NOAA FISHERIES SERVICE

Panama City Laboratory
National Marine Fisheries Service

Procedure Manual
for
Age, Growth, and Reproduction Lab

Fisheries Biology Group

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Preface

This manual was first constructed in May of 1999. Prior to this date, no manual or procedures existed for logging in data, or how to interpret the numerous datasheets received by our lab since the late 1970s by various sources (Trip Interview Program, Beaufort Head Boat Survey, Marine Recreational Fisheries Statistics Survey, and Observer Programs, and NOAA Fisheries Service and State of Florida Fishery Independent Surveys). It became crucial for us to document such procedures in order to establish guidelines for logging samples (Chapter 1 and Chapter 2), entering data (Chapter 3), proofing data (Chapter 4), short-term and long-term storage of age structure samples (Chapter 5), sectioning otoliths (Chapter 6), staging gonad samples (Chapter 7-9) and determining fecundity estimates (Chapter 10). In addition, there were no procedures to accurately log-in collections that had been sent to our lab and archived at our facility, with the establishment of source specific Log-In Procedures we are now able to provide an accurate list of age structure samples that exist in our archive (Table 5.1.1). Furthermore, all procedures listed in the following chapters are not only applicable to present day and future collections, but have been applied to historical samples (dating back to the late 1970s) as data is needed to be verified for such species. The justification of the time and effort devoted to this document is proven in the integrity and accuracy of our data.

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Chapter 1: Logging in Otoliths Updated January 2008

This chapter includes instructions on the procedures to log in samples from Panama City Laboratory (PCLAB), Beaufort Head Boat Survey (HB), Marine Recreational Fishery Statistics Survey (MRFSS), Trip Interview Program (TIP), Fishery Independent Survey (MSLAB), Cooperative Research Proposals (CO-OP), North Carolina Department of Marine Fisheries (NCDMR), and Shark Bottom Longline Observer Program (SBLOP). Several agencies are listed in this chapter as data sources (ALMR, FWCLE, SCDNR, VADMR), however due to the minimal number of samples received, they will not be mentioned further in the manual.

It is very important that all logs are correctly filled out and contain the correct information. The process of logging in is a species-specific task allowing for a a count of the number of ageing structure (otolith and/or spine) and gonads received at our facility by species each year, regardless if that ageing structure will be sectioned or aged in the present or future.

This chapter also includes copies of data sheets and envelopes to assist in identifying the materials that are sent to our lab to ensure samples are logged in properly. These processes have been incorporated with present and archive samples.

After the sample (referring to either an ageing structure and/or gonad) is properly logged in, it is necessary to store it in the correct box and location. The procedures for ageing structure storage are located in Chapter 5. The procedures for primary gonad storage are located in Chapter 7.

Also, included is this section is a list of species, abbreviations, and numerical codes (NMFS, NODC) for each species (Table 1).

A detailed explanation of source specific data sheets received are located in Chapter 2.

1.1 Steps to Logging in Otoliths

1.2 Key to Blank Log

Contributors: Meredith Burnette, Linda Lombardi, Beverly Barnett, Carrie Fioramonti

1.1 Steps to Logging in Otoliths

Step 1 Make sure the data sheet you are holding corresponds to the otoliths

Step 2 If there are more than 1 species, make copies and separate all otolith samples by species, highlighting the species on the data sheet that corresponds to the species you are logging in.

Step 3 Find the correct species log book and the next sequential Panama City Collection number for the appropriate year. If the log book does not contain a log sheet for that species, one may be printed out from the Blank Log Sheets folder located on the Age server (I:\Blank Log Sheets). If the species has not been incorporated into the log folder, it must be added to the log book and database (See senior technicians or your supervisor and Chapter 3 Section 3.10).

Step 4 Place a label (format below) on a small yellow post-it (stick on the upper left hand corner of the data sheet), and also write the label in the upper right hand corner of the data sheet with a RED pen.

The label is as follows: for example **98 RS 12**

98: The last two digits for the year of capture
RS: The 2 or 3 letter abbreviations for that species (see Table 1)
12: The corresponding species specific collection number

Step 5 Enter the log data into the log data sheet (Figure 1.1). Examples of data sheets from each source can be found in the appropriate section of Chapter 2: Data Sources.

Step 6 Fold the data sheet such that the collection numbers are visible on both sides after folding into quarters. Wrap all the corresponding otoliths inside the data sheet and place this in a zip-lock bag.

Step 7 Find the appropriate box for that species and collection number. Place the collection in the box.

★ Some species have a particular **INBOX**, located in the Age, Growth, and Reproduction Lab, in which to file the logged collections for further processing. Be aware that these species will change periodically. **king and Spanish mackerel** will always have an INBOX.

Figure 1.1: Blank Log Sheet
Common Name, Scientific Name
NODC #

__ __ __LOG__ __ __ __

PC Coll #	Date	Source	Source #	State	Location	Collector	# of Gonads	# of Otoliths	Cond. (S/W)	Mode	Gear	Comments
						Totals For Page # :						

1.2 Key for Blank Log

Year and Species	Write the last digits of the year and use the appropriate Species Abbreviation (see Table 1)	
PC Coll #	Panama City Laboratory collection number (species specific)	
Date	Date of collection from sampler (mo/da/yr) May be referred to as Date of Interview or Interview Date	
Source	ALMR:	Alabama Marine Resources
	CO-OP:	Cooperative Research Proposal
	FMRI:	Florida Marine Research Institute
	FWCLE:	Florida Wildlife Commission Law Enforcement
	HB:	Beaufort Head Boat
	MRFSS:	Marine Recreational Fisheries Statistical Survey
	MSLAB:	Mississippi Laboratory
	NCDMR:	North Carolina Department for Marine Resources
	PCLAB:	Panama City Laboratory
	POP:	Pelagic Observer Program
	RECFIN:	Recreational Fisheries Information Network
	SCDNR:	South Carolina Department for Natural Resources
	SBLOP:	Shark Bottom Longline Observer Program
	TIP:	Trip Interview Program
	USGS:	United States Geological Survey
	VADMR:	Virginia Department for Marine Resources
Source #	ALMR:	No Source #
	CO-OP:	First 4 digits = 4 digit year – Next 2 digits = first and last initial of captain – Last 2 digits = Trip #
	FMRI:	First 2 or 3 digits = boat name abbreviation – Next 2 or 3 digits = Cruise # – Last 2 or 3 digits = Station #
	FWCLE:	No Source #
	HB:	5 digit Collection number
	MRFSS:	First digit = assignment # (1 or 2) Next 4 digits = Interview # Last 2 digits = Intercept # (01, 02, 03, 04)
	MSLAB:	First 2 or 3 digits = boat name abbreviation –

Next 2 or 3 digits = Cruise # –
Last 2 or 3 digits = Station #

NCDMR 4 digit year
Next 5 digits = Interview #

PCLAB: For samples prior to 1999, see PC Lab section
For samples 1999 to present:
Leave blank, unless Trap Survey or CI Project:
Trap Survey:
First 2 digits = TS
Next 2 digits = 2 digit year (06, 07, 08)
Last 3 digits = Trap Survey #
CI Project
First 2 digits = CI
Next 2 digits – 2 digit year (06, 07, 08)
Last 3 digits = CI Station #

POP 6 digit Trip #

RECFIN First digit = assignment # (1 or 2)
Next 4 digits = Interview #
NIV = No Interview Number

SCDNR Should be associated with TIP (Chapter 2.12)

SBLOP Observer initials – Trip # – Haul #

TIP: For samples prior to 2004, see TIP section
For samples 2004 to present:
First 2 digits = agency #
Next 2 digits = State
Last 6 digits = Intercept #

USGS No Source #

VADMR No Source #

State

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina

	SC	South Carolina
	TX	Texas
	*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)	
	*For Mackerel Samples ONLY (Figure 1.2).	

Location	City/County samples where landed See Chapter 2 for source-specific instructions
Collector	First Initial and Last Name of sampler
# of Gonads**	Total number of gonads in the collection
# of Otoliths**	Total number of otoliths in the collection

**DO NOT record # of Gonads or # of Otoliths into the log if awaiting the samples. Once the samples are received, the # of Gonads and/or Otoliths will be recorded into the log.

Condition (S/W)	Condition of the otolith	
	S = Sectioned	W = Whole

Mode

Mode Abbreviations:

Abbreviation	Mode
CM	Commercial
CP	Charter Party
HB	Head Boat
PR	Private
TRN	Tournament
SS	Scientific Survey

Gear

Gear Abbreviations:

Abbreviation	Gear
HL	Hook & Line, Hand-line, Bandit
LL	Long-line
SP	Spearfish
TR	Trap
TRW	Trawl
GN	Gillnet, Trammel Net
CN	Castnet
VLL	Vertical Longline

Comments

Any other information not listed above
For example: Photos, Reef Site for PC Lab samples from Carrabelle or Steinhatchee, other location comments

Figure 1.2: Mackerel State Map

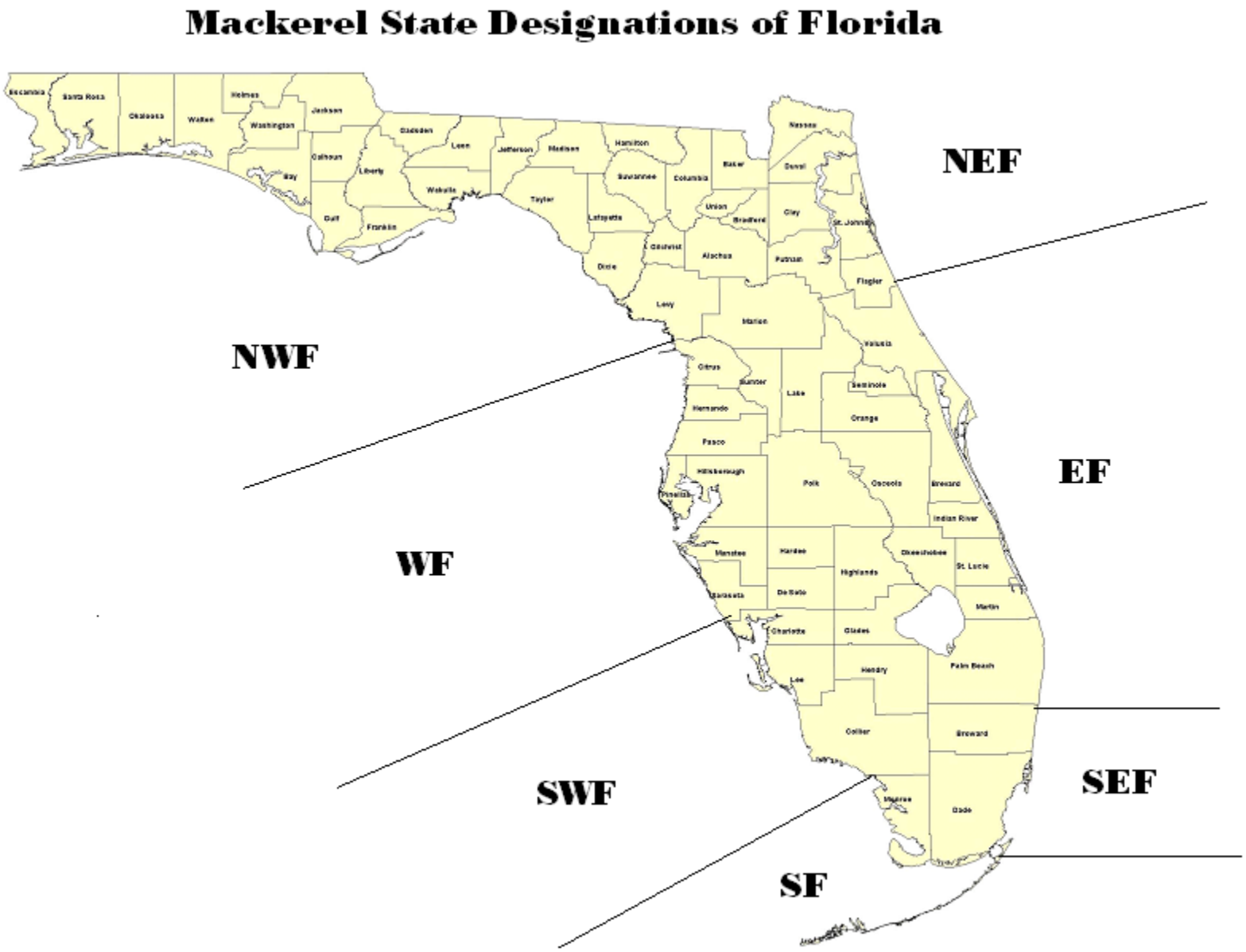

Table 1: List of Species, Abbreviation, and Numerical Codes (NMFS, NODC) in alphabetic order by Family and Common Name

Family Name	**Common Name**	**Scientific Name**	**Abbreviation**	**Previous ABREV**	**NMFS Code**	**NODC**
Balistidae	Filefish, Orange	*Aluterus schoepfi*	ORF			8860020101
Balistidae	Filefish, Planehead	*Monacanthus hispidus*	PHF			8860020301
Balistidae	Filefish, Unicorn	*Aluterus monoceros*	UFF			8860020103
Balistidae	Triggerfish, Gray	*Balistes capriscus*	GTR	GTF	4561	8860020201
Balistidae	Triggerfish, Ocean	*Canthidermis sufflamen*	OTR		4562	8860020502
Balistidae	Triggerfish, Queen	*Balistes vetula*	QTR		4563	8860020202
Bothidae	Flounder, Channel	*Syacium micrurum*	CHF	CF		
Bothidae	Flounder, Dusky	*Syacium papillosum*	DUF			
Bothidae	Flounder, Gulf	*Paralichthys albigutta*	GUF			8857030302
Bothidae	Flounder, Lefteyed		LFL			
Bothidae	Flounder, Ocellated	*Ancylopsetta quadrocellata*	OF			8857030506
Bothidae	Flounder, Southern	*Paralichthys lethostigma*	SFL	SOF		8857030304
Bothidae	Flounder, Summer	*Paralichthys dentatus*	SUF			8857030301
Carangidae	Amberjack, Greater	*Seriola dumerili*	GAM	AJ	1812	8835280801
Carangidae	Amberjack, Lesser	*Seriola fasciata*	LAM	LAJ	1815	8835280802
Carangidae	Banded rudderfish	*Seriola zonata*	BRF		1817	8835280804
Carangidae	Jack, Almaco	*Seriola rivoliana*	AJK	ALJ	1810	8835280803
Carangidae	Jack, Cotton Mouth	*Uraspis secunda*	CMJ			8835281701
Carangidae	Jack, Crevalle	*Caranax hippos*	JCR		0870	8835280303
Carangidae	Permit	*Trachinotus Falcatus*	PER		2550	8835280902
Carangidae	Pompano, Florida	*Trachinotus carolinus*	FLP		2720	8835280901
Carangidae	Runner, Blue	*Caranx crysos*	BLR		270	8835280306
Carangidae	Scad, Round	*Decapterus punctatus*	RSD			8835281202
Clupeidae	Sardine, Spanish	*Sardinella aurita*	SSD			8747011000
Coryphaenidae	Dolphin	*Coryphaena hippurus*	DOL		1050	8835290101
Emmelichthyidae	Crimson Rover	*Erythrocles monodi*	CRR		2525	8835350403
Ephippidae	Spadefish	*Chaetodipterus faber*	SP	SPF	3810	8835520101
Gadidae	Hake, Gulf	*Urophycis cirrata*	GUH	HAKE		8791031005

Family Name	Common Name	Scientific Name	Abbreviation	Previous ABREV	NMFS Code	NODC
Gadidae	Hake, Southern	*Urophycis floridanus*	SOH			8791031007
Haemulidae	Grunt, Bluestriped	*Haemulon sciurus*	BGR		1444	8835400113
Haemulidae	Grunt, Margate	*Haemulon album*	MGR	MRG	1442	8835400103
Haemulidae	Grunt, White	*Haemulon plumieri*	WGR	WG	1441	8835400102
Haemulidae	Pigfish	*Orthopristis chrysoptera*	PIG		2580	8835400201
Haemulidae	Porkfish	*Anisotremus virginicus*	PRK		2750	8835400306
Haemulidae	Tomtate	*Haemulon aurolineatum*	TOM	TT	1446	8835400101
Holocentridae	Soldierfish, Bigeye	*Ostichthys trachypoma*	BSF		0124	8810080301
Holocentridae	Squirrelfish	*Holocentrus adscensionis*	SQF			8810080101
Labridae	Hogfish	*Lachnolaimus maximus*	HOG		1790	8839010901
Labridae	Hogfish, Spanish	*Bodianus rufus*	SHF			8839010302
Labridae	Razorfish, Pearly	*Xyrichtys novacula*	PRF			
Lobotidae	Tripletail	*Lobotes surinamensis*	TRT	TRI	4590	8835380101
Lutjanidae	Snapper, Alfonsin	*Beryx splendens*	ALS		0016	8810050102
Lutjanidae	Snapper, Black	*Apsilus dentatus*	BS		3755	8835360201
Lutjanidae	Snapper, Blackfin	*Lutjanus buccanella*	BFS		3757	8835360106
Lutjanidae	Snapper, Cardinal	*Pristipomoides macrophthalmus*	CRS			8835360703
Lutjanidae	Snapper, Caribbean red	*Lutjanus purpureus*	CAS		3780	8835360111
Lutjanidae	Snapper, Cubera	*Lutjanus cyanopterus*	CS		3759	8835360101
Lutjanidae	Snapper, Dog	*Lutjanus jocu*	DS		3754	8835360109
Lutjanidae	Snapper, Gray (Black or Mangrove)	*Lutjanus griseus*	GS	MGS	3762	8835360102
Lutjanidae	Snapper, Lane	*Lutjanus synagris*	LS		3761	8835360112
Lutjanidae	Snapper, Mahogany	*Lutjanus mahogoni*	MHS		3772	8835360110
Lutjanidae	Snapper, Mutton	*Lutjanus analis*	MTS		3763	8835360103
Lutjanidae	Snapper, Queen	*Etelis oculatus*	QS		3770	8835360301
Lutjanidae	Snapper, Red	*Lutjanus campechanus*	RS		3764	8835360107
Lutjanidae	Snapper, Schoolmaster	*Lutjanus apodus*	SCS		3771	8835360104
Lutjanidae	Snapper, Silk	*Lutjanus vivanus*	SS		3758	8835360113

Family Name	Common Name	Scientific Name	Abbreviation	Previous ABREV	NMFS Code	NODC
Lutjanidae	Snapper, Vermilion	*Rhomboplites aurorubens*	VS		3765	8835360501
Lutjanidae	Snapper, Wenchman	*Pristipomoides aquilonaris*	WNS		3756	8835360701
Lutjanidae	Snapper, Yellowtail	*Ocyurus chrysurus*	YTS		3767	8835360401
Malacanthidae	Tilefish, Anchor	*Caulolatilus intermedius*	ATL		4479	8835220103
Malacanthidae	Tilefish, Blackline	*Caulolatilus cyanops*	BKT		4476	8835220102
Malacanthidae	Tilefish, Blueline (Grey)	*Caulolatilus microps*	BLT		4474	8835220104
Malacanthidae	Tilefish, Golden (Goldeneye)	*Lopholatilus chamaeleonticeps*	GTL		4470	8835220201
Malacanthidae	Tilefish, Goldface	*Caulolatilus chrysops*	GFT		4472	8835220105
Malacanthidae	Tilefish, Sand	*Malacanthus plumieri*	SDT		4478	8835220301
Mugilidae	Mullet, Redeye	*Mugil gaimardianus*	REM	RDM		
Mugilidae	Mullet, Striped	*Mugil cephalus*	STM		2341	8836010101
Ophidiidae	Brotula, Bearded	*Brotula barbata*	BRO		1144	8792010401
Percichthyidae	Wreckfish	*Polyprion americanus*	WRF		5131	8835022801
Pomatomidae	Bluefish	*Pomatomus saltatrix*	BF		0230	8835250101
Priacanthidae	Bigeye	*Priacanthus arenatus*	BGI	BGY	0140	8835170101
Priacanthidae	Bigeye, Short	*Pristigenys alta*	SBE	SBI	0145	8835170201
Priacanthidae	Snapper, Glasseye	*Priacanthus cruentatus*	GES		0147	8835170102
Rachycentridae	Cobia	*Rachycentron canadum*	COB		0570	8835260101
Sciaenidae	Croaker, Atlantic	*Micropogonias undulatus*	ACR		0900	8835440701
Sciaenidae	Drum, Black	*Pogonias cromis*	BKD		1081	8835440801
Sciaenidae	Drum, Blackbar	*Equetus iwamotoi*	BBD			
Sciaenidae	Drum, Red	*Sciaenops ocellatus*	RDM		1082	8835440901
Sciaenidae	Seatrout, Sand (White)	*Cynoscion arenarius*	SST		3455	8835440106
Sciaenidae	Seatrout, Silver	*Cynoscion nothus*	STR			8835440103
Sciaenidae	Striped croaker	*Bairdiella sanctaeluciae*	SC			8835440305
Scombridae	Little Tunny	*Euthynnus alletteratus*	LT		4653	8850030102
Scombridae	Mackere, Chub	*Scomber japonicus*	CBM		2150	8850030301
Scombridae	Mackerel, Cero	*Scomberomorus regalis*	CMK		1938	8850030503
Scombridae	Mackerel, King	*Scomberomorus cavalla*	KMK		1939	8850030501

Family Name	Common Name	Scientific Name	Abbreviation	Previous ABREV	NMFS Code	NODC
Scombridae	Mackerel, Spanish	*Scomberomorus maculatus*	SMK		3840	8850030502
Scombridae	Tuna, Blackfin	*Thunnus atlanticus*	BLK	BFT	4658	8850030404
Scombridae	Tuna, Bluefin	*Thunnus thynnus*	BFT		4652	8850030402
Scombridae	Tuna, Skipjack	*Katsuwonus pelamis*	KWP		4654	8850030101
Scombridae	Tuna, Yellowfin	*Thunnus albacares*	YFT		4655	8850030403
Scombridae	Wahoo	*Acanthocybium solanderi*	WAH		4710	8850030601
Scorpaenidae	Blackbelly Rosefish	*Helicolenus dactylopterus*	BBR		2420	8826010301
Scorpaenidae	Scorpion Fish (spinycheek)	*Neomerinthe hemingwayi*	SCF		3263	8826010402
Scorpaenidae	Scorpion Fish (spotted)	*Scorpaena plumieri*	SPS		3265	8826010614
Serranidae	Bass, Longtail	*Hemanthias leptus*	LTB		3374	8835021201
Serranidae	Bass, School	*Schultzea beta*	SB			
Serranidae	Coney	*Epinephelus fulvus*	CO		1429	8835020438
Serranidae	Creole-fish	*Paranthias furcifer*	CRE		1427	8835021701
Serranidae	Goliath fish (Jewfish)	*Epinephelus itajard*	GOG		1850	8835020401
Serranidae	Grouper, Black	*Mycteroperca bonaci*	BKG		1422	8835020502
Serranidae	Grouper, Comb	*Mycteroperca rubra*	COG			8835020510
Serranidae	Grouper, Gag	*Mycteroperca microlepis*	GAG		1423	8835020501
Serranidae	Grouper, Graysby	*Epinephelus cruentatus*	GSG	GRA	1428	8835020439
Serranidae	Grouper, Marbled	*Epinephelus inermis*	MBG		1417	8835020440
Serranidae	Grouper, Misty	*Epinephelus mystacinus*	MSG		1420	8835020409
Serranidae	Grouper, Nassau	*Epinephelus striatus*	NAG		1430	8835020412
Serranidae	Grouper, Red	*Epinephelus morio*	RDG		1416	8835020408
Serranidae	Grouper, Scamp	*Mycteroperca phenax*	SCA		1424	8835020505
Serranidae	Grouper, Snowy	*Epinephelus niveatus*	SNG		1414	8835020411
Serranidae	Grouper, Warsaw	*Epinephelus nigritus*	WRG		4740	8835020410
Serranidae	Grouper, Yellowedge	*Epinephelus flavolimbatus*	YEG		1415	8835020405
Serranidae	Grouper, Yellowfin	*Mycteroperca venenosa*	YFG		1426	8835020506
Serranidae	Grouper, Yellowmouth	*Mycteroperca interstitialis*	YMG		1425	8835020504
Serranidae	Hind, Red	*Epinephelus guttatus*	RDH		1413	8835020406

Family Name	Common Name	Scientific Name	Abbreviation	Previous ABREV	NMFS Code	NODC
Serranidae	Hind, Rock	*Epinephelus adscensionis*	RKH	ROH	1412	8835020402
Serranidae	Hind, Speckled	*Epinephelus drummondhayi*	SPH		1411	8835020404
Serranidae	Sand Perch	*Diplectrum formosum*	SPR		3110	8835021002
Serranidae	Sea Bass, Bank	*Centropristis ocyurus*	SBB	YSB	3375	8835020304
Serranidae	Sea Bass, Black	*Centropristis striata*	BSS	BSB	3360	8835020301
Serranidae	Sea Bass, Rock	*Centropristis philadelphica*	RSB		3362	8835020305
Serranidae	Spanish Flag	*Gonioplectrus hispanus*	SPF		3371	8835021101
Serranidae	Tattler	*Serranus phoebe*	TAT			8835022308
Sparidae	Pinfish (porgies)	*Lagodon rhomboides*	PIN		2670	8835430201
Sparidae	Porgy, Grass	*Calamus arctifrons*	GRP	GP	3305	8835430501
Sparidae	Porgy, Jolthead	*Calamus bajonado*	JHP		3312	8835430502
Sparidae	Porgy, Knobbed	*Calamus nodosus*	KBP		3308	8835430506
Sparidae	Porgy, Littlehead	*Calamus proridens*	LHP		3310	8835430508
Sparidae	Porgy, Longspine	*Stenotomus caprinus*	LSP		3299	8835430102
Sparidae	Porgy, Red	*Pagrus pagrus*	RDP	RP		8835430601
Sparidae	Porgy, Saucereye	*Calamus calamus*	SEP		3304	8835430503
Sparidae	Porgy, Sheepshead	*Calamus penna*	SHE			8835430510
Sparidae	Porgy, Whitebone (White)	*Calamus leucosteus*	WBP		3306	8835430505
Sphyraenidae	Barracuda, Great	*Sphyraena barracuda*	BAR			8837010104
Stromateidae	Barrelfish	*Hyperoglyphe perciformis*	BLF		0193	8851010201
Stromateidae	Driftfish, Black	*Hyperoglyphe bythites*	BDF		0192	8851010202
Synodontidae	Lizardfish, Inshore	*Synodus foetens*	INF			8762020101
Synodontidae	Lizardfish, Red	*Synodus synodus*	RLF			8762020106
Trichiuridae	Escolar	*Lepidocybium flavobrunneum*	ESC		2501	8850010301
Trichiuridae	Oilfish	*Ruvettus pretiosus*	OIL		2502	8850010401

Chapter 2: Data Sources Updated January 2008

Data is received from several different types of sources. These data sources include Cooperative Fishing Effort (CO-OP), Florida Marine Research Institute (FMRI), Beaufort Head Boat (HB), Marine Recreational Fisheries Statistical Survey (MRFSS), NOAA Fisheries Mississippi Laboratory (MSLAB), North Carolina Department of Natural Resources (NCDMR), No Data Sheet Provided, NOAA Fisheries Panama City Laboratory (PCLAB), Pelagic Observer Program (POP), Recreational Fisheries Information Network (RECFIN), Shark Bottom Longline Observer Program (SBLOP), Trip Interview Program (TIP), and United States Geological Survey (USGS).

Each section describes, in detail, each type of data source and their corresponding data sheets. This section is useful to answer questions when entering log or meristic data and/or proofing these data.

This chapter includes information on the following data sources:

2.1	CO-OP	Cooperative Fishing Effort
2.2	FMRI	Florida Marine Research Institute
2.3	HB	Beaufort Head Boat
2.4	MRFSS	Marine Recreational Fisheries Statistical Survey
2.5	MSLAB	Mississippi Laboratory-Fishery Independent Survey
2.6		No Data Sheet Provided
2.7	NCDMR	North Carolina Department of Natural Resources
2.8	PCLAB	Panama City Laboratory
2.9	POP	Pelagic Observer Program
2.10	RECFIN	Recreational Fisheries Information Network
2.11	SBLOP	Shark Bottom Longline Observer Program
2.12	TIP	Trip Interview Program
2.13	USGS	United States Geological Survey

Contributors: Meredith Burnette, Linda Lombardi, Beverly Barnett, Carrie Fioramonti

2.1 Cooperative Fishing Effort Samples (CO-OP)

Samples collected by Cooperative Research Proposals (CRP) are logged in using the following information. A copy of the appropriate data sheet (Figure 2.1.1) is also included. Data fields highlighted indicates data fields used to complete the log.

Key to CO-OP Data Sheet

PC Coll # — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Date — Date the sample was caught, __/__/__ (mo/da/yr)

Collector — Individual responsible for collection: First Initial. Last Name of sampler

Source — CO-OP

Source # — 4 digit year – First and Last Initial of sampler – Trip # (consecutive order for the year)
Example: year = 2001, First and Last Name Initial, Trip # 1
2001-ES-01 (please use preceding zeros)

State* — State Abbreviation

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location — City/Town or port name, see Table 2.12.2 for county numerical codes

Mode — Fishing Mode:

CM = Commercial	*Use with normal commercial catch
OB = Observer	*Use if sampled undersized fish with scientific permit
CP = Charter	
PR = Private	

Gear	Fishing Gear: HL Hand-line/ Bandit/ Hook and Line/ Troll LL Long-line TR Trap
Species Name	Corresponding species abbreviation (see Chapter 1, Table 1)
Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm)
FL (mm)	Fork Length (mm)
Whole WT (kg)	Weight (kg) of whole fish
Sex	F or ♀ Female M or ♂ Male I Immature * Leave blank if unable to identify
Samples Taken	Indicates whether each sample had otoliths, gonads or both
# of Gonads	Total number of gonads in the collection
# of Otoliths	Total number of otoliths in the collection
Condition (S/W)	Condition of the otolith S Sectioned W Whole
Latitude	Latitude recorded (degrees, minutes, degree minutes) where sample was caught at sea. During long-line haul backs, the pickup location of the first hi-flyer.
Longitude	Longitude recorded (degrees, minutes, degree minutes) where sample was caught at sea. During long-line haul backs, the pickup location of the first hi-flyer.
Maximum Depth	Maximum water depth (meters) Conversions: Meters = (fathoms) * 1.8288, Meters = (feet) * 0.3048
Time	Time of capture (long-line haul backs, the pickup time of the first hi-flyer)
Comments	Any additional comments e.g. color pattern of fish, other species characteristics

Figure 2.1.1. Data Entry Sheet for Samples Collected by Cooperative Fishing Effort

Date:____/____/_____Collector: _____Source: _____ Source #: _____-_____-____

Mode (circle one): CM OB Gear (circle one): HL LL TR

Species Name	Fish #	TL (mm)	FL (mm)	Whole Wt (kg)	Sex	Samples O	G	Latitude	Longitude	Depth (m)	Catch Time	Comments

2.2 Florida Marine Research Institute (FMRI)

Florida Marine Research Institute (FMRI) samples are collected during scientific surveys as part of the Fishery Independent Survey. A copy of the appropriate data sheet is also included (Figure 2.2.1). Data fields highlighted indicates data fields used to complete the log.

Key to FMRI Data Sheet

PC Coll #: A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Source: FMRI

Source # Boat Initials – Cruise # – Station #
(TM) (3 digits) (3 digits)
TM Tommy Monroe

Date Date the sample was caught, __/__/__ (mo/da/yr)

Latitude Latitude recorded (degrees, minutes, degree minutes) where sample was caught

Longitude Longitude recorded (degrees, minutes, degree minutes) where sample was caught

Time Time of capture.

Location* The approximate location where the sample was caught.
NE GOM: Latitude greater than 28°, Longitude less than 88°50
NW GOM: Latitude greater than 28°, Longitude greater than 88°50
SE GOM: Latitude less than 28°, Longitude less than 88°50
SW GOM: Latitude less than 28°, Longitude greater than 88°50

State* The approximate location where the sample was caught.
* The state will be determined by the degree of latitude and longitude.

Depth Water depth (meters)
Conversions: Meters = (fathoms) * 1.8288, Meters = (feet) * 0.3048

Mode SS Scientific Survey

Gear HL Hand-line/ Bandit/ Hook and Line/ Troll
LL Long-line
TR Trap
TRW Trawl

Comments	Additional comments provided by the processor	
Collector	First Initial and Last Name of cruise leader	
Species	Abbreviation of species being sampled, see Chapter 1, Table 1	
Fish #	Individual fish number supplied by sampler	
TL (mm)	Total Length (mm)	
FL (mm)	Fork Length (mm)	
Whole Wt (kg)	Weight whole fish	
Sex	F or ♀	Female
	M or ♂	Male
	I	Immature
Samples	O	Otolith
	G	Gonad

Figure 2.2.1 FWRI datasheet for fishery independent samples.

Data Entry Sheet for Samples Collected at Sea Station # _____

Source: __FMRI__ Source #:_TM-_____ Date: _____/_____/_____

Latitude: _____ Longitude: _____ Time Caught: _____

Location: SW GOM SE GOM NE GOM NW GOM State: _____ Depth (m): _____

Mode: SS Gear: HL LL TRP TRW

Comments: _____ Collector: _____

Species Name	Fish #	TL (mm)	FL (mm)	Whole Wt (kg)	Sex	Samples O	Samples G	Species Name	Fish #	TL (mm)	FL (mm)	Whole Wt (kg)	Sex	Samples O	Samples G

2.3 Beaufort Head Boat Survey Samples (HB)
Samples Received: 1980's - Present

Beaufort Head Boat (HB) samples are collected during interviews of registered Head Boats and samplers are supervised by biologist from NOAA Fisheries, Beaufort, NC. Only otolith envelopes are sent to our facility and we create a datasheet. Data fields highlighted indicates data fields used to complete the log.

COLLECTION NUMBER: _____

SPECIES: _____ DATE: _____

TOTAL LENGTH: _____ OT: _____

WEIGHT: _____ SP: _____

FORK LENGTH: _____ SC: _____

AREA: _____ GULF: _____ ATLANTIC: _____

Head Boat Envelope

All information is usually legible and correct, thus the sample can still be properly logged in without a data sheet.

If you are logging in Archive Beaufort Head Boat Samples, it is important to sort them by date and then by the Beaufort Collection number (our source #). There may be more than one Beaufort Collection number on one date. It is very crucial that these Beaufort Collection numbers are separated and that the log consists of each Panama City Collection number for each of the Beaufort collection number (our source #).

<u>Codes for the Beaufort Head Boat Survey Data File</u>

Vessel Type

1	Headboat
2	Research
3	Commercial
4	Charter boat
5	Seafood Houses
6	Unknown Vessel

Sex

0	Immature, sex undetermined
1	Female
2	Male
3	Immature Female
4	Immature Male

Sexual Maturity

1	Immature
2	Resting (not developing)
3	Developing
4	Spawning
5	Spent

Key to HB Data Sheet/Envelopes

Species — spelled out or provided in the appropriate abbreviation (see Chapter 1, Table 1)

PC Coll #: — A sequential species specific number is marked in the upper right-hand corner of the top envelope of each collection with a **RED** pen. This same number is marked on a small yellow post-it note and placed in the middle of the top envelope within the collection. Then place a rubber band around the envelopes, ensuring that all envelopes belonging to a specific collection remain together.

Date: — Date of collection from sampler __/__/__ (mo/da/yr)

Source: — HB

Source #: — Collection number (5-digit number, assigned by NOAA Fisheries Beaufort, NC)

Collector: — First Initial and Last Name of sampler

# of Gonads: — Total number of gonads in that sample

# of Otoliths: — Total number of otoliths in that sample

Condition (S/W): — Condition of the otolith
S = Sectioned W = Whole

Mode: — HB Head Boat

Gear: — HL Hand Line

Comments: — Any other information not listed above

Fish # — Allocated by sampler, either written next to the collection number or in the upper right hand corner of envelope.

Weight (kg) — whole weight (normally, confirm with sampler)

Total Length (mm)

Fork Length (mm)

Sex — F or ♀ Female
M or ♂ Male

Samples — OT Otolith
SP Spine
SC Scale

State*: State Abbreviation

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)

*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: City/Town or port name from the Area field, area is interpreted as follows:

Area	Location	State
01	Hatteras Offshore	NC
02	Cape Fear Inshore	NC
03	Cape Fear Offshore	NC
04	Inshore	SC
05	Offshore	SC
06	Savannah	GA
07	Jacksonville	E FL
08	Daytona to Ft. Pierce	E FL
09	Beaufort Inshore	NC
10	Beaufort Offshore	NC
11	Ft. Pierce to Miami	E FL
12	Florida Keys	E FL
13	Bahamas	
17	Dry Tortugas	FL
21	Naples	FL
23	Gulf Breeze	FL
23	Pensacola	FL
23	Destin	FL
23	Panama City	FL
23	Dauphin Island	AL
23	Orange Beach	AL
24	Grand Isle	LA
24	Fourchan	LA
24	Empire	LA
25	Freeport	TX
25	Galveston	TX
26	Port Aransas	TX
27	Port Isabel	TX

Area numbers 17 and greater refer to a Gulf of Mexico location.

Area numbers less than 17 refer to Atlantic Ocean.

Figure 2.3.1. **Beaufort HB Data Entry Sheet**

Species ___________
PC Coll # _________

Month / Day /Year
Date: ______/______/_____ Source: _________ Source #: _____________

Collector: ______________ State: _____ Location: _____________

Mode: HB Gear: HL
Comments: ____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G	Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G
					Total:								Total:		

Data Entered
Date: ______________ Initials: _____________

2.4 Marine Recreational Fishery Statistics Survey (MRFSS)/MACRO

The Marine Recreational Fishery Statistics Survey (MRFSS) is a nationwide program implemented to establish a reliable database for estimating the impact of recreational fishing on marine resources. Marine Recreational Fisheries Statistical Survey Samples were contracted through MACRO. There were two different data sheets for each of the two years, 1998 and 1999. Examples of these data sheets are included in this section (Figure 2.4.1 and 2.4.2). Data fields highlighted indicates data used to complete the log.

Key to MRFSS Data Entry
Samples Received 1998 – 1999:

Species — Species Name and Species Code are located on side 2 of the MRFSS data sheet.

PC Coll #: — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Collector: — First Initial and Last Name of sampler

Source: — MRFSS

Source #: — 1st digit = assignment No.
Next 4 digits = interviewer ID
Last 3 digits = intercept No.

Date: — year/month/day
On log sheet use the appropriate format: (mo/da/yr)

State*: — State Alpha Abbreviation

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: — Give the City where the SITE CODE is located.
Otherwise, use the COUNTY CODE, see Table 2.12.2

There is a detailed list of state, county, and site codes provided by Macro/MRFSS listed first by state and then by site code. For 1998, there is a list of state, county, and site codes for Alabama, Florida, Louisiana and Mississippi. Also, there are Quick Lists for 1999 created for Alabama, Florida, Louisiana and Mississippi. These lists are arranged by state code, county code and then finally by site code, these lists are located in the filing cabinet of the Age, Growth, and Reproduction laboratory. In addition, the county name that should appear in the Location column is listed. Please be aware that some sites in different states can have the same code. If it is possible, try to find out the appropriate site for the code while logging in; otherwise write in the code in the location.

Gonad #: Total number of gonads in the collection
MRFSS samplers do not supply us with gonads but should indicate sex with the data.

Otolith #: Total number of otoliths in the collection

Condition (S/W): Condition of the otolith

S	Sectioned
W	Whole

Mode: Fishing Mode

MRFSS Abbreviation	Our Abbreviation	Fishing Mode
PC	CP	Charter Party
HB	HB	Head Boat
PR	PR	Private Boat
SH	PR	Shore Fishing

Gear: HL Hand Line
A majority of MRFSS samples are from charter, head boat, or private boats that only use some type of hand lines, i.e. rod & reel.

Comments: Any other information not listed above
MRFSS often times collects 2 otoliths from each fish; this can be indicated in the comments.

Figure 2.4.1 1998 Marine Recreational Fisheries Statistics Survey Data Sheet

OMB NO. 0648-/ *EXP. 03/30/98)

MARINE RECREATIONAL FISHERIES STATISTICS SURVEY
1998 BIOLOGICAL SAMPLING QUESTIONNAIRE (5/98)

Hello, my name is _____ and I represent the Marine Sciences Group of QuanTech, Inc. We are collecting biological data for a study sponsored by the National Marine Fisheries Service of the US Department of Commerce. I'd like to ask you a few questions about your fishing and examine your fish.

S1. May I take measurements and remove scales/otoliths from your fish? Yes ... Continue No ... Terminate

1. ASSIGNMENT NO.
2. INTERVIEWER ID
3. YR/MO/DAY
4. INTERCEPT NO.
5. CONTROL NO.
6. STATE CODE
7. COUNTY CODE
8. SITE CODE

*9. Would you say you were fishing from...
1 Pier, Dock
2 Jetty, Breakwater, Breachway
3 Bridge, Causeway
4 Other Man-made Structure (Specify)_____
5 Beach or Bank
6 Headboat
7 Charterboat
8 Private or Rental Boat

*10. Was most of your (specify mode) fishing effort today in the...
1 Ocean/gulf/open bay
2 A sound (Other than those specified)
3 River (Other than those specified)
4 Bay (Other than those specified)
5 Other (Specify)
A Albermarle/Pamlico
B Biscayne Estuary
C Whitewater Estuary
D Sarasota Estuary
E Tampa Estuary
F Mobile Estuary
G Atchafalaya Estuary

*11. Was that
1 Three Miles or Less from Shore
2 More than Three Miles
3 Ten Miles or Less from Shore (West Florida)
4 More than Ten Miles (West Florida)

Side 1

SPECIES CODES:

BLACK SEABASS	8835020301	GAG GROUPER	8835020501
KING MACKEREL	8850030501	GREATER AMBERJACK	8835280801
RED SNAPPER	8835360107	RED DRUM	8835440901
SPANISH MACKEREL	8850030502	RED GROUPER	8835020408
BLUEFISH	8835250101	VERMILION SNAPPER	8835360501

FISH NO.	SPECIES	SPECIES CODE	STRAIGHT FORK LENGTH (mm)	WHOLE WEIGHT (Kg)	OTOLITH	SCALE
1.				.		
2.				.		
3.				.		
4.				.		
5.				.		
6.				.		
7.				.		
8.				.		
9.				.		
10.				.		
11.				.		
12.				.		
13.				.		
14.				.		
15.				.		

Side 2

DATE…….

INTERVIEWER…….

CONTROL #.....

DOCUMENT #.....

FISH # ……

SPECIES NAME
…….

SPECIES CODE……..
STRAIGHT FORKED
LENGTH ……
ISSUE TYPE…….

Front View

STATE CODE…….

COUNTY CODE…….

SITE CODE…….

Back View

1998 Marine Recreational Fisheries Statistics Survey Envelope

Figure 2.4.2 1999 Marine Recreational Fishery Statistics Survey Data Sheet

VERY IMPORTANT

MARINE RECREATIONAL FISHERY STATISTICS SURVEY
1999 BIOLOGICAL SAMPLING QUESTIONNAIRE (6/8/1999)

OMB NO. 0648-0052 (EXP. 10/30/01)

Hello, my name is _____ and I represent Macro International Inc. We are collecting biological data for a study sponsored by the National Marine Fisheries Service of the U.S. Department of Commerce. I'd like to ask you a few questions about your fishing and examine your fish.

