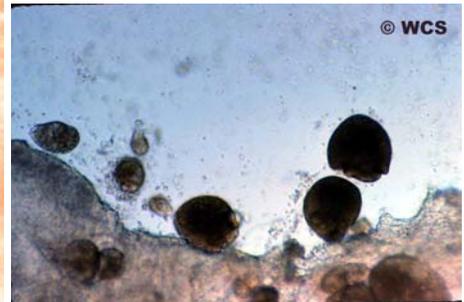
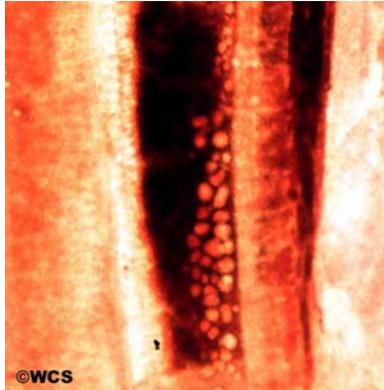


Fish Necropsy for students.

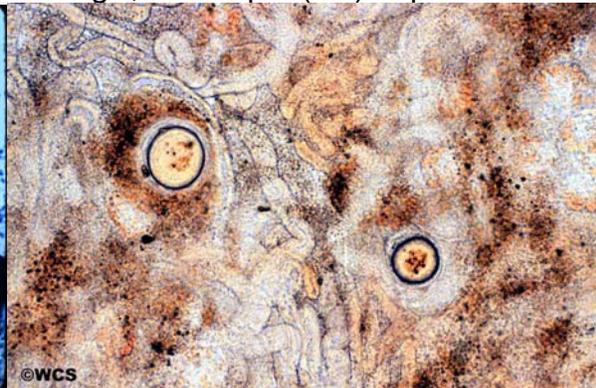
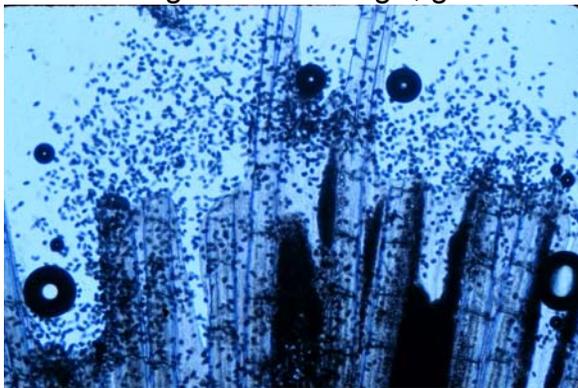
1. Identify the fish species
2. Measure the length and sex (if possible) and record on sheet along with the date, tank, keeper and your initials (most of this should already be on a mort card)
3. Place the fish in an appropriate container: petri dishes for small ones, trays for bigger, necropsy table for largest. If fresh, cover with water from the tank the fish was in, or aged tapwater if not.
4. Give the fish a good external look over. Is the colour normal for the species/sex? Is there a lot of mucus? Is the skin thickened, spotty, damaged or ulcerated? Are the eyes cloudy or haemorrhagic? Are there visible parasites or pathogens, or tufts of fungus (like soggy cotton wool)? Are the fins intact or are they split, eroded or ragged? Record these observations. Make sure to look at both sides of the fish, not just the one that faces upwards. Also look in the mouth, noting any parasites present, damage to the orobranchial chamber or blockages of the oesophagus.
5. Take a scrape of the skin using a round-bladed scalpel like a 22 or 23 (i.e. not an 11). Good places to scrape are the upper flanks, and at the base of the pectoral fin on the tail side (many parasites concentrate here). Place the scraped material on a clean slide with a 22 x 40 or 22 x 50 coverslip over and draw off excess fluid with a tissue. Place on a compound scope at x10 and x20 (rarely higher mag may be needed). Look for motile parasites or excess red blood cells. Record observations.
6. Snip some fin rays from the tip of the tail or anal fin and look at under high power with a coverslip on the slide. Look for motile parasites (especially *Neobenedenia* or *Gyrodactylus*, or white spot), or excess red blood cells. Record observations.
7. Using blunt or flat forceps, lift the trailing edge of the gill cover (operculum) and cut the gill cover from its upper caudal edge diagonally forwards and to the anterior end of the isthmus (on the ventral surface just behind the bottom lip. Lift the operculum away to expose the gills.
8. Snip the branchial arch bones of the top gill (arch 1) at top and bottom, and lift the gill arch out into a petri dish of clean water (fresh or marine, as per the fish in question). Cut the top end of the arch first as the bottom end is the afferent blood vessel and has higher blood pressure. If you cut it first, you risk flooding the gills with blood, which makes microscopical examination next to impossible. Look at the arch you have cut under the dissecting microscope, noting embolisms (bubbles of gas) or telangectasia (basically blood blisters), especially at the tips of gill filaments, epidermal thickening, mucus production, or any parasites present. Look at both sides. Record observations.
9. Snip several filaments from the gill and place on a slide with appropriate coverslip. Examine on compound microscope, preferably with nomarski, DIC or phase contrast interference. Look for parasitic worms (usually with little black eyespots and hooks), parasitic crustaceans (copepods or isopods), fungus, bacteria, proliferations of mucus cells (granular looking cells in the epidermis) or embolisms/telangectasia. Record results.
10. Return to the body, and pick it up in one hand. Make an incision from the trailing margin of the gill chamber, halfway up the flank, and cut through the bone. You may need big scissors or bone clippers for this. Keep the tip of the scissors up or you may damage the kidneys. Having cut through the body wall, continue cutting through ribs and muscle parallel to the spine, then follow the curve of the body cavity down towards the anus. Finish

