listed below can be used in the hospital and in the field.

Equipment and Supplies (not including most drugs and standard clinic supplies--a more detailed list appears in the Pet Fish Clinic Supplies section of this notebook)

Husbandry

Air pumps
Air tubing
Assorted plastic totes/sweater boxes
Assorted glass aquaria
Assorted sizes of plastic fish bags
Assorted nets
Commercial dechlorinator
Rubberbands
Sea Salt
Sponge filters
Water test kit
Water sample bottles (plastic, 250 ml)
5-gallon bucket(s)

Medical

Centrifuge
Complete dissecting kit
Compound microscope
Eugenol (clove oil) 1:9 with 95% ethanol (stock approx. 100 mg/ml)
Fish anesthesia machine
Gram scale (to 1 kg)
KG scale (to 10 kg)
MS-222 (10 mg/ml buffered stock solution)
Oxygen tank with regulator
Plastic surgical drapes
Refractometer
Sterile surgical pack(s)

Clinical Assessment

-A complete history should be taken on each patient or each population of patients.
-Fish should be first given a gross "in tank" or "in pond" inspection if appropriate followed by a thorough physical examination.
-In the majority of cases the aquarium or pond water should be tested. An accurate and properly functioning test kit is probably the fish clinician's most valuable diagnostic tool. The basic pet fish diagnostic laboratory must be equipped to test for temperature, ammonia, nitrite, nitrate, pH, dissolved oxygen and total alkalinity. Test kits that measure copper and chlorine are also desirable. Most state diagnostic labs can perform heavy metal and miscellaneous toxin testing. Several companies manufacture test kits that accurately and inexpensively test the appropriate water quality parameters.

Procedures
-The following list of procedures, while not applicable in every case, should be available and the practitioner should be comfortable with utilizing them or alternatively know where/when to refer.

Anesthesia/sedation
Cloacal lavage Celiotomy
Coelomic lavage
Enucleation
Eye aspiration
Fin biopsy
Gas bladder lavage
Gastric lavage
Gill biopsy
Intracoelomic (ICe/IP) and intramuscular (IM) injection sites
Microbiological sample collection (frequently kidney, spleen, liver)
Necropsy
Skin biopsy
Surgical wound repair
Tube-feeding
Venipuncture

Drugs (Refer to the Formulary section of this notebook for more details)
There are currently no drugs approved by the FDA for use in pet fish (although a small number are approved for use in fish intended for human consumption) in the United States. Many commonly employed veterinary compounds, including antibiotics, parasiticides, and disinfectants, should be on hand and available for use with pet fish patients. The fish practitioner should also be aware of the wide use of over-the-counter (OTC) drugs by pet fish hobbyists. An effort should be made to learn about these OTC compounds and have a basic understanding of their ingredients and impact on the patient and its environment.

Laboratory support
Fish clinicians should identify a laboratory that is familiar and comfortable handling fish samples, especially those related to clinical pathology and microbiology. In some cases this may require more than one laboratory.

Miscellaneous
With literally hundreds of species kept in captivity from a variety of (sometimes) unrelated families, it is understood that the clinician will not be familiar with every species of fish with which they are presented.

Fish clinicians should:
-Be familiar with zoonoses and be able to speak intelligently to their clients on this topic
-Cultivate a list of colleagues to share information and seek consultation

-Have a thorough understanding of the natural history, anatomy, and physiology of the major groups of pet fishes (eg. goldfish, koi, cichlids, livebearers, anabantoids).

Summary

Fish medicine is a growing and rewarding area of our profession. Many fish owners have strong emotional bonds to their fish and they seek state-of-the science care and support for their sick or injured pets. The fish medicine knowledge base is small compared to that of small animals or even some of the other exotic taxa. All fish practitioners are encouraged to contribute to this base of knowledge, either by publishing their findings or sharing information and discoveries with colleagues. The Fish Practice SOC document is not comprehensive or permanent and will continue to change, grow, and develop over time.