1. May I take measurements and remove otoliths/scales from your fish? ☐ Yes ☐ No - Terminate

2. ASSIGNMENT No. ☐ Please indicate if this is your first or second assignment today, by writing "1" or "2."

3. INTERVIEWER ID ☐☐☐☐

4. YR/MO/DAY 1 9 9 9 M M D D

5. INTERCEPT NO. ☐☐

6. INTERVIEW TIME (use 2400 clock) ☐☐☐☐ *Time this interview was completed.*

7. STATE CODE ☐☐ 8. COUNTY CODE ☐☐☐ 9. SITE CODE ☐☐☐☐

READ PRIVACY ACT. **This study is being conducted in accordance with the privacy act of 1974. You are not required to answer any question that you consider to be an invasion of your privacy.**

11. Would you say you were fishing from ...

- 1 ☐ Pier, Dock
- 2 ☐ Jetty, Breakwater, Breachway
- SH 3 ☐ Bridge, Causeway
- 4 ☐ Other Man-Made Structure (Specify) _____
- 5 ☐ Beach or Bank
- 6 ☐ Headboat
- PC 7 ☐ Charterboat
- PR 8 ☐ Private or Rental Boat

12. Was most of your (specify mode) fishing effort today in the ...

- 1 ☐ Ocean/gulf/open bay
- 2 ☐ A sound (*Other than those specified*)
- 3 ☐ River (*Other than those specified*)
- 4 ☐ Bay (*Other than those specified*)
- 5 ☐ Other (Specify) _____
- G ☐ Albemarle/Pamlico Estuary
- H ☐ Biscayne Estuary
- I ☐ Whitewater Estuary
- J ☐ Sarasota Estuary
- K ☐ Tampa Estuary
- L ☐ Mobile Estuary
- M ☐ Atchafalaya Estuary

13. Was that ...

- 1 ☐ Three miles or less from shore
- 2 ☐ More than three miles from shore
- 3 ☐ Ten miles or less from shore (WFL)
- 4 ☐ More than ten miles from shore (WFL)
- 8 ☐ NOT APPLICABLE Angler is not fishing in ocean/gulf/open bay, or angler is fishing in shore mode.

Side 1

SPECIES CODES:

RED SNAPPER	8835360107	VERMILION SNAPPER	8835360501
WARSAW GROUPER	8835020410	KING MACKEREL	8850030501
SNOWY GROUPER	8835020411	SPANISH MACKEREL	8850030502
RED GROUPER	8835020408	GREATER AMBERJACK	8835280801
GAG GROUPER	8835020501	RED DRUM	8835440901

FISH NO.	SPECIES NAME	SPECIES CODE	FORK LENGTH (mm) FL(mm)	WEIGHT (Kg)	OTOLITH ~~SCALE~~ NOT USED	SEX M F
1				.		
2				.		
3				.		
4				.		
5				.		
6				.		
7				.		
8				.		
9				.		
10				.		
11				.		
12				.		
13				.		
14				.		
15				.		

Side 2

Control #.....

Interviewer……..
……
Date……

Intercept #.....

State Code……..

County Code……..

Site Code……

Fish #.....

Species name……
……..
Species Code……

1999 Marine Recreational Fishery Statistics Survey Envelope

MFRSS envelopes

MRFSS envelopes are very easy to read and carry all of the same information that is found on the data sheet except for the Control #. The control # is for MACRO use only; all the other information printed on the envelope should be the same as found on the corresponding data sheet.

Fish #	Side 2	
Species Name	Side 2	
Species Code	Side 2	
Fork Length (mm)	Side 2	
Weight (kg)	Side 2	
Tissue Type	Side 2	O = Otolith
Sex	Side 2	F or ♀ Female M or ♂ Male

Table 2.4.1 MACRO International Inc. ID numbers and employee names for 1999 and earlier (on left) and 2000 through present (on right)

ID	NAME
0009	Steve Candileri, Regional Representative
2101	Jeff Persse
2335	Chuck Idelberger
2337	Bill Davis
2745	Curtis Butler, Regional Representative
2822	William Lake, Regional Representative
2921	Dave Newton
2933	Fred Scoggins
2943	Ken Searcy
2970	Charlie Broussard
2972	Bryan Batcher
2978	Dale Beter
2979	Charlie Gardner
2983	Ernie Nillen
2995	James Taylor
3003	Stanley Alba
3008	Brett Blackburn
3009	Johnie Heinle
3022	Greg Kolessar
3121	Brad Snell
5153	Wayne Primeaux
5161	Brett Hano
5164	Lynatta Jones
5166	Gregg Goodspeed
6026	Michael Metzger
6027	Brannon Wilson
6028	Norman Rollo

ID	NAME
0009	Steve Candileri,
2341	Gerry Carr
2341	Gerry Carr
4767	Joe DeRosa
5003	Lew Bullock
5071	Joe Borowicz
5108	Terri Menzel
5112	Eric Sander
5120	Ed Pulido
5136	Craig LaVine
5156	Mark Koryak
5215	Dave McGowen
5241	Billy Wheat
5242	Michelle Maki
5261	Carlos Llull
5279	Kelly Sullivan
5281	Sky Rudloe
5282	Jon Wolfson
5321	Ariel Poholek
5335	Gary Moore
5342	Joe Cioli
5345	Jennifer Bogdon
5347	Adam Purdy
5349	Brian Lowman
5354	Beverly Sauls
5357	Stephanie Wilson
5358	Tarsha Bradden
5359	Alyssa Jordon
5373	Sarah Maschal
5381	Terrie Weeks
5384	Joe Eskay
5385	Zeviel Persellin
5399	Holly Rolls
	Charlotte Mansfield
5419	Heather Rigney, Jason Herr
5414	Oscar Ayalas

Key to MRFSS Data Sheet
MRFSS Samples Received 2000 to present:

Currently, data sheets are computer generated (see Figure 2.4.3). Data fields highlighted indicates data used to complete the log.

Species — Abbreviation of species being sampled, see Chapter 1, Table 1

PC Coll #: — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Date: — Date of collection from sampler (mo/da/yr)

Source: — MRFSS

Source #: — 1st digit = assignment No.
Next 4 digits = interviewer ID
Last 3 digits = intercept No.

State*: — State Alpha Abbreviation

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: — City/County samples were landed, see Table 2.12.2

Collector: — First Initial and Last Name of sampler
See Table 2.4.1 For current list of samplers and ID #'s.

Gonad #: — Total number of gonads in the collection
MRFSS samplers do not supply us with gonads but should indicate sex with the data.

Otolith #: — Total number of otoliths in the collection

Condition (S/W):	Condition of the otolith S Sectioned W Whole
Mode:	CP Charter Party HB Head Boat PR Private Boat PR Shore Fishing
Gear:	HL Hand Line A majority MRFSS Samples are from charter, head boat, or private boats that only use some type of hand lines, i.e. rod & reel.
Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm)
FL (mm)	Fork Length (mm)
Whole Wt (kg)	Weight (kg) of whole fish
Sex	F or ♀ Female M or ♂ Male I Immature T Transitional stage
Comments:	Any other information not listed above MRFSS often times collects 2 otoliths from each fish; this can be indicated in the comments.

Figure 2.4.3. Example of computer generated datasheet constructed by Age, Growth, and Reproduction lab supervisor.

Samples from the State of Florida Samplers
Date Received: June 1, 2007

Year Species_____
PC Coll #_____

Year & Species	Date	Source	Source #	State	Location	Collector	Mode	Gear	Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	Sex	Comments
2007 RDG	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	671	485	482			F	
2007 RDG	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	672	517	511			F	
2007 RDG	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	673	815	742			F	
2007 KMK	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	674	880	800			F	
2007 KMK	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	675	665	600			M	
2007 KMK	3/25/2007	MRFSS	10009010	FL	Clearwater	S. Candileri	CP	HL	676	440	408			F	

2.5 NOAA Fisheries Mississippi Laboratory-Fishery Independent Survey (MSLAB)

Samples from the NOAA Fisheries Mississippi Laboratory are collected during scientific cruises at sea as part of the Fishery Independent Survey. Data fields highlighted indicates data used to complete the log. Either a copy of a data sheet (Figure 2.5.1) or a computer generated datasheet (Figure 2.5.2) is included with the biological samples.

Key to Fishery Independent Survey Data Sheet

PC Coll #	A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.
Station #	Corresponds to the station where sample was caught at sea
Source	MSLAB: Pascagoula Cruise
Source #	Boat Initials – Cruise # - Station # (3 digits) (3 digits) Oregon II OII Ferrel FER Caretta CAR Gordon Gunter GG HST HST Tommy Monroe TM Gandy GAN
Date	Date the sample was caught, __/__/__ (mo/da/yr)
Latitude	Latitude recorded (degrees, minutes, and seconds), where sample was caught at sea. During long-line haul backs, the pickup location of the first hi-flyer.
Longitude	Longitude recorded (degrees, minutes, and seconds), where sample was caught at sea. During long-line haul backs, the pickup location of the first hi-flyer.
Time	The approximate time of capture. During long line haul backs, the pickup time of the first hi-flyer.
Location	The approximate location where the sample was caught: NE GOM: Latitude greater than 28°, Longitude less than 88°50 NW GOM: Latitude greater than 28°, Longitude greater than 88°50 SE GOM: Latitude less than 28°, Longitude less than 88°50 SW GOM: Latitude less than 28°, Longitude greater than 88°50

State*	The approximate location where the sample was caught from the degree of longitude (find a map if there is a question): FL - approximately less than 87°50 AL - approximately greater than 87°50 and less than 88°50 MS - need to look at a map to distinguish actual location LA - greater than 88°50 and less than 94° TX - greater than 94° *NWF, WF, SWF, SF, SEF, EF, NEF (Florida) *For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)
Maximum Depth	Maximum water depth (meters) from CTD Data File
Mode	SS Scientific Survey
Gear	HL Hand-line/ Bandit/ Hook and Line/ Troll LL Long-line TR Trap TRW Trawl VLL Vertical Longline
Comments	Additional comments provided by the processor
Collector	First Initial and Last Name of cruise leader
Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm)
FL (mm)	Fork Length (mm)
Whole Wt (kg)	Weight (kg) of whole fish
Sex	F or ♀ Female M or ♂ Male
Samples Taken	O Otolith G Gonad S Spine
# of Gonads	Total number of gonads in the collection
# of Otoliths	Total number of otoliths in the collection
Condition (S/W)	Condition of the otoliths W Whole

Figure 2.5.1 Example of datasheet provided with MSLAB biological samples

Data Entry Sheet for Samples Collected at Sea Station # _____

Source:_____ Source #:_____ Date: _____/_____/_____

Latitude: _____ Longitude: _____ Time Caught: _____
Location: SW GOM SE GOM NE GOM NW GOM State: _____ Maximum Depth (m): _____
Mode: SS Gear: HL LL TR TRW VLL

Comments: _____ Collector: _____

Species Name	Fish #	TL (mm)	FL (mm)	Whole Wt (kg)	SEX	Samples Taken O	G	Species Name	Fish #	TL (mm)	FL (mm)	Whole Wt (kg)	SEX	Samples Taken O	G

Figure 2.5.2. Example of computer generated datasheet constructed by Age, Growth, and Reproduction lab supervisor.

2007 Oregon II
Reef Fish Cruise

Scientific Survey
Cruise Leader: Brani Noble

Year Species	PC Coll #	Fish #	Date	Source	Source #	State	Location	Latitude	Longitude	Depth	Gear	TL (mm)	FL (mm)	W WT (kg)	Sex
2007 RS	116	1	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	356	328	0.55	F
2007 RS	116	2	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	432	396	1.019	M
2007 RS	116	3	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	395	365	0.757	M
2007 RS	116	4	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	379	348	0.664	M
2007 RS	116	5	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	395	366	0.923	M
2007 RS	116	6	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	435	400	1.162	M
2007 RS	116	7	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	422	385	0.975	F
2007 RS	116	8	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	417	384	0.948	M
2007 RS	116	9	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	365	337	0.703	M
2007 RS	116	10	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	353	326	0.593	M
2007 RS	116	11	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	452	420	1.315	F
2007 RS	116	12	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	403	370	0.905	M
2007 RS	116	13	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	429	400	1.154	M
2007 RS	116	14	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	446	410	1.241	F
2007 RS	116	15	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	396	373	0.899	M
2007 VS	41	21	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	374	339	0.656	F
2007 VS	41	22	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	389	347	0.741	M
2007 VS	41	23	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	362	325	0.544	M
2007 VS	41	24	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	425	379	0.875	M
2007 VS	41	25	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	365	326	0.591	F
2007 VS	41	26	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	351	316	0.539	F
2007 VS	41	27	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	370	325	0.589	M
2007 VS	41	28	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	320	285	0.382	M
2007 VS	41	29	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	305	270	0.315	F
2007 VS	41	30	4/23/2007	MSLAB	OII-0275-018	TX	SW GOM	2702.49	09642.80	72.5	TR	285	254	0.287	M

2.6 No Data Sheet Provided

There are certain instances in which the sampler or collector does not complete or send a copy of their data sheet. It is necessary to complete a datasheet such that all questions (mode, gear, state, location, sampler's name, etc) regarding the collection of the samples can be obtained in the present. At these times, it is essential to complete a PC Lab Data Sheet (see Figure 2.6.1) to the best of your ability, making sure to complete all sections and blanks. If there is information which you cannot answer with 100% certainty, do NOT assume an answer. Promptly contact the lab manager and he/she will contact the sampler via e-mail, phone or regular mail. Data fields highlighted indicates data used to complete the log.

Key to PC Lab Data Entry Sheet

Species	Abbreviation of species being sampled, see Chapter 1, Table 1. Create a new datasheet for each species	
PC Coll#	A sequential species specific number is marked on the data sheet in the upper right-hand corner with a RED pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.	
Date	Date the sample was landed, mo / da /yr __/__/__	
Source	ALMR:	Alabama Marine Resources
	CO-OP:	Cooperative Research Proposal
	FMRI:	Florida Marine Research Institute
	FWCLE:	Florida Wildlife Commission Law Enforcement
	HB:	Beaufort Head Boat
	MRFSS:	Marine Recreational Fisheries Statistical Survey
	MSLAB:	Mississippi Laboratory
	NCDMR:	North Carolina Department for Marine Resources
	PCLAB:	Panama City Laboratory
	POP:	Pelagic Observer Program
	RECFIN:	Recreational Fisheries Information Network
	SCDNR:	South Carolina Department for Natural Resources
	SBLOP:	Shark Bottom Longline Observer Program
	TIP:	Trip Interview Program
	USGS:	United States Geological Survey
	VADMR:	Virginia Department for Marine Resources
Source #	ALMR:	No Source #
	CO-OP:	First 4 digits = 4 digit year – Next 2 digits = first and last initial of captain – Last 2 digits = Trip #

FMRI: First 2 or 3 digits = boat name abbreviation –
Next 2 or 3 digits = Cruise # –
Last 2 or 3 digits = Station #

FWCLE: No Source #

HB: 5 digit Collection number

MRFSS: First digit = assignment # (1 or 2)
Next 4 digits = Interview #
Last 2 digits = Intercept # (01, 02, 03, 04)

MSLAB: First 2 or 3 digits = boat name abbreviation –
Next 2 or 3 digits = Cruise # –
Last 2 or 3 digits = Station #

NCDMR 4 digit year
Next 5 digits = Interview #

PCLAB: For samples prior to 1999, see PC Lab section
For samples 1999 to present:
Leave blank, unless Trap Survey or CI Project:
- Trap Survey:
  - First 2 digits = TS
  - Next 2 digits = 2 digit year (06, 07, 08)
  - Last 3 digits = Trap Survey #
- CI Project
  - First 2 digits = CI
  - Next 2 digits – 2 digit year (06, 07, 08)
  - Last 3 digits = CI Station #

POP 6 digit Trip #

RECFIN First digit = assignment # (1 or 2)
Next 4 digits = Interview #
NIV = No Interview Number

SCDNR No Source # or Associated with TIP

SBLOP Observer initials – Trip # – Haul #

TIP: For samples prior to 2004, see TIP section
For samples 2004 to present:
First 2 digits = agency #
Next 2 digits = State
Last 6 digits = Intercept #

USGS	No Source #
VADMR	No Source #

Collector First Initial and Last Name of sampler

State* State where sample was caugh

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)

*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location City/county where samples were caught t, see Table 2.12.2

Bias Type

NB	No Bias
SB	Size Bias
EB	Effort Bias
SE	Size & Effort Bias
NI	No Information
TS	Targeting Sex Bias

Sample Type

QS	Quota Sampling
AA	As Available
SU	At Sea Sorted
LS	Landed Sorted
LU	Landed Unsorted

Mode

Abbreviation	Mode
CM	Commercial
CP	Charter Party
HB	Head Boat
PR	Private
TRN	Tournament
SS	Scientific Survey

Gear	Abbreviation	Gear
	HL	Hook & Line, Hand-line, Bandit
	LL	Long-line
	SP	Spearfish
	TR	Trap
	TRW	Trawl
	GN	Gillnet, Trammel Net
	CN	Castnet
	VLL	Vertical Longline

Comments	Any additional comments processor has
Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm); Conversions: mm = (inches)* 25.40
FL (mm)	Fork Length (mm); Conversions: mm = (inches)* 25.40
Whole WT (kg)	Weight (kg) of whole fish; Conversions: kg = (pounds)* .4536
Gutted WT (kg)	Weight (kg) of gutted fish; Conversions: kg = (pounds)* .4536
Sex	F or ♀ Female M or ♂ Male I Indeterminate T Transition
Samples Taken	Indicates whether each sample had otoliths, gonads, or both
# of Gonads	Total number of gonads in the collection
# of Otoliths	Total number of otoliths in the collection
Condition (S/W)	Condition of the otolith S = Sectioned W = Whole
Data Entered	This information is recorded when data on this sheet is entered into the computer in the appropriate database at the PC Lab.
Initials	Initials of the PC Lab personnel who entered the data into the computer
Date	Date when data has been completely entered into the computer

Figure 2.6.1 Datasheet to be completed for samples received without an accompanying datasheet

PC Lab Data Entry Sheet

Species _____

PC Coll # _____

Month / Day / Year

Date: _____/_____/_____ Source: _____ Source #: _____

Collector: _____ State: _____ Location: _____

Bias: NB SB EB SE NI TS Sample Type: QS AA SU LS LU

Mode: CM CP HB PR TRN SS Gear: HL LL SP TR TRW GN CN VLL

Comments: _____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G	Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G
				Total:								Total:			

Data Entered

Initials: _____ Date:_____

2.7 North Carolina Department of Marine Fisheries (NCDMR)

A division of the North Carolina Department of Marine Environment and Estuarine Resources

King and Spanish mackerel are the only species received from the North Carolina Department of Marine Fisheries (NCDMF), formerly the North Carolina Department of Marine Resources (NCDMR), for processing and ageing. These data sheets are unique to NCDMR and examples are included at the end of this section (Figures 2.7.1 & 2.7.2). Data fields highlighted indicates data used to complete the log. The Species Name and Species Code are located on side 2 of the NCDMR data sheet. Be aware, that gear codes differ from those used by TIP.

Key to NCDMR Data Sheet

Species: Abbreviation of species being sampled, see Chapter 1, Table 1

PC Coll #: A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Date: Date of collection from sampler (mo/da/yr)

Source: NCDMR

Source #: 9-digit number, assigned by NCDMR
4-digit Year + 5-digit Sequence #

Mackerel State: NC (North Carolina)

Location: City/Town or port name recorded on data sheet

Collector: First Initial and Last Name of sampler

Initials	Collector
CC	Chip Collier
CY	Carole Yoder
FS	Felix Del Toro Silva
JC	Jean Conway
NC	Nick Chaplinski
RG	Randy Gregory
RC	Rich Carpenter

Since only the first and last name initials of the sampler (collector) is indicated on the datasheet, use this table of collector's full names.

# of Otoliths: Total number of otoliths in that collection

Condition (S/W): Condition of the otolith
S Sectioned
W Whole

Mode:

TRN	Tournament, KMT, Day 1 or 2
CM	Commercial
CP	Charter
PR	Private

If mode or gear not recorded, please contact NCDMR

Gear: North Carolina gear codes differ greatly with those used for TIP samples

Numeric Code	Gear Description	Gear Code
130	Poundnet	CN
200	Gill Net (unclassified)	GN
220	Gill Net (mono/sink/anchor)	GN
241	Gill Net (mono/sink/run-around)	GN
245	Gill Net (mono/float/anchor)	GN
248	Gill Net (run-around/single panel)	GN
249	Gill Net (run-around/double panel)	GN
260	Trammel Net	CN
320	Haul Seine	LH
341	Beach stop net	CN
352	Commercial Long Haul	LH
353	Swipe Net	CN
371	Cast Net	CN
420	Hook and Line (unclassified)	HL
422	Trolling	HL
500	Trawl (unclassified)	TRW
543	Flounder trawl (deep water)	TRW
556	Juvenile Trawl	TRW
560	Shrimp Trawl (commercial)	TRW
800	Hand Gear (unclassified)	HL
835	Gig	SP
927	Dive/Spear	SP
928	Dive/Gig	SP
940	Electroshock	N/A

Comments: Any other information not listed above

Fish # Fish # is the Line Number

Fork Length (mm) Length is Fork Length

Weight (kg) *usually* whole weight

Samples O otoliths

Sex
1 Male
2 Female
3 Indeterminate

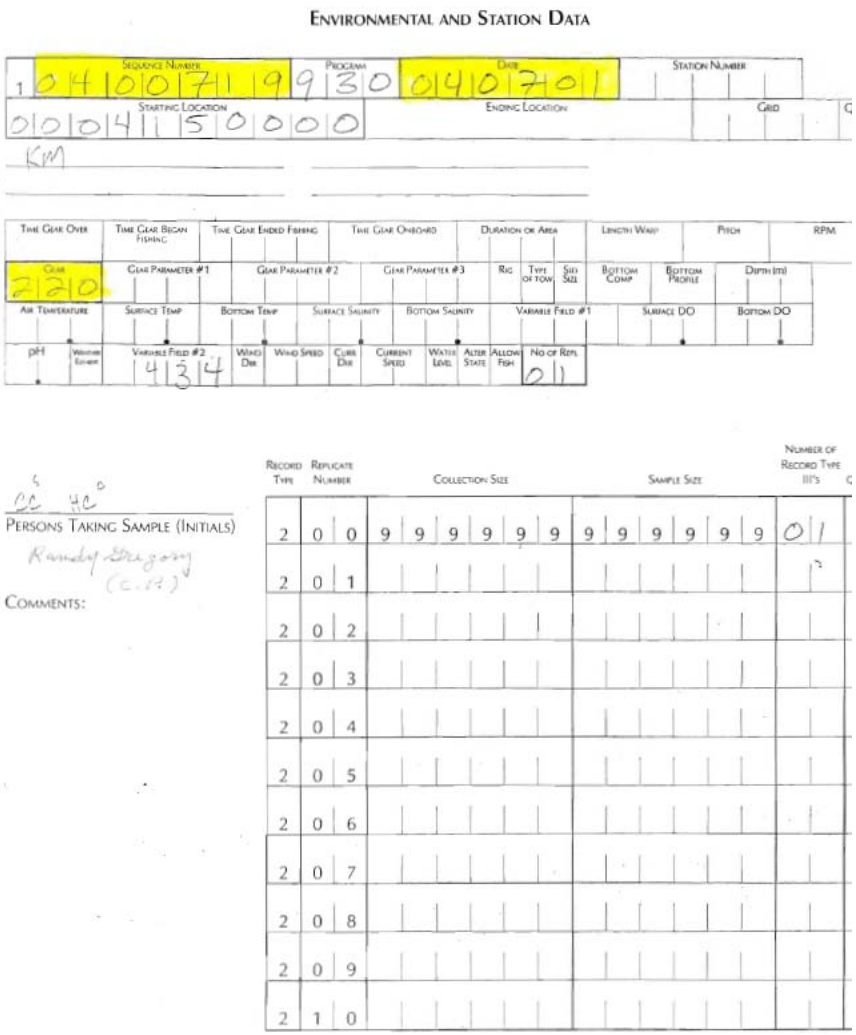

Figure 2.7.1. NCDMR data sheet example- date, sequence number, mode, gear, collector, and vessel data

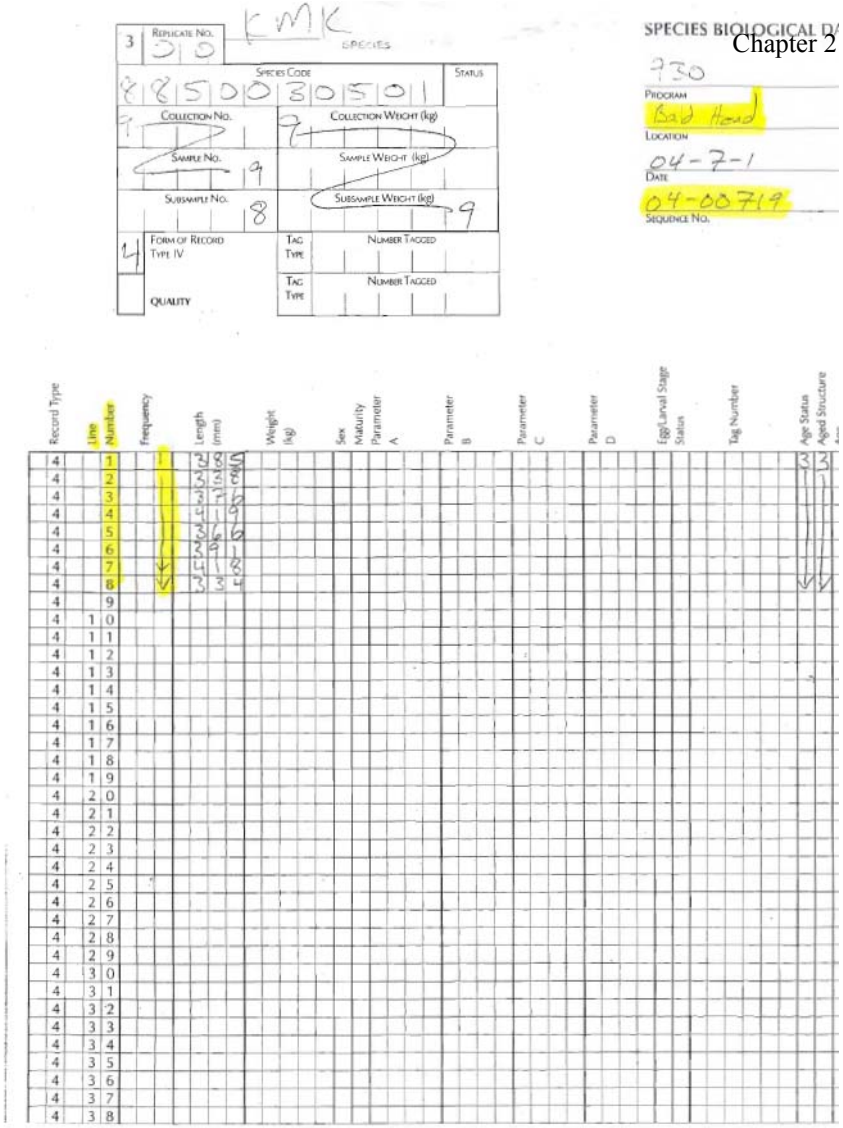

Figure 2.7.2. NCDMR data sheet example-location, sequence number, and individual fish data

2.8 Panama City Laboratory Samples (PCLAB)

Panama City Laboratory collects commercial, recreational, and scientific survey samples. Prior to 1998, a computerized board generated a computer printout to record sampling data, see 2.12 TIP for a key to interpreting the computer printout (Figure 2.12.2). Also prior to 1999, a TIP Recreational Data Sheet was used to record data. Currently, two data sheets are being used to record data; one for only otoliths samples (PC Lab Data Entry Sheet, Figure 2.8.1) and the other for both otolith and gonad samples (Gonad Processing Data Sheet, Figure 2.8.2). Examples of previous and current forms are included in this section. Data fields highlighted indicates data used to complete the log.

Key to PC Lab Data Entry Sheet

Species — Abbreviation of species being sampled, see Chapter 1, Table 1

PC Coll #: — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a RED pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Date: — Date of collection from sampler (mo/da/yr)

Source: — PCLAB

Source #: — Prior to 1999, use Interview Number
1999 to present, leave blank, unless Trap Survey or CI Project

Trap Survey:
- First 2 digits = TS
- Next 2 digits = 2 digit year (06, 07, 08)
- Last 3 digits = Trap Survey #

CI Project
- First 2 digits = CI
- Next 2 digits – 2 digit year (06, 07, 08)
- Last 3 digits = CI Station #

Collector: — (Interviewer Name) First Initial and Last Name of Sampler

State

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina

SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)

*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: City/County samples were landed,
Fishery Independent Survey
If area fished/trapped off of Panama City: Location is reef site.
If area fished/trapped off of another site (Carrabelle, Steinhatchee):
Location is City and reef site in comments.
Commercial/Recreational Sample
Do not enter the fishhouse, use Panama City as Location

Bias Type		
	NB	No Bias
	SB	Size Bias
	EB	Effort Bias
	SE	Size & Effort Bias
	NI	No Information
	TS	Targeting Sex Bias

Sample Type		
	QS	Quota Sampling
	AA	As Available
	SU	At Sea Sorted
	LS	Landed Sorted
	LU	Landed Unsorted

Mode	Abbreviation	Mode
	CM	Commercial
	CP	Charter Party
	HB	Head Boat
	PR	Private
	TRN	Tournament
	SS	Scientific Survey

Gear	Abbreviation	Gear
	HL	Hook & Line, Hand-line, Bandit
	LL	Long-line
	SP	Spearfish
	TR	Trap
	TRW	Trawl
	GN	Gillnet, Trammel Net
	CN	Castnet
	VLL	Vertical Longline

Comments Any additional comments processor has

Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm); Conversions: mm = (inches)* 25.40
FL (mm)	Fork Length (mm); Conversions: mm = (inches)* 25.40
Whole WT (kg)	Weight (kg) of whole fish; Conversions: kg = (pounds)* .4536
Gutted WT (kg)	Weight (kg) of gutted fish; Conversions: kg = (pounds)* .4536
Sex	F or ♀ Female M or ♂ Male I Indeterminate T Transition
Samples Taken	Indicates whether each sample had otoliths, gonads, or both
# of Gonads	Total number of gonads in the collection
# of Otoliths	Total number of otoliths in the collection
Condition (S/W)	Condition of the otolith S Sectioned W Whole
Data Entered	This information is recorded when data on this sheet is entered into the computer in the appropriate database at the PC Lab.
Initials	Initials of the PC Lab personnel who entered the data into the computer
Date	Date when data has been completely entered into the computer

Figure 2.8.1 Datasheet to be completed for samples received without an accompanying datasheet

PC Lab Data Entry Sheet

Species _____

PC Coll # _____

Month / Day / Year

Date: _____/_____/_____ Source: _____ Source #: _____

Collector: _____ State: _____ Location: _____

Bias: NB SB EB SE NI TS Sample Type: QS AA SU LS LU

Mode: CM CP HB PR TRN SS Gear: HL LL SP TR TRW GN CN VLL

Comments: _____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G	Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G
				Total:								Total:			

Data Entered

Initials: _____ Date:_____

Key for Gonad Processing Data Sheet

Species	Abbreviation of species being sampled, see Chapter 1 for appropriate abbreviations.
PC Coll#	Panama City lab collection number (Sequentially numbered for each species); assigned at the lab after samples are received
Date	Date the sample was landed
Source	PC LAB: Panama City Laboratory
Source #	Prior to 1999, use Interview Number 1999 to present, leave blank, unless Trap Survey or CI Project Trap Survey: First 2 digits = TS Next 2 digits = 2 digit year (06, 07, 08) Last 3 digits = Trap Survey # CI Project First 2 digits = CI Next 2 digits – 2 digit year (06, 07, 08) Last 3 digits = CI Station #
Collector	(Interviewer Name) First initial and last name of sampler
State	State where sample was landed
Location	City/county where samples were landed
Latitude	Latitude recorded (degrees, minutes, degree minutes) where sample was caught at sea.
Longitude	Longitude recorded (degrees, minutes, degree minutes) where sample was caught at sea.
Bottom Temp (^oC)	Bottom sea water temperature (^oC)
Depth (m)	Depth, if range less than 5 fathoms take average Conversions: meters = (fathoms) * 1.8288 meters = (feet) * 0.3048
Start Depth (m)	Depth, if range greater than 5 fathoms enter minimum Conversions: meters = (fathoms) * 1.8288 meters = (feet) * 0.3048

End Depth (m)	Depth, if range greater than 5 fathoms enter maximum Conversions: meters = (fathoms) * 1.8288 meters = (feet) * 0.3048	
Time Caught	An approximate time of fish capture. If a range of time is given, take average, using 24 hour clock. Rather than using the number of minutes in the hour, it is recorded as the fraction of the hour. For example, 1:30pm would be recorded as 13.50	
Mode	CM	Commercial
	CP	Charter Boat Party
	HB	Headboat
	PR	Private
	TRN	Tournament
	SS	Scientific Survey
Gear	HL	Handline/ Bandit/ hook and line/ troll
	LL	Long line
	TR	Trap
	SP	Spearfish
	GN	Gillnet
Processed By	Initials of Panama City personnel who processed sample	
Process Date	Date the sample was processed by Panama City Lab	
Preservation Type	Method by which gonad samples were preserved on arrival to lab See Chapter 5, Gonad Procession for code interpretation	
Gonad Comments	Any additional comments processor has. For example, gonad preserved at sea in formalin	
Fish #	Individual fish number supplied by sampler	
TL	Total Length (mm)	
FL	Fork Length (mm	
Whole Wt	Weight (kg) of whole fish	
Gutted Wt	Weight (kg) of gutted fish	
Sex	F or ♀	Female
	M or ♂	Male
	I	Indeterminate

Samples	O	otolith
	G	gonad
	S	spine

Bias Type R/S	Sampler will indicate whether individual samples were randomly taken (R) or selected (S).

For further explanation and code interpretation of the following data fields, see Chapter 5: Gonad Processing.

Macro Class	Preliminary maturation stage
Max OD	Maximum oocyte diameter, taken from largest oocyte in gonad sample, calculated in ocular micrometer units (=OMUs) not MMs
Micro Stage	Most advanced maturation stage of gonad sample
Gonad Cond. (previous form)	Gonad Condition, variations in the physical appearance of gonad
Gonad WT (g)	Weight of gonad to the nearest 0.1 g
% Gonad	Estimate of amount (in whole %) of gonad provided
Remarks (previous form)	Qualifying remarks concerning the condition of the gonad sample
Kept (Y/N)	Kept? yes or no
Histo #	Sequential PCL Cassette # assigned during blocking
Gonad Comment	Notes and observations about each gonad
Total	Total number of otoliths and/or gonads in sample
Data Entered	This information is recorded when data on this sheet is entered into the computer in the appropriate database.
Initials	The initials of the PC Lab personnel who entered the data into the computer
Date	Date when data has been completely entered into the computer

Figure 2.8.2. Gonad Processing Data sheet ,use this datasheet with both otoliths and gonads collected.

Gonad Processing Data Sheet

Species: _____
PC Coll #: _____

Date: _____/_____/_____ Source: _____ Source#: _____
Collector: _____ State: _____ Location:_____

Latitude: _____ Longitude: _____ Bottom Temp (^{o}C): _____ Time Caught: _____
Depth (m): _____ Start Depth (m):_____ End Depth (m):_____

Mode: CM CP HB PR TRN SS Gear: HL LL TR SP GN

Processed By: _____ Process Date: ____/____/____ Preservation Type: _____

Collection Comments: _____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G	Bias Type R/S	Macro Class (gonad)	Max OD	Micro Stage (oocyte)	% Gonad	Gonad WT (g)	Kept Y/N	Histo #	Gonad Comment
					Total:											

Data Entered: Initials: _____ Date: _____

After data has been entered into the appropriate annual database this data sheet is initialed and dated by the technician. This data sheet, along with a copy of the original field datasheet is filed in the species specific Reef Fish Reproductive Study Notebook.

2.9 Pelagic Observer Program (POP)

Pelagic Observer Program samples are collected by fishery observers from US commercial fishing vessels. Currently, Blue Fin Tuna samples are being collected by observers and shipped to NMFS Panama City. As soon as samples arrive please contact the lab manager. Data fields highlighted indicates data used to complete the log.

Key to Pelagic Observer Program Data Sheets

Species	Bluefin Tuna, BFT (see Chapter 1, Table 1)
PC Coll #	Assigned by species and grouped by Trip ID #. A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.
Collector	First initial and last name of observer
Source	POP
Source #	Trip ID # Assigned to each observer by the NOAA Fisheries, Southeast Fisheries Science Center, Miami by Program Supervisor Larry Beerkircher, Lawrence.R.Beerkircher@noaa.gov See lab manager for current observer name and source # list.
Date	Date of Capture
Carcass Tag #	Each observer begins at 1 for each Bluefin tuna sampled and sequentially number the proceeding Bluefin tuna sampled during the observational period. A sequential alphanumeric on the Animal Log's "Carcass Tag" data field. For example: BFT01, BFT02, BFT101, BFT102, etc.
Sampling Date	Date sample taken from fish
State	FL (Florida) or LA (Louisiana) State to which the observer is assigned.
Location	Left Blank
Mode	CM Commercial
Gear	LL Longline

Biological Sampling Checklist:

Each observer needs to complete a 'Biological Sampling Checklist' for each Bluefin Tuna sampled. The checklist will indicate which samples were collected. Verify that each sample is labeled with the Trip ID #, date of capture, and Carcass Tag #. Each sample should have a tag inside the appropriate container (vial, ziplock, etc) and each container should be labeled with the same information (Trip ID#, date of capture, and Carcass Tag #).

Otoliths w/o head*		Gonad subsample 1 (hist)*	
Otoliths w/ head[1]		Gonad subsample 2 (hist)*	
Dorsal Spine[1]		Gonad subsample 3 (hist)*	
Vertebrae[1]		Gonad subsample 4 (hist)*	
Muscle[1]		Gonad subsample 5 (hist)*	
Skin[1]		Gonad subsample 6 (hist)*	
Liver[1]		Gonad subsample 7 (frozen)[2]	
Liver subsample (hist)*		Other (ex. Whole gonad)	

* These samples should be stored and shipped at room temperature, **do not** refrigerate or freeze.

[1] These samples can be stored either on ice or frozen while on the vessel, but should only be stored and shipped frozen after return to port.

[2] These samples should be frozen as soon as possible after collection, and should remain frozen during storage and shipping.

Other	Any additional samples taken describe here
Whole Gonad Weight:	Gonad Weight (kg)
Comments	Any other information pertaining to the collection not listed
Date shipped	Found on shipping label or the date delivered by observer

2.10 Recreational Fisheries Information Network (RECFIN)

Recreational Fisheries Information Network (RECFIN) is a sampling program similar to MRFSS which intercept biological samples from recreational vessels. RECFIN is a Gulf State Marine Fisheries Commission program and each state has its own sampling supervisor. Most of the RECFIN samples are sent to our facility by the state of Florida, supervised by Richard Cody (FWC/FWRI). We only receive RECFIN data digitally (Figure 2.10.1). Data fields highlighted indicates data used to complete the log.

Key to RECFIN Data Sheet

Species — Abbreviation of species being sampled, see Chapter 1, Table 1

PC Coll #: — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Date: — Date of collection from sampler (mo/da/yr)

Source: — RECFIN

Source #: — 1st digit = assignment No.
Next 4 digits = interviewer ID
Last 3 digits = NIV (No Intercept #)

State*: — State Alpha Abbreviation

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: — City/County samples were landed

Collector: — First Initial and Last Name of sampler
See Table 2.4.1 For current list of samplers and ID #'s.

Gonad #: — Total number of gonads in the collection

Otolith #: Total number of otoliths in the collection

Condition (S/W): Condition of the otolith
S Sectioned
W Whole

Mode:
CP Charter Party
HB Head Boat
PR Private Boat
PR Shore Fishing

Gear:
HL Hand Line
A majority of MRFSS samples are from charter, head boat, or private boats that only use some type of hand lines, i.e. rod & reel.

Fish # Individual fish number supplied by sampler

TL (mm) Total Length (mm)

FL (mm) Fork Length (mm)

Whole Wt (kg) Weight (kg) of whole fish

Sex
F or ♀ Female
M or ♂ Male
I Immature
T Transitional stage

Comments: Any other information not listed above
MRFSS often times collects 2 otoliths from each fish; this can be indicated in the comments.

Figure 2.10.1. Example of computer generated datasheet constructed by Age, Growth, and Reproduction lab supervisor.

Samples from the State of Florida Samplers
Date Received: June 1, 2007

Year Species_____
PC Coll #_____

Year & Species	Date	Source	Source #	State	Location	Collector	Mode	Gear	Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	Sex	Comments
2007 RDG	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	671	485	482			F	
2007 RDG	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	672	517	511			F	
2007 RDG	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	673	815	742			F	
2007 KMK	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	674	880	800			F	
2007 KMK	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	675	665	600			M	
2007 KMK	3/25/2007	RECFIN	10009NIV	FL	Clearwater	S. Candileri	CP	HL	676	440	408			F	

2.11 Shark Bottom Longline Observer Program (SBLOP)

Otoliths and gonads collected from NMFS Shark Observer Program are collected on-board commercial fishing vessels. This program is supervised by John Carlson (NOAA Fisheries, Panama City, FL). These samples are accompanied by at least three completed datasheets entitled 'Trip Summary' (Figure 2.11.1), 'Haul Log' (Figure 2.11.2) and 'Animal Log' (Figure 2.11.3). Otoliths and gonads are logged in according to the species and unique OBS/TRIP ID-HAUL #. Data fields highlighted indicates data used to complete the log.

Species	Animal Log: Species – use valid PC LAB species abbreviations Note: Shark Observer Program has different species abbreviations for some species (See Table 2.11.4).
PC Coll #	Assigned by species and grouped by haul # per trip. A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.
Date	Haul Log: Set/Hal Info – begin of set
Source	SBLOP – Shark Bottom Longline Observer Program
Source #	Observer initials – Trip # – Haul # (i.e. JLC, SBC)
State	Determined by the latitude and longitude of fish caught
Location	Trip Summary: Returning dock
Collector	First Name Initial, Last Name (observer)
# of Gonads	Total number of gonads in collection
# of Otoliths	Total number of otoliths in collection
Condition (S/W)	Condition of the otoliths W Whole
Mode	CM Commercial
Gear	LL – Haul Long: Gear Code 675, 676
Collection Comments	Such as 'Gonads preserved at sea'

Start Latitude	Haul Log: Set Haul Into – beginning and end of gear set
End Latitude	
Start Longitude	
End Longitude	
Vessel Name	Trip Summary: Vessel Name or Haul Log: Vessel Name
Fish #	Animal Log: Specimen Number
Fork Length (mm)	Animal Log: Length
Samples	Animal Log: Samples Taken – O, G
Sex	Animal Log: Sex – M, F, U
# otoliths per fish	Number of otoliths per fish –1-2
Otolith Comments	Are the otoliths whole, chipped or broken?
Preservation Type	3 – Preserved in 10% buffered formalin ASAP aboard ship

Figure 2.11.1. NMFS Shark Bottom Longline Observer Program Trip Summary

TRIP SUMMARY
(This will be the cover sheet to your trip datasheets)

Trip ID: _____ **Vessel name:** _____ **Doc #:** _____

Incidental Take: Y / N If Yes what haul number(s): _____

Biological Samples Taken: Y / N

Departure Date: _____ **Departure Port:** _____
mm/dd/yyyy City, State

Return Date: _____ **Return Port:** _____
mm/dd/yyyy City, State

Sea Days: _____ **Number of Hauls:** _____

Weigh out:

Carcass lbs.: _____ **Fins lbs.:** _____ **Ratio:** _____ **Copy included** Y / N

Invoice: place 'x' where appropriate

____ Enclosed with trip data

____ Left completed with captain/owner

____ Enclosed with Trip ID(s) _____.
For multiple trips per invoice.

Vessel accommodations:
Head: Y / N **AC and/or Heat:** Y / N (circle which applies)

Bunk: Y / N If no comment:_____
Comments:

Office use only:

Data received: _____ Trip Number: _____

Debrief: _____/_____ Dbase proofed: _____

Data entry: _____ Animal Log proof: _____

Figure 2.11.2. NMFS Shark Bottom Longline Observer Program Haul Log

BOTTOM LONGLINE HAUL LOG *NOAA Fisheries Panama City Laboratory SBLOP*

1 OBS/TRIP ID	2 VESSEL NAME	3 VESSEL NUMBER	4 DATE LANDED mm/dd/yyyy	5 HAUL #	6 GEAR CODE	7 PAGE NUMBER 1 OF _____

8 HAUL OBS? (Y/N)	9 CATCH? (Y/N)	10 INC TAKE? (Y/N)	11 WEATHER	12 WIND SPEED	12 WIND DIRECTION	13 MAX WAVE HEIGHT	14 REVERSE HAUL? (Y/N)	15 GEAR COND	16 STRING #

17 MAINLINE LENGTH	18 SET SPEED	19 BOTTOM DEPTH RANGE	20 TOTAL ADD. WEIGHT	21 TARGET SPECIES ABBR	22 SOAK DURATION
. NM	. KN	____ FM			

23 GEAR USED

	Y/N	NUMBER
FLOATS	_____	_____
HIGH FLYERS	_____	_____
ADD LINE WEIGHTS	_____	_____

24 NUMBER OF HOOKS

SET _____

LOST _____

25 BAIT INFORMATION

	NUMBER	LBS	KIND	TYPE	COND
1	_____	_____	_____	_____	_____
2	_____	_____	_____	_____	_____
3	_____	_____	_____	_____	_____
4	_____	_____	_____	_____	_____
5	_____	_____	_____	_____	_____

26 SET/HAUL INFO		DATE mm/dd/yyyy	TIME 24 hours	LATITUDE HH	LATITUDE MM	LONGITUDE HH	LONGITUDE MM	TEMP (F)	DURATION hours
SET	BEGIN		:					.	
	END		:					.	.
HAUL	BEGIN		:					.	
	END		:					.	.