the cut before the anus is reached. Then turn the body around and cut forwards towards the head along a midventral line. Either come up the flank to the origin of the cut at the level of the pelvic fins (leaving the heart cavity undisturbed), or continue midventrally the full length of the isthmus. You may need to pry apart, but you should be able to lift the whole side of the fish away, revealing the viscera.

11. Note the nature of the body cavity fluid, is it haemorrhagic? If so, take a swab for bacteriology. Put some on a slide with slip and look for bacteria (usually rods, usually motile) or excess blood cells.
12. Identify the oesophagus and sever.
13. Identify the anus and sever the rectum at the body wall.
14. Teasing away any mesenteries, remove the viscera to a separate dish, and this time fill the dish with saline.
15. Leaving that aside, remove the heart to a small dish of saline. Look for haemorrhages on the surface of the heart chamber. Open the heart with scissors and look for clots or parasitic worms (sanguinicolids), which will be flat and move with a quivering swimming motion.
16. Return to the body and identify the head kidneys (if the fish has bipartite kidneys), which are normally a plum colour and lie along either side of the spine in front of the swim bladder. Take a sample with forceps and squash on a slide with a coverslip. Look for granulomas, motile parasitic protozoans, tubule degeneration, bacteria, neoplasias (tumours), or haemorrhage. Take also a sample from the trunk kidneys, further back along the spine.
17. Return to the viscera. Identify the various organs: the liver, which has two lobes (one larger than the other) and is usually a peachy color; the gall bladder, a dark green bag that lies under the liver; the stomach (if present), which is a muscular bag behind the oesophagus; the intestine, winding from stomach towards anus; the rectum, a darker section of intestine just before the anus; and the spleen, a dark red organ much smaller than the liver normally.
18. Look for pale spots, granulomas, cysts, haemorrhage, discoloration, dilation of blood vessels and neoplasias on the liver and spleen.
19. Note the size, shape and colour of the liver. Grossly enlarged or atrophied livers indicate chronic problems. Take a liver squash, see 15. Do the same for the spleen.
20. Note the size of the gall bladder. Large GB indicates failure to eat. Sample the gall bladder bile (or open the bladder itself), see 15.
21. Open the gut from one end to the other, noting the locations of any parasites (usually worms) or blockages identified. To be a blockage, the obstruction must distort the walls of the gut in all directions in its normal state (food will often be present and should not be misinterpreted). Take a scrape from the lining of the intestine or rectum, and examine on a slide for motile protozoan parasites or coccidian oocysts.
22. When finished with the viscera, peel back some skin on one side of the body and look for cysts or irregularities in the musculature, which should be translucent and whitish, except for a darker band along the sides parallel with the spine.
23. Record all details and clean down the dissecting station, making sure to put dirty glassware into soak and rinsing dissecting tools for later washing. Remove slides from microscopes and wipe down if any saline has made its way onto the stage. Dispose of the body in the freezer or appropriate infectious waste disposal.