EVENT PARTICIPANT LIST

Fish Health Management
July 21, 2005 - July 23, 2005

1. Jerome Albright
   Priority One Services
   6600 Fleet Dr
   Alexandria, VA 22310
   703-971-5504
   jerome@priorityoneservices.com

2. Geni Barnes
   Virginia Living Museum
   308 Mary Bierbauer Way
   Yorktown, VA 23693
   757-595-8326
   geni@cox.net

3. Heather Bates
   NC Aquarium at Roanoke Island
   8 Croatan Ct
   Manteo, NC 27954
   252-202-1604
   heather.bates@ncmail.net

4. Steve Buchholz
   Apet
   515 Dublin
   Mundelein, IL 60060
   874-877-0505
   sbuchholz@apetinc.com

5. L. Adam Church
   NC Museum of Natural Science
   1021 Avent Hill Apr B3
   Raleigh, NC 27606
   919-200-8945
   church_a@hotmail.com

6. Jason Crichton
   South Carolina Aquarium
   100 Aquarium Wharf Road
   PO Box 130001
   Charleston, SC 29412
   843-720-1990
   jcrichton@scaquarium.org

7. Dr. Stephen Dempsey - IACUC
   NC State University
   2701 Sullivan Dr Box 7514
   Raleigh, NC 27695
   919-513-8142
   steve_dempsey@ncsu.edu

8. Dr. Thomas Edling
   PETCO Animal Supplies
   9125 Rehco Road
   San Diego, CA 92121
   858-453-7845 x4385
   thomase@petco.com

9. Dr. Karen J. Fischer
   I-20 Animal Medical Center
   5820 W I-20
   Arlington, TX 76016
   817-492-8621
   karen.fischer@comcast.net

10. Kate Freeman
    CVM-Class of 2008
    4700 Hillsborough Street
    Raleigh, NC 27606

EVENT PARTICIPANT LIST

Fish Health Management
July 21, 2005 - July 23, 2005

11. Dr. Rae Gandolf
Pittsburgh Zoo & PPG Aquarium
One Wild Place
Pittsburgh, PA 15206
412-365-2561
rgandolf@pittsburghzoo.org

12. Dr. Kari Granier
USDA-APHIS-VS Georgia
129 Colonial Cir NW
Cartersville, GA 30120
770-363-8376
kari.l.granier@aphis.usda.gov

13. Amanda Grant
University of Illinois
406 Park Cir
St. Joseph, IL 61873

algrant@uiuc.edu

14. Dr. Lori J. Heintzelman
P.O. Box 2049
Lebanon, VA 24266

tirlith@hotmail.com

15. James Irving
Whetstone Creek Koi
1028 NC 704
Madison, NC 27025
336-613-7527
support@whetstonecreekkoi.com

16. Robert B. Irving
Irving Fish Hatchery
663 NC 704
Madison, NC 27025
336-427-7171
irving_fish_hatchery@yahoo.com

17. Jack Jewell
Shark Reef at Mandalay Bay
3950 Las Vegas Blvd S
Las Vegas, NV 89119
702-632-4560
jjewell@mrgmail.com

18. Brian Joseph
3990 Old Town Ave #208A
San Diego, CA 92110
619-686-5706
josephbri@saic.com

19. Philip Jull
Old Marple Veterinary Clinic
10 S Kirklyn
Upper Derby, PA 19082-2602
610-457-8160
pjull@hotmail.com

20. Rachel Kalisperis
South Carolina Aquarium
100 Aquarium Wharf
PO Box 130001
Charleston, SC 29412
843-720-1990
rkalisperis@scaquarium.org

EVENT PARTICIPANT LIST

Fish Health Management
July 21, 2005 - July 23, 2005

21. Melanie Landry
CVM-Class of 2008
4700 Hillsborough Street
Raleigh, NC 27606

mmlandry@ncsu.edu

22. Allen Lee
900 Club Dr
Monroe, NC 28112

allen.lee@siemens.com

23. Raymond Massey
New Year Tech at NIEHS
111 TW Alexander Dr MC: C0-01
RTP, NC 27709
919-541-3676
massey2@niehs.nih.gov

24. Todd Matejovich
Priority One Services
6600 Fleet Dr
Alexandria, VA 22310
703-971-5504
todd@priorityoneservices.com

25. Kevin Mathis
NC Aquarium at Fort Fisher
900 Loggerhead Road
Kure Beach, NC 28449
910-458-8257
kevin.mathis@ncmail.net