COMMENTS

Figure 2.11.3. NMFS Shark Bottom Longline Observer Program Animal Log

BOTTOM LONGLINE ANIMAL LOG

NOAA Fisheries Panama City Laboratory SBLOP

OBS/TRIP ID	VESSEL NAME	VESSEL NUMBER	DATE OF HAUL mm/dd/yyyy	HAUL NUMBER	PAGE _____ of _____

Specimen Number	*SPECIES* Name (Abbr)	*SPECIES* Code	*STATUS* Unk (0) Alive (1) Dead (2) Damaged (3)	*ACTION* Unk (0) Kept (1) Released dead (2) Released alive (3) Finned (4) Lost (5) Tended (6) Kept as bait (7) Part of carcass kept (8)	*LENGTH* (cm) FL Straight line (1) Curved line (2) Estimated (3) FL	*LENGTH* Code	*SEX* (M, F, U)	*TAG NUMBER OR COMMENT*	*TAG CODE* Released (1) Retagged (2) Recaptured (3)	*SAMPLES TAKEN* (Check appropriate boxes) Vert	Stom	Repro	Fin	Pic

Comments:

Table 2.11.4 Shark Bottom Longline Observer Program Species List

Common Name	Scientific Name	Code	NMFS #
AMBERJACK GREATER	*Seriola dumerili*	GAJ	1812
AMBERJACK LESSER	*Seriola fasciata*	LAJ	1815
AMBERJACKS	*Seriola* sp.	AMJ	0030
ANGELFISH BLUE	*Holocanthus bermudensis*	BAF	0579
ANGELFISH FAMILY	*Pomacanthidae*	ANF	0578
ANGLERFISH MONKFISH	*Lophius sp.*	AGL	0121
BALLOONFISH	*Diodon holocanthus*	BAL	0031
BARRACUDA FAMILY	Sphyraenidae	BAR	0180
BARRACUDA GREAT	*Sphyraena barracuda*	GBA	0181
BATFISH FAMILY	Ogcocephalidae	BAT	0032
BATFISH POLKA-DOT	*Ogcocephalus radiatus*	BPD	0033
BILLFISH FAMILY	Istiophoridae	BIL	0190
BLUEFISH	*Pomatomus saltatrix*	BLU	0230
BONITO	*Sarda sarda*	BON	0330
BRYOZOA	Bryozoa	BRY	7200
BUMPER ATLANTIC	*Chloroscombrus chrysurus*	ABU	0040
BUTTERFISH	*Peprilus triacanthus*	BUT	0037
BUTTERFISH GULF	*Peprilus burti*	BGF	0038
CATFISH GAFFTOPSAIL	*Bagre marinus*	CGF	0035
CATFISH HARDHEAD	*Arius felis*	CHH	0036
CIGARFISH BIGEYE	*Cubiceps* sp.	CUB	0530
COBIA	*Rachycentron canadum*	CBA	0570
CORAL	Anthozoa	COR	8160
CORNETFISH BLUESPOTTED	*Fistularia tabacaria*	FIS	0010
COWFISH SCRAWLED	*Lactophrys quadricornis*	COW	0028
CRAB ATLANTIC ROCK	*Cancer irroratus*	CAI	7120
CRAB BLUE	*Callinectes sapidus*	CBL	0114
CRAB CANCER	*Cancer* spp.	CAC	7140
CRAB FLAME BOX	*Calappa flammea*	CAF	7130
CRAB JONAH	*Cancer borealis*	CAB	7110
CRAB KING	*Paralithodes sp.*	CAK	7090
CRAB PORTUNIDAE	Portunidae	CPO	7020
CRAB SPIDER	Majidae	MAJ	7187
CROAKER ATLANTIC	*Micropogonias undulatus*	CRO	0041
CUBBYU	*Equetus umbrosus*	CYU	0042
CUTLASSFISH ATLANTIC	*Trichiurus lepturus*	CAT	0009
DEALFISH FAMILY	Trachipteridae	DEA	0985
DOLPHIN FISH	*Coryphaena hippurus*	DOL	1050

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
DRUM RED	*Sciaenops ocellatus*	RDD	1082
DRUM STAR	*Stellifer lanceolatus*	DST	0045
EEL BEARDED BROTULA	*Brotula barbata*	BBR	1145
EEL CONGER FAMILY	Congridae	CNG	1136
EEL KING SNAKE	*Ophichthus rex*	KSE	1144
EEL MORAY FAMILY	Muraenidae	MEL	1139
EEL PALE-SPOTTED	*Ophichthus ocellatus*	PSE	1140
EEL RETICULATE MORAY	*Muraena retifera*	RMO	1141
EEL SPOTTED MORAY	*Gymnothorax moring*	SMO	1142
EELS	Anguilliformes	EEL	1143
ESCOLAR (SMOOTH SKIN)	*Lepidocybium flavobrunnrum*	GEM	2501
FILEFISH UNICORN	*Aluterus monoceros*	FUN	0109
FILEFISH WHITESPOTTED	*Catherines macrocerus*	FIW	0046
FILEFISHES	*Aluterus* sp.	FLE	0047
FLOUNDER	*Paralichthys* sp.	FLO	0048
FLOUNDER GULF	*Paralichthys albigutta*	FLG	0049
FLOUNDER OCELLATED	*Anclyopsetta quadrocellata*	FLC	1250
FLOUNDER SOUTHERN	*Paralichthys lethostigma*	FLS	0050
GROUPER BLACK	*Mycteroperca bonaci*	**BLG**	**1422**
GROUPER FAMILY	Serranidae	**GRP**	**1410**
GROUPER GAG	*Mycteroperca microlepis*	**GAG**	**1423**
GROUPER GOLIATH	*Epinephelus itajara*	**GOL**	**1421**
GROUPER GRAYSBY	*Epinephelus cruentatus*	**GSG**	**1428**
GROUPER MARBLED	*Epinephelus inermis*	**MBG**	**1417**
GROUPER MISTY	*Epinephelus mystacinus*	**MSG**	**1420**
GROUPER RED	*Epinephelus morio*	**RGR**	**1416**
GROUPER SCAMP	*Mycteroperca phenax*	**CGR**	**1424**
GROUPER SNOWY	*Epinephelus niveatus*	**OGR**	**1414**
GROUPER WARSAW	*Epinephelus nigritus*	**WGR**	**4740**
GROUPER YELLOWEDGE	*Epinephelus flavolimbatus*	**YEG**	**1415**
GROUPER YELLOWFIN	*Mycteroperca venenosa*	**YFG**	**1426**
GROUPER YELLOWMOUTH	*Mycteroperca interstitialis*	**YMG**	**1425**
GRUNT BARRED	*Conodon nobilis*	BGU	1427
GRUNT BLUESTRIPED	*Haemulon sciurus*	SGU	0051
GRUNT CAESAR	*Haemulon carbonarium*	CGU	1429
GRUNT MARGATE	*Haemulon album*	MGT	1442
GRUNT WHITE	*Haemulon plumieri*	WGT	1441
GUITARFISH ATLANTIC	*Rhinobatos lentiginosus*	GUI	0052

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
HAKE	*Merluccius* spp.	GAD	5070
HAKE SILVER	*Merluccius bilinearis*	HSL	5090
HAKE SOUTHERN	*Urophycis floridana*	HAK	3901
HARVESTFISH	*Peprilus alepidotus*	HAR	0053
HERRING ATLANTIC THREAD	*Opisthonema oglinum*	HAT	0054
HERRING FAMILY	Clupeidae	**HER**	**0055**
HIND RED	*Epinephelus guttatus*	**REH**	**1413**
HIND ROCK	*Epinephelus adscensionis*	**RHI**	**1412**
HIND SPECKLED	*Epinephelus drummondhayi*	**SHI**	**1411**
HOUNDFISH	*Tylosurus crocodilus*	HOU	0056
JACK ALMACO	*Seriola rivoliana*	AJC	1810
JACK BLUERUNNER	*Caranx crysos*	JBR	0270
JACK CREVALLE	*Caranx hippos*	JCR	0870
JACK FAMILY	Carangidae	JKF	0057
JACKS	*Caranx* sp.	JAK	0034
KINGFISH	*Menticirrhus* sp.	KIG	1811
KINGFISH GULF	*Menticirrhus littoralis*	KGU	0058
KINGFISH NORTHERN	*Menticirrhus saxatilis*	KNO	0059
KINGFISH SOUTHERN	*Menticirrhus americanus*	KSO	0060
LADYFISH	*Elops saurus*	LAD	0111
LANCETFISH	*Alepisaurus* sp.	LAX	2035
LEATHERJACKET FAMILY	Balistidae	LEA	0061
LITTLE TUNNY	*Euthynnus alletteratus*	LTA	4653
LIZARDFISH FAMILY	*Synodontidae*	LIZ	0029
LIZARDFISH INSHORE	*Synodus foetens*	LZD	0062
LIZARDFISH SANDDIVER	*Synodus intermedius*	LSD	0039
LOBSTERS	Nephropidae	LOB	0113
LOOKDOWN	*Selene vomer*	LKD	0063
MACKEREL ATLANTIC	*Scomber scombrus*	AMK	0064
MACKEREL BULLET	*Auxis rochei*	BMK	0065
MACKEREL CERO	*Scomberomorus regalis*	MCE	0066
MACKEREL CHUB	*Scomber japonicus*	CHM	2150
MACKEREL FRIGATE	*Auxis thaza*	FRM	1900
MACKEREL KING	*Scomberomorus cavalla*	KGM	1940
MACKEREL SNAKE	*Trichiuridae* sp.	TRX	2504
MACKEREL SPANISH	*Scomberomorus maculatus*	SMK	3840
MARLIN BLUE	*Makaira nigricans*	BUM	2179
MARLIN WHITE	*Tetrapturus albidus*	WHM	2177

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
MENHADEN	*Brevoortia* sp.	MEN	0067
MENHADEN ATLANTIC	*Brevoortia tyranus*	MAT	0068
MENHADEN GULF	*Brevoortia patronus*	MGU	0069
MENHADEN YELLOWFIN	*Brevoortia smithi*	MYF	0070
MIX	Mixed Species	MIX	0001
MOLLUSC	Mollusca	MOL	0002
MOONFISH	*Selene setapinnis*	MNF	0071
NEEDLEFISH ATLANTIC	*Strongylura marina*	ATN	0190
OILFISH (ROUGH SKIN)	*Revetus pretiosus*	OIL	2502
OPAH	*Lampris guttatus*	OPA	2503
PERCH SAND	*Diplectrum formosum*	PSA	3110
PERCH SILVER	*Bairdiella chrysoura*	PSI	0072
PERMIT	*Trachinotus falcatus*	PER	0073
PIGFISH	*Orthopristis chrysoptera*	PIG	0074
PILOTFISH	*Naucrates ductor*	PLF	0075
PINFISH	*Lagodon rhomboides*	PIN	2670
POMFRETS	*Brama* sp.	POA	2710
POMPANO AFRICAN	*Alectis ciliaris*	PAF	2719
POMPANO FLORIDA	*Trachinotus carolinus*	PFL	2720
PORCUPINEFISH	*Diodon hystrix*	POQ	3579
PORGY FAMILY	Sparidae	PRG	3580
PORGY JOLTHEAD	*Calamus bajonado*	JPO	3312
PORGY LITTLEHEAD	*Calamus proridens*	POL	0076
PORGY RED	*Pagrus pagrus*	PRD	3300
PORGY SAUCEREYE	*Calamus calamus*	POS	0077
PORGY SILVER	*Diplodus argenteus*	SPR	3313
PORGY WHITEBONE	*Calamus leucosteus*	POW	0078
PUFFER FAMILY	Tetraodontidae	PUX	2760
PUFFER SPINY FAMILY	Diodontidae	PUS	0079
RAINBOW RUNNER	*Eleganyis bipinnulata*	RUN	1814
RAY BULLNOSE	*Myliobatis freminvillei*	RBU	3652
RAY BUTTERFLY	*Gymnura* sp.	RBT	3651
RAY COWNOSE	*Rhinoptera bonasus*	RCN	3653
RAY DEVIL	*Mobula hypostoma*	DEV	3654
RAY EAGLES	*Myliobatis* sp.	EAG	3655
RAY LESSER ELECTRIC	*Narcine brasiliensis*	RLE	0080
RAY MANTA	*Manta birostris*	RMA	0081
RAY SPOTTED EAGLE	*Aetobatis narinari*	SPE	3656

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
REMORA	*Remora remora*	RRM	0082
REMORA FAMILY	Echeneidae	REM	2865
RUDDERFISH BANDED	*Seriola zonata*	RUD	1817
SAILFISH	*Istiophorus platypterus*	SAL	0083
SAILFISH ATLANTIC	*Istiophorus albicans*	SAI	3026
SAND FLEA	Crustacea	SFE	7109
SAWFISH LARGETOOTH	*Pristis pristis*	LSW	3506
SAWFISH SMALLTOOTH	*Pristis pectinata*	SSW	0084
SCORPIONFISH FAMILY	Scorpaenidae	SCO	0085
SCORPIONFISH SPINYCHEEK	*Neomerinthe hemingwayi*	SCS	0086
SEABASS BLACK	*Centropristis striata*	SBL	0087
SEABASS FAMILY	*Serranidae*	SBF	0088
SEAHORSE LINED	*Hippocampus erectus*	SHL	0112
SEAROBIN LEOPARD	*Prionotus scitulus*	SEL	0008
SEAROBINS	*Prionotus* sp.	SER	0089
SEATROUT SILVER	*Cynoscion nothus*	STS	0091
SEATROUT WEAKFISH	*Cynoscion regalis*	STW	0092
SEATROUTS	*Cynoscion* sp.	STT	0090
SHAD	*Alosa sp.*	SHA	3474
SHARK ATLANTIC ANGEL	*Squatina dumerili*	ANG	3582
SHARK ATLANTIC SHARPNOSE	*Rhizoprionodon terraenovae*	SAS	3518
SHARK BIGEYE SIXGILL	*Hexanchus vitulus*	BSX	3529
SHARK BIGNOSE	*Carcharhinus altimus*	SBG	3491
SHARK BLACKNOSE	*Carcharhinus acronotus*	SBN	3485
SHARK BLACKTIP	*Carcharhinus limbatus*	SBK	3495
SHARK BLUE	*Prionace glauca*	BSH	3504
SHARK BONNETHEAD	*Sphyrna tiburo*	BHH	3483
SHARK BULL	*Carcharhinus leucas*	SBU	3497
SHARK CROCODILE	*Pseudocarcharias kamoharai*	SCR	3578
SHARK DOGFISH CHAIN CATSHARK	*Scyliorhinus retifer*	DGC	3520
SHARK DOGFISH FAMILY	Squalidae	SDG	3503
SHARK DOGFISH SMOOTH	*Mustelus canis*	DGS	3511
SHARK DOGFISH SPINEY	*Squalus acanthias*	DGY	3521
SHARK DUSKY	*Carcharhinus obscurus*	DUS	3514
SHARK FINETOOTH	*Carcharhinus isodon*	SFT	3481
SHARK GALAPAGOS	*Carcharhinus galapagensis*	GAL	3492
SHARK GREENLAND	*Somniosus microcephalus*	SGR	3532
SHARK HAMMERHEAD	*Sphyrna* sp.	XHH	3516

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
SHARK HAMMERHEAD GREAT	*Sphyrna mokarran*	GHH	3524
SHARK HAMMERHEAD SCALLOPED	*Sphyrna lewini*	SPL	3523
SHARK HAMMERHEAD SMOOTH	*Sphyrna zygaena*	SHH	3522
SHARK LEMON	*Negaprion brevirostris*	LEM	3517
SHARK MACKEREL FAMILY	Lamnidae	SMF	0093
SHARK MAKO	*Isurus* sp.	XMA	3571
SHARK MAKO LONGFIN	*Isurus paucus*	LMA	3502
SHARK MAKO SHORTFIN	*Isurus oxyrinchus*	SMA	3505
SHARK NIGHT	*Carcharhinus signatus*	SNI	3494
SHARK NURSE	*Ginglymostoma cirratum*	NUR	3480
SHARK OCEANIC WHITETIP	*Carcharhinus longimanus*	OCS	3498
SHARK PORBEAGLE	*Lamna nasus*	POR	3501
SHARK REEF	*Carcharhinus perezi*	SRF	3490
SHARK REQUIEM FAMILY	Carcharhinidae	SRQ	0094
SHARK SAND TIGER	*Carcharhias taurus*	SST	3482
SHARK SANDBAR	*Carcharhinus plumbeus*	SSB	3513
SHARK SEVENGILL	*Heptranchias perlo*	SEV	3587
SHARK SILKY	*Carcharhinus falciformis*	FAL	3493
SHARK SIXGILL	*Hexanchus griseus*	SIX	3528
SHARK SMOOTHHOUND FLORIDA	*Mustelus norrisi*	SFL	3507
SHARK SPINNER	*Carcharhinus brevipinna*	SSP	3496
SHARK THRESHER	*Alopias* sp.	XTH	3500
SHARK THRESHER BIGEYE	*Alopias superciliosus*	BTH	3510
SHARK THRESHER COMMON	*Alopias vulpinus*	PTH	3509
SHARK TIGER	*Galeocerdo cuvier*	TIG	3515
SHARK WHITE	*Carcharodon carcharias*	GWS	3512
SHARKS	Elasmobranchii	SHX	3508
SHARKSUCKER	*Echeneis naucrates*	SUK	2863
SHARKSUCKER WHITEFIN	*Echeneis neucratoides*	WSK	2864
SHEEPSHEAD	*Archosargus probatocephalus*	SHE	0095
SKATE CLEARNOSE	*Raja eglanteria*	CLE	3657
SKATES AND RAYS	Rajiformes	SRX	3650
SNAKEFISH	*Trachinocephalus myops*	SKF	0108
SNAPPER BLACK	*Apsilus dentatus*	**BSN**	**3755**
SNAPPER BLACKFIN	*Lutjanus buccanella*	**BFS**	**3757**
SNAPPER CARDINAL	*Pristipomoides macropthalmus*	**CRS**	**3773**
SNAPPER CUBERA	*Lutjanus cyanopterus*	**CSN**	**3759**
SNAPPER DOG	*Lutjanus jocu*	**DSN**	**3754**

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
SNAPPER FAMILY	*Lutjanidae*	**SNA**	**0096**
SNAPPER GRAY	*Lutjanus griseus*	**SNG**	**3762**
SNAPPER LANE	*Lutjanus synagris*	**LUL**	**3761**
SNAPPER MAHOGANY	*Lutjanus mahogoni*	**MHS**	**3772**
SNAPPER MUTTON	*Lutjanus analis*	**MSN**	**3763**
SNAPPER QUEEN	*Etelis oculatus*	**QSN**	**3770**
SNAPPER RED	*Lutjanus campechanus*	**RSN**	**3764**
SNAPPER SCHOOLMASTER	*Lutjanus apodus*	**SMS**	**3771**
SNAPPER SILK	*Lutjanus vivanus*	**SNS**	**3758**
SNAPPER VERMILLION	*Rhomboplites aurorubens*	**SNV**	**3765**
SNAPPER WENCHMAN	*Pristipomoides aquilonaris*	**WNS**	**3756**
SNAPPER YELLOWTAIL	*Ocyurus chrysurus*	**YTS**	**3767**
SPADEFISH	*Chaetodipterus faber*	SPD	0100
SPEARFISH LONGBILL	*Tetrapturus pfluegeri*	SPF	4010
SPEARFISH ROUNDSCALE	*Tetrapturus georgei*	SPG	4009
SPEARFISHES	*Tetrapturus* sp.	SPX	4000
SPONGE	Porifera	PRF	8200
SPOT	*Leiostomus xanthurus*	SPO	0101
SQUIRRELFISHES	*Holocentrus sp.*	SQU	4120
STARFISH	Asteroidea	STF	8280
STINGRAY ATLANTIC	*Dasyatis sabina*	SAT	0102
STINGRAY BLUNTNOSE	*Dasyatis sayi*	SBO	3659
STINGRAY ROUGHTAIL	*Dasyatis centroura*	SRO	3658
STINGRAY SOUTHERN	*Dasyatis americana*	SSO	3660
STINGRAYS	*Dasyatis* sp.	STR	2862
STURGEON ATLANTIC	*Acipenser oxyrhyncus*	STA	0103
STURGEONS	*Acipenser* sp.	STU	0104
SUNFISHES	*Mola* sp.	MOX	4260
SWORDFISH	*Xiphius gladius*	SWO	4320
TARPON	*Megalops atlanticus*	TAR	4350
TILEFISH	*Lopholatilus chamoeleonticeps*	**TIL**	**4471**
TILEFISH BLACKLINE	*Caulolatilus cyanops*	**BKT**	**4476**
TILEFISH BLUELINE (GRAY)	*Caulolatilus microps*	**BLT**	**4474**
TILEFISH GOLDFACE	*Caulolatilus chrysops*	**GFT**	**4472**
TILEFISH SAND	*Malacanthus plumieri*	**MAL**	**4478**
TOADFISH FAMILY	Batrachoididae	TOD	4500
TOADFISH GULF	*Opsanus beta*	TOG	4501
TOADFISH LEOPARD	*Opsanus pardus*	TOL	4502

Table 2.11.4 Shark Bottom Longline Observer Program Species List (cont'd)

Common Name	Scientific Name	Code	NMFS #
TOMTATE	*Haemulon aurolineatum*	TOM	0105
TRIGGERFISH GRAY	*Baslistes capriscus*	**TRG**	**0106**
TRIGGERFISH QUEEN	*Balistes vetula*	TRQ	4563
TRIPLETAIL	*Lobotes surinamensis*	TRI	0107
TUNA ALBACORE	*Thunnus alalunga*	ALB	4651
TUNA BIGEYE	*Thunnus obesus*	BET	4657
TUNA BLACKFIN	*Thunnus atlanticus*	BLK	4658
TUNA SKIPJACK	*Euthynnus pelamis*	SKJ	4654
TUNA YELLOWFIN	*Thunnus albacares*	YFT	4655
TUNAS	*Thunnus* sp.	TUN	4656
UNCODED ANIMAL	Uncoded animal	UNC	9999
UNKNOWN	Unknown animal	UNK	0000
URCHIN SEA	Echinodermata	URC	0003
WAHOO	*Acanthocybium solanderi*	WAH	4710
WHIFF BAY	*Citharichthys spilopterus*	WFB	0110
WORM	Polychaeta	WOR	0004

2.12 Trip Interview Program (TIP)

TIP Port Agents collect commercial and recreational samples. Depending upon the TIP sampler, any one of the following data forms can be used to record data.

Prior to 2004: TIP Recreational Data sheet (Figure 2.12.1)
TIP Computer Print out (Figure 2.12.2)

Post 2004: Reporting Form for the Trip Interview Program (Figure 2.12.3)
Summary Sheet for Biological Samples (Figure 2.12.4)
TIP Online Data Form (Figure 2.12.5)

In 2004, the Trip Interview Program made some adjustments to the Source #, replaced the NMFS State and County Codes with FIPS State and County Codes, all species NODC codes were adapted from 10 to 12 digits, and tag #s include the four digit year, Port Agents special code, and a sequential number. Specific changes to how 2004 samples are logged will be found later in this section.

TIP Samples Received: 1991-2003

The following section includes keys to TIP Data Sheets: 1991-2004. Data fields highlighted indicate data used to complete the log regardless of which data sheet is being used. Only pertinent fields are included.

Key to TIP Recreational Data Sheet

PC Coll #: A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Source: TIP

Source #: Interview Number

1st 2 digits = state # (Table 2.12.2)
Next 2 digits = Interview # (Sampler # and Duty Station)
Last 5 digits = Intercept #

For example: 114700023
11 = FL West Coast
47 = Interview #
00023 = 5 digit sequence number

Mode:

Abbreviation	Mode
CM	Commercial
CP	Charter Party
HB	Head Boat
PR	Private
TRN	Tournament
SS	Scientific Survey

Collector: First Initial and Last Name of Port Agent. If a code is used, see Table 2.12.4 for Interview # of Port Agent.

Date: Date of Interview (mo/da/yr).

State*: Reporting Area/State. If a Reporting State Code is given, see Table 2.12.2.
State Alpha Abbreviation as shown in accompanying list

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: Reporting Area/County. If Reporting County Code is given see Table 2.12.2. A Reporting Port Code may be given.
For example 6386, Bay County.

Bias Type

NB	No Bias
SB	Size Bias
EB	Effort Bias
SE	Size & Effort Bias
NI	No Information
TS	Targeting Sex Bias

Gear:

Code	General Description	TIP Code	Gear Description
HL	Hook & Line	600	Troll & Hand Lines
	Hand-line	610	Lines Hand
	Bandit	611	Rod and Reel
	Troll	612	Manual Reel
		613	Electric or Hydraulic Reel
		616	Electric Reel
		660	Lines Troll, other top waters
		665	Mackerel Troll Lines
LL	Long-line	675	Lines Long set w/ hooks top waters
		676	Lines Long, reef fish bottom waters
SP	Spearfish	760	Spears
TR	Trap	295	Floating Traps
		345	Pots & Traps
		355	Pots & Traps, Spiny Lobster
TRW	Trawl	210	Otter Trawl, Bottom, Fish
		215	Otter Trawl, Bottom, Shrimp
		220	Otter Trawl Bottom, Other
GN	Gillnet,	425	Gill nets
	Trammel Net	475	Gill nets, Drift, Runaround
CN	Castnet	735	Castnet

Weight Type Weights may be recorded as rounded or gutted, pounds, grams, or kilograms

Abbreviation	Weight type
RK	Round/Whole Kilograms
GK	Gutted Kilograms
RP	Round/Whole Pounds
GP	Gutted Pounds
RG	Round/Whole Grams
GG	Gutted Grams

Species: Abbreviation of species being sampled, see Chapter 1 for appropriate abbreviations.

Code Each species has a NMFS code. This may be found in Table 1.1.

# of Gonads: Total number of gonads in the collection

# of Otoliths: Total number of otoliths in the collection

Condition (S/W): Condition of the otolith
S Sectioned
W Whole

Comments: Any other information not listed above

Fish # Fish Tag ID #. Individual fish number supplied by sampler

TL (mm) Total Length (mm); May be recorded in cm or inches:
Conversions: mm = (cm) *10
(inches)* 25.40

FL (mm) Fork Length (mm); May be recorded in cm or inches:
Conversions: mm = (cm) *10
(inches)* 25.40

Whole WT (kg) Weight (kg) of whole fish
Conversion: pounds (lb) * .4536 = kg

Gutted WT (kg) Weight (kg) of gutted fish
Conversion: pounds (lb) * .4536 = kg

Sex F or ♀ = Female
M or ♂ = Male
I = Indeterminate
T = Transition

Figure 2.12.1. TIP Recreational Data Sheet 1991-2004

INTERVIEW NUMBER _____

TARGET FISHERY: CP RF OP IN ML BF EG MX

FISHING MODE: CM CP HB PR TR SS

Agent:_____ Date:_____

REPORTING AREA

State:_____ County:_____ Port:_____

SAMPLE SITE

County:_____ Port:_____ Location:_____

INFORMATION SOURCE

SITESTATE _____ REGION _____ SR LB SS

TRIP TYPE

30 100 200 300 400 600 675 676 900

TIME OF COLLECTION

Begin:__:__ End:__:__

BIAS TYPE

NB SB EB SE NI

INTERVIEW TYPE

FS DS TS AT

LANDING TYPE (Weight)

CL IL NL

CREW SIZE: _____ DAYS OUT: ___.__ DAYS FISHED: ___.__

TERMINATION: UK NR WR

SIZE CODE: UK S M L J

VESSEL ID :_____

VESSEL LENGTH:_____

GEAR

220 425 610 611 612 613 660 675 676 760

Hrs Fished:_____ Area:_____

VESSEL NAME :_____

WEIGHT TYPE: RK GK RP GP RG GG

CATCH STATUS Y N

SAMPLE TYPE: QS AA SU LS LU

LANDING WEIGHT:_____

SPECIES_____ CODE_____ REPLICATE_____ SAMPLE_____ MEASUREMENT: FC FI TC TI FT

FISH TAG ID #	LENGTH FORK (mm) CM	LENGTH TOTAL (mm) CM	WEIGHT (lb) KG	SEX M F U	SAMPLES TAKEN				FISH TAG ID #	LENGTH FORK (mm) CM	LENGTH TOTAL (mm) CM	WEIGHT (lb) KG	SEX M F U	SAMPLES TAKEN			
					O	G	T	D S						O	G	T	D S
1									21								
2									22								
3									23								
4									24								
5									25								
6									26								
7									27								
8									28								
9									29								
10									30								
11									31								
12									32								
13									33								
14									34								
15									35								
16									36								
17									37								
18									38								
19									39								
20									40								

Key to Computer Printout

Prior to 1999, Computer Printouts could be used by all Port Agents that had a Trip Interview Program data-collecting computerized board. The following information explains how to interpret this printout. The highlighted fields should be completed on the log regardless of which data sheet is being used.

PC Coll #: Group the individual fish together that belong to the collection you are logging in by placing a bracket, i.e. } to indicate the fish included in this collection number. A sequential species specific number is marked on the data sheet beside the bracket with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

Source: Trip Interview Program (TIP)

Collector: First Initial and Last Name of Sampler (name or initials of sampler usually handwritten on printout)

Date: A '**d**' indicates the date of sample.

Source #: The new interview number is indicated with '**i**' and the first two digits will be the same as the year of collection.

Location: Next the letter '**c**' indicates the location; each location is represented by 4 or less numbers. Most locations show a code of 6386, which is Panama City.

Bias Type: A '**b**' indicates the bias type; NB = no bias

Mode: The letter '**m**' indicates the mode.

Abbreviation	Mode
CM	Commercial
CP	Charter Party
HB	Head Boat
PR	Private
TRN	Tournament
SS	Scientific Survey

Gear: After mode is the gear indicated by a '**g**' and will be a series of numbers, representing:

TIP Code	Abbreviation	Gear
600, 610-613, 616	HL	Hand-line, Bandit
675, 676	LL	Long-line
760	SP	Spearfish

TIP Code	Abbreviation	Gear
295, 345, 355	TR	Trap
220	TRW	Trawl
425, 475	GN	Gillnet, Trammel Net

The '**e**' indicates the type of weight.

RK – Round/Whole in kilograms
GK – Gutted in kilograms
RP – Round/Whole in pounds (convert to kg)
GP – Gutted in pounds (convert to kg)
RG – Round/Whole in grams (covert to kg)
GG –Gutted in grams (convert to kg)

On that same line the '**r**' states the sample type of effort that was conducted.

QS – Quota Sampling
AA – As Available
SU – At Sea Sorted
LS – Landed Sorted
LU – Landed Unsorted

Species On the following line, starting with an '**s**' indicates the species with the appropriate abbreviation. See the complete list of species and their abbreviations.

An '**n**' and a fish or tag number indicate each fish.

On that same line all lengths, weight, sex, and samples taken from the fish will be indicated.

'**l**' represents the fork length

'**t**' represents the total length

'**w**' indicates the weight

'**x**' is followed by the appropriate sex either F = Female or M = Male

# of Gonads & # of Otoliths = '**a**' indicates what samples were taken:
O = Otolith
G = Gonad

A new line starting with an '**i**' indicates a new trip or interview number.

Figure 2.12.2 TIP Computer Printout/Data Sheet 1991-1998

Source #

COMPUTER PRINTOUT

```
F4093H
03102
14:20:29 07/19/98
d19980718
i9843036c6386vSSyFSbNBmCPg611eRKpNAqNrLU
sSMK
n00110360.0t0000.0w    0.000xMaO
n00210394.0t0000.0w    0.000xMaO
n00310381.0t0000.0w    0.000xFaO
n00410349.0t0000.0w    0.000xMaO
n00510412.0t0000.0w    0.000xMaO
n00610482.0t0000.0w    0.000xFaO
n00710451.0t0000.0w    0.000xFaO
n00810420.0t0000.0w    0.000xFaO
n00910443.0t0000.0w    0.000xFaO
n01010460.0t0000.0w    0.000xMaO
n01110410.0t0000.0w    0.000xFaO
n01210376.0t0000.0w    0.000xMaO
n01310373.0t0000.0w    0.000xMaO
d19980718
i9843037c6386vSSyFSbNBmHBg611eRKpOQqNrLU
sRDP
n01410273.0t0309.0w    0.425xFaOG
n01510251.0t0288.0w    0.345xMaOG
n01610272.0t0305.0w    0.435xMaOG
d19980718
i9843038c6386vSSyFSbNBmCPg611eRKpNAqNrLU
sRS
n01710438.0t0468.0w    1.430xFaOG
n01810362.0t0384.0w    0.775xMaOG
n01910478.0t0502.0w    1.830xMaOG
n02010405.0t0430.0w    1.210xFaOG
n02110598.0t0642.0w    3.880xFaOG
n02210435.0t0463.0w    1.595xFaOG
n02310381.0t0406.0w    1.030xMaOG
n02410544.0t0577.0w    3.190xFaOG
n02510414.0t0440.0w    1.245xFaOG
sGS
n02610491.0t0516.0w    1.750xMaOG
n02710477.0t0500.0w    1.710xMaOG
sRDG
n02810597.0t0629.0w    3.680xFaOG
sKMK
n02910705.0t0000.0w    2.565xFaO
sSCA
n03010440.0t0468.0w    1.130xFaOG
sRDG
n03110734.0t0772.0w    7.695xFaOG
n03210497.0t0516.0w    2.230xFaOG
n03310570.0t0593.0w    3.145xFaOG
sSCA
n03410458.0t0495.0w    1.365xFaOG
n03510576.0t0633.0w    2.400xFaOG
sRDG
n03610783.0t0818.0w    9.260xFaOG
sRDP
n03710378.0t0428.0w    1.085xMaOG
sRDG
n03810755.0t0790.0w    6.340xFaOG
```

DATE
INTERVIEW NUMBER
LOCATION
MODE GEAR WEIGHT TYPE
SPECIES
FISH # FORK LENGTH
START NEW INTERVIEW #
TOTAL LENGTH
WEIGHT
SEX
SAMPLES TAKEN
O = OTOLITH
G = GONAD

d = date
i = interview #
c = location
b = bias type
NB = no bias
m = mode
g = gear
e = weight type
s = species abbreviation
n = fish # or tag #
l = fork length
t = total length
w = weight
x = sex
a = samples taken

TIP Samples Received: 2004-Present

In 2004, the Trip Interview Program made some adjustments to the Source #, replaced the NMFS State and County Codes with FIPS State and County Codes, all species NODC codes were adapted from 10 to 12 digits, and tag #s include the four digit year, interviewer special code, and a sequential number.

TIP Port Agents collect commercial and recreational samples. Our facility has requested that all TIP Port Agents provide a computerized print out of all data from TIPONLINE. Depending upon the TIP sampler, any one of the following forms may be included with the data: "Reporting Form for the Trip Interview Program (TIP), a Summary Sheet for Heads, Otoliths, Spines, and Tissue Samples, and printout from TIPONLINE. Copies of these forms are being included at the end of this section.

The following section includes a key to TIP Reporting Forms: Post-2004 and follows the format for the TIP Online. The . Data fields highlighted indicate data used to complete the log regardless of which data sheet is being used.

Key to TIP Reporting Forms:

Species — Abbreviation of species being sampled, see Chapter 1 for appropriate abbreviations.

PC Coll #: — A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.

<u>In Interview Section of TIP Online:</u>

Collector: (Username) — First Initial and Last Name of sampler

Source: — TIP

Source #: — Interview Number
- 1st 2 digits = agency # (Table 2.12.1)
- Next 2 digits = State Abbreviation
- Last 5 digits = Interview #

Date: — Date of Interview (mo/da/yr)

Mode: Mode Abbreviations:

Abbreviation	Mode
CM	Commercial
CP	Charter Party
HB	Head Boat
PR	Private
TRN	Tournament
SS	Scientific Survey

State*: Landed, State Alpha Abbreviation as shown in accompanying list of state and county codes (Table 2.12.2).

Abbreviation	State
AL	Alabama
E FL	East coast Florida
FL	West coast Florida
GA	Georgia
LA	Louisiana
MS	Mississippi
NC	North Carolina
SC	South Carolina
TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)

*For Mackerel Samples ONLY (See Chapter 1, Figure 1.2)

Location: Landed, County.
If Reporting Area Zip Code is given, see the complete Zip Code list and assign corresponding location. If a Reporting County Code is given, see the list of county codes, both NMFS codes and FIPS codes are provided in this section (see Table 212.2).

Bias Type

NB	No Bias
SB	Size Bias
EB	Effort Bias
SE	Size & Effort Bias
NI	No Information
TS	Targeting Sex Bias

Condition (S/W): Condition of the otolith

S	Sectioned
W	Whole

In Effort Section of TIP Online:

Gear: Gear Information, Code

Code	General Description	TIP Code	Gear Description
HL	Hook & Line	600	Troll & Hand Lines
	Hand-line	610	Lines Hand
	Bandit	611	Rod and Reel
	Troll	612	Manual Reel
		613	Electric or Hydraulic Reel
		616	Electric Reel
		660	Lines Troll, other top waters
		665	Mackerel Troll Lines
Code	General Description	TIP Code	Gear Description
LL	Long-line	675	Lines Long set w/ hooks top waters
		676	Lines Long, reef fish bottom waters
SP	Spearfish	760	Spears
TR	Trap	295	Floating Traps
		345	Pots & Traps
		355	Pots & Traps, Spiny Lobster
TRW	Trawl	210	Otter Trawl, Bottom, Fish
		215	Otter Trawl, Bottom, Shrimp
		220	Otter Trawl Bottom, Other
GN	Gillnet,	425	Gill nets
	Trammel Net	475	Gill nets, Drift, Runaround
CN	Castnet	735	Castnet

Latitude — Latitude recorded (degrees, minutes, and seconds), where caught

Longitude — Longitude recorded (degrees, minutes, and seconds), where caught

Area/Grid — Corresponds to NMFS Shrimp Fishing Zones.

Depth — Approximate depth where sample captured

In Sample Section of TIP Online:

Species Code — Each species has a NMFS code and PC Lab Abbreviation. These may be found in Table 1.1.

# of Gonads: Total number of gonads in the collection

# of Otoliths: Total number of otoliths in the collection

<u>In Observation Sample Section of TIP Online:</u>

Tag #	Individual Fish number supplied by sampler Make sure the tag #s labeled on the envelopes match the tag #s on the datasheet. Each tag number is composed of 3 parts: For example: 200400100001 2004 4 digit year 001 3 digit sampler code (Table 3) 0001 4 digit consecutive tag number
TL (mm)	Total Length (mm); Conversions: mm = (cm) *10 (inches)* 25.40
FL (mm)	Fork Length (mm); Conversions: mm = (cm) *10 (inches)* 25.40
Whole WT (kg)	Weight (kg) of whole fish Conversion: pounds (lb) * .4536 = kg
Gutted WT (kg)	Weight (kg) of gutted fish Conversion: pounds (lb) * .4536 = kg
Sex	F or ♀ = Female M or ♂ = Male I = Indeterminate T = Transition
Samples Taken	Indicates whether each sample had otoliths, gonads, spines, or a combination O = otolith G = gonad S = spine

Comments: Any other information not listed above

Figure 2.12.3.

Data Collected Pursuant to OMB APPROVAL 0648-0013 - Expires 12/31/2001

REPORTING FORM FOR THE TRIP INTERVIEW PROGRAM (TIP)

Next Row is for Data Entry Personnel Use Only

PC Data Entry by:		Date:		Batch		Interview #:	

SECTION I

Interview Number										
Fishery Codes	CP	RF	OP		IN	ML	BF		EG	MX	SL
Trip Type	030	100	200	300	400	600	675	676	735	900	
Agent Code or Name			OR								
Date of Interview											
	Month		Day		Year						
Reporting Area of Landings											
	Reporting State				Reporting County				Reporting Area Zipcode		
Sampling Site											
	State		County		Zipcode		Sampling Location Code (Dealer or MRFSS)				
Start / End Date of Trip											
	Start Month		Start Day		Start Year		End Month		End Day		End Year
Information Source	SR		LB	CI	SS		SI		OD		SO
	Sales Records		Logs		Site Sampling		Recs and Int		Observer Data		Recs and Observ
Fishing Mode	CM		CP		HB		PR		TR		SS
	Commercial		Charter/Party		Head Boat		Private Rec		Torunament		Scientific Survey
Time of Data Collection -24hr	Begin	:			End	:					
		Hour	Minutes			Hour	Minutes				
Bias Type	NB		SB		EB		SE		NI		
	No Bias		Size Bias		Effort Bias		Size & Effort		No Information		
Interview Type	FS			DS			TS			AT	
	Fisherman Sample			Dealer Sample			Trip Survey			Angler Trip	
Landings Type	CL			IL			NL			NF	
	Complete Landings (weight)			Incomplete Landings			No Landings			No Fish Caught	
Crew Size				(The number of crew including the captain)							
Total Effort	_ _ _._		_ _ _._	UK	NR	WR	EQ	NF	SA	WI	OB	QR
	Days Out		Days Fished	Termination Code							
Vessel Information											
	Vessel ID			Vessel Length (feet)					Vessel Name		

SECTION II

Gear Information				Effort / Location		
Code	Number	Quantity	Other	Soak Time (Hours)	Area Fished	Depth Range (Fathoms)
_ _ _	_ _ _ _ _	_ _ _ _ _	_ _ _._ _	_ _ _ _ _._	_ _ _ _ _._	_ _ _._ / _ _ _._
_ _ _	_ _ _ _ _	_ _ _ _ _	_ _ _._ _	_ _ _ _ _._	_ _ _ _ _._	_ _ _._ / _ _ _._
_ _ _	_ _ _ _ _	_ _ _ _ _	_ _ _._ _	_ _ _ _ _._	_ _ _ _ _._	_ _ _._ / _ _ _._
_ _ _	_ _ _ _ _	_ _ _ _ _	_ _ _._ _	_ _ _ _ _._	_ _ _ _ _._	_ _ _._ / _ _ _._
_ _ _	_ _ _ _ _	_ _ _ _ _	_ _ _._ _	_ _ _ _ _._	_ _ _ _ _._	_ _ _._ / _ _ _._

Figure 2.12.3.

SECTION III

Note	Spec Code	Size Code	Gear Code	Area Fished	Landing Weight	Weight Type	Value	Price	Number of Trips	Replicate Number
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+
___	_____	__	___	_____-_	_____	__	_____	___.__	___	__+

SECTION IV

Rep #	Note	Species Code	Samp #	Samp Type	Catch Status	FISH IN SAMPLE			FISH IN SUB-SAMPLE			Form Rec 41
						Weight	Number	Type	Weight	Number	Type	
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_
__	___	_____	__	__	_	_____.___	____	__	_____.___	____	__	_

Figure 2.12.3.

SECTION V													
Rep #	Sam #	Species Code	Line #	Num	Lower Length	Upper Length (If range)	Leng Type	Weight	Wt Type	Sex	Age Struc	Age Status	Tag #
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___
__	__	_____	___	____	____.__	____.__	__	____.___	__	_	__	_	___

Figure 2.12.4. TIP **Summary Sheet for heads, otoliths, spines, and tissue samples.**

Interview # _____ **State** _____ **County** _____ **Month** _____ **Day** _____ **Year** _____

Gear type _____ **Sampler** _____

All measurements should be in fork (centerline) length for fin fish. Make note if otherwise. Check types of samples taken (tissue, otolith). Indicate weight type (WT). Indicate Sex as M, F, or U (Unknown).