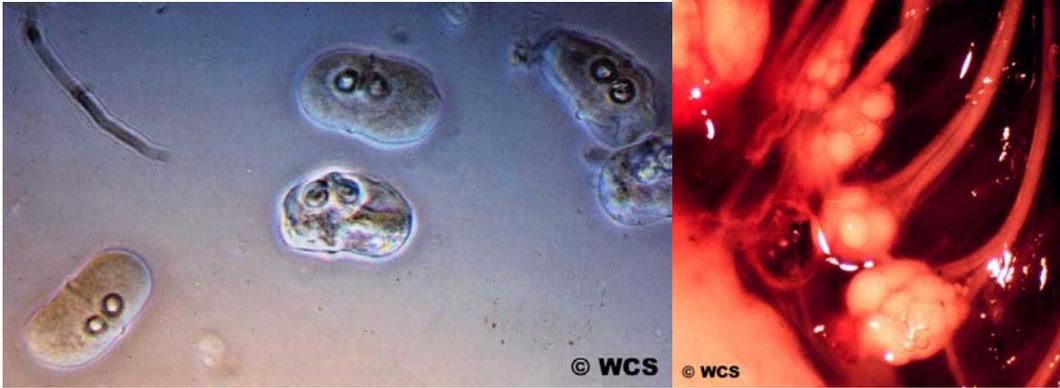
L-R: granulomas in gill, gas embolism in gill, white spot (Ich) trophonts



L-R: Uronema on eroded fin, kidney granulomas



L-R: many monogeneans on a gill (dissector), a monogenean on a gill (high power)

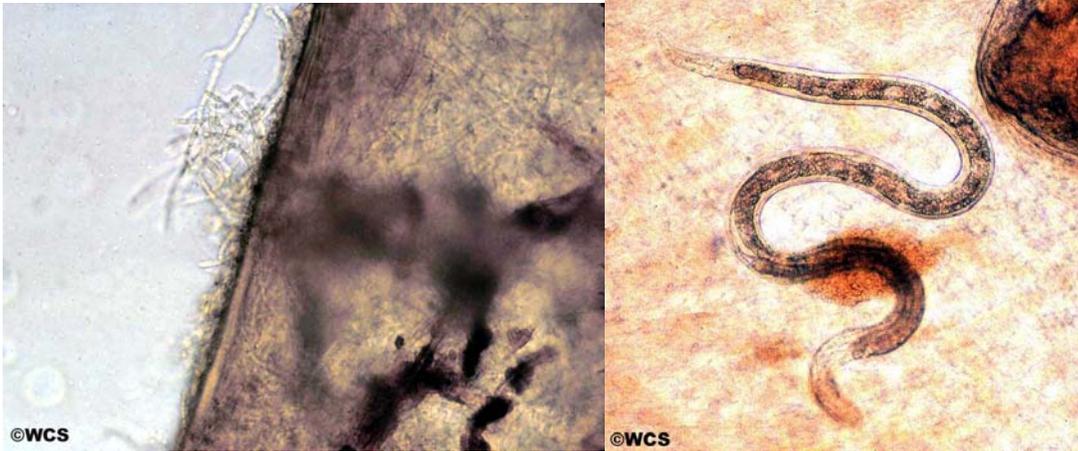


L-R: myxozoans from a gall bladder (common), microsporidian cysts on lateral nerves

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L-R: Sarcoma on swordtail, melanoma on fish tail



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L-R: saprolegnious fungus, nematode from gut



L-R: tapeworms in fish intestine, digenean flukes from fish intestine



L-R: scat with *Lymphocystis* virus, fish heart with necrotic (pale) patches & some haemorrhage.