26. Ryan McAlarney
NC Aquarium at Fort Fisher
900 Loggerhead Road
Kure Beach, NC 28449
910-458-8257
ryan.mcalarney@ncmail.net

27. Dr. Brigitte Mercier
SPSNQ - Jardin Zoologique & Parc
9530, rue de la Faune
Charlesbourg, QC G1G 5H9
418-622-0313 x261
brigitte.mercier@spsnq.qc.ca

28. Mitchell A. Morton
Foster Lake & Pond
183 Denmoor Ct
Garner, NC 27529
919-772-8548
mitch@fosterlake.com

29. Laura Muha
133 W Millstream Rd
Cream Ridge, NJ 08514
609-758-6533
lmuha@aol.com

30. Mary Orr
Tetra
3001 Commerce St
Blacksburg, VA 24060
540-951-5518
mary.orr@tetra.net

EVENT PARTICIPANT LIST

Fish Health Management
July 21, 2005 - July 23, 2005

31. Dr. Richard O. Overton
USDA
190 Miles Rd
Metter, GA 30439-5508
912-685-9742
notrevorich_2000@yahoo.com

32. Dr. James W. Provo
USDA-APHIS-VS
4639 Long Valley Road
Greensboro, NC 27410
336-643-5061

33. Britton N. Purtee
PO Box 2606
Manteo, NC 27954
252-473-3233
wolf_obsidian@hotmail.com

34. Lory Scott
Virginia Living Museum
3510 Fontwell Ct
Suffolk, VA 23435
757-534-7484
lory.scott@valivingmuseum.org

35. Maria Serrano
CVM-Class of 2008
4700 Hillsborough Street
Raleigh, NC 27606

meserran@ncsu.edu

36. Brian Shannon
NC State University
207 Research Station Rd
Plymouth, NC 27962
252-793-4428 x162
dbshanno@unity.ncsu.edu

37. Dr. Julie Sheldon
Oklahoma State University, College of
1807 N Hartford Apt 914
Stillwater, OK 74075
405-269-6555
juliesheldon@aol.com

38. Laura Shewmon
1315 Pineview Dr
Raleigh, NC 27606

lshewmo@unity.ncsu.edu

39. Dr. Raghu Shivappa
CVM-PHP Department
4700 Hillsborough Street
Raleigh, NC 27606
919-513-6608
raghunath_shivappa@ncsu.edu

40. Dr. Rosemary B. Sifford
USDA-APHIS-VS
930 Main Campus Dr Ste 200
Raleigh, NC 27606
919-855-7711
rosemary.sifford@aphis.usda.gov

EVENT PARTICIPANT LIST

Fish Health Management
July 21, 2005 - July 23, 2005

41. Jeffrey Smith
    NC Museum of Natural Science
    5740 Poolside Dr
    Raleigh, NC 27612
    919-733-7450 x519
    jeffrey.smith@ncmail.net

42. Jessica Sprague
    University of Illinois - Urbana-Champaign
    209 S Dodson Dr
    Urbana, IL 61802

    jlyons2@uiuc.edu

43. Dr. Suzanne Storey - Drug Approval
    FDA-CVM
    HFV-131
    Rockville, MD 20855
    301-827-7581
    sstorey@cvm.fda.gov

44. Dr. Rhonda Washington
    Alamance Community College
    320 Severin Street
    Chapel Hill, NC 27516
    919-929-4682
    rlwashin@uncg.edu

45. Dr. Christopher E. Yach
    West Flamingo Animal Hospital
    5445 W Flamingo Rd
    Las Vegas, NV 89103
    702-876-2111
    wfah@earthlink.net

Participant Count: 45