Tag #	Spec	Tissue	Oto	Sex	Length (cm)	Weight	WT	Tag #	Spec	Tissue	Oto	Sex	Length (cm)	Weight	WT
1								26							
2								27							
3								28							
4								29							
5								30							
6								31							
7								32							
8								33							
9								34							
10								35							
11								36							
12								37							
13								38							
14								39							
15								40							
16								41							
17								42							
18								43							
19								44							
20								45							
21								46							
22								47							
23								48							
24								49							
25								50							

Figure 2.12.5 TIP ON-LINE computer generated datasheet, highlighted fields reflect those used for logging in biological samples.

```
================
INTERVIEW SECTION
=================
USERNAME       : DFABLE                          INTERVIEW      : 000286224                       INTERVIEW DATE : 2008/04/28
MODE           : COMMERCIAL                      INFO           : SALES RECORDS                   TYPE           : FISHERMAN SAMPLE
LANDED         : FLORIDA                         COUNTY         : BAY                             PLACE          : NONE
SAMPLED        : FLORIDA                         COUNTY         : BAY                             PLACE          : PANAMA CITY
DEALER         : GREG ABRAMS                     CODE           : WD0003898                       #VESSELS       : 01
TRIP#          : 00                              CREW SIZE      : 003                             HAS EFFORT?    : Y
LANDING TYPE   : COMPLETE LANDINGS               BIAS TYPE      : NO BIAS KNOWN                   TERMINATION    : NORMAL
START TIME     : 00:00                           END TIME       : 00:00
START DATE     : NONE                            END DATE       : NONE
DAYS OUT       : 07.50                           DAYS FISHED    : 07.00
LICENSE AGENCY : NONE                            LICENSE        :
VESSEL         : 923621                          NEW NAME       : NONE
TICKET AGENCY  : NONE                            TICKET         : NONE
COMMENTS       :

==============
EFFORT SECTION
==============
GEAR QTY   FREQ  SIZE  AVGINFO SET   LNGTH MI/SET SOAK   AREA       Q SQ LAT/LONG             MINDPTH MAXDPTH DIST
---- ----- ----- ----- ------- ----- ----- ------ ------ ---------- - -- -------------------- ------- ------- ------
613                                                        8.0000      29.00.00N:085.00.00W   33.00

===============
LANDING SECTION
===============
LAND_ID GEA AREA       SPECIES_CODE NAME                           SIZE            GRADE           NUMBER WEIGHT   UNIT            CONDITION       MIN MAX
------- --- ---------- ------------ ------------------------------ --------------- --------------- ------ -------- --------------- --------------- --- ---
 989386 613     8.0000 1423         GAG                            MIXED/UNSIZED   NO GRADE                2078.00 POUNDS          GUTTED - HEAD O
 989387 613     8.0000 1416         RED GROUPER                    MIXED/UNSIZED   NO GRADE                 434.00 POUNDS          GUTTED - HEAD O
 989388 613     8.0000 1811         BAR JACK                       MIXED/UNSIZED   NO GRADE                  91.00 POUNDS          GUTTED - HEAD O
 989389 613     8.0000 3764         RED SNAPPER,LUTJANUS CAMPECHAN MEDIUM OR #2    NO GRADE                 257.00 POUNDS          GUTTED - HEAD O
 989390 613     8.0000 3764         RED SNAPPER,LUTJANUS CAMPECHAN LARGE OR #1     NO GRADE                 256.00 POUNDS          GUTTED - HEAD O
 989391 613     8.0000 3760         GRAY SNAPPER                   MIXED/UNSIZED   NO GRADE                  47.00 POUNDS          GUTTED - HEAD O
 989392 613     8.0000 1424         SCAMP                          MIXED/UNSIZED   NO GRADE                  67.00 POUNDS          GUTTED - HEAD O
 989393 613     8.0000 0570         COBIA                          MIXED/UNSIZED   NO GRADE                  17.00 POUNDS          GUTTED - HEAD O
 989394 613     8.0000 3765         VERMILION SNAPPER              SMALL OR #3     NO GRADE                  36.00 POUNDS          GUTTED - HEAD O
 989395 613     8.0000 3765         VERMILION SNAPPER              MEDIUM OR #2    NO GRADE                  41.00 POUNDS          GUTTED - HEAD O
 989396 613     8.0000 4561         GRAY TRIGGERFISH               MIXED/UNSIZED   NO GRADE                  10.00 POUNDS          GUTTED - HEAD O
 989397 613     8.0000 3302         RED PORGY                      MIXED/UNSIZED   NO GRADE                 182.00 POUNDS          GUTTED - HEAD O

==============
SAMPLE SECTION
==============
LAND_ID SAMP_ID SPECIES_CODE NAME                           SIZE            GRADE           NUMBERWEIGHT   UNIT            CONDITION       C SAMPLE_TYPE   R
------- ------- ------------ ------------------------------ --------------- --------------- --- -------- --------------- --------------- - ------------- -
 989386  605110 1423         GAG                            MIXED/UNSIZED   NO GRADE         30          NO WEIGHT       GUTTED - HEAD O N LANDED UNSORT Y

==================
SUB-SAMPLE SECTION
==================
No sub-sample information entered for this interview.

===================
OBSERVATION SECTION
===================
SAMP_ID OBS_ID  SPECIES_CODE NAME                 SIZE            GRADE           SEX NUM WEIGHT   UNIT  CONDITION       LENGTH1 TYPE1    LENGTH2 TYPE2    LENGTH3 TYPE3
------- ------- ------------ -------------------- --------------- --------------- --- --- -------- ----- --------------- ------- -------- ------- -------- ------- ----
605110 5328835 1423         GAG                  MIXED/UNSIZED   NO GRADE        U     1     .000 NW    GUTTED - HEAD O   70.50 FL CM            NL NL            NL NL
605110 5328836 1423         GAG                  MIXED/UNSIZED   NO GRADE        U     1     .000 NW    GUTTED - HEAD O   59.30 FL CM            NL NL            NL NL
605110 5328837 1423         GAG                  MIXED/UNSIZED   NO GRADE        U     1     .000 NW    GUTTED - HEAD O   67.20 FL CM            NL NL            NL NL
605110 5328838 1423         GAG                  MIXED/UNSIZED   NO GRADE        U     1     .000 NW    GUTTED - HEAD O   70.50 FL CM            NL NL            NL NL
```

Figure 2.12.5 TIP ON-LINE computer generated datasheet, highlighted fields reflect those used for logging in biological samples.

605110	5328839	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.20	FL CM	NL	NL		NL	NL
605110	5328840	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.10	FL CM	NL	NL		NL	NL
605110	5328841	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	66.80	FL CM	NL	NL		NL	NL
605110	5328842	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	76.50	FL CM	NL	NL		NL	NL
605110	5328843	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.00	FL CM	NL	NL		NL	NL
605110	5328844	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.00	FL CM	NL	NL		NL	NL
605110	5328845	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.80	FL CM	NL	NL		NL	NL
605110	5328846	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	65.70	FL CM	NL	NL		NL	NL
605110	5328847	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	63.50	FL CM	NL	NL		NL	NL
605110	5328848	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	78.00	FL CM	NL	NL		NL	NL
605110	5328849	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	69.90	FL CM	NL	NL		NL	NL
605110	5328850	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	65.70	FL CM	NL	NL		NL	NL
605110	5328851	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	75.10	FL CM	NL	NL		NL	NL
605110	5328852	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	92.40	FL CM	NL	NL		NL	NL
605110	5328853	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	71.00	FL CM	NL	NL		NL	NL
605110	5328854	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	68.20	FL CM	NL	NL		NL	NL
605110	5328855	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	61.20	FL CM	NL	NL		NL	NL
605110	5328856	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	69.90	FL CM	NL	NL		NL	NL
605110	5328857	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.30	FL CM	NL	NL		NL	NL
605110	5328858	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	85.70	FL CM	NL	NL		NL	L
605110	5328859	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.70	FL CM	NL	NL		NL	NL
605110	5328860	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.00	FL CM	NL	NL		NL	NL
605110	5328861	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	71.40	FL CM	NL	NL		NL	NL
605110	5328862	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.80	FL CM	NL	NL		NL	NL
605110	5328863	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.40	FL CM	NL	NL		NL	NL
605110	5328864	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	73.20	FL CM	NL	NL		NL	NL

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OBSERVATION SAMPLE SECTION
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OBS_ID	SPECIES_CODE	NAME	SIZE	GRADE	SEX	NUM	WEIGHT	UNIT	CONDITION	LENGTH1	TYPE1	TAG1	S1	TAG2	S2	TAG3	S3
5328835	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.50	FL CM	20080291546	O				
5328836	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	59.30	FL CM	20080291547	O				
5328837	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.20	FL CM	20080291548	O				
5328838	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.50	FL CM	20080291549	O				
5328839	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.20	FL CM	20080291550	O				
5328840	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.10	FL CM	20080291551	O				
5328841	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	66.80	FL CM	20080291552	O				
5328842	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	76.50	FL CM	20080291553	O				
5328843	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	70.00	FL CM	20080291554	O				
5328844	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.00	FL CM	20080291555	O				
5328845	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.80	FL CM	20080291556	O				
5328846	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	65.70	FL CM	20080291557	O				
5328847	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	63.50	FL CM	20080291558	O				
5328848	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	78.00	FL CM	20080291559	O				
5328849	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	69.90	FL CM	20080291560	O				
5328850	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	65.70	FL CM	20080291561	O				
5328851	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	75.10	FL CM	20080291562	O				
5328852	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	92.40	FL CM	20080291563	O				
5328853	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	71.00	FL CM	20080291564	O				
5328854	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	68.20	FL CM	20080291565	O				
5328855	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	61.20	FL CM	20080291566	O				
5328856	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	69.90	FL CM	20080291567	O				
5328857	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.30	FL CM	20080291568	O				
5328858	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	85.70	FL CM	20080291569	O				
5328859	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.70	FL CM	20080291570	O				
5328860	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.00	FL CM	20080291571	O				
5328861	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	71.40	FL CM	20080291572	O				
5328862	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.80	FL CM	20080291573	O				
5328863	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.40	FL CM	20080291574	O				
5328864	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	73.20	FL CM	20080291575	O				

This report contains information for all sections of TIPONLINE.

Table 2.12.1 List of TIP Agency and associated ID numbers.

Agency ID #	Agency Name
1	MAINE
2	NEW HAMPSHIRE
3	MASSACHUSETTS
4	RHODE ISLAND
5	CONNECTICUT
6	NEW YORK
7	NEW JERSEY
8	PENNSYLVANIA
9	DELAWARE
10	MARYLAND
11	VIRGINIA
12	NORTH CAROLINA
13	SOUTH CARLOLINA
14	GEORGIA
15	**FLORIDA**
16	NMFS-NER
17	NMFS-HQ
18	**NMFS-SER**
19	ASMFC
20	NEFMC
21	MAFMC
22	SAFMC
23	USFWS
24	DISTRICT OF COLUMBIA
25	POTOMAC RIVER RC
26	ALABAMA
27	MISSISSIPPI
28	**LOUISIANA**
29	TEXAS
30	GSMFC
31	PUERTO RICO
32	USVI

The agency names in **BOLD** send samples to our facility.

Table 2.12.2 List of County Codes

This list of county codes assists in the assignment of the appropriate county landed location to be used in the log and data files. This list is in order by State, NMFS County Codes, FIPS County Codes, and County Name.

If a FIPS County Code is not listed, then use the web site:
http://www.zipinfo.com/search/zipcode.htm
to locate the appropriate landing location or reporting county.

Due to the changes in the Trip Interview Program in 2004, NMFS State and County Codes were replaced with the FIPS State and County Codes. Therefore, this list can be used for all years of Trip Interview Program Sampling.

ALPHA STATE	NMFS STATE	NMFS COUNTY	FIPS STATE	FIPS COUNTY	COUNTY NAME
AL	01	00	01	000	NOT CODED
AL	01	01	01	003	BALDWIN
AL	01	03	01	097	MOBILE
E FL	10	00	12	000	NOT CODED
E FL	10	01	12	009	BREVARD
E FL	10	03	12	011	BROWARD
E FL	10	07	12	025	DADE
E FL	10	08	12	031	DUVAL
E FL	10	09	12	035	FLAGLER
E FL	10	15	12	061	INDIAN RIVER
E FL	10	19	12	085	MARTIN
E FL	10	21	12	089	NASSAU
E FL	10	23	12	099	PALM BEACH
E FL	10	25	12	107	PUTNAM
E FL	10	27	12	109	ST JOHNS
E FL	10	29	12	111	ST LUCIE
E FL	10	33	12	127	VOLUSIA
FL	11	00	12	000	NOT CODED
FL	11	01	12	005	BAY
FL	11	03	12	015	CHARLOTTE
FL	11	05	12	017	CITRUS
FL	11	07	12	021	COLLIER
FL	11	09	12	029	DIXIE
FL	11	11	12	033	ESCAMBIA
FL	11	13	12	037	FRANKLIN
FL	11	14	12	039	GADSDEN
FL	11	15	12	045	GULF
FL	11	17	12	053	HERNANDO

ALPHA STATE	NMFS STATE	NMFS COUNTY	FIPS STATE	FIPS COUNTY	COUNTY NAME
FL	11	19	12	057	HILLSBOROUGH
FL	11	21	12	065	JEFFERSON
FL	11	23	12	071	LEE
FL	11	25	12	075	LEVY
FL	11	27	12	081	MANATEE
FL	11	29	12	087	MONROE
FL	11	31	12	091	OKALOOSA
FL	11	33	12	101	PASCO
FL	11	35	12	103	PINELLAS
FL	11	37	12	113	SANTA ROSA
FL	11	39	12	115	SARASOTA
FL	11	41	12	123	TAYLOR
FL	11	43	12	129	WAKULLA
FL	11	45	12	131	WALTON
LA	21	00	22	000	NOT CODED
LA	21	01	22	001	ACADIA
LA	21	03	22	005	ASCENSION
LA	21	05	22	007	ASSUMPTION
LA	21	07	22	019	CALCASIEU
LA	21	09	22	023	CAMERON
LA	21	11	22	045	IBERIA
LA	21	13	22	051	JEFFERSON
LA	21	15	22	053	JEFFERSON DAVIS
LA	21	17	22	055	LAFAYETTE
LA	21	19	22	057	LAFOURCHE
LA	21	21	22	071	ORLEANS
LA	21	23	22	075	PLAQUEMINES
LA	21	25	22	087	ST BERNARD
LA	21	27	22	089	ST CHARLES
LA	21	29	22	095	ST JOHN THE BAPTIST
LA	21	31	22	099	ST MARTIN
LA	21	33	22	101	ST MARY
LA	21	35	22	103	ST TAMMANY
LA	21	37	22	105	TANGIPAHOA
LA	21	39	22	109	TERREBONNE
LA	21	41	22	113	VERMILION
LA	21	43	22	009	AVOYELLES
LA	21	45	22	013	BIENVILLE
LA	21	46	22	121	WEST BATON ROUGE
LA	21	47	22	015	BOSSIER
LA	21	48	22	033	EAST BATON ROUGE
LA	21	49	22	017	CADDO

ALPHA STATE	NMFS STATE	NMFS COUNTY	FIPS STATE	FIPS COUNTY	COUNTY NAME
LA	21	51	22	021	CALDWELL
LA	21	53	22	025	CATAHOULA
LA	21	55	22	029	CONCORDIA
LA	21	57	22	035	EAST CARROLL
LA	21	59	22	041	FRANKLIN
LA	21	60	22	043	GRANT
LA	21	61	22	047	IBERVILLE
LA	21	63	22	059	LA SALLE
LA	21	65	22	065	MADISON
LA	21	67	22	067	MOREHOUSE
LA	21	69	22	069	NATCHITOCHES
LA	21	71	22	073	OUACHITA
LA	21	72	22	091	ST HELENA
LA	21	73	22	077	POINTE COUPEE
LA	21	75	22	079	RAPIDES
LA	21	76	22	027	CLAIBORNE
LA	21	77	22	081	RED RIVER
LA	21	78	22	085	SABINE
LA	21	79	22	083	RICHLAND
LA	21	80	22	037	EAST FELICIANA
LA	21	81	22	097	ST LANDRY
LA	21	82	22	127	WINN
LA	21	84	22	031	DE SOTO
LA	21	85	22	107	TENSAS
LA	21	86	22	115	VERNON
LA	21	87	22	111	UNION
LA	21	88	22	061	LINCOLN
LA	21	89	22	119	WEBSTER
LA	21	90	22	049	JACKSON
LA	21	91	22	123	WEST CARROLL
LA	21	92	22	011	BEAUREGARD
LA	21	93	22	125	WEST FELICIANA
LA	21	94	22	093	ST JAMES
LA	21	95	22	063	LIVINGSTON
LA	21	96	22	117	WASHINGTON
LA	21	97	22	003	ALLEN
LA	21	98	22	039	EVANGELINE
MS	27	00	28	000	NOT CODED
MS	27	01	28	045	HANCOCK
MS	27	03	28	047	HARRISON
MS	27	05	28	059	JACKSON

ALPHA STATE	NMFS STATE	NMFS COUNTY	FIPS STATE	FIPS COUNTY	COUNTY NAME
TX	46	00	48	000	NOT CODED
TX	46	01	48	007	ARANSAS
TX	46	03	48	039	BRAZORIA
TX	46	05	48	057	CALHOUN
TX	46	07	48	061	CAMERON
TX	46	09	48	071	CHAMBERS
TX	46	11	48	167	GALVESTON
TX	46	13	48	201	HARRIS
TX	46	15	48	245	JEFFERSON
TX	46	17	48	273	KLEBERG
TX	46	19	48	321	MATAGORDA
TX	46	21	48	355	NUECES
TX	46	23	48	361	ORANGE
TX	46	25	48	391	REFUGIO
TX	46	27	48	409	SAN PATRICIO
TX	46	29	48	489	WILLACY

Table 2.12.3. List of TIP Port Agents and their unique sampler numbers

Collector (First Last)	Sampler Code Name	Unique sampler #
Charlie Schaefer	CSCHAEFE	004
Tom Herbert	THERBERT	006
	AMARTINE	015
Bryan Summerland	BSUMMERL	017
Graham Cole	GCOLE	018
Chad Hanson	HCHAD	019
Jeff Sauer	JSAUER	020
Laura Baird	LBAIRD	021
Lew Bullock	LBULLOCK	022
Lisa Hallock	LHALLOCK	023
Laura Lambremont	LLAMBREM	024
Rick Beaver	RBEAVER	025
Steve Brown	SBROWN	026
Wendy Dyer	WDYER	027
Claudia Dennis	CDENNIS	028
Debbie Fable	DFABLE	029
Ed Little	ELITTLE	030
Jay Boulet	JBOULET	032
June Weeks	JWEEKS	033
Kit Doncaster	KDONCAST	034
Keith Roberts	KROBERTS	035
Michelle Gamby	MGAMBY	037
Pam Eyo	PEYO	038
Michelle Padgett	PMICHELL	039
Gary Rousse	RGARY	040
Rene Labadens	RLABADEN	041
Rene Roman	RROMAN	042
Roy Spears	SROY	043
Ted Flowers	TFLOWERS	044
Pam Machunga	PMACHUNG	068
Albert Lefort	ALEFORT	069
	BYEISER	070
Gary Haddle	GHADDLE	072
Vanessa Maxwell	VMAXWELL	073
Chris Bohnsack	CBOHNSAC	074

Highlighted collectors belong to Agency – Florida (Agency ID #15)

All other collectors belong to Agency – NMFS SER (Agency ID #18)

Source #
Year –
Agency ID –
state alpha abbr.-
6 digit sequential #

Beginning in 2004, all TIP tag numbers will appear as follows:
4-digit year 3-digit unique sampler # last 4-digits tag number (consecutively numbered)
For example, Claudia Dennis' first otolith sample would be tagged: 20040280001

Table 2.12.4 Trip Interview Program Sampler State Assignment, Duty Station Assignments, Contact Information (Prior to 2004)

Interview #

S	DS	Port Agent Name	Address	Telephone # Fax #
1	8	Wiggers, Robert SCDNR	217 Fort Johnson Road P.O. Box 12559 Charleston, SC 29422	Phone: (843) 953-9363 Fax: (843) 953-9362
2	9	Califf, Julie GDNR	Coastal Resources Div. One Conservation Way Brunswick, GA 31523	912-264-7218
3	A	Riser, Beau	No longer @ F.M.R.I.	
3	7	Brown-Eyo, Pam NMFS	SEFSC 75 Virginia Beach Drive Miami, FL 33149	Phone: (305) 361-4565 Fax: (305) 361-4460
3	8	Schaefer, Charles Gamby, Michelle (start 2002) NMFS	P.O. Box 3478 19100 S.E. Federal Hwy Tequesta, FL 33469	Phone: (561) 575-4461 Fax: (561) 743-1583
3	9	Dennis, Claudia Greenwood, Annjanette Haddle, Greg (start 2002) NMFS	P.O. Box 2025 New Smyrna Beach, FL 32170	Phone: (386) 427-6562 Fax: (386) 427-6562
3	T	Baird, Laura Jones, Angela FDEP	F.M.R.I. 1220 Prospect Ave., Suite #285 Melbourne, FL 32901	
3	W	Myers, Robbyn FDEP	F.M.R.I. 7825 Baymeadows Way, Suite #B-200 Jacksonville, FL 32256	Phone: (904) 807-3300 Fax: (904) 448-4366
4	2	Roman, Renee Machuga, Pam (start 2003) Fairclough, Greg (pre 2002) NMFS	S.E. Regional Office 263 13th Avenue South St. Petersburg, FL 33701	Phone: (727) 570-5393 Fax: (727) 570-5300
4	4	Walling, Bill (pre 1999) NMFS	Panama City Laboratory 3500 Delwood Beach Rd. Panama City, FL 32407	Phone: (850) 234-6541 Fax: (850) 235-3559
4	5	Little, Ed Machuga, Pam (only 2002) NMFS	P.O. Box 269 301 Simonton St., Room 208 Key West, FL 33040	Phone: (305) 294-1921 Fax: (305) 294-1921
4	6	Herbert, Tom NMFS	2000 Main St., Room 409A Ft. Myers, FL 33902	Phone: (239) 334-4364 Fax: (239) 334-4364

S	DS	Port Agent Name	Address	Telephone # Fax #
4	7	Fable, Debbie Weeks, June NMFS	Panama City Laboratory 3500 Delwood Beach Rd. Panama City, FL 32407	Phone: (850) 234-6541 Fax: (850) 235-3559
4	B	Hanson, Chad FWC	F.M.R.I. 350 Carroll St. East Point, FL 32328	Phone: (850) 670-4783 Fax: (850) 670-4324
4	E	Bullock, Lew FDEP	F.M.R.I. 100 8th Avenue, S.E. St. Petersburg, FL 33701	Phone: (727) 896-8626 Fax: (727) 823-0166
4	F	Hallock, Lisa FDEP	Charlotte Harbor Field Lab 1481-A Market Circle Port Charlotte, FL 33953	Phone: (941) 255-7403 Fax: (941) 255-7400
4	R	Dyer, Wendy Beaver, Rick FDEP	2796 Overseas Hwy. Suite #119 Marathon, FL 33050	Phone: (305) 289-2330 Fax: (305) 289-2334
4	S	Summerlin, Bryan FDEP	F.M.R.I. 12418 State Road 24 P.O. Box 909 Cedar Key, FL 32625	Phone: (352) 543-9200 Fax: (352) 543-6958
5	7	Flowers, Ted NMFS	USCG/ATC P.O. Box 97 Mobile, AL 36608	Phone: (251) 441-6193 Fax: (251) 441-6193
5	8	Estes, Noel AMRD	P.O. Box 189 Dauphin Island, AL 36548	334-861-2882
5	9	Roberts, Barry AMRD	P.O. Drawer 458 Gulf Shores, AL 36542	334-968-7576
6	8	Cuevas, Kerwin MDMR	Bureau of Marine Resources 2620 Beach Boulevard Biloxi, MS 39531	
6	9	Labadens, Rene Armstrong, Charles NMFS	3209 Frederic Street Pascagoula, MS 39567	Phone: (228) 762-7402 Fax: (228) 762-9200
7 7 7	C 6 L	Rousse, Gary	P.O. Box 623 Golden Meadow, LA 70357	Phone: (985) 632-4324 Fax: (985) 632-4324
7	2	Dzuryachko, Jill LDWF	1600 Canal Street, Suite #316 New Orleans, LA 70112	Phone: (504) 556-9897 Fax: (504) 568-2048
7 7	3 B	Moss, Ed (early 1990's) LDWF	P.O. Box 189 Bourg, LA 70343	
7	B	Boulet, Jay NMFS	2626 Charles Dr., Suite 201 Chalmette, LA 70043	Phone: (504) 277-0294 Fax: (504) 279-0343
7	6	Kubel, Karen LDWF	1600 Canal Street, Suite #316 New Orleans, LA 70112	504-556-9897

S	DS	Port Agent Name	Address	Telephone # Fax #
8	2	Roberts, Keith NMFS	Galveston Laboratory 4700 Avenue U, Bldg. 306 Galveston, TX 77551	Phone: (409) 766-3515 Fax: (409) 766-3543
8	3	Padgett, Michelle Mauermann, Tom NMFS	P.O. Box 2533 Texas Gulf Bank, Suite 213 200 W. 2nd St. Freeport, TX 77542	Phone: (979) 233-4551 Fax: (979) 233-4551
8	4	Spears, Roy NMFS	P.O. Box 1815 132 Cleveland Blvd Aransas Pass, TX 78336	Phone: (361) 758-0436 Fax: (361) 758-1043
8	9	Doncaster, Kit Lopez, Edie NMFS	HC 70 Box 15 Corner @ Angler & Evergreen Brownsville, TX 78521	Phone: (956) 548-2516 Fax: (956) 548-2516
I	C	Ventura, Willy VIDFW	Lagoon Street Leg. Complex Rm 203 Frederiksted, St. Croix, USVI 00840	340-772-1955
P	0	Matos, Daniel PRDRN	Fisheries Research Lab P.O. Box 3665 Marina Station Mayaguez, PR 00709	787-833-2025
		LeFort, Albert LDWF	P O Box 189 Bourg, LA 70343	Phone: (985) 594-4139 Fax: (985) 594-7317
		Kowal, Kelly		Phone: (727) 896-8626

2.13 United States Geological Survey- Fishery Independent Survey (USGS)

Samples from the United States Geological Survey (USGS) are part of the Fishery Independent Survey and collected during scientific cruises at sea (see list of sources). These samples are no longer being collected. Data fields highlighted in yellow indicates data used to complete the log.

Species	Abbreviation of species being sampled, see Chapter 1, Table 1
PC Coll #:	A sequential species specific number is marked on the data sheet in the upper right-hand corner with a **RED** pen. This same number is marked on a small yellow post-it note and placed on the upper left-hand corner of the data sheet.
Date:	Date of collection from sampler (mo/da/yr)
Source:	FMRI
Source #	Boat Initials – Cruise # – Station # (TM) (3 digits) (3 digits) Tommy Monroe TM
Date	Date the sample was caught, __/__/__ (Month / Day / Year)
Latitude	Latitude recorded (degrees, minutes, and seconds)
Longitude	Longitude recorded (degrees, minutes, and seconds)
Time	The approximate time of capture.
Location*	The approximate location where the sample was caught.
State*	The approximate location where the sample was caught. * The state and location will be determined by the degree of latitude and longitude.
Depth	Water depth (meters)
Mode	SS Scientific Survey
Gear	HL Hand-line/ Bandit/ Hook and Line/ Troll LL Long-line TR Trap TRW Trawl

Comments	Additional comments provided by the processor
Collector	First Initial and Last Name of cruise leader
Fish #	Individual fish number supplied by sampler
TL (mm)	Total Length (mm)
FL (mm)	Fork Length (mm)
Whole Wt (kg)	Weight (kg) of whole fish
Sex	F or ♀ Female M or ♂ Male I Immature
Samples Taken	Indicates whether each sample had otoliths, gonads, and/or both

Chapter 3: Data Entry

Updated January 2008

This chapter includes procedures for accessing the databases and the appropriate procedures for data entry. Over the course of 6 years the data entry procedures have been updated and assessed for quality control. In 1998, all databases were imported from Microsoft Excel to Microsoft Access for better accessibility and accuracy. In addition, in 2001 all databases were transitioned to a secured server and a network of workstations was established. By establishing an Age, Growth, and Reproduction Workgroup (network of workstations established on the server) one level of security was added to the databases. In addition, all databases are secured within two levels in Microsoft Access primarily, with user names with set permissions and secondarily, with database passwords. Therefore, databases can only be accessed by persons within the Age, Growth, and Reproduction Workgroup and with the appropriate password. Currently, all species data per year have been combined to establish Annual Group Databases (years: 1990-present) which include fields with increased validation rules and drop lists to add in the accuracy and efficiency of data entry.

This Chapter includes:

3.1 Overall Database Structure

3.2 General Procedures for Opening Annual Group Database

3.3 Procedures for Entering Log Data

3.4 Procedures for Entering Age and Growth Data

3.5 Procedures for Entering Reproduction Data

3.6 Procedures for Entering Histological Data

3.7 Procedures for Entering Mackerel Data

3.8 Procedure for Entering Blue Fin Tuna Data

3.9 Procedures for Proofing Archival Data

3.10 Procedures for Adding New Species to the Database

Contributors: Linda Lombardi-Carlson, Carrie Fioramonti, Chris Palmer

3.1 Overall Database Structure

Microsoft Access is very powerful and user friendly database software. Each annual group database consists of one main data table, supporting data tables, numerous queries, data entry forms, reports that tally totals such as the number of samples by species, etc, and macros that assist in conversion calculations.

Each Annual Group Database opens to a menu (Figure 3.2.1) that allows the user to choose either a form to enter data, or the option to open queries and/or reports. The type of form to use and what other materials are needed to complete data entry will be discussed further in this chapter.

3.2. General Procedures for Opening an Annual Group Database

- Click on Age, Growth, and Reproduction Server Folder
- The server will prompt user for username and password
- Click on Annual Group Database
- Enter Microsoft Access Username
- Enter Microsoft Access Password
- Enter Annual Group Database Password

Annual Group Database Menu will appear:

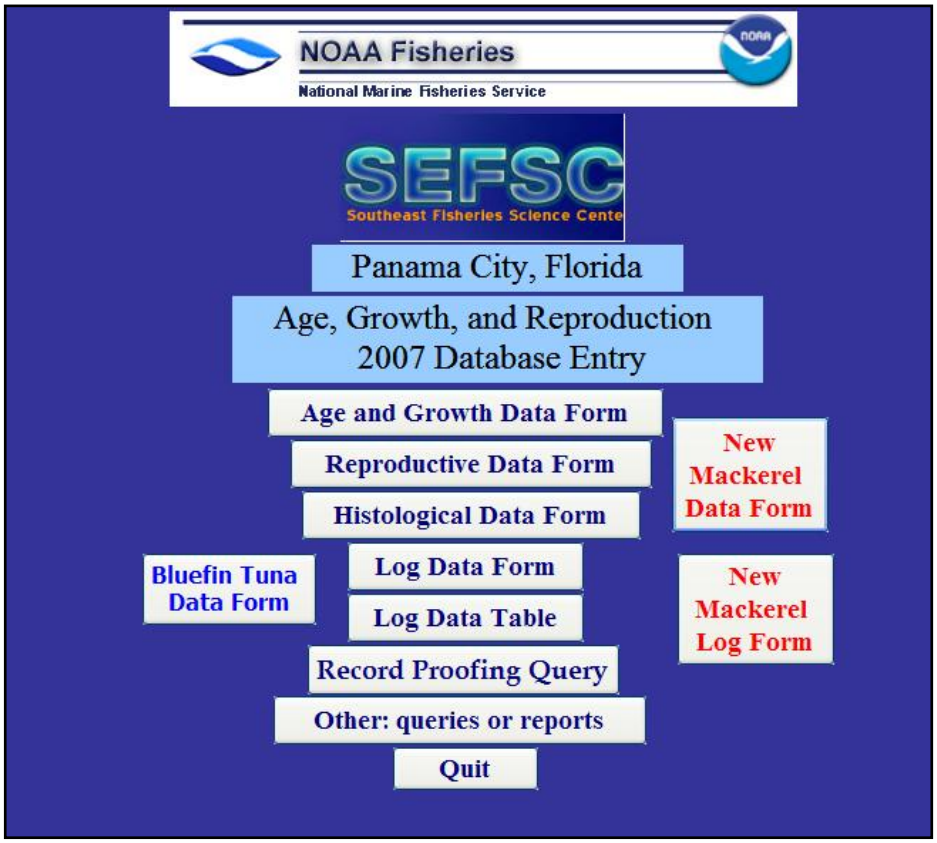

Figure 3.2.1 Annual Group Database Menu

3.3. Procedures for Entering Log Data

Gather the following materials:

- Species specific written log

The following instructions are for all species
EXCEPT KING AND SPANISH MACKEREL
If entering MACKEREL, please see Section 3.7
Mackerel data entry has its own data entry form.

Step 1 Open Annual Group Database and Menu will appear
Click on the Log Data Form action button

- Immediately the Log Data Form (Figure 3.3.1) will appear

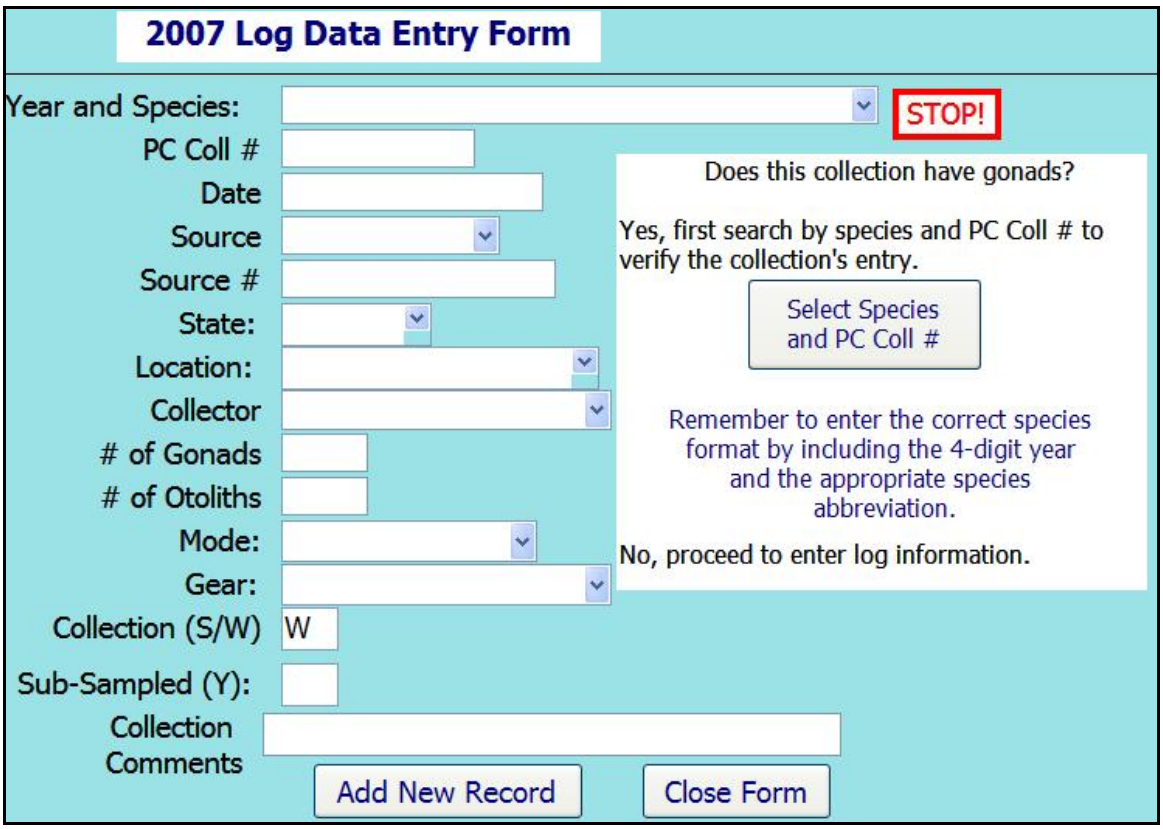

Figure 3.3.1 Log Data Entry Form

Step 2 **STOP**

Does this collection have gonads?
If **YES**, click on the action button
Select Species and PC Coll #

1. Enter Desired Species (do not forget the 4 digit year, species abbreviation does not have to be in capital letters) and press enter

- For Example: 2007 YTS, 2007 GAG, 2007 RS

2. Enter Desired PC Coll # and press enter

3. If a blank screen appears this indicates that the species and collection number entered does **NOT** appear in the database.

   Now you need to enter the following information, go to Step 3

4. If the species specific collection number appears, double check to see if the number of gonads and otoliths were entered for **ONLY ONE** of the records in the collection. The form's bottom legend displays the number of records and what number is currently viewed.

If **NO** gonads were collected, then proceed to enter the log information.

Step 3 Click on action button ADD NEW RECORD
Now the following information needs to be entered:

Year and Species	controlled by a validated list, use drop arrow to view list
PC Coll #	double check this number
Date	enter as month/day/year
Source	controlled by a validated list of sources, use drop list
Source #	see the procedures in Chapter 1 for the appropriate values
State	controlled by a validated list of states, use drop list
Location	use drop list for common locations or verify spelling of location city, county, or port
Collector	controlled by a validated list of samplers, use drop list
# of Gonads	leave blank if there are no gonads
# of Otoliths	verify this value the number of otoliths in the collection
Mode	controlled by a validated list of modes, use drop list
Gear	controlled by a validated list of gears, use drop list
Collection (S/W)	Are the samples 'W' whole or 'S' sectioned?
Collection Comments	any other important information For Example: gonads were preserved in formalin and stored at another location, etc.

Step 4 When you are completely finished entering data, click on the action button Close Form. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

Step 5 Finally, use a pencil and initial next to the collection number on the written log, verifying the information was entered and is correct. Also, make sure the collection is placed into the appropriate INBOX in the Age, Growth, and Reproduction lab or its correct box in the archive (see Chapter 5).

3.4 Procedures for Entering Age and Growth Data

The Age and Growth Data Form is used when entering data for a particular species for the entire year. This form allows for easy entry of capture information along with individual meristic data for each fish in the collection. To enable a higher efficiency to data entry special features have been added to the form. These features are in the form of action buttons that complete specific functions such as conversions, copying and/or adding records (the action buttons will be explained in further detailed below). Note: Due to the various data sheets used by various port agents these procedures will only be describing how data is entered from the Trip Interview Program data sheet (provided in Chapter 2, Section 12).

Gather the following materials:

- Specific species collections (data sheet & otolith samples)
- Annual log for specific species

The following instructions are for all species
EXCEPT KING AND SPANISH MACKEREL
If entering MACKEREL, please see Section 3.7
Mackerel data entry has its own data entry form.

Step 1 Make sure all collections have been logged in
See previous section, Procedures for Entering Log Data

Step 2 Open the Annual Group Database

- Annual Group Database Menu will appear
- Click on the Age and Growth Data Form
- Immediately the Age and Growth Data Entry Form (Figure 3.4.1.)will appear

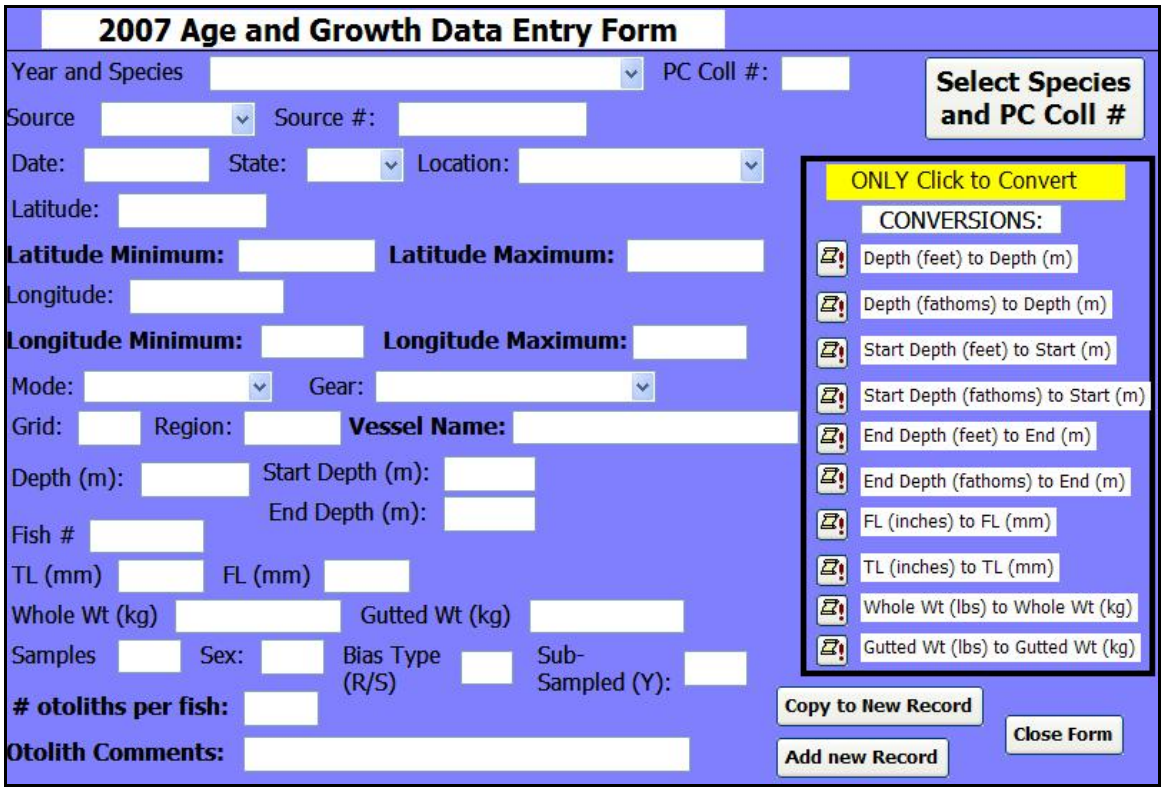

Figure 3.4.1 Age and Growth Data Form

Step 3 Click on the action button Select Species and PC Coll #

1. Enter Desired Species (do not forget the 4 digit year, species abbreviation does not have to be in capital letters) and press enter
   - For Example: 2007 YTS, 2007 GAG, 2007 RS

2. Enter Desired PC Coll # and enter

3. The form should have the following fields filled in with the appropriate information for the collection from the log data:

   Year and Species
   PC Coll #
   Source
   Source #
   Date
   State
   Location
   Mode
   Gear

   Since the database is organized from one main table, data entered for the log is recovered and ready to be used for individual fish records.

4. If a blank screen appears this indicates that the species and collection number entered does **NOT** appear in the database.

Double check the species log to see if the log information was entered into the database. If it was, search the database again, there may have been a clerical error.

Step 4 Now enter the following data from the data sheet:
Record catch location data as follows:

★ It is important to enter the most accurate and descriptive capture location, however, please follow these guidelines:

Latitude 2800
Longitude 8400
} For example, Area Fished = 42884

Grid 6 Corresponds to NMFS Shrimp Fishing Zones
(see Figure 3.4.2 or Table 3.4.1)

Region N Degree of Latitude greater than 2800 = N,
Degree of Latitude less than 2759 = S

★ If specific latitude and longitude is given for the capture location, then enter the exact location including degrees, minutes and seconds

For Example: NMFS Panama City Site 3 x 5
Latitude = 2933.06, Longitude = 8558.51, Grid = 8, Region = N

★ If a range of latitude and longitude is provided on the datasheet, then record this data in the latitude/longitude minimum/maximum fields.

For Example: on datasheet 27-28, 83-84
Enter:
Latitude Minimum = 2700 Latitude Maximum = 2800
Longitude Minimum= 8300 Longitude Maximum= 8400

Unable to determine Region and Grid…Leave these fields blank.

★ If a grid is the only capture location data provided on the datasheet, then enter:
For Example: on datasheet Area Fished = 5
Enter in the database: Grid = 5, Region = S
No data is entered in the latitude and longitude fields.

Here are two examples of catch location (effort data) on TIP data forms:

TIP Reporting Form, Section II:

SECTION II			
	Effort / Location		
Other	Soak Time (Hours)	Area Fished	Depth Range (Fathoms)
___-__	_____-_	_____-_	____/____
___-__	_____-_	_____-_	____/____
___-__	_____-_	_____-_	____/____

TIP Online Data Sheet, Effort Section:

```
==============
EFFORT SECTION
==============
GEAR QTY   FREQ  SIZE  AVGINFO SET   LNGTH MI/SET SOAK   AREA       Q SQ LAT/LONG             MINDPTH MAXDPTH DIST
---- ----- ----- ----- ------- ----- ----- ------ ------ ---------- - -- --------------------- ------- ------- ------
613                                                        8.0000      29.00.00N:085.00.00W   33.00
```

Depth (m) — If depth range less than or equal to 5 fathoms, then enter the average of the start and end depths in meters.

Start depth (m)
End depth (m)
} If depth range is greater than 5 fathoms, then enter both the minimum and maximum depths.

If depth data is not in meters, then use the conversion action buttons to calculate the depths in the appropriate unit. Simply place the cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the mouse.

★ If depth is not reported in fathoms and the depth is a range, then first convert the depth into fathoms to determine if the range is less than or greater than 5 fathoms.

For Example: Depth Range 100-150 ft

→ Conversion factor: 1 fathom = 6 feet

Start 100 ft = 16.7 ftm, End 150 ft = 25 ftm
Depth Range 8.3 ftm (greater than 5 fathoms), thus
Enter Start Depth (m) = 30.48
Enter End Depth (m) = 45.72

For Example: Depth Range 50-58 m

→ Conversion factor 1 fathom = 1.8288 meter

Start 50 m = 27.34 ftm, End 58 m = 31.71 ftm
Depth Range 4.37 ftm (less than 5 fathoms), thus calculate the average depth:
Depth (m) = 54

Figure 3.4.2 National Marine Fisheries Service Shrimp Fishing Grid Zones for Atlantic Ocean and Gulf of Mexico

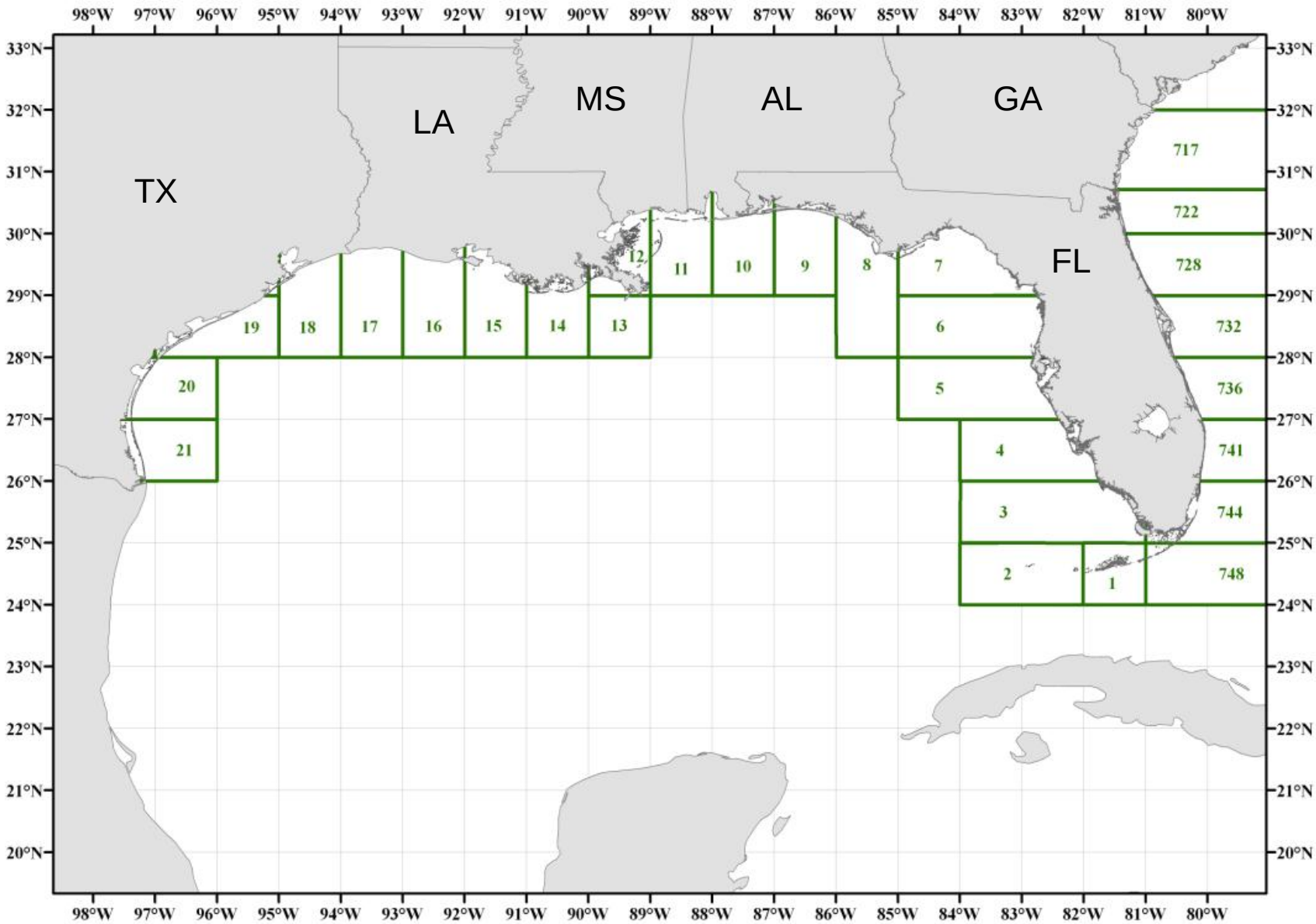

Table 3.4.1 Allocation of NMFS Shrimp Fishing Grid Zones described by the degree of latitude and longitude and/or written description.

Region	Grid	Latitude	Longitude	Written Description
S	1	24	81	
S	2	24	82, 83, 84	west of Dry Tortugas, FL
S	3	25	81, 82, 83, 84	
S	4	26	82, 83, 84	west of Ft. Myers, FL
S	5	27	82, 83, 84	west of Tampa Bay, FL
N	6	28	82, 83, 84	Florida Middle Grounds
N	7	29	83, 84	
N	8	28, 29, 30	85	
N	9	28, 29, 30	86	
N	10	28, 29, 30	87	
N	11	28, 29, 30	88	
N	12	29, 30	89	
N	13	28, 29	89	
N or S	14	27, 28, 29	90	Region depends on the degree of latitude N ≥ 28, S ≤ 27
N or S	15	27, 28, 29	91	
N or S	16	27, 28, 29	92	
N or S	17	27, 28, 29	93	
N or S	18	27, 28, 29	94	
N	19	28	95, 96	
S	20	27	95, 96, 97	
S	21	26	95, 96, 97	
S	22	25	96, 97	
S	23	24	96, 97	

Vessel Name and Captain

Enter this information in this field, if available.

DO NOT ENTER VESSEL #'S

Vessel #'s are confidential data.

Here are two examples of meristic data from TIP data forms:

TIP Reporting Form, Section V:

SECTION V													
Rep #	Sam #	Species Code	Line #	Num	Lower Length	Upper Length (If range)	Leng Type	Weight	Wt Type	Sex	Age Struc	Age Status	Tag #
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---
--	--	----------	---	----	----.--	----.--	--	----.---	--	-	--	-	---

TIP Online Data Sheet, Observation Section:

OBSERVATION SAMPLE SECTION

OBS_ID	SPECIES_CODE	NAME	SIZE	GRADE	SEX	NUM	WEIGHT	UNIT	CONDITION	LENGTH1	TYPE1	TAG1	S1
5297827	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	82.20	FL CM	20080290498	O
5297828	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	90.50	FL CM	20080290499	O
5297829	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	89.50	FL CM	20080290500	O
5297830	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	101.50	FL CM	20080290501	O
5297831	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	73.50	FL CM	20080290502	O
5297832	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	75.80	FL CM	20080290503	O
5297833	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	92.40	FL CM	20080290504	O
5297834	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	77.70	FL CM	20080290505	O
5297835	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	67.10	FL CM	20080290506	O
5297836	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	76.00	FL CM	20080290507	O
5297837	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	86.50	FL CM	20080290508	O
5297838	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	69.80	FL CM	20080290509	O
5297839	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	86.70	FL CM	20080290510	O
5297840	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	79.60	FL CM	20080290511	O
5297841	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	74.20	FL CM	20080290512	O
5297842	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.30	FL CM	20080290513	O
5297843	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	96.00	FL CM	20080290514	O
5297844	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	75.50	FL CM	20080290515	O
5297845	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	75.80	FL CM	20080290516	O
5297846	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	88.20	FL CM	20080290517	O
5297847	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	86.50	FL CM	20080290518	O
5297848	1423	GAG	MIXED/UNSIZED	NO GRADE	U	1	.000	NW	GUTTED - HEAD O	72.40	FL CM	20080290519	O

Record meristic data as follows:

Fish # Enter given Tag #

For TIP Tag # (2004 – present) enter only the last 4 digits

TL (mm) if in centimeters, then multiple the length by 10

FL (mm) if in centimeters, then multiple the length by 10

Trip Interview Program Length Types:

FC Fork in Centimeters
FI Fork in Inches
TC Total in Centimeters
TI Total in Inches
FT Fork & Total in Centimeters

If the length data is not in millimeters, then use the conversion action buttons to calculate the lengths in the appropriate unit. Simply place the cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the tab key or the mouse.

Whole Weight (kg) or Gutted Weight (kg)

Trip Interview Program Weight Types:

RK Round/Whole in Kilograms
GK Gutted in Kilograms
RP Round/Whole in Pounds
GP Gutted in Pounds
RG Round/Whole in Grams
GG Gutted in Grams

If the weight data is not in kilograms, then use the conversion action buttons to calculate the weights in the appropriate unit. Simply place the

cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the mouse.

Samples — Protected by validating rule, enter one of the following:
- O — Otolith
- G — Gonad
- S — Spine
- OG — both otolith and gonad

Other combinations of these variables

Sex — Protected by validating rule, enter one of the following:
- F — Female
- M — Male
- I — Indeterminate
- or — Leave blank

Bias Type (R/S) — Refer to TIP form Page 1, Section I
- R — No bias, No Information
- S — Size Bias

Sub-Sampled — Put a "Y" in the box, if this collection is sub-sampled from the entire year of samples.

Otoliths per fish — Enter # of otoliths per fish
If only one otolith is present, then in otolith comments record whether it is the left or right otolith.

Otolith Comment — Record if otoliths are whole, chipped, or broken

Step 5 If there are more than one fish in this collection, then press the action button Copy to New Record.

Access will create a new record with the exact same data in the fields as the previous record, simply change the following data for the new record:

Fish #
TL (mm)
FL (mm)
Whole Weight (kg) or Gutted Weight (kg)
Samples
Sex
Sub-Sampled
Otoliths per fish
Otolith Comment

Step 6 Once the entire collection's data is entered the legend on the bottom of the form will indicate how many records have been entered. Make sure the number of records matches the number of fish in the collection. To proof the data entered simply press the back arrow on the form's bottom legend. In the event a duplicate entry is made, a query must be created in order to delete the record. **SEE YOUR SUPERVISOR** before doing so, if you are not authorized to do this!

Step 7 When you are completely finished entering data, click on the action button Close Form. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

3.5 Procedures for Entering Reproduction Data

The Reproduction Data Form is used when entering data for gonad samples received by our lab. Fresh gonads are sent to our facility by port agents and fresh gonads must be processed immediately. If gonads are not processed immediately the decay of the gonad tissue will prohibit the analysis of the gonad tissue. This form allows for easy entry of the data specific to initial processing of gonads. These forms are filled out by the person initially processing the gonads (e.g. Bill Walling). These datasheets are filled by species in the horizontal file folders in the Age, Growth, and Reproduction Lab.

Gather the following materials:

- Gonad Processing datasheet
- Collection data sheet

The following instructions are for all species
EXCEPT KING AND SPANISH MACKEREL
If entering MACKEREL, please see Section 3.7.
Mackerel data entry has its own data entry form.

Step 1 Open the Annual Group Database

- Annual Group Database Menu will appear
- Click on the Reproduction Data Form
- Immediately the Reproduction Data Entry Form (Figure 3.5.1) will appear

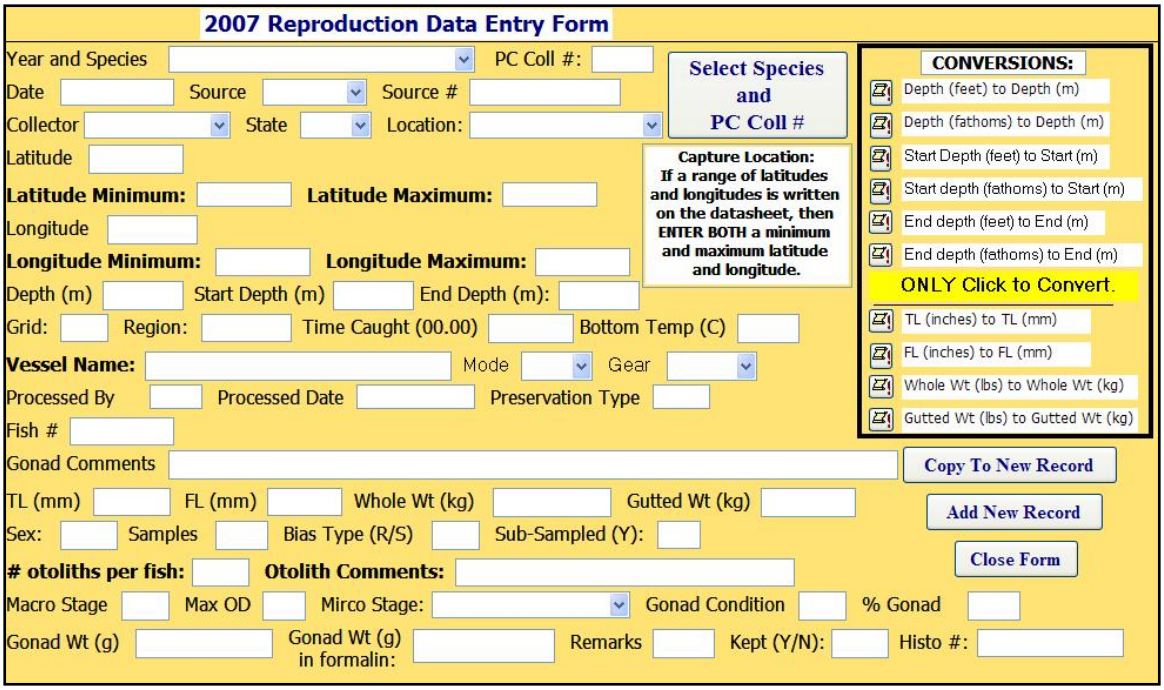

Figure 3.5.1 Reproduction Data Entry Form

Step 2 Click on the action button Select Species and PC Coll #

2. Enter Desired Species (do not forget the 4 digit year, species abbreviation does not have to be in capital letters) and enter
   - For Example: 2007 YTS, 2007 GAG, 2007 RS

2. Enter Desired PC Coll # and enter

3. The form may be blank if the collection log data has not been entered.

Step 3 All data fields should be filled on the Gonad Processing Data Sheet from the original field data sheet from the port agent and from the initial processing of the gonads. A copy of the original data sheet should be stapled to the back of the Gonad Processing Data Sheet. Please make sure the Gonad Processing Data Sheet has been filled out completely, including capture location (latitude, longitude, depth, etc.).

The Reproduction Data Entry Form is arranged in the same order as the data appears on the Gonad Processing Data Sheet to allow for ease in data entry. Simply enter the data and use the tab button, arrow keys, enter or the mouse to scroll through the data fields.

Year and Species	controlled by a validated list, use drop list
PC Coll #	double check this number
Date	enter as month/date/year

Source	controlled by a validated list of sources, use drop list
Source #	See Chapter 1 for the appropriate values by source
Collector	controlled by a validated list of samplers, use drop list
State	controlled by a validated list of states, use drop list
Location	Verify spelling of location city, county, or port

If gonads are from the Trip Interview Program, see Section II of original data sheet for effort data

Latitude 2800
Longitude 8400
} For example, Area Fished = 42884

Grid NMFS Shrimp Fishing Zones (Figure 3.4.2)

Region Latitude greater than 2800=N, less than 2799 = S

- ★ If specific latitude and longitude is given for the capture location, then enter the exact location including degrees, minutes and seconds

  For Example: NMFS Panama City Site 3 x 5
  Latitude = 2933.06, Longitude = 8558.51, Grid = 8, Region = N

- ★ If a range of latitude and longitude is provided on the datasheet, then record this data in the latitude/longitude minimum/maximum field.

  For Example: on datasheet 27-28, 83-84
  Enter:
  Latitude Minimum = 2700 Latitude Maximum = 2800
  Longitude Minimum = 8300 Longitude Maximum = 8400

  Unable to determine Region and Grid…Leave these fields blank.

- ★ If a grid is the only capture location data provided on the datasheet, then enter:
  For Example: on datasheet Area Fished = 5
  Enter in the database: Grid = 5, Region = S
  No data is entered in the latitude and longitude fields.

Bottom Temp (C) Bottom sea water temperature (ºC)

Depth (m) If depth range less than 5 fathoms, then enter average

Start Depth (m)
End Depth (m)
} If depth range is greater than 5 fathoms, then enter both the minimum and maximum depths.

If depth data is not in meters, then use the conversion action buttons to calculate the depths in the appropriate unit. Simply place the cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the mouse.

★ If depth is not reported in fathoms and the depth is a range, then first convert the depth into fathoms to determine if the range is less than or greater than 5 fathoms.

For Example: Depth Range 100-150 ft

→ Conversion factor: 1 fathom = 6 feet

Start 100 ft = 16.7 ftm, End 150 ft = 25 ftm
Depth Range 8.3 ftm (greater than 5 fathoms), thus
Enter Start Depth (m) = 30.48
Enter End Depth (m) = 45.72

For Example: Depth Range 50-58 m

→ Conversion factor 1 fathom = 1.8288 meter

Start 50 m = 27.34 ftm, End 58 m = 31.71 ftm
Depth Range 4.37 ftm (less than 5 fathoms), thus calculate the average depth:
Depth (m) = 54

Vessel Name and Captain

Enter this information in this field, if available.
DO NOT ENTER VESSEL #'S
Vessel #'s are confidential data

Time Caught	Use 24 hour time clock
Mode	controlled by a validated list of modes, use drop list
Gear	controlled by a validated list of gears, use drop list
Processed By	Enter initials of gonad processor
Processed Date	Date gonads were processed
Preservation Type	See list in Chapter 7

Fish #
TL (mm)
FL (mm)
Whole Wt (kg)

If the length or weight data is not in millimeters or kilograms, respectively, then use the conversion action buttons to calculate the lengths and weights in the appropriate unit, simple place the cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the mouse.

Gutted Wt (kg) Fish collected with gonads should *not* have a gutted weight

Sex — Protected by validating rule, enter one of the following:

F	Female
M	Male
I	Indeterminate
or	Leave blank

Samples — Protected by validating rule, enter one of the following:

O	Otolith
G	Gonad
S	Spine
OG	both otolith and gonad

Or a combinations of these variables

Bias Type (R/S)

R	No bias, No Information
S	Size Bias

Sub-Sampled Put a "Y" in the box, if this collection is sub-sampled from the entire year of samples.

Macro Stage
Max OD
Micro Stage
Gonad Condition
% Gonad
Gonad Wt (g)
Gonad Wt (g) in formalin
Remarks
Kept (Y/N)

Verify all information in the correct format as pertains to the Key to Processing Gonads, **only** numerical values will be accepted by the database, except for the Micro Stage and the Kept field (see Chapter 7 for further information regarding these fields).

Histo # — Leave blank until the gonad is blocked.

Gonad Comments — Anything that pertains to the collection, if comment pertains to one fish only include in specific record

Step 4 If there are more than one fish in this collection, then press the action button Copy to New Record.

Access will create a new record with the exact same data in the fields as the previous record. Simply change the following data for the new record:

Fish #
TL (mm)
FL (mm)
Whole Wt (kg)

Gutted Wt (kg)
Sex
Samples
Bias Type (R/S)
Sub-Sampled
Macro Stage
Max OD
Micro Stage
Gonad Condition
% Gonad
Gonad Wt (g)
Gonad Wt (g) in formalin
Remarks
Kept (Y/N)
Histo #
Gonad Comments

Step 5 Once the entire collection's data is entered the legend on the bottom of the form will indicate how many records have been entered. Make sure this matches the number of fish in the collection. To proof the data entered simply press the back arrow on the form's bottom legend.

Step 6 When you are completely finished entering data, click on the action button Close Form. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

Step 7 At the bottom of the Gonad Processing Data Sheet, enter initials and date of computer entry. Finally, use the large hole puncher and file the Gonad Processing Data Sheet in the appropriate year and species folder located in the Age, Growth, and Reproduction Lab.

3.6 Procedures for Entering Histological Data

Gather the following materials:

- Annual Histology Green Book
- Annual Species Specific RFRS Binder (Reef Fish Reproductive Study)
- Locate species specific 'Gonad Processing Data Sheet'

  If no 'Gonad Processing Data Sheet' can be located, then:
  Was the gonad processed (fixed in formalin) at sea?

  If yes, fill out a gonad processing data sheet and make sure all appropriate fields are completely filled.
  ➡ If no, contact the Age, Growth, & Reproduction lab manager.

Step 1 Open the specific Annual Group Database
- Database Menu will appear
- Click on the action button Histological Data Form
- Immediately the Histological Data Form (Figure 3.6.1) will appear

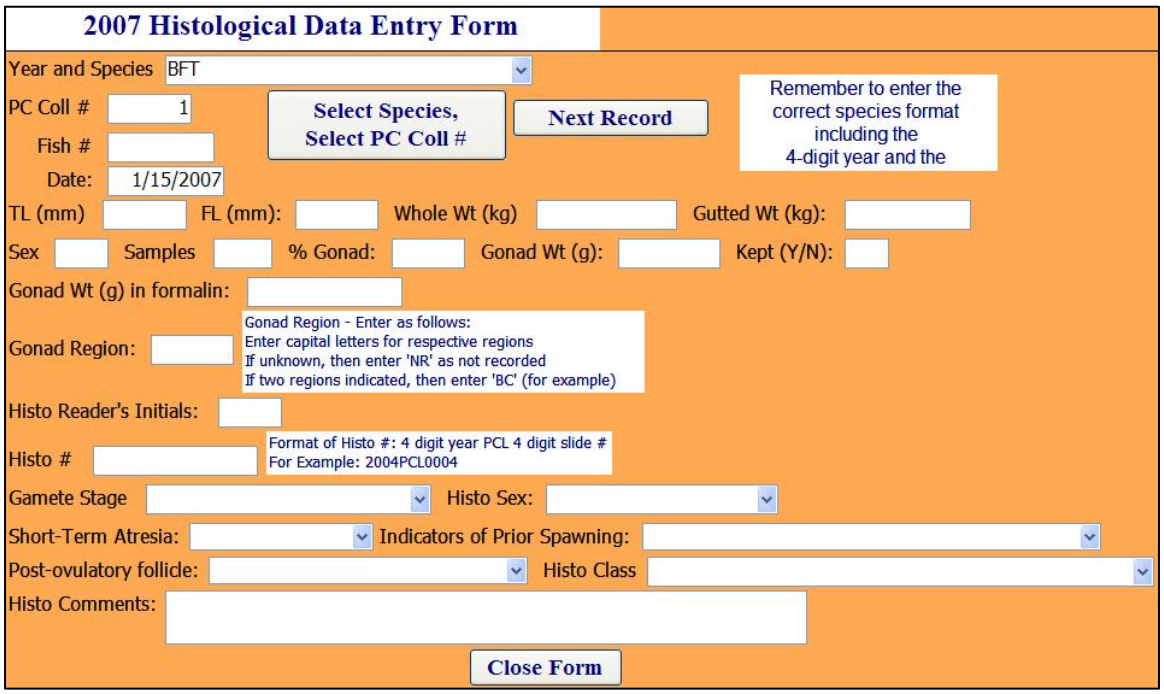

Figure 3.6.1 Histological Data Form- New Classifications

Step 2 Click on the action button, Select Species and PC Coll #

Enter Desired Species (do not forget the 4 digit year, species abbreviation does not have to be in capital letters) and enter
- For Example: 2007 YTS, 2007 GAG, 2007 RS

Enter Desired PC Coll # and enter

Step 3 The corresponding fish meristic (lengths and weights) data along with other information concerning that sample should appear.

Use the action button, Next Record to view other fish in the collection

Possible problems if your select fish does not appear:
1. Spelling error on species abbreviation
2. Incorrect PC Coll #
3. Incorrect Fish #

} **Try search again.**

4. Individual Fish Data was never entered into Database
   ➡ Bring this to the attention of AGR lab manager.

Step 4 Begin entering and/or verifying data that corresponds to the specific fish you selected. Use tab, enter, or the arrow keys to move to your desired fields

TL (mm)	
FL (mm)	
Whole Wt (kg)	
Sex	
Samples	- **G** should at least appear, along with **O** or **S**
% Gonad	
Gonad Wt (g)	- fresh weight without formalin
Kept (Y/N)	- **Y** should be in blank if gonad was blocked
Gonad Wt (g) in formalin	-from green notebook
Gonad Region	- first region**,** second region
Histo Reader's Initials	- first last in capital letters
Histo #	- enter as: 2007PCL#### (slide #)

Prior to 2007, Histological Data has the following fields and criteria:
(However, all database forms reflect current histological classifications.)

For the following criteria, see Chapter 7 for definitions

Gamete stage	- PG, CA, V, LV, H, PS, SS, ST, SZ
Histo sex	- F, E, T, M
Short-term Atresia	-0, 1, 2, 3, 4
Long-term Atresia	- BB0, BB1, BB2
Post-ovulatory follicle	- P0, P1, P2, P3
Spawning State	- INI, INU, INM, AMN, AMS
Muscle Bundles	-0, 1
Histo Comments	- any other information written by reader

After 2007, Histological Data has the following fields and criteria:

For the following criteria, see Chapter 7 for definitions

Gamete stage	- PG, CA, V, LV, H, LH, PS, SS, ST, SZ
Histo sex	- F, E, T, M
Short-term Atresia	- 0, 1, 2, 3
Post-ovulatory follicle	- P0, P1, P2, P3
Indicators of Prior Spawning	- 0, 1, 2, 3
Histo Class	- I, U, A, S, D, DV, P, R, RS
Histo Comments	- any other information written by reader

Step 5 Add Histo # to Gonad Processing Data Sheet.

Step 6 Use the action button, Next Record to view other fish in the collection or Use the action button, Close Form to stop or Click on the action button, Select Species and PC Coll # to search for more records in other collections.

3.7 Procedures for entering Mackerel Data

This section includes procedures for entering log and meristic data for king mackerel and Spanish mackerel. These species are unique in that their seasons do not correspond to the calendar year, the state of Florida has been sub-divided into fishing areas (see Mackerel State) and the Gulf of Mexico, Atlantic and a mixing region are assessed as separate stocks.

3.7.1 Entering Mackerel Log Data

Gather the following materials:

- Species specific written log

Step 1 Open Annual Group Database

- Annual Group Database Menu will appear
- Click on the New Mackerel Log Form
- Immediately the Mackerel Log Data Entry Form (Figure 3.7.1) will appear

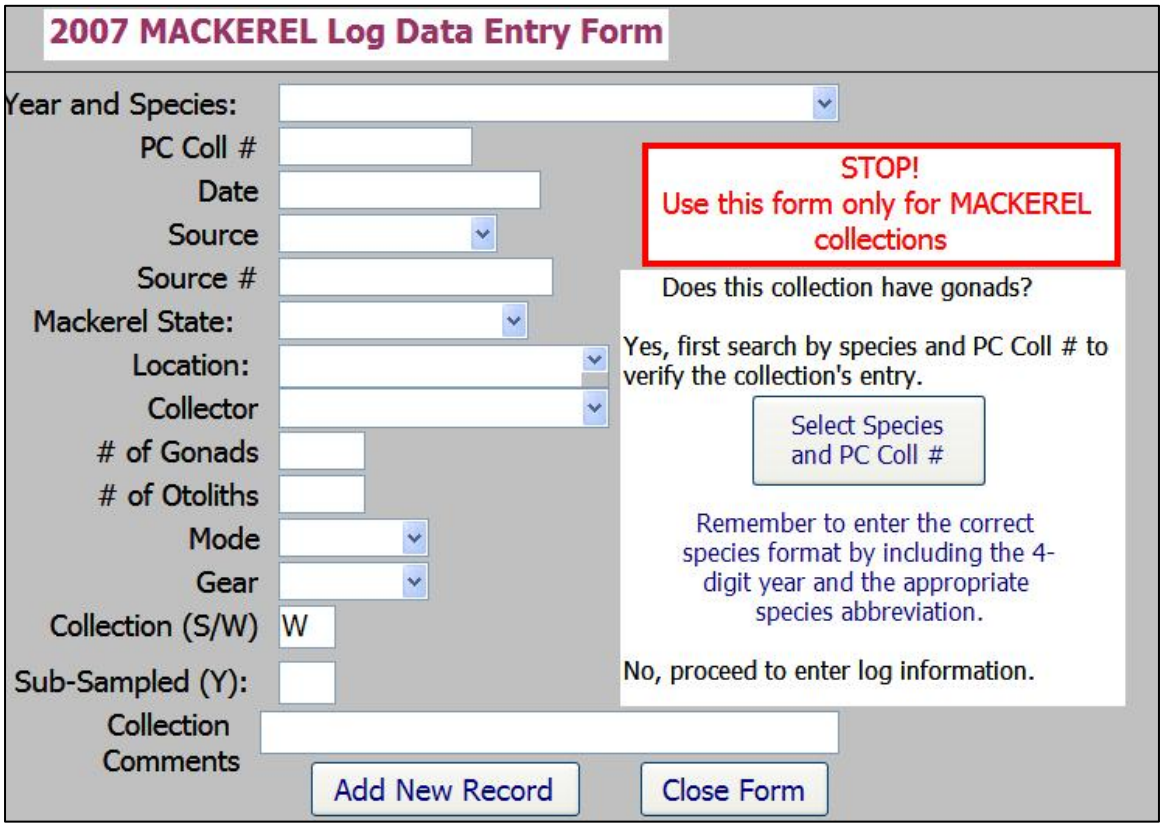

Figure 3.7.1 Mackerel Log Data Entry Form

★ You will notice that the Mackerel Log Data Form is identical to the Log Form **EXCEPT** for the field "Mackerel State". Mackerel are assigned a more specific "Mackerel State" rather than simply "State" due to the

distinct stocks and fisheries, (see Chapter 1, Figure 1.2, map of Mackerel States.

Step 2 **STOP**

Does this collection have gonads?
If **YES**, click on the action button

Select Species and PC Coll #

1. Enter Desired Species (do not forget the 4 digit year, species abbreviation does not have to be in capital letters) and press enter
- For Example: 2007 KMK, 2007 SMK

2. Enter Desired PC Coll # and press enter

3. If a blank screen appears this indicates that the species and collection number entered does **NOT** appear in the database.

Now you need to enter the following information, go to **Step 3**

4. If the species specific collection number appears, double check to see if the number of gonads and otoliths were entered for **ONLY ONE** of the records in the collection. At the bottom of the form, the legend displays the number of records and what number of the entire collection is currently viewed.

If **NO** gonads were collected, then proceed to enter the log information.

Step 3 Click on action button ADD NEW RECORD
Now the following information needs to be entered:

Year and Species	controlled by a validated list, use drop arrow to view list
PC Coll #	double check this number
Date	enter as month/day/year
Source	controlled by a validated list of sources, use drop list
Source #	see Chapter 2 the appropriate values
Mackerel State	controlled by a validated list, use drop arrow to view list

Location	use drop list for common locations or verify spelling of location city, county, or port
Collector	controlled by a validated list of samplers, use drop list
# of Gonads	leave blank if there are no gonads
# of Otoliths	verify this value the number of otoliths in the collection
Mode	controlled by a validated list of modes, use drop list
Gear	controlled by a validated list of gears, use drop list
Collection (S/W)	Are the samples 'W' whole or 'S' sectioned?
Collection Comments	any other important information For Example: gonads were preserved in formalin and stored at another location, etc.

Step 4
When you are completely finished entering data, click on the action button Close Form. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

Step 5
Finally, use a pencil and initial next to the collection number on the written log, verifying the information was entered and is correct. Also, make sure the collection is placed into the appropriate INBOX in the Age, Growth, and Reproduction lab.

3.7.2 Entering Mackerel Age, Growth, and Reproduction Data

Gather the following materials:

- Mackerel collections (data sheet & otolith samples)
- Annual log for specific species

Step 1 Make sure all collections have been logged in
See previous section, Procedures for Entering Mackerel Log Data.

Step 2 Open Annual Group Database

- Annual Group Database Menu will appear
- Click on the New Mackerel Data Form
- Immediately the Mackerel Age, Growth, and Reproduction Data Entry Form (Figure 3.7.2) will appear.

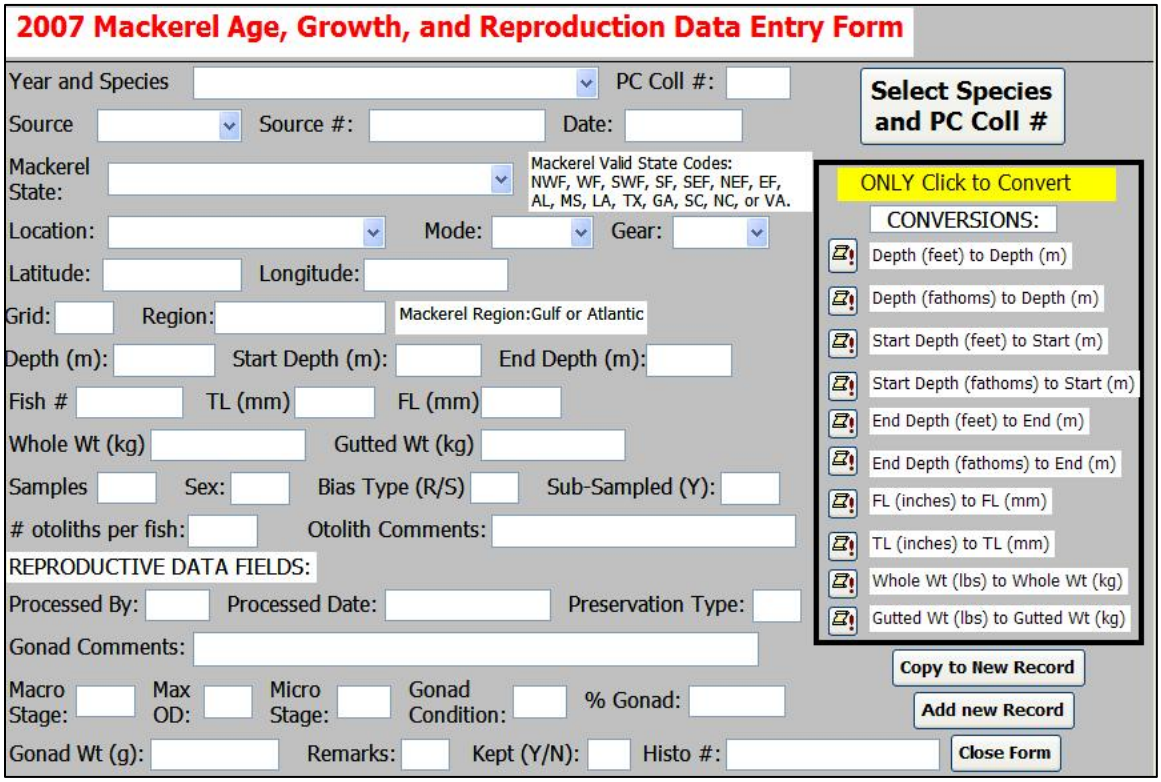

Figure 3.7.2 Mackerel Age, Growth, Reproduction Data Entry Form

Step 3 Click on the action button Select Species and PC Coll #

1. Enter Desired Species- (do not forget the 4 digit year, species abbreviation does not have to be in capital letter) and enter
   - For Example: 2007KMK, 2007SMK

2. Enter Desired PC Coll # and enter

3. The form should have the following field filled in with the appropriate information for the collection:

Year and Species
PC Coll #
Source
Source #
Date
Mackerel State
Location
Mode
Gear

Since the database is organized from one main table, data entered for the log is recovered and ready to be used for individual fish records.

4. If a blank screen appears this indicates that the species and collection number entered does **NOT** appear in the database

Double check the species log to see if the log information was entered into the database. If it was, search the database again, there may have been a clerical error.

Step 4
The meristic data for age, growth, and reproduction is entered in the same manner as sections 3.4-3.5 of this chapter with the following exceptions:

Grid/Area — Corresponds to NMFS Shrimp Fishing Zones
See Figure 3.4.2 or Table 3.4.1

Region — **FOR MACKEREL ONLY**
Corresponds to Body of Water
Either Gulf (of Mexico) or Atlantic (Ocean)

For Example:
On datasheet Area Fished = 3 x 5's
Enter in the database: Grid = 8, Region = Gulf

On datasheet Area Fished = Jupiter, Palm Beach Co.
Enter in the database: Grid = 741, Region = Atlantic

If a grid is the only capture location data provided on the datasheet, then enter:

For Example:
On datasheet Area Fished = 5
Enter in the database: Grid = 5, Region = Gulf
No data is entered in the latitude and longitude fields.

Step 5
If there are more than one fish in this collection, then press the action button Copy to New Record.

Access will create a new record with the exact same data in the fields as the previous record; simply change the following data for the new record:

Fish #
TL (mm)
FL (mm)
Whole Weight (kg) or Gutted Weight (kg)
Samples
Sex

Step 6
Once the entire collection's data is entered the legend on the bottom of the form will indicate how many records have been entered. Make sure this matches the number of fish in the collection. To proof the data entered simply press the back arrow on the form's bottom legend.

Step 7
When you are completely finished entering data, click on the action button Close Form. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

3.7.3 Entering Mackerel Histological Data

This data must be entered in the Histological Data Form found on the Annual Group Database Menu (See Section 3.6).

3.8 Entering Blue Fin Tuna Data

This section includes procedures for entering Bluefin tuna log and biological samples.

Gather the following materials:

- Species specific written log

Step 1 Open Annual Group Database

- Annual Group Database Menu will appear
- Click on the Bluefin Tuna Data Form

Immediately the Bluefin Tuna Data Entry Form (Figure 3.8.1) will appear.

Step 2 Click on the action button Select Species and PC Coll #

1. Enter Desired Species- (do not forget the 4 digit year, species abbreviation does not have to be in capital letter) and enter
    - For Example: 2007 BFT
2. Enter Desired PC Coll # and enter
3. A blank form should appear. This is done to avoid duplicating collections. If the collection is already entered, verify data and proceed to next collection.

Step 3 Click on action button ADD NEW RECORD

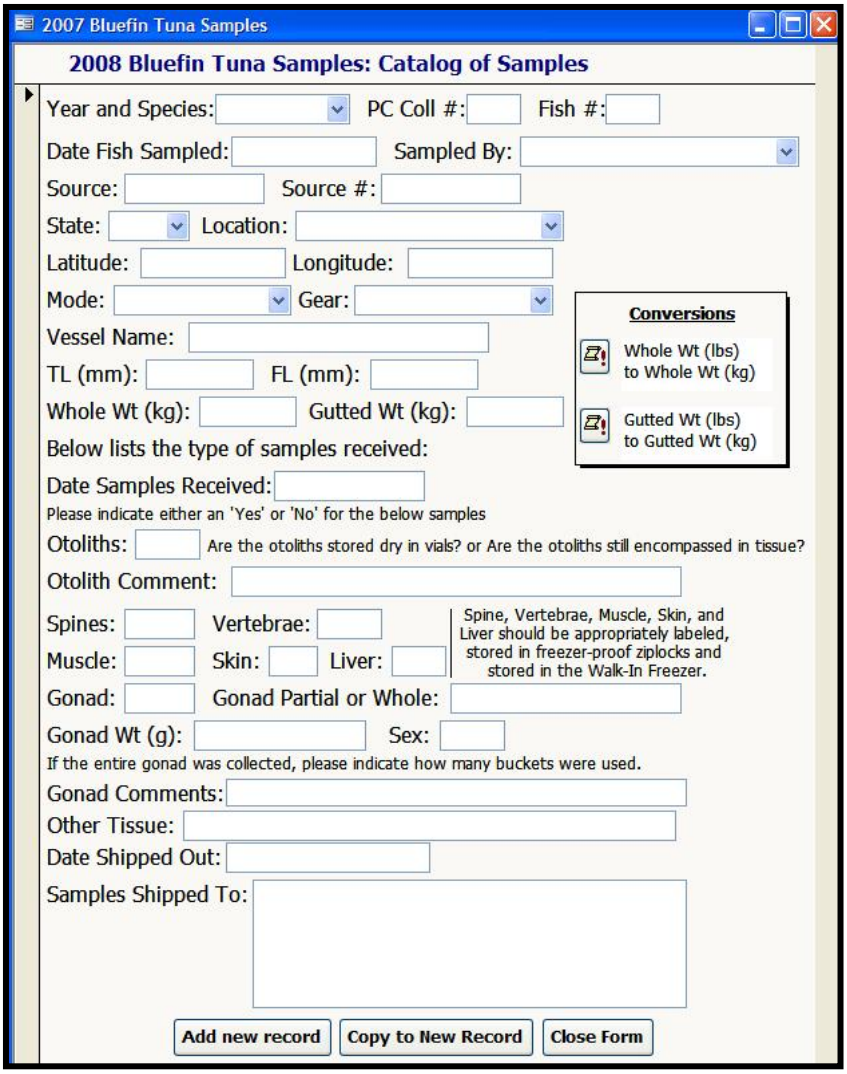

Figure 3.8.1 Bluefin Tuna Catalog of Samples

Now the following information needs to be entered:

Year and Species	controlled by a validated list, use drop arrow to view list
PC Coll #	double check this number
Fish #	sequential alphanumeric
Date Fish Sampled	date of capture month/day/year
Sampled By	first initial and last name of observer Controlled by a validated list of samplers, use drop list
Source	controlled by a validated list of sources, use drop list

Source #	See the procedures in Chapter 1 for the appropriate values
State	controlled by a validated list of states, use drop list
Location	generally blank
Latitude	location including degrees, minutes and seconds
Longitude	location including degrees, minutes and seconds
Mode	controlled by a validated list of modes, use drop list
Gear	controlled by a validated list of gears, use drop list
Vessel Name	Enter this information in this field, if available. **DO NOT ENTER VESSEL #'S** **Vessel #'s are confidential data**

TL (mm)
FL (mm)
Whole Wt (kg)
Gutted Wt. (kg)

If the length or weight data is not in millimeters or kilograms, respectively, then use the conversion action buttons to calculate the lengths and weights in the appropriate unit, simple place the cursor in the desired field, enter measurement, and press the appropriate conversion button, then move to next field by using the mouse.

Date Samples Received	date samples received by PC Lab month/day/year
Otoliths	**Y** is sample taken/received
Otolith Comment	Record if otoliths are whole, chipped, or broken
Spines, Vertebrae, Muscle, Skin, Liver	**Y** is sample taken/received
Gonad	**Y** is sample taken/received
Gonad Partial or Whole	Indicate whether whole gonad or only partial 'plugs' were sampled from the gonad
Gonad Wt. (g)	if available, fresh weight of gonad as recorded by observer
Sex	Protected by validating rule, enter one of the following: F Female M Male I Indeterminate

	or Leave blank
Gonad Comments	Anything that pertains to the collection, if comment pertains to one fish only include in specific record For example, 'samples in vials'
Other Tissue	comments pertaining to other tissue or condition of the fish For example, 'gonad/liver vials frozen'
Date Shipped Out	Date when samples are shipped to other researchers
Samples Shipped To	Full address with name of researcher samples was sent to
Collection (S/W)	Leave blank
Collection Comments	any other important information For Example: gonads were preserved in formalin and stored at another location, etc.

3.9 Procedures for Proofing Archival Data

Proofing archival (pre-1998) data can be a tedious and stress-inducing task but is extremely useful in creating large datasets of age, length, and reproductive data. The main rule of thumb is to be consistent.

Gather the following materials:

- Previous written logs
- Sectioned and/or whole otoliths
- Reproduction processing sheets (RFRS LAC binders)
- Other original datasheets (see file cabinets)

Step 1 If otoliths have been sectioned, then put slides in order by date and by fish number. If otoliths are still whole make sure a written log has been completed.

Step 2 Obtain a print out of the whole otolith log, make sure it is in order by date.

Step 3 Leave extra room at the end of the whole log so that collections of sectioned otoliths can be added to the log (such that a completed log of both whole and sectioned otoliths and gonads is created).

Step 4 Open the associated annual group database and click on action button Proofing Data Form (Figure 3.9.1). This form contains 36 fields that need to be verified, edited, and/or data added to the

database. This form presents the data in order by date and by fish number in chronological order.

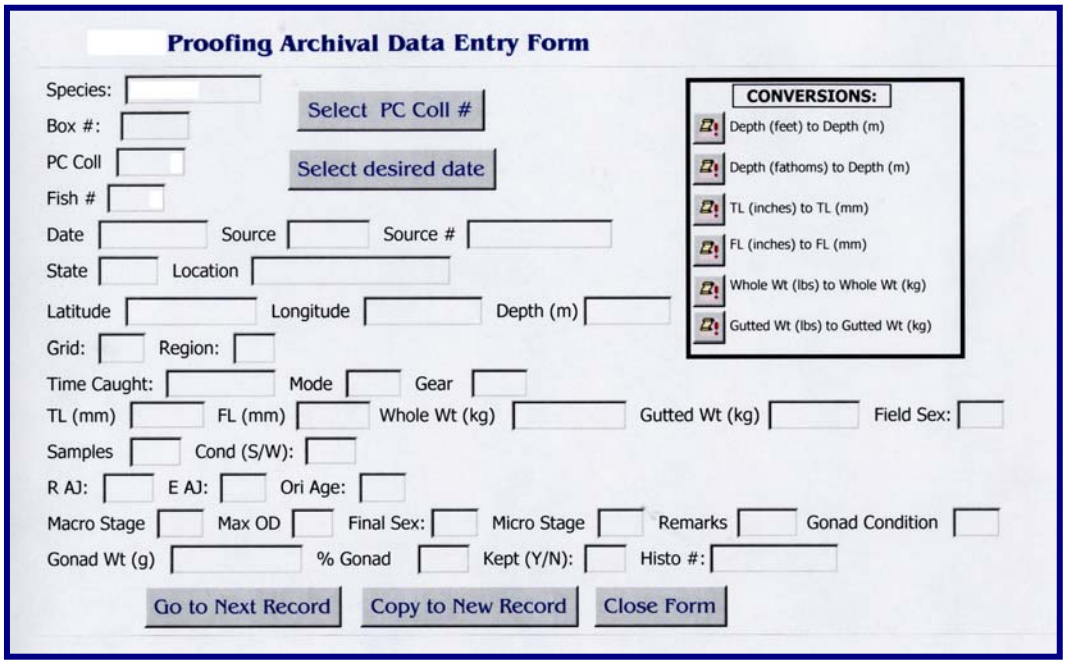

Figure 3.9.1 Proofing Archival Data Entry Form

Step 5 First, compare the data on the slide with the data in the database.

1. Search the database by date by clicking on the action button Select desired date (in this format: M/DD/YYYY). For example: Enter 1/25/1994
   – If the data on the slide matches the data in the database, find the associated 'Reproduction Processing Datasheet'.

Now to allocate a PC Coll #:
Check the log and make sure that the slide does not belong to a collection of whole otoliths (look at the whole otolith log and run through the dates).

2. If the slide belongs to a whole collection, then give the slide the associated PC Coll. # and verify the data on the original datasheet.

3. If the slide **DOES NOT** belong to a whole collection, then give the slide the next sequential PC Coll. # and

4. Add the following information to the log:
   - Date
   - Source (TIP, HB, etc.)
   - Source # (see key for appropriate values)

- State
- Location
- Mode
- Gear
- Collector's Name (if known)
- total # of gonads
- total # of otoliths
- Condition of the otoliths (S/W)
- Any other Comments

By comparing the data in the database, slides, and the original Reproduction Processing sheets, count how many samples were in the collection.

A collection consists of samples collected on the same day with the same source #, mode and gear.

5. Now add, verify or edit the following data on the 'Proofing Form':
(Use the original data sheet as the official data)
    - Year & Species
    - Box #
    - PC Coll #
    - Fish #
    - Date
    - Source
    - Source #
    - State
    - Location
    - Latitude
    - Longitude
    - Depth (m)
    - Time Caught
    - Mode
    - Gear
    - TL (mm)
    - FL (mm)
    - Whole Weight (kg)
    - Gutted Weight (kg)
    - Field Sex = Sex Pr.
    - Samples (Otoliths and/or gonads)
    - Condition of the otoliths (S/W)
    - R AJ (see slide)
    - E AJ (see slide)
    - Ori Age AJ

If 2 Readings or ? on slide, leave R AJ blank.
Ori Age AJ field should match the R AJ field.

See Gonad Processing Sheet for these fields

- Macro Stage = Stage Prl
- Max OD = Max OD
- Final Sex = Sex Fnl
- Micro Stage = Stg Fnl
- Remarks = Remarks
- Gonad Condition = Nad. Cond.
- Gonad Weight (g) = Nad. Wt.
- % Gonad = % of Nad.
  - Refer to Gonad Cond. code
- Kept (Y = gonad was kept & blocked, N = gonad discarded, not blocked)
- Histological Slide #

Step 6 Finally, if the data on the slide does not match any data found in the database, then allocate a new PC Coll # and add the associated information to the log and data file (see above).

Step 7 Write the year, species abbreviation, collection # - fish # on slide and put in the next available slide box.
For Example: 1994 RS Coll # 1-Fish # 1
1994RS01-01 or 94RS01-01

Step 8 Repeat above steps until all slides are properly checked and labeled. Click on Close Form when done. This button will bring you back to the database menu. Then click on Quit and Microsoft Access will close down.

3.10 Adding New Species to the Age, Growth, and Reproduction Databases

As the stock assessment effort at NMFS increases and more comprehensive fishery independent sampling efforts are made new species will need to be added to our database and archive collection. The following section list the procedure for this task.

Step 1 Open Archive Database

1. Open Query: Species List

2. Search Query by Common Name for the new species

3. If the New Species ***does not*** exist:
   a. Create Abbreviation* for new species

   b. Search Query: Species List to check the new abbreviation is unique

c. Add New Species to Archive Database:

- Family Name
- Scientific Name
- Common Name
- Abbreviation*
- NMFS Code*
- NODC Code*

*Search SAMPLE_ABBREVS_FINAL_TIP.exl
If the new species is not included in this file, then please see supervisor.

d. If your New Species does exist, go to Step 2.

Step 2 Open Annual Databases

1. Open Table: Species List

2. Add new Species to Annual Database:
   - Year (space) Abbreviation (in all caps)
   - Abbreviation (in all caps)
   - Common Name

Step 3 Create Log Sheet for New Species

1. Go to Folder I:\Blank Log Sheets

2. Choose Appropriate Folder: Grouper Logs, Snapper Logs, Other Species

3. Open one of the word documents

4. Save as 'new species name'

5. Correct the following (upper left hand corner):
   - Scientific Name
   - Common Name
   - NMFS Code
   - NODC Code

6. Print out New Species Log Sheet and file in 3-ring Binder.

Chapter 4: Proofing Data Written January 2008

It is of vital importance to "proof" or check all data entry to help ensure scientific integrity at the Panama City Laboratory. There are several types of data entered into the databases. These types of data are Log, Effort, Meristic, Reproductive, Histological, and Age Data.

This chapter contains the following:

4.1 Preparing and Printing Proofing Sheets

4.2 Proofing the Data

4.3 Making Corrections to Databases

4.4 Printing the Proofed Log

4.5 Conversion Factor Table

Contributor: Carrie Fioramonti

4.1 Preparing and Printing Proofing Sheets

Refer to the example spreadsheets (Figure 4.1.1) for proofing sheet templates. This is only a template for all data fields entered for each category of data. Occasionally, more or less data will need to be included in the proofing sheets per biologists' requests or data from more than one category may be combined onto one data sheet for efficiency. Age data is typically not proofed from hardcopy sheets, but rather from the computer data entry screen itself.

Steps to Printing Log Data Proofing Sheets

Step 1 Open appropriate Annual Group Database (see Chapter 3).

Step 2 Choose "Reports" from the Objects Menu

Step 3 Choose "Log: Species Select" and type in the year and species

Step 4 Print log report

Steps to Printing Proofing Sheets for Meristic, Effort, Reproductive, and Histological Data

Step 1 Open appropriate Annual Group Database (see Chapter 3).

Step 2 Create a query or modify an existing query to include all desired fields/data to be proofed (see Figure 4.1.1)

Step 3 Import data from Access into an Excel file.

Step 4 Customize the proofing sheets (landscape, margins, header/footer, gridlines, repeating rows, font, etc)

Step 5 Print proofing sheets.

Step 6 Create a checklist for proofreaders to initial, indicating that data has been proofed and/or changes entered (Figure 4.1.2)

Step 7 Create project notebook. Insert proofing sheets and checklist.

Figure 4.1.1 Example proofing spreadsheets, including typical data fields.

Meristic & Effort Data

PC Coll #	Fish #	Date	Latitude	Longitude	Depth (m)	Grid	Region	Vessel Name	Bias Type	Total Length (mm)	Fork Length (mm)	Whole Wt (kg)	Gutted Wt (kg)	Samples	Sex	# of otoliths per fish	Otolith Comm ent

Reproductive Data

PC Coll #	Fish #	Date	Samples	Sex	Macro Stage	Max OD	Micro Stage	Gonad Condition	% Gonad	Gonad Wt. (g)	Gonad Wt. in Formalin (g)	Remarks	Kept	Histo #	Gonad Comments
.															.

Histological Data

Histo #	PC Coll #	Fish #	% Gonad	Gonad Wt. In Formalin (g)	Gonad Region	Leading Gamete Stage	Histo Sex	Short-Term Atresia	Post-Ovulatory Follicles	Indicators of Prior Spawning	Histo Class	Histo Comments

Figure 4.1.2 Example of checklist for proofreaders for the Log

2006 Log Proofing
Groupers

Please initial and date

Species	**Proofing Started**	**Proofing Completed**	**Corrections Entered**
Black Grouper			
Creolefish Grouper			
Gag Grouper			
Graysby Grouper			

2006 Log Proofing
Snappers

Please initial and date

Species	**Proofing Started**	**Proofing Completed**	**Corrections Entered**
Blackfin Snapper			
Cardinal Snapper			
Cubera Snapper			
Dog Snapper			

4.2 Proofing the Data

- Gather the following materials:
    1. Proofing sheets
    2. Original data sheets from otolith collections
    3. Handwritten log books
    4. RFRSLAC Notebooks, and/or Green Histological Records.

- Compare data on the proofing sheets to the ***original*** data sheets. If necessary, refer to Chapter 2: Data Sources for interpretation.

- Using a **RED** ink pen, make necessary corrections to data proofing sheets. Initial and date the lower right hand corner of each proofing sheet.

- Put a red check mark at the beginning of the record to indicate that the data has been proofed

- Note ANY discrepancies or omissions between the original data and the database on the proofing data sheets.

- Correct **handwritten** log book AT THIS TIME. If log sheets for previous years have been printed out, correct these, as well.

- Upon completion of a section/year, initial checklist.

4.3 Making Corrections to Databases

Step 1 Open Annual Group Database (see Section 3.1 & 3.2).

Step 2 Open the query created to print out proofing sheets

Step 3 Enter the corrections directly into the query.
Occasionally, corrections are more easily made from the form (rather than queries). An example would be when a fish record is completely missing. The form allows you to copy an entire record (automatically enters all log, effort, meristic, reproductive, histological data) and change the meristic, reproductive, and/or histological data accordingly.

Step 4 Once all corrections have been made, initial checklist.

4.4 Printing the Proofed Log

Once all log data has been proofed and the corrections entered into the database and handwritten log, a final log report for the year may be printed. These reports are kept in binders in the Age, Growth, and Reproduction Lab arranged by species in chronological order. Follow this procedure to print out the log report.

Step 1 Open the Annual Age, Growth, and Reproduction (AGR) Database

- Open the Age, Growth, and Reproduction Server Folder
- Open the specific year AGR database
- The server will prompt user for username and password
- Enter Microsoft Access Username
- Enter Microsoft Access Password

Step 2 Click on Other: queries and/or reports

Step 3 Open Report 'Log: Select Species'

Step 4 Enter year and species abbreviation

Step 5 Print out the report

Step 6 File report into appropriate log binder

4.5 Conversion Factor Table

Measurement	Multiply by	Converts to
Centimeters (cm)	0.1	Millimeters (mm)
Kilometers (km)	0.5396	Miles, nautical
Inches (in)	25.4	Millimeters (mm)
Feet (ft)	0.3048	Meters (m)
Fathoms (ftm)	1.8288	Meters (m)
Yards (yd)	0.914	Meters (m)
Grams (g)	1000	Kilograms (kg)
Kilograms (g)	0.001	Grams (g)
Pounds (lb)	0.4536	Kilograms (kg)

Chapter 5: Storage of Otoliths Updated January 2008

This chapter includes procedures for storing whole and sectioned otoliths, including the most recent samples and backdating to those collected in the 1980s (Table 5.1.1). All samples are located in the Amesbury Age Structure Archive of the Panama City Laboratory, unless requested to be sent to another location. The most recent samples (current year) are grouped in one location within the Amesbury Age Structure Archive in species specific boxes (Figure 5.1.1). Samples from previous years are grouped together by species and arranged in chronological order. This chapter also discusses the methods used in the storing of sectioned otoliths.

This chapter contains the following:

5.1 Storing Whole Otolith Samples

5.2 Storing Sectioned Otoliths (both present and archive)

5.3 Storing Archive Whole Otolith Samples (before present year)

5.4 Creating Archive Box Labels

Contributors: Meredith Burnette, Linda Lombardi, Nancy Evou, Natalie Amesbury, and Carrie Fioramonti

5.1 Storing Whole Otolith Samples (present year).

Step 1 After the otolith sample has been properly logged in and given species specific Panama City Collection # (PC Coll #); the entire collection is placed in its corresponding species specific box, in consecutive order, which is located in the archive or the appropriate INBOX in the Age and Growth Lab.

Step 2 If more than one storage box is needed per species, create a new box label (Figure 5.1.2), including the range of Collection #s found in the box, as well as the number of the box corresponding to the species for that present year.

Note: Due to spacial constraints, only one box per species is located within the present year grouping. Full boxes from the present year should be moved to the archive space at the end of the species grouping.

Figure 5.1.1 Diagram of the Ambesbury Age Structure Archive

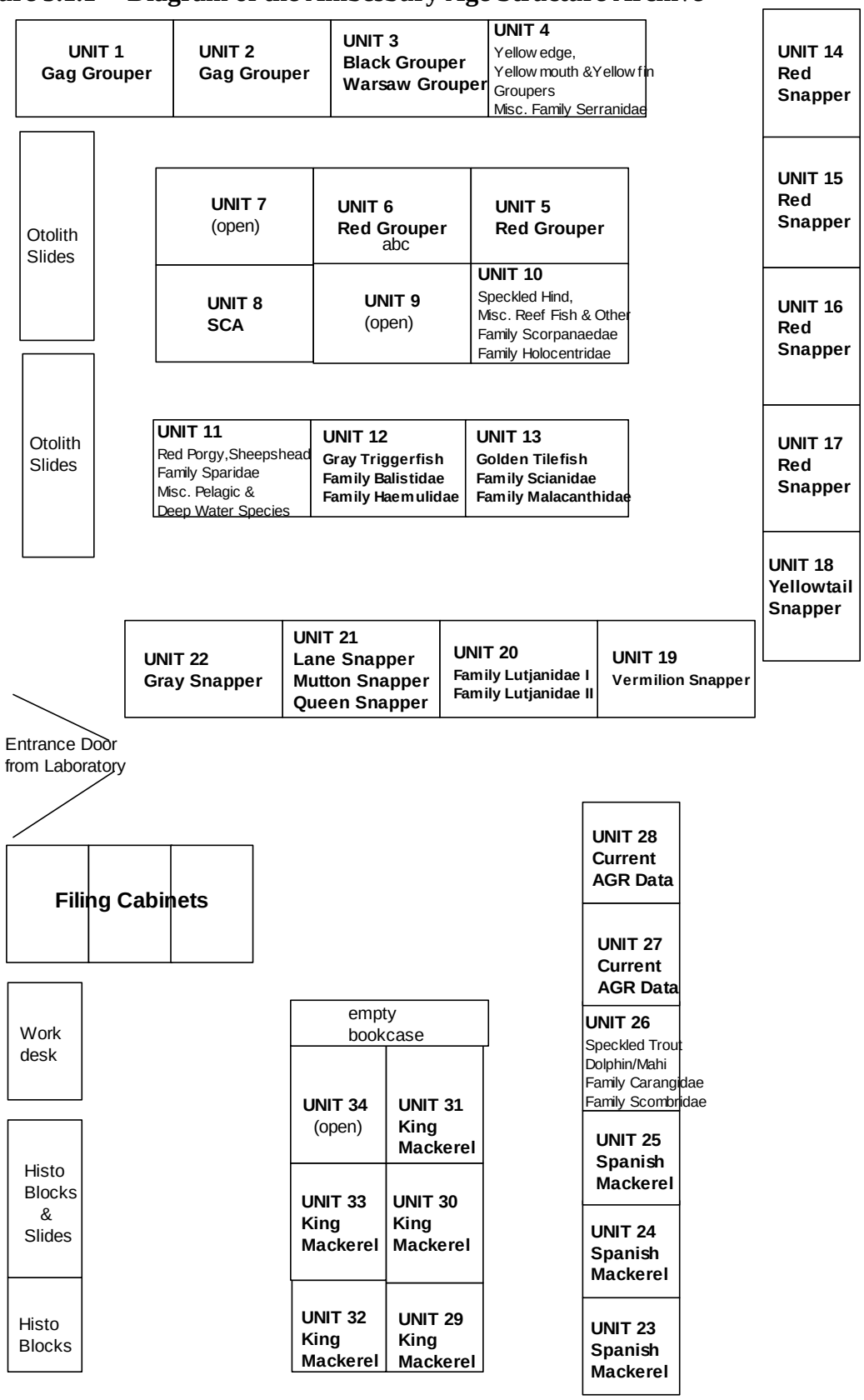

Table 5.1.1. Complete list of species and number of age structures currently in the Amesbury Age Structure Archive, arranged in alphabetic order by Family and Common Names.

Family Name	Common Name	1990	1991	1992	1993	1994	1995	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007	Total
Balistidae	Filefish, Orange		1																	1
	Filefish, Planehead			1																1
	Filefish, Unicorn		1	5		2	2													10
	Triggerfish, Gray	17	90	682	410	482	258	179	59	74	109	341	326	260	154	176	291	277	224	4,409
	Triggerfish, Ocean												2							2
	Triggerfish, Queen												6	6	16	1	2	4		35
Bothidae	Flounder, Channel					1														1
	Flounder, Dusky		5	12	4	1														22
	Flounder, Gulf				2	5	1	1							1			2	2	14
	Flounder, Lefteyed			1																1
	Flounder, Ocellated				1															1
	Flounder, Southern					2											13			15
	Flounder, Summer		7	10	8	1														26
Carangidae	Amberjack, Greater	47	8	3	1	25	17	28	17	5	4	33	44	75	181	115	75	31	24	733
	Amberjack, Lesser		1	1	2							1				10		6	26	47
	Banded rudderfish			18	5	5													2	30
	Jack, Almaco			9	3	3	3		1	1		2	6		9	1	3	6	45	92
	Jack, Cotton Mouth			1	1															2
	Jack, Crevalle																		1	1
	Permit																		1	1
	Pompano, Florida																		30	30
	Runner, Blue																		5	5
Coryphaenidae	Dolphin																			
Emmelichthyidae	Crimson Rover				2															2
Ephippidae	Spadefish			2											1				14	17
Gadidae	Hake, Gulf																	1	6	7
	Hake, Southern																	6	5	11
Haemulidae	Grunt, Bluestriped														1					1
	Grunt, Margate									1			5	22	29	11	6	21	11	106
	Grunt, White		2	136	91	19	1	3	17	2			5	38	12	14	379	185	176	1,080
	Pigfish		1	12	4															17
	Porkfish																2			2

Family Name	Common Name	1990	1991	1992	1993	1994	1995	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007	Total
	Tomtate		8	23								3			1		3		8	46
Holocentridae	Soldierfish, Bigeye																		1	1
	Squirrelfish							1					8				1	7	6	23
Labridae	Hogfish													6	5			1	2	14
	Hogfish, Spanish				1								1							2
	Razorfish, Pearly																			
Lobotidae	Tripletail			1											4	3		1	3	12
Lutjanidae	Snapper, Alfonsin						1													1
	Snapper, Black								17	1										18
	Snapper, Blackfin		6	2	2	1			1	1	11	1	22	30	55	108	72	108	165	585
	Snapper, Cardinal												1		4	3		9	2	19
	Snapper, Cubera			3	1	1		3	1	1	1		6	15	29	10	3	15	12	101
	Snapper, Dog		1	1								1	1	13	14	13	3	29	22	98
	Snapper, Gray	13	46	174	268	647	293	362	565	193	186	100	252	600	747	599	842	728	627	7,242
	Snapper, Lane	15	160	358	230	252	130	146	124	88	135	69	130	318	203	202	501	421	448	3,930
	Snapper, Mahogany		9	1																10
	Snapper, Mutton			16		68	4	50	37	3	5	12	77	157	168	193	184	451	246	1,671
	Snapper, Queen		112	4	34							9	34	19	51	60	38	30	33	424
	Snapper, Red	32	1,659	3,816	3,861	2,693	908	372	383	13,140	11,803	6,921	7,646	10,135	7,524	6,178	7,027	7,462	8,370	99,930
	Snapper, Schoolmaster			1		9	1		1				1						3	16
	Snapper, Silk		10	1	25	5	4	2	26	1	38	11	26	46	80	103	86	90	174	728
	Snapper, Vermilion	21	624	1,016	1,141	881	339	454	97	202	390	1,594	2,374	1,987	3,335	2,693	4,608	4,035	4,889	30,680
	Snapper, Wenchman							1	1					6		4			120	132
	Snapper, Yellowtail		3	129	235	331	434	609	590	197	176	165	233	696	442	342	236	246	280	5,344
Malacanthidae	Tilefish, Anchor																		1	1
	Tilefish, Blackline														1	2		3	3	9
	Tilefish, Blueline												16	3	5	136	34	23	202	419
	Tilefish, Golden				3			1	51	5		23	131	147	316	555	723	932	1,546	4,433
	Tilefish, Goldface								43	5				1		4	3	4		60
	Tilefish, Sand														2		3	1	3	9
Mugilidae	Mullet, Redeye							4		1				1,146	3					1,154
	Mullet, Striped														1		30	30		61
Ophidiidae	Brotula, Bearded				2							1	3		6	1	21	33	15	82
Percichthyidae	Wreckfish															1				1
Pomatomidae	Bluefish				2	1									2	3	16	1	2	27

Family Name	Common Name	1990	1991	1992	1993	1994	1995	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007	Total
Priacanthidae	Bigeye				1										1	1		4	1	8
	Bigeye, Short		3	4	1												3			11
	Snapper, Glasseye												1							1
Rachycentridae	Cobia			7	2	6	4	1		4	6		1	3			2	3	4	43
Sciaenidae	Croaker, Atlantic																34	84	44	162
	Drum, Black												3		2		41	1		47
	Drum, Blackbar																4			4
	Seatrout, Sand	6	6	1	1									5			139	156	171	485
	Seatrout, Silver														3					3
	Striped croaker			1	1															2
Scombridae	Little Tunny														8	18	21	1		48
	Mackerel, Cero									5		1	1		1	5		20	1	34
	Mackerel, King	1	7				1		7	3	287	558	2,856	3,238	4,591	3,637	3,078	4,197	2,812	25,273
	Mackerel, Spanish			5							496	221	1,012	1,226	1,525	1,496	1,206	1,317	1,150	9,654
	Tuna, Blackfin														1			3	1	5
	Tuna, Bluefin																		29	29
	Tuna, Skipjack			6	1	3		1									1			12
	Tuna, Yellowfin															14	9		2	25
	Wahoo			2							8		3	11	6	6	6	16	2	60
Scorpaenidae	Blackbelly Rosefish												14			2	1	1	2	20
	Scorpion Fish (spinycheek)						1						1	1	23	14	29	37	50	156
	Scorpion Fish (spotted)																		1	1
Serranidae	Bass, Longtail				17									1		2	22	21	30	93
	Bass, School				20															20
	Coney								2							1	2		1	6
	Creole-fish	1		3	1	2			3				12	2	3	3	23	25	61	139
	Grouper, Black		1	6	11	1		3	3	2	15	10	59	79	146	155	213	299	185	1,188
	Grouper, Comb				1		1								1					3
	Grouper, Gag	12	363	559	940	904	667	983	412	352	531	652	1,838	2,348	2,066	2,633	3,439	2,360	1,513	22,572
	Grouper, Graysby	2		7	3		7	5	2				2	25	11	2	6	9	16	97
	Grouper, Marbled		7										1	1	1	1	2	3	15	31
	Grouper, Misty												2	2	9	2	1	5	8	29
	Grouper, Nassau																1			1
	Grouper, Red		159	296	502	537	583	470	181	306	906	809	2,091	2,919	3,367	4,082	4,396	4,464	3,919	29,987
	Grouper, Scamp		290	194	439	323	249	285	106	128	179	230	1,239	1,896	3,136	2,218	2,027	1,548	1,898	16,385

Family Name	Common Name	1990	1991	1992	1993	1994	1995	1996	1997	1998	1999	2000	2001	2002	2003	2004	2005	2006	2007	Total
	Grouper, Snowy		102	87	42	45	6	5		20	32	22	126	112	317	407	326	436	788	2,873
	Grouper, Warsaw	2	23	19	6	3	1	6	2	9	9	18	34	87	130	111	97	74	64	695
	Grouper, Yellowedge		302	74	26	2				50	100	227	701	259	914	659	774	547	1,445	6,080
	Grouper, Yellowfin		1		1							2	7	1	4	11	6	23	9	65
	Grouper, Yellowmouth	1	10	2	2	5		1			5	2	6	18	8	16	3	12	15	106
	Hind, Red		3	1		1				1			18	20	20	7	7	53	7	138
	Hind, Rock	2	23	36	18	10	22	23	6	4	2	1	7	21	25	18	27	46	52	343
	Hind, Speckled	1	3	9	2	4	2	2	4	16	19	26	125	298	892	569	507	503	527	3,509
	Sand Perch		26	30	3							4	4				4	1	29	101
	Sea Bass, Bank		28	14							10	81	24	1			2		7	167
	Sea Bass, Black		1	32	19	9	1	6	2	543	20			1	16	5	87	49	39	830
	Sea Bass, Rock											1								1
	Spanish Flag																	2	1	3
	Tattler											3	11						1	15
Sparidae	Pinfish (porgies)		1		1															2
	Porgy, Grass			6		1											1			8
	Porgy, Jolthead								11		1		1	1		20		3	1	38
	Porgy, Knobbed							4					1	11	2			5	4	27
	Porgy, Littlehead	2		21	11	1						17	19	6	3	2				82
	Porgy, Longspine													1					1	2
	Porgy, Red		5	85	65	35	383	214	141	116	163	173	271	103	191	130	137	151	399	2,762
	Porgy, Saucereye		1	9	3	5	1													19
	Porgy, Sheepshead														4		115	30		149
	Porgy, Whitebone		6	25	12	8	9	4		3	4	9	8				2	1	4	95
Sphyraenidae	Barracuda, Great												2		2		6	10	13	33
Stromateidae	Barrelfish				5											16			1	22
	Driftfish, Black												4			2			5	11
Synodontidae	Lizardfish, Inshore																		5	5
	Lizardfish, Red																		1	1
Trichiuridae	Escolar																15			15
	Oilfish													1						1
Grand Total		175	4,125	7,980	8,496	7,340	4,334	4,229	2,913	15,483	15,651	12,354	21,861	28,424	30,831	27,806	32,027	31,719	33,089	288,837

Figure 5.1.2 Current year otolith storge box label.

Species: _____

Year: _____

Box #: _____

Collection #s _____

5.2 Storing Sectioned Otoliths (present and archive)

Slides of sectioned otoliths from fish under ***current*** study are kept on shelves in the Age, Growth and Reproduction Lab. Each species is separated and arranged in chronological order. For the most part, each slide box contains only samples only from one year, regardless if there is 40 or 90 in one year.

Each slide box is labeled
As the diagram shows:

1
9
9
9

R
S

B
O
X

1

Year
Species Abbreviation
Box #

The slides are arranged by Collection number and then fish number within each box. The box number is another number that is used to reference the location of that slide. This number is entered in the database. If there are any questions that might rise concerning one fish, it can be easily located and the problem resolved.

All ***archived*** slides of sectioned otoliths are kept on bookshelves in the Amesbury Archive. Slides are arranged first by species, then in chronological order. Each box holds 1000 slides of a single species and may contain multiple years.

5.3 Storing Archive Whole Otolith Samples (before present year).

Archive otolith samples are species that are not currently under investigation. In 1999, this process was first begun and numerous samples were organized, first separating collections by species and then by year. Each species collection was then logged in (see Chapter 1 for procedures) by year and a tally of whole otoliths was calculated. Our main goal was to have a complete list of the number of samples by species per year.

Step 1 Once log data entry from the most recent year has been proofed, the boxes are arranged first by species, then by chronological order in the archive (see archive diagram).

Step 2 Create labels for the new boxes to the archive (see section 5.4)

Step 3 Finally, update the Otolith Inventory List (Table 5.1.1).

5.4 Creating Archive Labels

Labels are created assuming that the content of the archive box is complete and will not change. Generally, the labels are updated after annual log proofing has been completed, or samples have been pulled for sectioning. Examples of labels can be seen in Figure 5.4.1.

Step 1 Record the label information from unlabeled archive boxes onto an Archive Label Info Table form, I:\Other files\Archive Labels. Be sure to reassign the box with a new box number that is contiguous with the entire species grouping (i.e. the last box of labeled Red Snapped is BOX 71, therefore the next box will be BOX 72).

Step 2 Enter the new label information in the Excel Spreadsheet entitled Archive Labels Spreadsheet, I:\Other files\Archive Labels. An example of the spreadsheet is shown in Figure 5.4.2. This spreadsheet represents what is contained within the entire archive. BE CAREFUL WITH THIS FILE! Work with it from your machine until labels are complete, before updating the server file!

a) When entering data into the spreadsheet, be sure to enter ALL TEXT within that field that needs to be printed on the label. For example, in the "Collections" field, record data as "Colls. 1-43" as opposed to only"1-43". The program simply places the text in its appropriate position and font.

b) If multiple years will be included on one label, use the subsequent fields (Year1, Coll1, Notes1; Year 2, Coll 2, Notes2; Year 3, Coll 3, Notes3) to record data, so the program will arrange the information in the correct format.

c) Save the updated spreadsheet to the server (and your desktop).

d) Close the Excel spreadsheet prior to creating the labels with Mail

Step 3 Using Microsoft Mail-Merge to create labels for new archive boxes

a) In Microsoft Word, under the **Tools** drop-down menu, select "letters and mailing" and "Mail Merge". Select **document type** as "labels". Click "next: starting document"

b) Change the **document layout** by selecting "label options".
   1. A window will appear, giving a selection of sizes for labels.
   2. Choose "3260 Notecard".

c) Click "next: choose recipients". **Use an existing list** by choosing "browse". Choose the Archive Label Spreadsheet file. A window will appear, giving a selection of records from the excel file. Choose the appropriate species spreadsheet. Some spreadsheets have only one species, some have multiple species. Note: only one spreadsheet can be accessed at one time.

d) Choose the corresponding box labels to be printed. Click "next: arrange your labels" Choose "more items". A window will appear listing each field of the spreadsheet. "Insert" all of the fields listed. Close the window.

e) Create your own "macro" by positioning each category and giving it the appropriate font (style and size). Every label will be created according to this format.

f) If all labels are of similar format, click on "update all labels". This will assigned the macro to all labels in the document. If the labels ARE NOT of similar format, you will have to create two different documents.
   1. Click on "next: preview your labels". Inspect your labels. If corrections must be made, use the "previous" selection link.

g) If the labels are ready for printing, click on "next: complete the merge".
   1. Then choose "edit individual labels". A window will appear, choose "all" and "OK" to create a new, separate document containing the archive labels ready for printing.

Note: A file containing examples of labels can be saved as templates. However, it can be difficult to amend and print from these files, as only entire documents can be printed and the copy/paste function does not always perform properly when trying to add new labels to the document.

Step 4 Printing Archive Labels

Note: Only entire **documents** can be printed using Mail Merge.

a) Using an inkjet printer, print labels on solar (yellow) paper
b) Cut each label to fit inside of a pint-sized Ziploc bag
c) Be sure to put the printed side of the label on the clear side of the Ziploc bag, so that the label clearly shows through.
d) Put the labels inside the front of the corresponding box.

Figure 5.4.1 Examples of Archived Slide Box Labels Created by Microsoft Mail Merge.

Box 86

King Mackerel (KMK)

Season 2005-2006 Coll.05KMK310-05KMK360

Box 4

Gray Snapper (GS)

Year 2002 Coll. 177-188
Year 2003 Coll. 1-80

Box 1 **All Years- 2005**

Family Lutjanidae I

Black Snapper
Blackfin Snapper
Cubera Snapper

Box 1 **All Years**

Misc. Pelagic Species

Pomatomidae
Bluefish

Rachycentridae
Cobia

Figure 5.4.2 Example of Archive Labels Spreadsheet

BOX #	ABBREV	SPECIES	YEAR1	COLLS	NOTES	YEAR2	COLLS2	NOTES2	YEAR3	COLL3	NOTES3
Box 1	(GTR)	Gray Triggerfish	Year 1980-1991	All Collections		Year 1992	Coll. 1-160				
Box 2	(GTR)	Gray Triggerfish	Year 1992	Coll. 161-268		Year 1993	Coll. 1-96				
Box 3	(GTR)	Gray Triggerfish	Year 1993	Coll. 97-182		Year 1994	Coll. 1-225				
Box 4	(GTR)	Gray Triggerfish	Year 1995-1998	All Collections							
Box 5	(GTR)	Gray Triggerfish	Year 1999	Coll. 1-24		Year 2000	Coll. 1-82		Year 2001	Coll. 1-21	
Box 6	(GTR)	Gray Triggerfish	Year 2001	Coll. 22-89		Year 2002	Coll. 1-40				
Box 1	(RDG)	Red Grouper	Year 1981-1993	All Collections							
Box 2	(RDG)	Red Grouper	Year 1994-1998	All Collections							
Box 3	(RDG)	Red Grouper	Year 1999	All Collections							
Box 4	(RDG)	Red Grouper	Year 2000	All Collections							
Box 5	(RDG)	Red Grouper	Year 2001	Coll. 1-115							
Box 6	(RDG)	Red Grouper	Year 2001	Coll. 166-240							
Box 7	(RDG)	Red Grouper	Year 2001	Coll. 241-284		Year 2002	Coll. 1-55				
Box 8	(RDG)	Red Grouper	Year 2002	Coll. 56-166							
Box 9	(RDG)	Red Grouper	Year 2002	Coll. 167-244							
Box 10	(RDG)	Red Grouper	Year 2002	Coll. 245-338							
Box 1	(MTS)	Mutton Snapper	Years 1981-2006	All Collections	On Loan J. Ohop in St Pete.						

Chapter 6: Preparing Otoliths for Microscopic Examination Updated January 2008

The otoliths of bony fish contain a wealth of information. Locked in the recesses of its size, shape and form one can discern its chemical and physical properties, its intricate patterns of opaque and translucent zones, and possible clues to the morphology, physiology, and age of the fish it came from. Scientists around the world are engaged in a concerted effort to uncover these clues and render them into useful and reliable information. However, nature does not give up secrets easily. A large number of environmental and biological factors presumably affect the formation and growth of otoliths, and that confounds efforts to decipher the information locked within. Investigation into these variables is an ongoing and consuming process that is taking place in laboratories both here and abroad. Only after years of dedicated study and practical experience do these little stones begin to yield their information.

One of the most useful applications to date to come from the study of otoliths is in determining of the age of individual fish. The interpretation of the opaque and translucent patterns (annuli) seen in most fish otoliths is far from perfect. Considerable variation in patterns from one species to another, and difficulty correlating annuli with specific years, has made otolith interpretation as much an art as it is a science. Nevertheless, despite all the obstacles to a standardized and definitive procedure for aging fish, considerable progress has been made in determining the age structure and distribution of a variety of fish species. This information is invaluable in managing and promoting the long-term health of our commercial and recreational fisheries.

This chapter outlines the general procedure for sectioning otoliths. Otoliths differ greatly in size and form. Consequently, there are differences in the procedures and techniques used to prepare otoliths for microscopic study. Even after establishing an accepted procedure for a specific species of fish, there may still be procedural differences from one technician to another. However, regardless of these procedural details, the basic requirements of otolith preparation remain the same. This section will discuss these requirements, and outline some of the generally accepted procedures for preparing an otolith for microscopic examination.

This chapter contains the following:

6.1 Weighing Otoliths

6.2 Steps to Sectioning Otoliths on Hillquist High-Speed Saws

6.3 Trouble-shooting for Sectioning Otoliths on Hillquists

6.4 Steps to Sectioning Otoliths on Buehler Isomet Slow-Speed Saws

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6.1 Weighing Otoliths

In many fish species, there is a strong correlation between otolith weight, and other attributes, such as age, length, weight, etc. The following four steps will help you obtain and record accurate otolith weights.

Step 1: Obtain otolith weight sheets from Access database.

Step 2: Tare (zero out) the scale before weighing and thereafter as required. Due to sensitivity of our scale, it may take from thirty seconds to a minute for the scale to return to zero once a sample is removed. Give it time to return to zero on its own. Forcing it to zero too soon could introduce error into the measurement. If after a reasonable time, if the scale reading still does not return to zero, use the Tare button.

Step 3: Only weigh **whole** otoliths.
A good rule is to weigh an otolith if at least 99% of the otolith is present. If a very small portion of the tip is missing you can still record the weight; sometimes the tip is still in the envelope so be sure all pieces are removed.

DO NOT weigh the otolith if the otolith is broken and pieces are missing.

Step 4: Our scale is very accurate; sensitive to one ten thousandth of a gram. It is therefore very important to remove (as much as possible) any dried up organic material that may still be clinging to the otolith.

Step 5: Record:

- Weight of left and/or right otolith (see Figure 6.1.1)
- Number of otoliths collected from each fish (1 or 2)
- Any comments regarding otolith (i.e. otolith in pieces, otolith broken in half, tip broken off, etc)

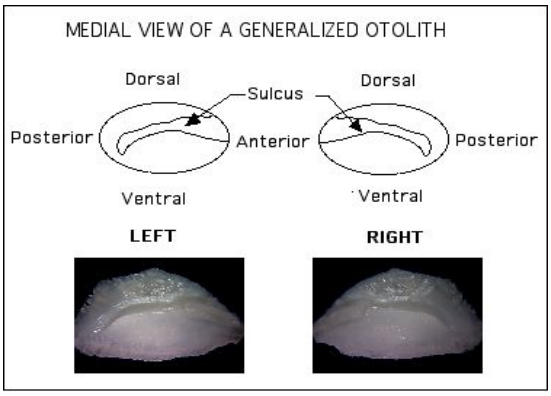


Figure 6.1.1 Determination of left versus right otolith (i.e if the bend in the sulcus is on the left side of the otolith, then it is a left otolith).

6.2 Steps to Sectioning Otoliths on Hillquist High-Speed Saws:

Step 1: Prepare the sectioning area with all supplies:

Slides – Star frost, or some suitable alternative, with a white frosted area at one end of the slide for labeling

Lab Markers – Black, solvent resistant markers used to label slides
DO NOT USE SHARPIE brand (alcohol soluble!)

Pencils – Used to mark the core of the otolith

Styrofoam Tray - Used to hold the slides

UV Glue – Used to secure the otolith half to the slide

Cytoseal- used to seal the final section

Step 2: Prepare the slides:

The number of otoliths selected for mounting at one time may vary, however it is usually convenient to limit the number of otoliths to the number of slides that can fit in one styrofoam tray, (approximately 28).

In addition, for ease of record keeping, and to facilitate continuity between the various technicians working on the same project, it is best to work with whole numbers of collections. That is, if a collection contains 20 otoliths, prepare them all at the same time. If the collections are small, several collections can be prepared at the same time (trying not to exceed the capacity of the Styrofoam tray). For collections exceeding 28 fish, two or more trays will be necessary.

Place the blank slides vertically on the table with the white label at the top. Using the Lab Marker write the following information across the top of each slide (Figure 6.2.1):

- The last two digits of that year (01, 02, etc.)

- The appropriate species abbreviation (RS, GAG, etc.)

- The collection number (1, 2, . . . etc.)

-The fish or tag # (1, 2, 3,. . . etc.)
Be careful here. The numbers are not always sequential, nor do they always start with one, "1". Check the data sheet that came with the collection to make sure that you use the right fish numbers.)

Here is an example of a slide label:

Capture year is 2002
Red Snapper sample
Collection number is 468
Fish number is 2

02 RS 468-02

When all the slides have been labeled, place them in a styrofoam tray in sequential order and proceed to step 3.

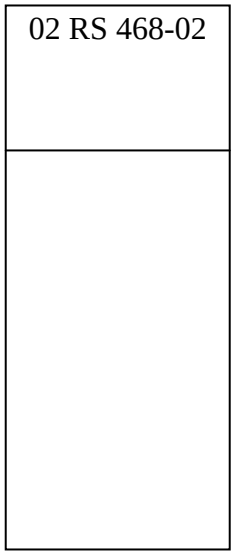


Figure 6.2.1
Labeled otolith slide

Step 3: Prepare the otoliths:

Open the envelopes/vials containing the otoliths. If there are two otoliths in one envelope, choose the **left** otolith for preparation. It is not difficult to distinguish between the two (See Figure

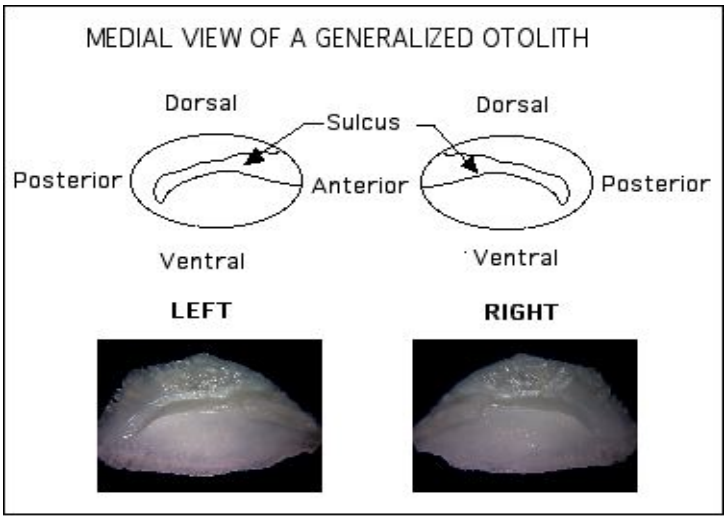


Figure 6.2.2 Comparison of left versus right otoliths

6.2.2). In most fish species, the gross features of the otolith are similar. The sulcus, a feature common to most otoliths, is a channel that runs along the proximal, or convex, side of the otolith. The end of the sulcus toward the anterior end of the otolith usually extends all the way to the anterior edge, and widens out into a delta-like formation. The opposite end of the sulcus is narrower, bent, and usually does not quite reach the posterior edge. Place the otolith flat on a table with the convex side up, and the bend in the sulcus pointing toward you. If the bend in the sulcus is on the left side of the otolith, then it is a left otolith, and vice versa.

Next, the core, or nucleus, of the otolith must be located. This is an important but not always straightforward procedure. The location of the core varies from species to species. In most cases the core is somewhere in the general area of the geometric center of the otolith, but there are exceptions. One technique for finding the core is to hold the otolith up to a strong light. A series of concentric rings paralleling the perimeter of the otolith is sometimes visible. At the center of these rings is a dense area, which is (in most cases) the core. If there is uncertainty, or dealing with an unfamiliar species, get help from someone with more experience.

Once the core is located, draw a pencil line across the otolith from dorsal to ventral edge, and just posterior to the core (See Figure 6.2.3). This line should be used as a guide when making your initial cut on the Hillquist saw. After marking the otolith, place it on its corresponding slide in the styrofoam tray. Before moving on to the next step, finish marking all otoliths in the selection.

Figure 6.2.3
Marking the core of the otolith

CAUTION - From this point until the end of Step 6 be very careful not to shake, jar, drop, or disrupt the tray in any way. The otoliths are loose on their designated

slides. If they are disturbed in any way that causes the otoliths to separate from their designated slides, and consequently their identifying labels, the otoliths must be reweighed and matched to the appropriate fish.

Step 4: First Cut:

The otolith is now ready for the first cut on the Hillquist saw. There are two switches located on the top, below the light. The switch on the left turns on the water pump, and the switch on the right turns on the saw. Next, turn on both water controls by turning the dials counterclockwise a few times. These are the black knobs on the back of the green tub.

STOP: Wear both eye protection and earplugs before proceeding!

While holding the otolith in both hands, align the line on the otolith with the blade on the left side of the saw. Cut through the otolith with light but steady pressure (See Figure 6.2.4). It is the anterior half (the half where the sulcus forms a delta; See Figure 6.2.2) that will be kept for further processing. Discard the other half.

Figure 6.2.4 . Make the first cut

Next move to the grinding blade on the left side of the Hillquist saw. While holding the half section in your right hand, carefully and slowly grind down the cut edge (See Figure 6.2.5).

Figure 6.2.5. Finding the core

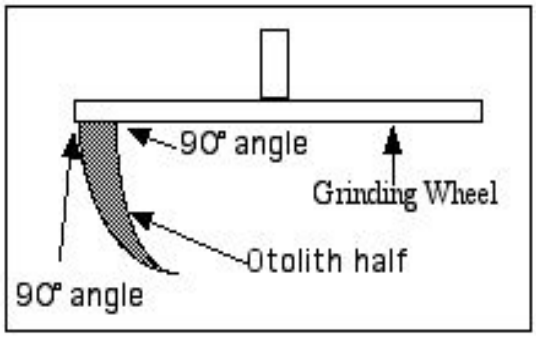


Figure 6.2.6 Proper cutting angle

Take care to assure that the cut surface is flat on the grinding wheel, and that the proximal and distal sides of the otolith half are at right angles to the grinding surface (See Figure 6.2.6). Remember, the objective is to get a good cross section of the core onto the slide, so at this point try to get as close as possible without actually going through or beyond the core. Grind the edge down in small increments, stopping frequently to examine the edge closely under a bright light (a small hand lens may be helpful here). As you approach the core, the cross section of the sulcus will usually take on a characteristic tornado shape (See Figure 6.2.7).

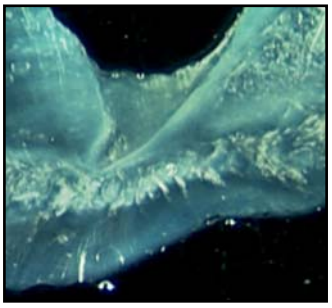


Figure 6.2.7 Tornado-shaped sulcus as the core is approached.

Moving closer and closer to the core, the sides of the tornado will begin to straighten out (See Figure 6.2.8) until, at the core itself, the cross section of the sulcus will form a perfect “V” shape (See Figure 6.2.9).

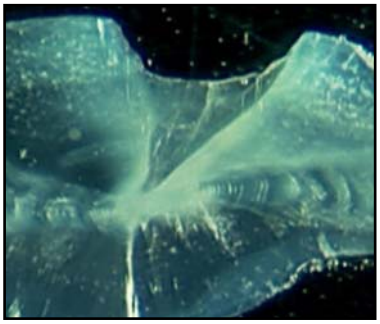


Figure 6.2.8 Tornado-shaped sulcus straightens at as you approach the core

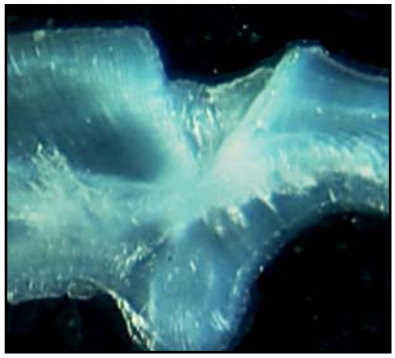


Figure 6.2.9 Ideal V-shaped sulcus at the core of the otolith

Figure 6.2.10 U-shaped sulcus if you grind past the core

If you inadvertently grind past the core, the sulcus will take on a characteristic "U" shape (See Figure 6.2.10). Ideally, you want to stop just before you get to the perfect “V". Do not get discouraged if you cannot hit the core every time. In most cases, the otolith can still be aged, although missing the core does make the job a bit more difficult. When you have finished grinding, place the half otolith back on its designated slide. Continue this process until all the otoliths on your tray have been cut and ground; then proceed to the next step.

Step 5: Sanding and Polishing:

In this step, you will sand and polish the cut and grounded edge of the otolith from Step 4 to a smooth surface. There are two objectives: the first is to provide a smooth surface that will adhere better when glued to the glass slide; the second is to provide an alternate surface (top and bottom so to speak) for microscopic examination. This is particularly important if the otolith was ground down to the core, or slightly past it in Step 4.

Sanding - The objective is to remove the rough grinding marks from otolith cross-section. Place a scrap piece of sand paper flat on the table and apply a small amount

Figure 6.2.11. Sanding the otolith surface by hand

of water to the paper. Grasp the otolith half firmly between the thumb and forefinger. Be sure the otolith is flat on the paper to avoid beveling. Rub the cut and ground surface of the otolith across the wet and paper (See Figure 6.2.11).

STOP: Wear eye protection before proceeding!

Figure 6.2.12. Polishing the surface of the otolith with the wheel

Polishing - The polishing wheel helps to clean off and remove the fine lines left by sanding. Turn on the polishing wheel, apply a little water and Micropolish to the spinning wheel, and reapply as needed. Hold the otolith half, flat as you did for sanding, and press it to the spinning wheel. There is little danger here of changing the shape or orientation of the cut and sanded surface, so it is acceptable to move the otolith in a back and forth, or in a circular motion in order to facilitate polishing. Be sure to continuously apply water. After about five to ten seconds, examine the cut surface of the otolith. If you can see reflected light, from the window or a light bulb, flash evenly and uniformly off the cut surface, your polishing is complete. The otolith half is now ready to for gluing to a glass slide.

Figure 6.2.13 UV light tray

Step 6: Gluing the Otolith onto the Slide: The otoliths will be glued to their respective slide, with the use of UV-154, ultraviolet sensitive adhesive. Turn on the UV light (to let it warm up). Clean and dry off the cut and polished cross section, using a Kim Wipe, or some suitable tissue or cloth. Also, double check that the slide is clean.

Place the slide on the table with the label end pointing toward you. Apply a thin line (about as long as the otolith is wide) of the UV glue to the outer edge of the slide opposite the label. Next place the half otolith onto the slide, making sure that the concave side (the side with the sulcus) is facing the outer edge of the slide, away from you. If the otolith section has been prepared with a flat cross section that is perpendicular to the axis of the sulcus, then it should stand easily on its cut end. Continue this step with all the sectioned otoliths. Place each slide on to the UV light tray (See Figure 6.2.13). Check to see that none of your otoliths have fallen over. Place the tray under the UV light for approximate 30 minutes. This is all the time that UV glue needs to fix. Once the UV glue fixes, the otolith half is secure, and there is no way to remove it from the slide without breaking it. However, any excess UV glue around the edges may still be a little soft. You may remove excess glue in a later step. Once the UV glue has had time dry, the otolith is ready for the final cut.

If, by chance, an otolith has fallen over from its upright position on the slide after the first cut (contact a technician that has been successful in this task).

Step 7: Final Cut:

There are two steps for making the final cut. First, the cutting blade on the left side of the Hillquist saw removes most of the excess otolith, leaving only a thin section containing the otolith core. Then the grinding wheel on the right side of the Hillquist grinds the thin section down to its final thickness.

STOP: Wear both eye protection and earplugs before proceeding!

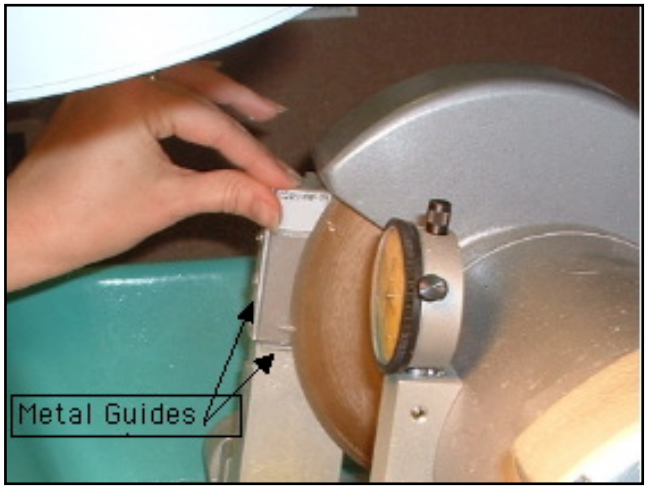


Figure 6.2.14. Making the final cut

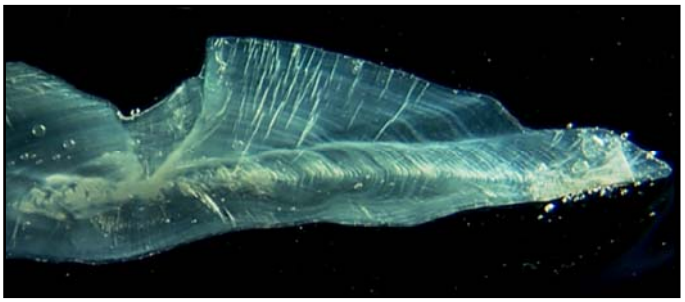


Figure 6.2.15 Fracture lines in otolith section caused by hasty cutting

Cutting off excess otolith - Turn on the saw as in Step 4. Make sure that there is a good flow of water on both cutting and grinding wheels. Place the slide on the left arm pad and guide. Make sure that the bottom of the slide and the side of the slide toward you is resting squarely on the metal guides (See Figure 6.2.14). Hold the upper edge of the slide and slowly move the arm towards the blade. Cut through the otolith with a slow and smooth movement. If you go too fast you could leave small fracture lines (See Figure 6.2.15) in the remaining thin section making later analysis difficult.

Grinding the thin section - Unlike the left cutting blade, the right grinding wheel is adjustable. This allows for discretion as to the final thickness of the thin section. First, check the calibration of the grinding wheel. Turn the adjustment knob, located on the left side of the saw just below the cutting blade, until the metal guide of the right arm pad is just touching the side of the grinding wheel. The dial indicator should read zero. If it does not, calibrate the saw before proceeding. If you are not familiar with this procedure, ask for help, or refer to the instructions that came with the Hillquist saw. Be aware that as we begin to process new species, we may calibrate the saw. In this instance, a sign is placed on the saw identifying that species. If you must change the calibration for a different species, affix a notice of the change to the saw. The saw will be re-calibrated once cutting for the previous species resumes.

Once you have calibrated the saw, adjust the thickness according the species. There are no absolute guidelines as to how thick your thin section should be. As a starting point adjust the thickness until the indicator dial reads + .45 (the indicator dial is graduated in .01mm increments). Next, place the slide in the right arm pad and guide. Make sure that the bottom of the slide and the surface of the slide furthest from you are resting squarely on the metal guides. Note that on the grinding side of the saw the metal guide is on the far side of the arm pad rather than the near side (See Figure 6.2.16). Hold the upper edge of the slide and slowly move the arm towards the blade at a steady and constant rate in order to avoid fracturing the thin section. After completing one or two slides, check the thickness of your thin sections by placing a drop of water on your section and viewing under a microscope. What you are looking for is good contrast between the opaque and translucent bands in your thin section. If you don't see a clear demarcation among bands, adjust the saw to make a slightly thinner grind. Continue this process until you get optimum thickness (it is a good idea to ask the advice of an experienced slide reader at this point). Once you have achieved the proper thickness, no further adjustment should be necessary. Continue cutting and grinding the remainder of the selected otoliths.

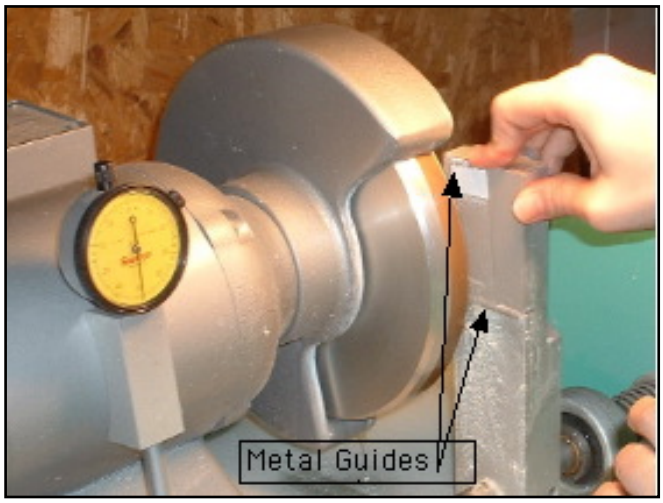


Figure 6.2.16. Adjusting the thickness of the section

Step 8: Final Sanding:

As with the first cut, you will have to sand and polish the grinding marks from the second cut to a smooth surface. Make sure the sandpaper is wet, using only distilled water. Grit of 400 is especially good. Hold the label end of the slide with your thumb

and outer fingers, leaving your index finger free to apply pressure directly over the middle of the thin section (See Figure 6.2.17). It is important to place the thin section of the otolith flat against the sanding wheel. Failure to do so will result in a thin section surface that slopes from left to right, or top to bottom, or both. Sand with a slightly circular motion for about five to ten seconds if the sandpaper is new, longer as the sand paper wears down. A good way to judge your progress is to look for the lines left by the grinding wheel.

Figure 6.2.17. Sanding the otolith slide

The grinding wheel will leave vertical lines on the surface of the cross section. The sanding wheel will sand away the vertical lines and replace them with a finer set of horizontal lines. Examine the cross section. Sanding is usually complete once horizontal lines have replaced all of the vertical lines unless the section is too thick.

Step 9: Final Polishing:

Final polishing removes those fine horizontal lines left from sanding. Turn on the polishing wheel, apply a little water and/or Micropolish to the spinning wheel, and reapply as needed. Hold the slide in the same manner you did for final sanding (See Figure 6.2.18). Try to hold the slide flat against the polishing wheel. There is little chance here of changing the slope of the thin section, but polishing will proceed quicker if the slide is held flat. After five to ten seconds, examine the surface of the thin section. If light reflects evenly off the surface, polishing is complete.

Figure 6.2.18. Polishing the otolith slide

Figure 6.2.19. Cleaning the excess glue from the otolith slide

Step 10 Cleaning the slide:

The process of cutting, grinding, sanding and polishing leaves various residues on the otolith section. You must remove these prior to sealing your otolith cross section. Excess UV glue, grit from the sand paper, and excess polishing compound can be cleaned off using Isopropyl alcohol and a Kim Wipe. Dip the slide into a beaker of alcohol, and with a Kim Wipe, wipe the area of the cross section using a circular motion to remove the debris that has accumulated along the edge of the cross section. Make sure the Kim Wipe is wet with alcohol, as using a dry Kim Wipe will only tend to smear the residue across the surface of the otolith cross section and leave bothersome fibers. Use the Bausch & Lomb microscope to examine the slide (See Figure 6.2.19). The surface of the cross section should be free of any residue that would obstruct viewing for aging.

NOTE: It is difficult to make the “perfect”.

Step 11: Sealing the otolith cross section:

Figure 6.2.20. Sealing the otolith section with Cytoseal

The final step in preparing an otolith for microscopic examination is to seal the thin section with a thin layer of Cytoseal. Cytoseal dries very quickly when exposed to air. For this reason, you must apply the Cytoseal smoothly and quickly. Putting a small amount of Cytoseal in a small glass vile will prevent rapid dry out, and facilitate application to the cross section. Dip a bent teasing needle into the small glass vile to collect one drop of Cytoseal. While viewing the otolith cross section under the microscope touch the drop of Cytoseal on the teasing needle to one end of the cross section (Figure 6.2.20). With a smooth motion of the needle, pull the drop of Cytoseal from one end of the otolith cross section to the other. If any air bubbles are trapped over the surface of the cross section, quickly try to push them aside with the teasing needle. Place each slide back in a new, clean Styrofoam tray once sealed. When finished, cover the tray to prevent dust particles from settling in the Cytoseal, and set it aside to dry overnight (or a minimum of three hours). The otolith cross sections are ready for microscopic examination.

Following the steps as outlined above will generally produce high quality preparations suitable for aging and/or for other studies requiring precision measurements. However, specific techniques can and do vary from one technician to another. Many technicians prefer to group several steps together. For example, a technician may chose to make a first cut, sand, polish and glue one otolith before moving to the next otolith. This is a perfectly acceptable procedure. Technicians should feel free to adjust their technique to whatever is most comfortable for them, as long as all of the preceding steps are including in their procedure and that a high quality preparation suitable for detailed study is the ultimate goal.

6.3 Trouble-shooting for Sectioning Otoliths on Hillquists

The Panama City laboratory uses otoliths to study the age structure of fish. This study begins where your job of preparing otolith slides for microscopic examination leaves off. Understanding the job of the analysts and slide-readers, and what they look for, will help you to turn out a better final product. Some of the more common problems encountered in slide preparation have already been alluded to in section 6.2. In this section, we will give further examples of these problems, and offer some suggestions for avoiding them. First, a list of common preparation problems:

1. Off center (missing the core)
2. Thickness (too thick, or too thin)
3. Sloped surface
4. Broken surface/edges
5. Fractured surfaces
6. Air bubbles obscuring the surface
7. Residue (glue, grit, polishing compound, etc.)

Now, let us look at some examples:

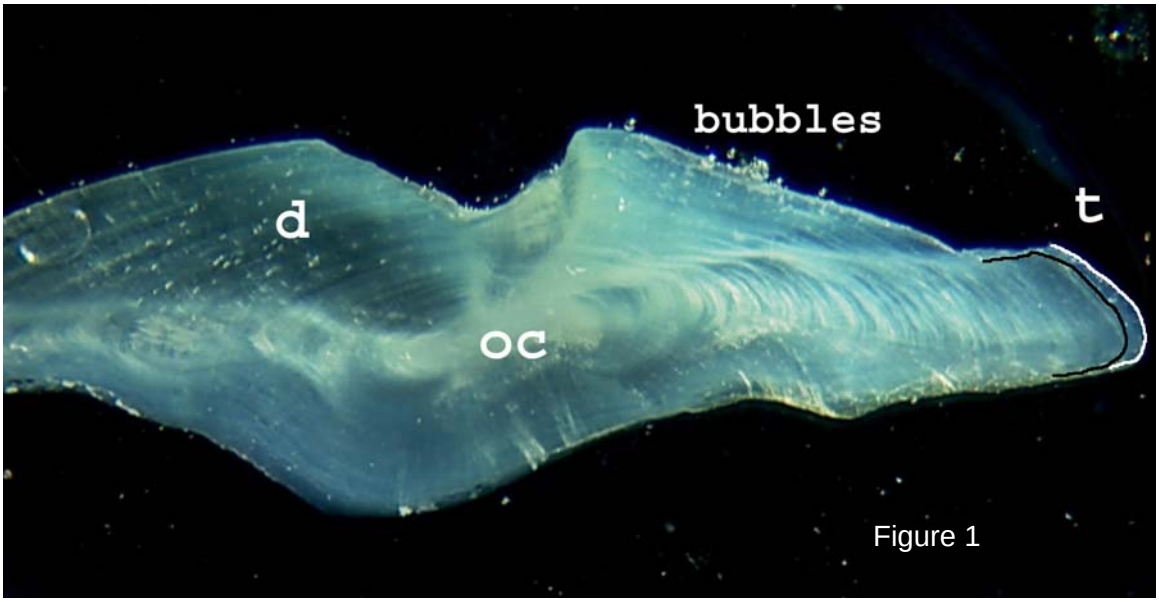


Figure 1

In this example (Figure 1), there are multiple problems. It is "off-center" (oc), polishing dust (d) remains on the surface, and Cytoseal has bubbles along the proximal dorsal edge. Also, the edge thickness is pronounced (t) because of the off-center cut making the edge type difficult to determine. We will address these problems one at a time.

1. Off Center - notice the characteristic tornado shape of the sulcus. As explained in section 6.2, this indicates that the first cut and grind (Step 4), stopped short of reaching the core of the otolith.

   Solution - The transition between the tornado and the perfect "V" can happen very quickly. Grind down in small increments, and examine the cut surface frequently. As you approach the core, the sides of the tornado will straighten out. You may find it helpful to use a hand lens or microscope until you gain experience.

2. Polishing Dust - Polishing dust, as well as debris from sanding and excess glue, can obscure the surface of the otolith, making it difficult to read and measure.

   Solution - In addition to following the procedures outlined in section 4.1, Step 10, use clean alcohol. Residue from the slides will accumulate in your beaker of alcohol. After a time the alcohol may become so contaminated with residue that it could contribute to the problem rather than solve it. If the otolith has already been sealed with Cytoseal, you will need to repeat sanding and polishing in section 4.1 Steps 8 through 1.

3. Air Bubbles - There are two potential sources of air bubbles; the UV glue used to glue the otolith to the slide, and the Cytoseal used to seal the polished surface of the otolith thin section. Both can obscure critical features of the otolith, detracting from its usefulness.

   Solution - After applying a line of UV glue to the end of the slide, look for bubbles in the glue. In the slow drying UV glue these bubbles can easily be removed with a teasing needle. When placing the otolith half on the line of glue, place it so that the outside edge (the sulcus edge) touches the glue first; then, pivot it forward until the cut edge is flat on the surface of the slide. This will force any bubbles out from under the otolith. Eliminating bubbles from the Cytoseal is a more difficult problem. Cytoseal becomes viscous very rapidly when exposed to air. Follow the procedures outlined in Step 11 to minimize air exposure to the Cytoseal. If the bubbles persist, or the Cytoseal becomes too viscous to work with, use the sanding wheel to remove the Cytoseal, re-polish, and try again.

4. Thick Cross Section - As Figure 1 clearly shows, the thickness of a cross section that is slightly off center can easily be mistaken for additional surface area, and subsequently lead to erroneous edge determination, and /or faulty measurements.

   Solution - Make sure to calibrate the grinding side of the Hillquist properly. Examine the first few slides under a microscope to assure good contrast between opaque and translucent zones, and to look for the type of edge problems shown in Figure 1. When in doubt, ask for help!

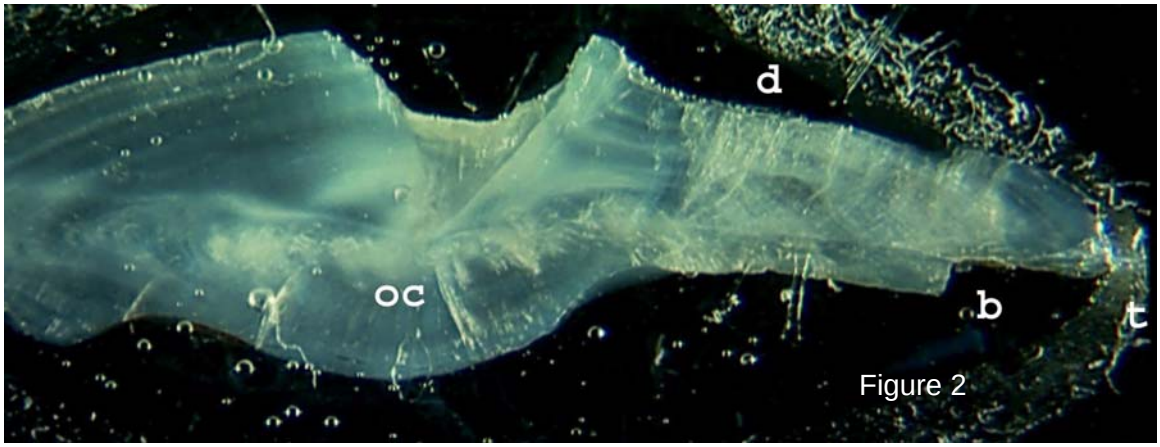


Figure 2

The example in Figure 2 also has multiple problems. It is "off-center"(oc) exhibiting the "tornado" effect, the Cytoseal obscures the dorsal tip (t), and the proximal and distal dorsal edges appear to be broken. In addition, there are numerous stress cracks and unpolished areas obscuring the central area on the dorsal side (d).

5. Broken Otolith - In some cases this problem is beyond your control. Otoliths are easily chipped or broken during removal from fish, shipping, or handling. Once in the lab, a few simple precautions may reduce breakage.

Solution - Depending upon the fish species, otoliths can be very robust, or very fragile. Know what species you are working with and take the appropriate precautions to mitigate breakage. Do not force the otolith through the first cut and grind (Step 4). Let the Hillquist do the work. Applying too much pressure during sanding, and to a lesser extent, polishing, can result in chipped and broken thin sections.

6. Fractures - Fractures can sometimes get so bad that the cross section becomes useless. There is a conundrum here in that while "thin" preparations are usually easier to read, they fracture more easily.

Solution - Following the same suggestions for broken otoliths can also help reduce fracturing. Additionally, during Step 7 (Final Cut) you should look for good contrast between the opaque and translucent bands in your thin section. If the bands tend to blend in with one another, adjust the saw to make a slightly thinner grind. On the other hand, if fracture lines begin to appear, you have gone *too* thin. The balance between thick and thin will vary from species to species. When working with an unfamiliar species, get the advice of an experienced person.

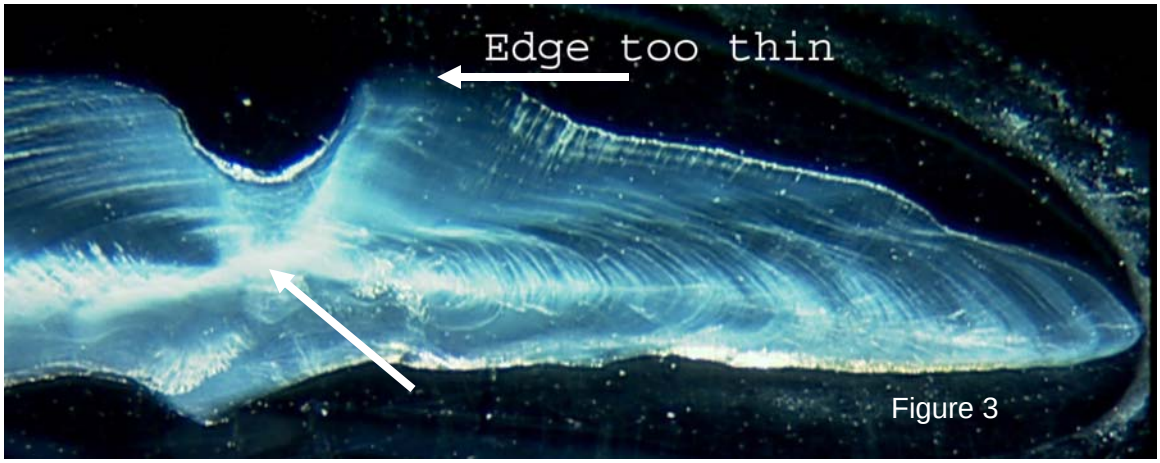


Figure 3

In this example (Figure 3), the otolith has been tilted in sanding, and as a result, the proximal edge has been thinned to the point of being unreadable. In addition, the cut is "off-center" (arrow) so that the first year is ambiguous.

7. Plane of the Cross Section not Perpendicular to the Axis of the Sulcus - Figure 3 is a good example of what can result during final sanding (Step 8) if slide is not held flat to the surface of the sanding wheel.

   Solution - Make a concerted effort to hold the slide flat. Using finer grain sand paper (1200 grit, for example) will slow down the sanding process and allow you more time to adjust the manner in which you hold the slide. The finer grit will also make the job of polishing much easier.

Study the following Figures 4 through 8 for more examples of the problems cited above. Keeping attuned to these potential problems will help you avoid them.

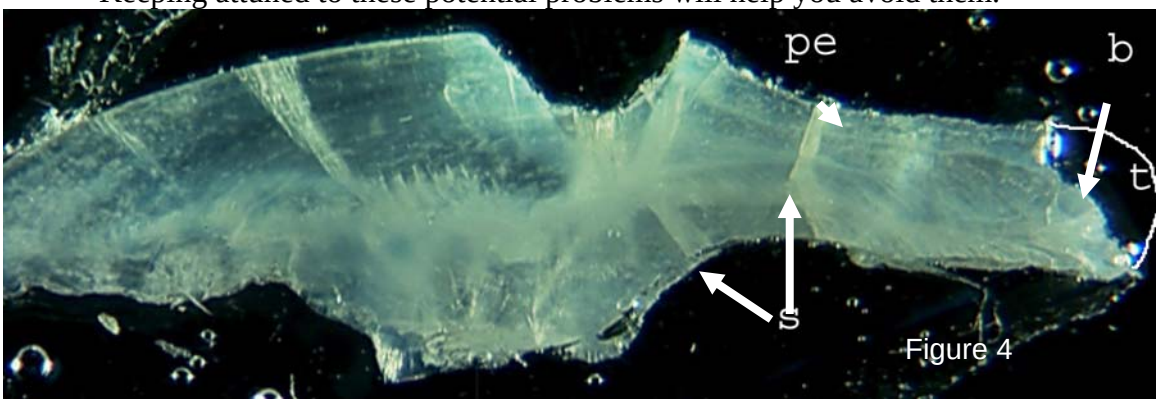


Figure 4

Here the otolith is broken (b) and contains numerous stress cracks (s). There could be another ring concealed along the proximal dorsal edge (pe) or emerging on the missing dorsal tip (t).

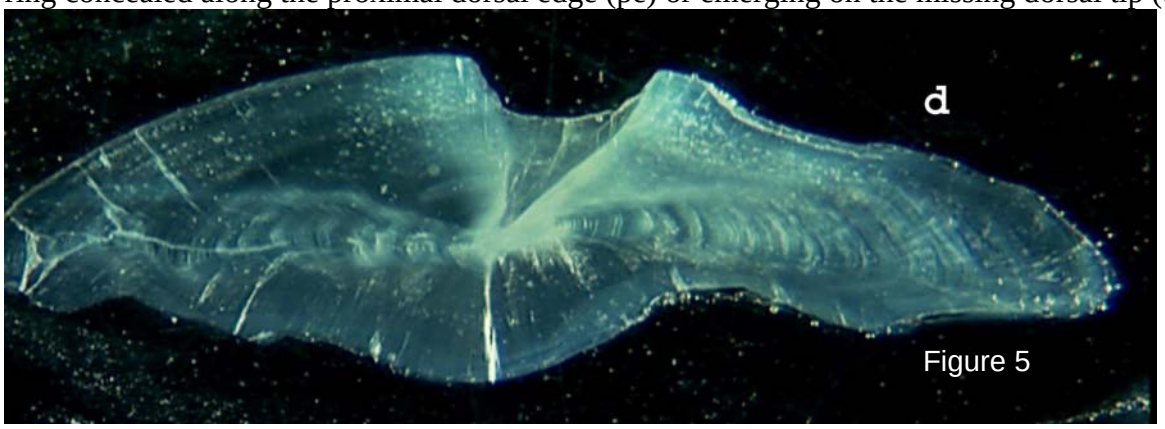


Figure 5

Besides being "off center" this otolith has polishing dust, (d) obscuring rings 2 & 3.

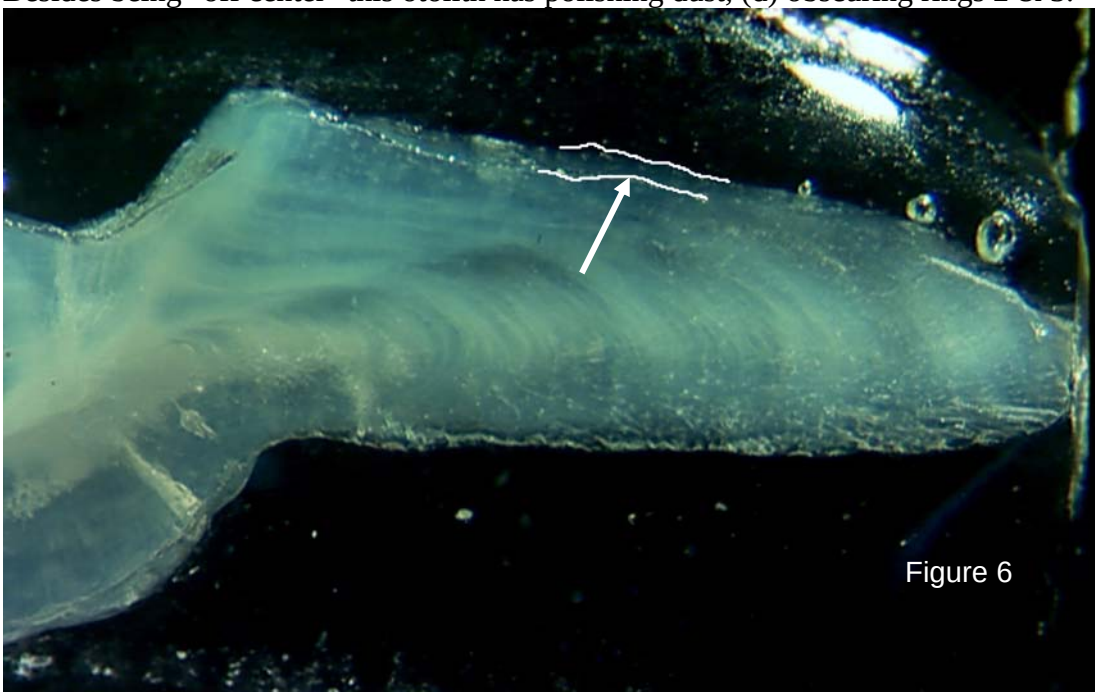
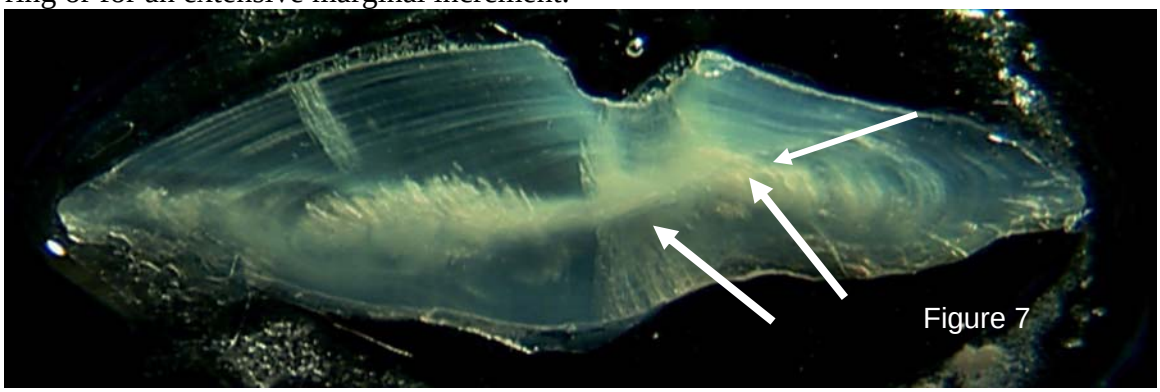
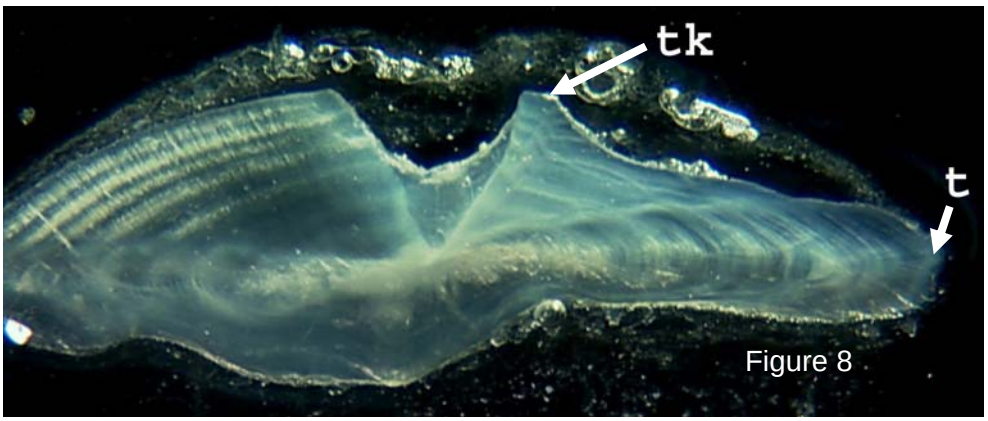


Figure 6

This otolith section is too thick (arrow). The upper surface edge may be mistaken for another ring or for an extensive marginal increment.

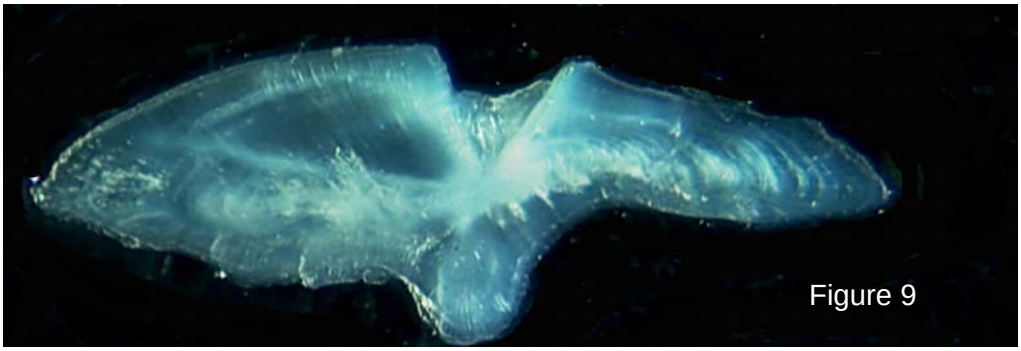


"Off-center", Core and 1st annulus are uncertain (arrows).



The tip of the dorsal side of the sulcus (tk) has been truncated in preparation.
Thus, the marginal increment as read along this axis will miss the new ring forming at the dorsal tip (t) end. See arrows.

Now that you know what an otolith preparation *should not* look like, here is an example of what one *should* look like.



This is a well-prepared otolith sample. The cut is exactly through and perpendicular to the core. The marginal increment is clearly discernable. It is free of dust and stress cracks, and both ends of the otolith are available for interpretation. With a little practice and experience, otolith preparations like the one in Figure 9 will be the rule rather than the exception.

6.4 Steps to Sectioning Otoliths on Buehler Isomet Slow-Speed Saws

This section will use mackerel otoliths as an example species to section on the Buehler Isomet slow-speed saw. Young mackerel are better read "whole". Generally, these fish are below a certain fork length. Therefore, the following is a guideline for otoliths that an accurate age from a whole otolith is possible.

Step 1: Determine which otoliths need to be sectioned

- King Mackerel (KMK):

    1. Determine Season
        - Atlantic Ocean: **April 1 - March 31**
        - Gulf of Mexico: **July 1 - June 30**

    2. Open sub-sampling query and set criteria:
        - Date: dates for specific season
        - PC Collection #: Set criteria according to sub-sample list
        - Fish #
        - Fork Length: Set criteria as

          Females ≥ 900mm
          Males ≥ 800mm

- Spanish Mackerel (SMK**):**

    1. Determine Season
        - Atlantic Ocean and Gulf of Mexico

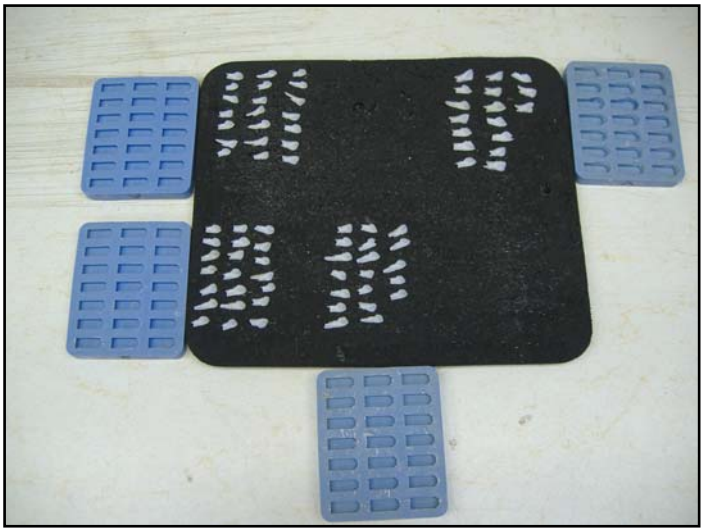
          April 1 – March 31

    2. Open sub-sampling query and set criteria:
        - Date: dates for specific season
        - PC Collection #: Set criteria according to sub-sample list
        - Fish #
        - Fork Length: Set criteria as

          Females ≥ 550mm
          Males ≥ 450mm

Step 2: Casting molds

Arrange molds (regular sized and enlarged) around pad.



BE SURE THAT THIS SETUP IS NOT AT RISK FOR BEING BUMPED BY ANYTHING OR ANYONE AS THESE OTOLITHS ARE NOT WEIGHED AND THEIR IDENTITY CANNOT BE DETERMINED IF MIXED UP!

Figure 6.4.1 Otolith molds and pad arrangement with otoliths positioned next to respective molds for embedding.

1. Record the mold #'s on the Molding Grid Worksheet.

Mold #	X1	
111-239		
112-726		
114-831		

Figure 6.4.2 Molding Grid

2. Carefully remove otoliths from vials using forceps and determine into which mold the otolith will be cast (best fitx). Some otoliths are wider at the posterior end and will require the larged mold to lay flat inside the grooves of the mold.

3. Place otoliths onto the black pad next to the mold, recording the Collection # and Fish # on the Molding Grid Worksheet in the appropriate Mold Grid and Box (See Figures 6.4.1- 6.4.2).
4. Into the designated container, dispense transparent epoxy, taking care that liquid from each tube dispenses in equal amounts. Using a metal spatula, **gently** stir the epoxy until it is a uniform color. Rapid mixing creates air bubbles!

5. Fill each groove of mold approximately ½ full with epoxy. Let this stand for at least ten (10) minutes.

6. Using forceps, place otoliths into ½ filled molds. Be sure to place the posterior (See Figure 6.4.4) end as far over to the beveled end as possible. Be careful not to press down on the otolith, but be sure that to insert otolith fully into the groove.

7. Finish filling the grooves with epoxy, leaving no air pockets. Look for a "glossy sheen" atop all otoliths to ensure coverage by the epoxy. The epoxy should be flush with the top of the groove and covering the entire otolith (See Figure 6.4.3). DO NOT allow the epoxy to overflow over out of the groove creating a rounded surface.

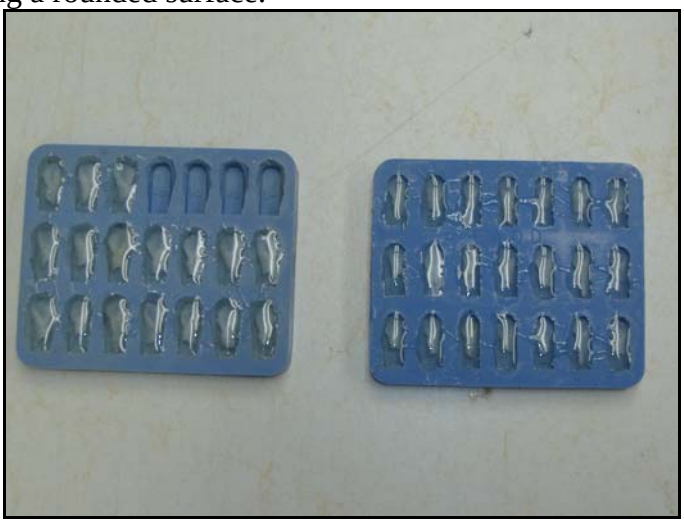


Figure 6.4.3 Otoliths embedded in epoxy within molds

8. Allow molds to air dry for at least 24 hours.

9. Once set (they may be slightly tacky), mark the core (Figure 6.4.4) for each otolith with a fine-tipped Sharpie marker.

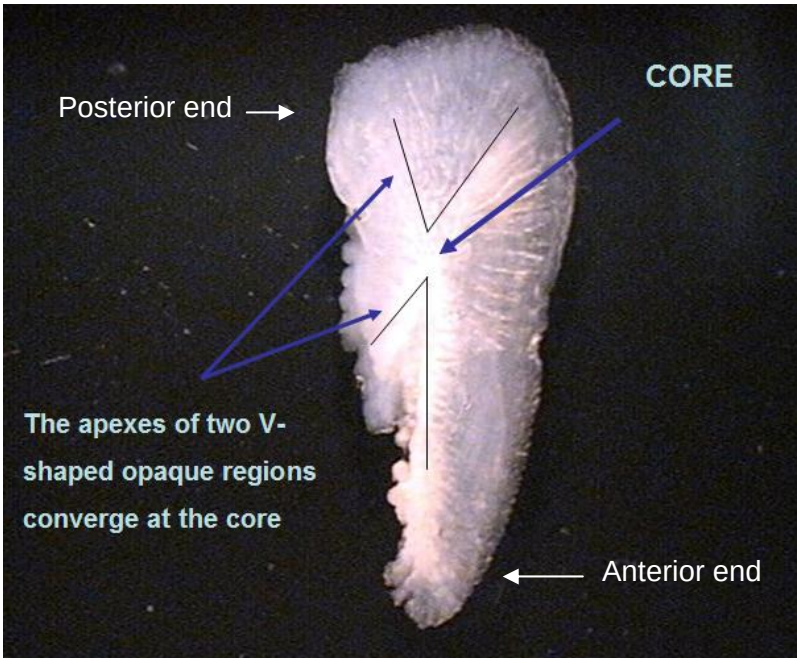


Figure 6.4.4 Mackerel core

10. Transfer molded otoliths into cell trays. Using a Sharpie marker, write the Collection # and Fish # corresponding for each otolith on the top of each cell of each tray (See Figure 6.4.5). Secure the tray and its cover with a rubberband to prevent spillage.

Figure 6.4.5 Embedded otolith labeled and contained with cell tray

Step 3: Sectioning with Isomets

IMPORTANT: The blades are VERY expensive, so: never allow the arm to slam down onto the blade whilst holding an otolith. It will break the $100 blade. Always prop the arm up on the sponges when not in use, to prevent blade breakage. Always use two hands to turn the dial, etc, to prevent blade breakage.

1. Insert the mold into the chuck rostrum end first. Make sure that the section and the blade are as close to perpendicular as possible (Figure 6.4.6). Be sure to keep track of which otolith is being cut by writing the fish number on a small piece of paper and placing it on the saw. Tighten chuck with hex wrench. NOT TOO tight! The epoxy will melt and the otolith will slip out of the chuck, breaking the $100 blade!

2. Make sure that the mold is not too far ahead or to far behind the blade. Adjust the angle of the chuck if necessary.

3. Lift the arm and line the blade up with the small ink mark on the mold by rotating the dial

4. If the dial reads between 0 and 15, turn the dial towards you, pass 0 and stop on 0 again.

5. If the dial reads between 16 and 24 then turn the dial towards you and stop on zero.

6. Make the first cut. Discard the posterior end after the first cut. Adjust the auto-stop dial to stop the saw automatically for the sections.

Figure 6.4.6. King mackerel embedded otolith mounted for cutting on isomet saw

7. Lift the arm and turn the dial away from you, stopping on 20. Make the second cut. This will produce the first section. The section will either hang up between the blade and the mold or end up on the blade. Remove the section with forceps and dip into fresh water. Rub the section gently between your fingers and place in the appropriate cell.

8. Lift the arm and turn the dial away from you to 15 and make the third cut to produce the second section.

9. Lift the arm and turn the dial away from you to 10 and make the forth cut to produce the third section.

10. Discard the remaining part of the mold.

Step 4: Making Slides

1. Perform this portion of the processing under a chemical/exhaust hood, as a noxious compound will be used to mount the otoliths to the slides.

2. Place a corresponding number of slides onto slide trays. Using a fine-tipped Sharpie, label the frosted side as follows:

	For example:
Season:	04-05
Species:	KMK
Coll/Fish#	06KMK123-1

PC Coll # Fish #

3. Using forceps, place otolith sections onto black pad in proper orientation for slide mounting (See Figure 6.4.1). Be sure otoliths are placed with sulcal groove to the LEFT and the dorsal portion on TOP.

4. Place slides on dark paper. Put two (2) drops of Cytoseal in the center of each slide.

5. Using forceps, place the sections in the Cytoseal. Press the sections onto the slide with a probe to remove all air pockets. Be sure that the cores of each section are in a straight line. Cover the section with Cytoseal from the surrounding area (Figure 6.4.7).

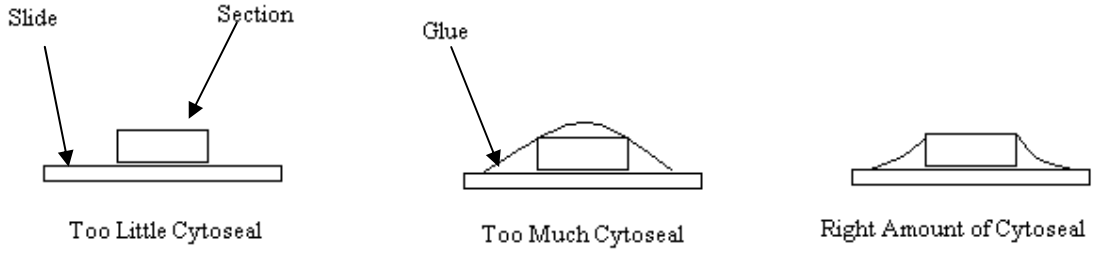


Figure 6.4.7. Correct amounts of Cytoseal/glue

6. Allow the slides to dry for at least 24 hours.

7. Put dry slides into appropriate slide boxes and label (see Chapter 5 for labeling slide boxes).

DO NOT STACK TRAYS! CYTOSEAL IS GLUE

Chapter 7: Processing Gonads Updated June 2008

Gonad samples have been sent here principally by two different sources: the Beaufort Head Boat Survey (HB), and the Trip Interview Program (TIP). Increasingly, samples are available from scientific surveys (MSLAB, PCLAB, and FMRI), at-sea observers (SBLOP and POP) and Cooperative Fishing Effort (CO-OP). This chapter describes the instructions for logging in gonad samples that are provided to the Panama City Laboratory by various sources, along with our own collection of gonads from local docks/fish houses.

This chapter also describes the procedures of initial assessment of reproductive tissues. We use the term "class" to describe gonadal development and "stage" for gamete development (see Chapter 9 on histology). Thus, the initial assessment includes both macroscopic classification (gonad observed with the naked eye) and microscopic staging of female oocytes using a tissue smear of oocytes observed under magnification. These initial (macro/micro) observations are often compared to final histological examination (as described in Chapter 9). Note that gamete staging for males is very difficult to do using a tissue smear. Reliable assessment of male gamete stages requires a histological preparation.

This chapter contains the following sections:

7.1 Logging in Gonads

7.2 Processing Sheet and Initial Examination of Fresh Gonads

7.3 Macroscopic and Microscopic Examination of Gonads
- 7.3.1 Procedures for Macroscopic Class Examination
- 7.3.2 Procedures for Measuring Max OD of Oocytes
- 7.3.3 Procedures for Microscopic Stage Examination
- 7.3.4 Procedures for Measuring Gonad Weight (g)

7.4 Primary Storage of Gonads

7.5 Treatment of Preserved Gonads Received From an Outside Lab Source

7.6 Historical Reproductive Codes

Contributors: Alan Collins, Bill Walling, Gary Fitzhugh, Carrie Fioramonti, Melissa Cook

7.1 Logging in Gonad Samples

Step 1 Locate original data sheets sent with gonads. If otoliths are to be received at a later date (usual protocol), make a copy of the original data sheet and put into the "awaiting otoliths" file in age, growth, reproduction lab.

Step 2 Using the Species Log-In Notebooks, log in each sample. If there is more than one species listed on a data sheet, make a copy of the original data sheet (provided by the port agent) for each species. Each species will receive its own unique PC Coll #.

Please refer to Chapter 1 for the appropriate procedures for logging in samples. Make sure all information is provided accurately and that all data fields are completed.

List of Log Data Fields

PC Coll #	Panama City Laboratory collection number (species specific)	
Date	Date of collection from sampler (mo/da/yr)	
Source	CO-OP	Cooperative Fishing Effort
	FMRI:	Florida Marine Research Institute
	HB:	Beaufort Head Boat
	MSLAB:	Pascagoula Cruise
	PCLAB:	Panama City Laboratory
	POP:	Pelagic Observer Program
	SBLOP:	Shark Bottom Longline Observer Program
	TIP:	Trip Interview Program
Source #	CO-OP	First 4 digits = 4 digit year – Next 2 digits = first and last initial of captain – Last 2 digits = Trip #
	FMRI:	First 2 or 3 digits = boat name abbreviation – Next 2 or 3 digits = Cruise # – Last 2 or 3 digits = Station #
	HB:	5 digit Collection number
	MSLAB:	First 2 or 3 digits = boat name abbreviation – Next 2 or 3 digits = Cruise # – Last 2 or 3 digits = Station #
	PCLAB:	For samples prior to 1999, see PC Lab section For samples 1999 to present: Leave blank, unless Trap Survey or CI Project:

Trap Survey:
First 2 digits = TS
Next 2 digits = 2 digit year (06, 07, 08)
Last 3 digits = Trap Survey #
CI Project
First 2 digits = CI
Next 2 digits – 2 digit year (06, 07, 08)
Last 3 digits = CI Station #

	POP	Trip ID #
	SBLOP	Observer initials – Trip # – Haul #
	TIP:	For samples prior to 2004, see TIP section For samples 2004 to present: First 2 digits = agency # Next 2 digits = State Last 6 digits = Intercept #
State	AL	Alabama
	FL	West Coast Florida
	E FL	East Coast Florida
	LA	Louisiana
	MS	Mississippi
	TX	Texas

*NWF, WF, SWF, SF, SEF, EF, NEF (Florida)
*For Mackerel Samples ONLY (Figure 1.2).

Location	City/County samples were landed (see list of zip codes Chapter 1)
Collector	First Initial and Last Name of sampler
# of Gonads	Total number of gonads in the collection
# of Otoliths	Total number of otoliths in the collection (If no otoliths, write 'Awaiting Otoliths' in the Log Comments)
Condition (S/W)	Condition of the otolith S = Sectioned or W = Whole (leave blank if not present)

Mode	CM	Commercial
	CP	Charter Party
	HB	Head Boat
	PR	Private
	TRN	Tournament
	SS	Scientific Survey

Gear	HL	Hook & Line, Hand-line, Bandit
	LL	Long-line
	SP	Spearfish
	TR	Trap
	TRW	Trawl
	GN	Gillnet, Trammel Net

Comments Any other information not listed above
For example:

If otoliths did not accompany gonad samples, write ‘Awaiting Otoliths’ in comments.

If the number of otoliths and gonads does not equal, state the fish # that does not have a gonadal sample in the comments.

If the gonads were fixed in formalin at sea, write ‘Gonads processed at sea’ in comments.

If the gonads were selected or the sample collection was somehow brought to the attention of the port agent rather than as a random sample, indicate ‘gonad selected’ or ‘collection non-randomly identified’ in comments. For example, someone (usually a fisherman) requests the sex, age or information about a fish or collection of fish from a port agent (see bias type below).

7.2 Processing Sheet and Initial Examination of Fresh Gonads

This section will describe the initial classes/stages of gonad tissue and how to fill out the Gonad Processing Sheet (Figure 7.2.2). This form was updated in 2008 (Figure 7.2.3).

At this time, gather the following materials:

- Data sheet (sent with gonads from port agent)
- Gonad Processing Data Sheet and Key

Step 1
Fill out the Gonad Processing Data Sheet as completely as possible for each collection before processing the gonads.

From the data sheet provided by the port agent, fill out these blanks:

Species	Abbreviation of species being sampled, see Chapter 1 for appropriate abbreviations.	
Date	Date the sample was landed	
Source	CO-OP	Cooperative Fishing Effort
	HB:	Beaufort Head Boat
	TIP:	Trip Interview Program
	PC LAB:	Panama City Laboratory
	MS LAB:	Pascagoula Cruise
	FMRI:	Florida Marine Research Institute
	SBLOP:	Shark Bottom Longline Observer Program
Source #	CO-OP	4 digit year –Sampler first & Last Initial – Trip #
	HB:	5 digit Collection number
	TIP:	1st 2 digits = state #
		Next 2 digits = Interview #
		Last 5 digits = Intercept #
	PCLAB:	Leave blank
	MS LAB:	Boat Initials – Cruise # - Station #
		For Boat Initials see Chapter 2.6
	FMRI:	No Source # is given
	SBLOP	Observer initials – Trip # – Haul #
Collector	First initial and last name of sampler	
State	State where sample was landed	
Location	City or county or port where samples were landed	

Latitude	Latitude where sample was caught at sea
Longitude	Longitude where sample was caught at sea
Bottom Temp (^{o}C)	Bottom sea water temperature (^{o}C)

May be recorded as an abbreviation (i.e. 42884) or a range on data sheet. Transcribe this EXACTLY.

Depth (m)	Depth, if range less than 5 fathoms take average
Start Depth (m)	Depth, if range greater than 5 fathoms enter minimum
End Depth (m)	Depth, if range greater than 5 fathoms enter maximum
Time Caught	An approximate time of fish capture. If a range of time is given, take average, using 24 hour clock. For example, 1:30pm would be recorded as 13.30

Mode

CM	Commercial
CP	Charter Boat Party
HB	Headboat
PR	Private
TRN	Tournament
SS	Scientific Survey

Gear

HL	Handline/ Bandit/ hook and line/ Troll
LL	Long line
TR	Trap
SP	Spearfish
GN	Gillnet, Trammel net
TRW	Trawl

Processed By — Initials of Panama City personnel who processed sample

Process Date — Date the sample was processed by Panama City Lab

Preservation Type — How the gonad samples were preserved on arrival to lab:

1. Ice, then premixed formalin
2. 10% premixed formalin ASAP aboard ship
3. Ice, temporarily in 37% formalin and then changed to 10% formalin
4. Ice, 10% mixed formalin (dilution of 37% formalin by Hunter's formula by PC Lab)
5. 50% mixed formalin ASAP aboard ship
6. At sea: ½ of gonad in formalin ASAP, then 20% sections of gonad from ice to formalin in 12 or 24 hour increments
7. Frozen first

Collection Comment	Any additional comments processor has. For example, gonad preserved at sea, or otolith missing
Fish #	Individual fish number supplied by sampler
TL	Total Length (mm)
FL	Fork Length (mm)
Whole Wt (rounded)	Weight (kg) of whole fish (RK)
Gutted Wt	Weight (kg) of gutted fish (GK)
Sex	F Female M Male I Indeterminate/Immature T Transitional stage (note: this would be difficult to discern with macroscopic staging—more of a guess)
Samples Taken	Check the appropriate box: otoliths, gonads or both
Bias Type R/S	Sampler will indicate whether individual samples were randomly sampled (R) or selected (S).

These may be recorded as cm or inches (FC/FI, TC/TI).

These may be recorded as pounds or grams (RP/RG, GP, and GG)

The following data will be recorded as the gonads are processed in Steps 2-7.

Macro Class	Assign appropriate class (1-5, see section 7.3).
Max OD	Record maximum diameter in omu of oocyte in sample
Micro Stage	Assign appropriate class, ovaries only (PG, CA, V, H) (See section 7.3.3)
Gonad Wgt	Weight of gonad to the nearest 0.1 g
% Gonad	Estimate % gonad (in whole %) of gonad provided
Kept (Y/N)	Indicates whether or not that particular gonad sample was kept or was discarded. Yes = Gonad was kept No = Gonad Discarded
Gonad Comment	Comments pertaining to individual fish gonads

Histo #	Leave blank. Assigned during blocking of gonads (See Chapter 8)
Total	Total number of otoliths and/or gonads in the collection

Gonad bags should be removed from the shipping container and put into a leak-proof tray. Best practice is to perform Steps 2-8 on one gonad before examination of the next gonad. This reduces damage to the gonads from mechanical and temperature exposure. All examinations should be performed within the Age, Growth, and Reproduction laboratory.

Step 2 Match the tag #s in the gonad bags with the original data sheet and the tag #s on the Gonad Processing Data Sheet.

Step 3 Assign a Macroscopic Class to each gonad. This examination is made with the naked eye. Section 7.3 describes each macroscopic class for both females and males. Repeat this step with each gonad. Always assume that macroscopic class is based upon fresh gonads unless otherwise noted in the comments. See section 7.5 for treatment of fixed gonads but classes and stages would remain the same and section 7.3, Macroscopic and Microscopic Examination of Gonads.

Step 4 Prepare a tissue smear of the gonad, ONLY if it is an ovary**.**
For protogynous hermaphrodites, take the sample from either the C or F lobe (Figure 2.1), as this region is typically where transition from female to male occurs. For gonochoristic species, using a scalpel, cut through the tunica of the gonad in a random region of the gonad using a six-sided die to randomly select the gonad location in which the sample will be taken. The locations are A-F (1-6, respectively for die rolling purposes), with locations A and D marking the right and left anterior ends. Locations C and F, the right and left posterior ends, usually contain the connection between the lobes. The top of the gonad will usually have connective tissue attached and the bottom of the gonad will not have any connective tissue.

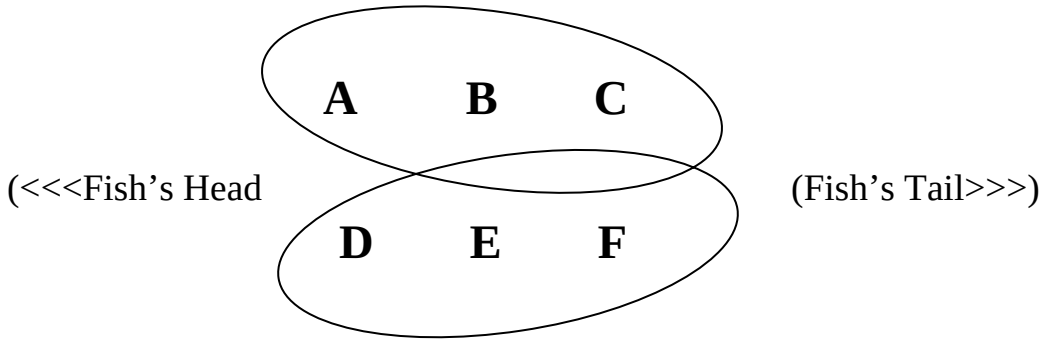


Figure 7.2.1 Partitioned sections of gonad in relation to the position of the gonad in the body cavity of a fish

Place the tissue smear into a clean Petri dish with a small amount of water. Put the Petri dish under the dissecting scope and immediately proceed to Step 5. Exposure to the light/heat of the microscope will affect the oocyte diameter measurement.

Step 5 Measure and record Max OD. Take the maximum oocyte diameter from the largest oocytes in the ovary sample, calculated in ocular micrometer units (=OMU's) not millimeters (1 OMU = 0.04 mm), using Heerbrugg WILD Model # M5-46824 dissecting microscope with micrometer in 10X ocular and microscope magnification set at 25X (see 7.3.2 Procedures for Measuring Max OD of Oocytes).

Step 6 Assign a Microscopic Stage to each gonad. This examination is made with the dissecting microscope (see above description). Section 7.2.3 describes, in detail, each microscopic stage for females. Repeat this step with each gonad. We always assume that microscopic stage is based upon fresh gonads unless otherwise noted in the comments. See section 7.4 for treatment of fixed gonads but classes and stages would remain the same. (see 7.3.3 Procedures for Microscopic Stage Examination).

Step 7 Estimate % gonad (in whole %) of gonad provided

Step 8 Gonad Wt (g) Weight of gonad to the nearest 0.01 g
(see 7.3.4 Procedures for Measuring Gonad Weight)

Step 9 Prepare gonads for long-term storage, see Section 7.4

Figure 7.2.2 Gonad Processing Data Sheet (prior to 2008)

Gonad Processing Data Sheet

Species: _____
PC Coll #: _____

Date: _____/_____/_____ Source: _____ Source #: _____
Collector: _____ State: _____ Location: _____

Latitude: _____ Longitude: _____ Bottom Temp (^{o}C): ____ Depth (m): _____
Start Depth (m): _____ End Depth (m): _____ Time Caught : _____

Mode: CM CP HB PR TRN SS Gear: HL LL TR SP GN

Processed By: _____ Process Date: ____/____/____ Preservation Type: _____
Comments: _____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	Sample Taken G	Bias Type R/S	Macro Stage	Max OD	Micro Stage	Gonad Cond.	% Go-nad	Gonad WT(g)	Re-marks	Kept Y/N	Histo #
					Total:												

Data Entered: **Initials: _____** **Date: _____**

After data has been entered into the appropriate annual database this data sheet is initialed and dated by the technician. This data sheet, along with a copy of the original datasheet is filed in the appropriate Reef Fish Reproductive Study Notebook, located in the Age, Growth, and Reproduction Laboratory.

Figure 7.2.3 Gonad Processing Data Sheet (2008 to present)

Gonad Processing Data Sheet

Species: _____
PC Coll #: _____

Date: ____/____/_____ Source: _____ Source#: _____
Collector: _____ State: _____ Location: _____

Latitude: _____Longitude: _____ Bottom Temp (^{o}C): _____ Depth (m): _____
Start Depth (m): _____ End Depth (m): _____ Time Caught : _____

Mode: CM CP HB PR TRN SS Gear: HL LL TR SP GN

Processed By: _____ Process Date: ____/____/____ Preservation Type: _____
Collection Comments: _____

Fish #	TL (mm)	FL (mm)	Whole WT (kg)	Gutted WT (kg)	SEX	Sample Taken O	G	Bias Type R/S	Macro Class (gonad)	Max OD	Micro Stage (oocyte)	Gonad WT(g)	Kept Y/N	Histo #	Gonad Comment
				Total:											

Data Entered: **Initials: _____** **Date: _____**

After data has been entered into the appropriate annual database this data sheet is initialed and dated by the technician. This data sheet, along with a copy of the original datasheet is filed in the appropriate Reef Fish Reproductive Study Notebook

7.3 Macroscopic and Microscopic Examination of Gonads

7.3.1 Procedures for Macroscopic Class Examination

A. Female Macroscopic Class (description pertains to ovary)
Classified as one of the following:

1 Immature or regressed
2 Early maturing
3 Late maturing
4 Ripe
5 Spent

Reproduction of marine fishes studies by the SEFSC may be of two classifications: gonochorists or protogynous hermaphrodites. The gonochorists are fish that are born as one gender, either male or female, and remain that gender throughout their lifetime. Examples of gonochoristic fish studied by this laboratory include snappers and mackerels.

The protogynous hermaphrodites are fish that are born as females and change to males. Therefore, all immature fish of these species will be female. Examples of protogynous hermaphrodites studied by this laboratory include the majority of groupers and *Calamus* sp. of porgies.

For any ovary that upon examination cannot be categorized into classes 1-5, a note should be made regarding the unusual or unknown characteristics in the comments.

The following pages have give descriptions and characteristics for each macroscopic class. Use these as a guideline to assign macroscopic class.

1. Immature or resting (ribbon-like, no visible oocytes to naked eye)

Figure 7.3.1.1 Stage 1, immature or resting female gonad tissue

2. Early maturing - oocytes barely seen with naked eye (no Photo Provided).

3. Late maturing oocytes easily seen with naked eye

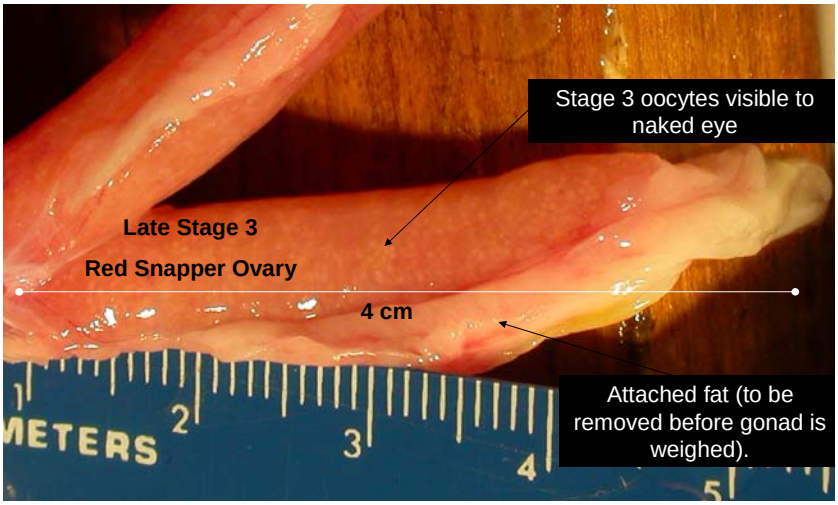


Figure 7.3.1.2 Stage 3, late maturing oocytes visible for gonochoristic fish.

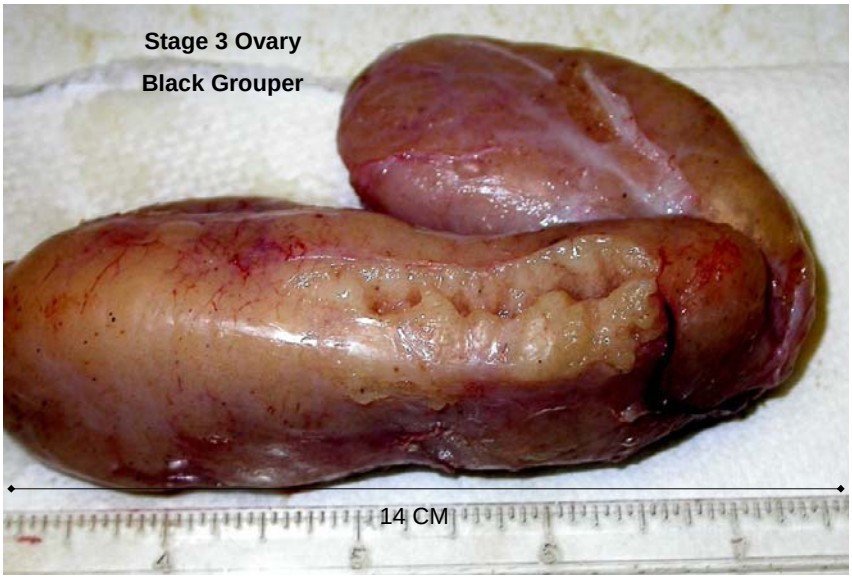


Figure 7.3.1.3 Stage 3, late maturing oocytes visible for hermaphroditic fish.

4. Ripe - some clear hydrated oocytes seen through tunicate

Figure 7.3.1.4 Stage 4, ripe clear hydrated oocytes visible through tunicate for gonochoristic fish.

Figure 7.3.1.5 Stage 5, ripe clear hydrated oocytes visible through tunicate for gonochoristic fish.

5. Spent - can be very red and flaccid, recently spawned, (no photo provided).

Table 7.3.1.1 Table of Female Macroscopic Class Description and Characteristics

Class	Description	Characteristics
1	Immature	Small and/or ribbon-like; Opaque and jellied Grayish color; No visible oocytes to naked eye
1	Regressed	Macroscopic distinctions between regressed (resting) and immature ovaries are imprecise but the regressed ovary is likely to more opaque and jellied and with a more reddish-grey cast than the immature ovary.
2	Early Maturing	Granulated; Small opaque oocytes emerge towards the end of the stage, barely seen with naked eye.
3	Late Maturing	Granulated; Oocytes easily seen with naked eye.
4	Ripe (hydrated)	Some clear oocytes (hydrated) seen through tunicate
5	Spent	Reddish; Flaccid

B. Male Macroscopic Class

Description pertains to testis (Brown-Peterson and Wyanski 2006[1])

Classified as one of the following:
1 Immature
2 Developing
3 Developing
4 Spawning
5 Spent

For any testis that upon examination cannot be categorized into classes 1-5, a note should be made regarding the unusual or unknown characteristics in the comments.

Table 7.3.1.2 Table of Male Macroscopic Class Descriptions and Characteristics

Class	Description	Characteristics
1	Inactive	Testis is small, then, ribbon like, often translucent or pink in color. No spermatozoa evident.
2	Developing	Testis small but firm, white and often triangular shaped. Spermatozoa not released when testis is cut.
3	Developing	Testis small, but slightly larger than class 2, firm, white and often triangular shaped. Spermatozoa not released when testis is cut.
4	Spawning	Testis is very thick and extrudes milt. Often maximum size for species; spermatozoa released when pressure applied to abdomen or when testis is cut.
5	Spent	Testis is elongated, flaccid and may be reddish, no spermatozoa released.

7.3.2 Procedures for Measuring Max OD of Oocytes

Maximum oocyte diameter is measured from the largest oocyte in the tissue smear and is calculated in ocular micrometer units (=OMU's) not millimeters (1 OMU = 0.04 mm) Using Heerbrugg WILD Model # M5-46824 dissecting microscope with micrometer in 10X ocular and microscope magnification set at 25X, place the tissue smear under the microscope. Remember DO NOT leave the dish under the light and microscope as it will cause the oocytes to swell. Looking into the ocular micrometer, measure several oocytes. Each hash mark on the micrometer is 1 omu. Record the Max OD for that sample.

[1] Brown-Peterson, N.J. and D.W. Wyanski. 2006. A proposed "straw man" reproductive classification for male teleosts. Pp 9-11 in: Proceedings of the 3rd Workshop on Gonadal Histology of Fishes, New Orleans, La., July 2006.

7.3.3 Procedures for Microscopic Stage Examination

Female Microscopic Stage (Description pertains to oocytes)
Microscopic stages are examined and assigned using the Heerbrugg WILD model # M5-46824 microscope with micrometer in 10X ocular and scope magnification set at 25X. (1 OMU = .04mm). Prior to 2007, Microscopic stages were numeric, 1-4, with Stage 1 corresponding to PG, Stage 2 corresponding to CA, and so forth.

PG **Primary growth**
Diameter up to ~4 omu, slightly opaque or clear, no nucleus or yolk present

CA **Cortical Alveolar**
Diameter up to ~ 8 omu, oocyte more opaque, with nucleus and/or yolk

V **Vitellogenesis**
Diameter up to ~20 omu, oocytes more opaque, mostly filled with yolk globules, may have thin, clear oocyte perimeter

H **Hydration or Spawning**
Diameter ~ 33 omu, oocytes much less opaque, may have oil globules, may be partially collapsed

If microscopic examination results in oocyte diameters up to ~20 omu with 'ragged' appearance of follicles and a lot of debris in ovary, this should aid the macroscopic classification of a spent ovary (formerly microscopic stage 5). For any sample that cannot be categorized into microscopic oocyte stages 1-4, a note should be made regarding the unusual or unknown characteristics in the comments.

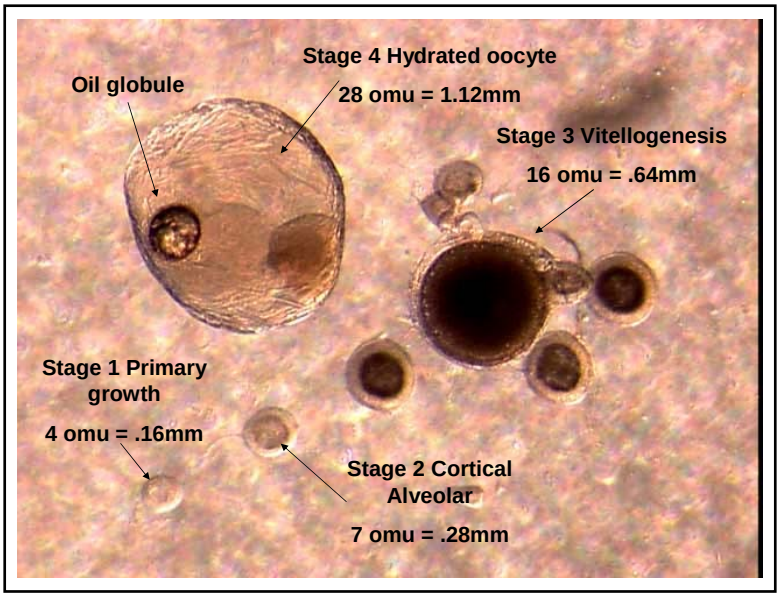


Figure 7.3.3.1 Microscopic Gonad Stages PG – H (1 – 4), see text for descriptions

7.3.4 Procedures for Measuring Gonad Weight (g)

Step 1 Trim excess fat or connective tissue from gonad. Most gonads are received with excess connective tissue and/or attached fat. This tissue and fat must be trimmed off as much as possible so an accurate gonad weight can be determined.

Figure 7.3.4.1 Gonad, connective, and fat tissue

Step 2 Blot dry any excess moisture off the gonad using paper towels.

Step 3 Tare weight boat on scale. Place gonad in weight boat on scale, and record weight of gonad in grams (g), to the nearest 0.01 g, on gonad processing sheet.

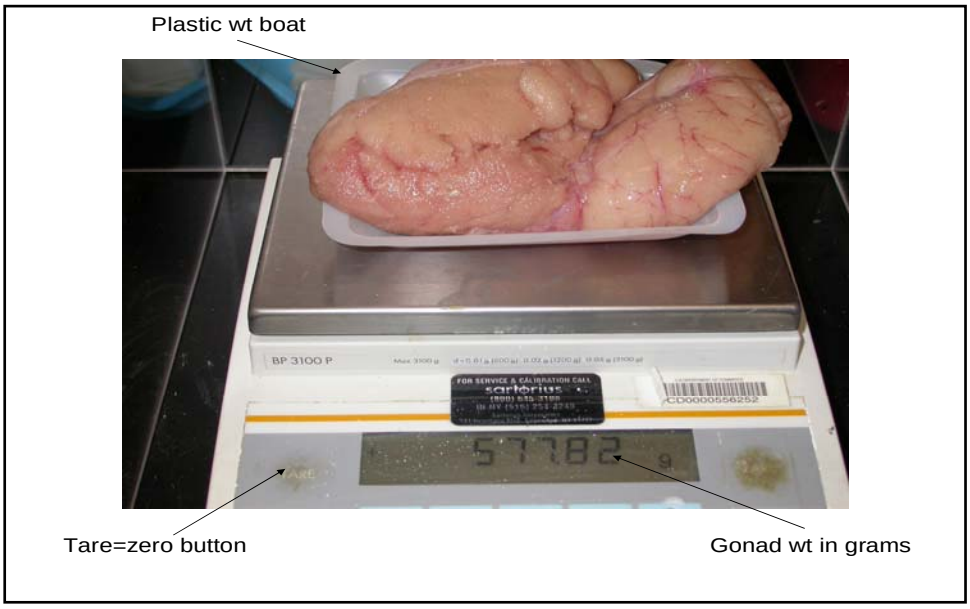


Figure 7.3.4.2 Image of entire gonad tissue being weighed

Step 4 After the examination and weighing of gonads they must be preserved and stored **immediately**, see Section 7.4 for Primary Gonad Storage.

Note: 10 % formalin is made by diluting 37-40 percent “formaldehyde”, the concentrated liquid form of formaldehyde, by 10X and so (10% formalin) is actually a 4 percent solution of formaldehyde. Since formaldehyde is actually a gas it is made useful for us by dissolving it in water; 37-40 percent formaldehyde in water is a saturated solution and is properly called “formalin” (any solution of formaldehyde in water is “formalin”).

7.4 Primary Storage of Gonads

Each collection of gonads will be placed into a gallon size zip-loc freezer bag with the species specific PC Collection Number, corresponding fish numbers, date, mode, gear, and location written in pencil on a water proof piece of paper and inserted into the gallon zip-loc freezer bag. This information should also be written on the outside of the zip-lock freezer bag (or Whirlpak) using only Sharpie Fine Point **Black** Marker as all other colors will rub off bag (Figure 7.4.1).

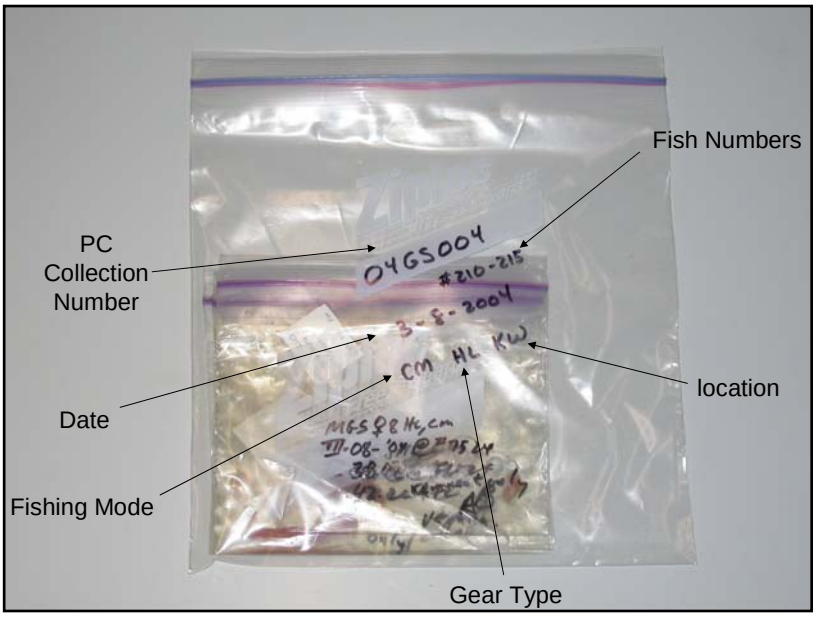


Figure 7.4.1 Example of labeled Ziploc bag for gonad long-term gonad storage

Gonads are preserved in 10% formalin for a minimum soak time of 14 days. This is to ensure that all of the gonad tissue is properly fixed in formalin. We use 10% neutral buffered formalin purchased from Sigma Aldrich (www.sigma-aldrich.com/order, #HT50-10640).

Add enough formalin to the Ziploc bag (or Whirlpak) to <u>completely immerse</u> the gonads.

CAUTION: When working with formalin, you are required to have the proper ventilation. This is accomplished by using the exhaust hood in the laboratory or working outside. Be sure to have a water source for contamination of skin or eyes. Also, be certain to wear the proper protective equipment, such as eye protection, apron, long pants, and closed toed shoes/rubber boots.

The gonad collection then goes into the appropriate bucket by species and year. The above, for example Gray Snapper, would go into the 2004 Mix #1 Bucket.

Species that have their own buckets, due to the large volume of those species are:

Species	Abbreviation	Species	Abbreviation
Red Snapper	RS	Red Grouper	RDG
Yellowtail Snapper	YTS	Gag	GAG
		Scamp	SCA
King Mackerel	KMK		
Golden Tilefish	GTL		

Gonad samples collected in fishery independent surveys have their own buckets, and are marked with the year, vessel name, cruise dates, species and PC collection numbers.

All other species of reef fish and pelagic gonads go into the current "Mix Species" Bucket.

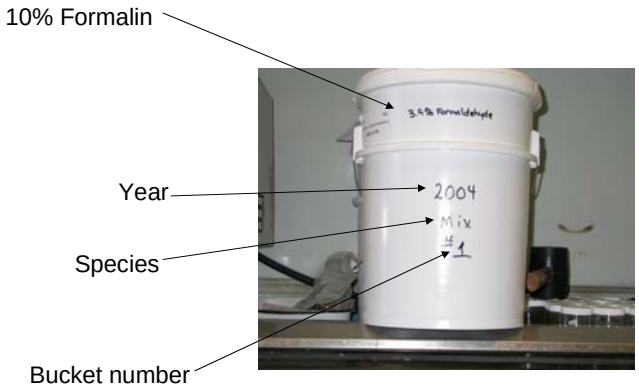


Figure 7.4.2 Appropriate labeling of 5-gallon bucket for the primary storage of gonads

7.5 Treatment of Preserved Gonads Received From an Outside Lab Source

In some circumstances, gonad samples are received at Panama City that have already been preserved elsewhere and information that could be gathered from observing fresh tissue is unavailable (this often occurs on fishery independent surveys and observer programs). When gonads are received fixed in formalin or any substitute fixative they can be microscopically examined, but it is time consuming and formalin handling protocols must be followed and thus, not typically completed. For these samples, the procedure for processing deviates from section 7.2 in the following manner:

Step 1 Determine whether gonads are part of a collection that has already been cataloged. This can be accomplished by searching the Age, Growth, and Reproduction Databases and log data for the source number.

Step 2 If the gonads have already been cataloged, proceed to Step 4. If not, proceed to step 3.

Step 3 Gonads that do not belong to an existing collection, should be logged in according to procedures in Chapter 1.

Step 4 If the gonads belong to an existing collection:

a) Be sure that log data (handwritten and database) includes accurate # of gonads
b) Obtain a copy of the original data sheet
c) Generate a Gonad Processing Sheet (Figure 7.2.3) with correct species and PC Collection # by filling out the following fields:

Date
Source
Source #
Collector
Lat/Long/Temp/Depths (if available)
Mode
Gear
Processed by:
Process Date:
Preservation Type:
Gonad Comments (if applicable)
Fish #
TL/FL (mm)
Whole/Gutted Wt (kg)
Sex
Sample
Kept

Step 5 Inventory preserved gonads

If no fish # is indicated on the gonad sample, then use the length and/or weight data to determine the fish #.

Step 6 A water proof tag with the following information is written in number 1 or 2 pencil (Figure 7.4.1) and placed in the freezer proof Ziploc (or Whirlpak):

Species abbreviation, Fish number, Catch Date, PC collection number, fish number, Fishing mode, Gear type, and Vessel Name

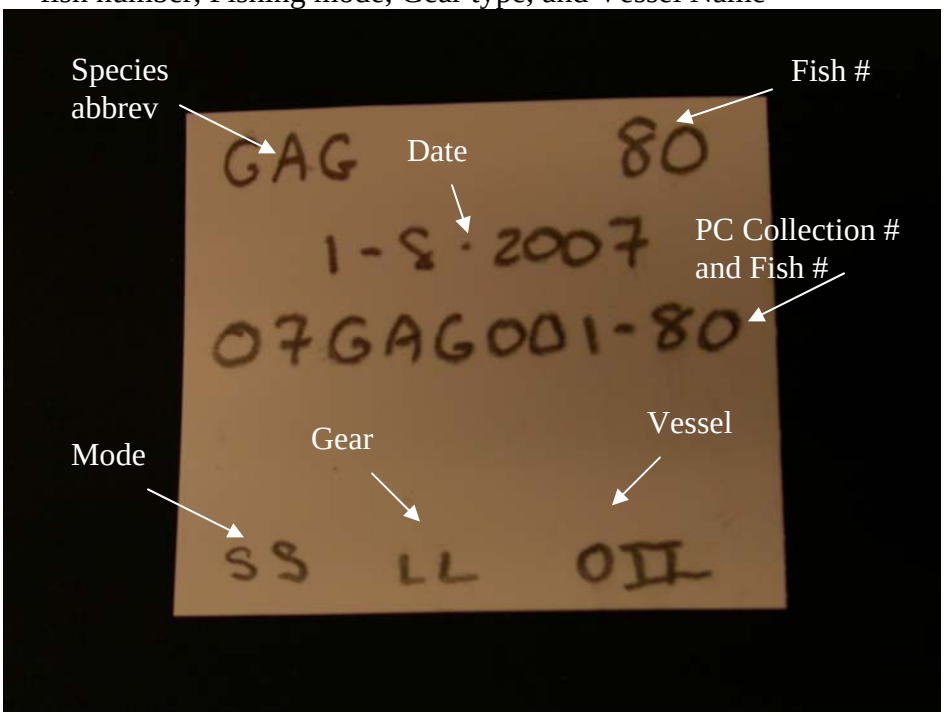


Figure 7.5.1 Example of a gonad tag with the appropriate data

Step 7 Each collection should be grouped together in a freezer proof Gallon Ziploc bag. The Ziploc bag should be marked using a <u>black</u> Sharpie Marker.

The following information should be written on the zip-loc bag:

PC collection number, Fish numbers, Catch Date, Fishing mode, Gear type, and Vessel

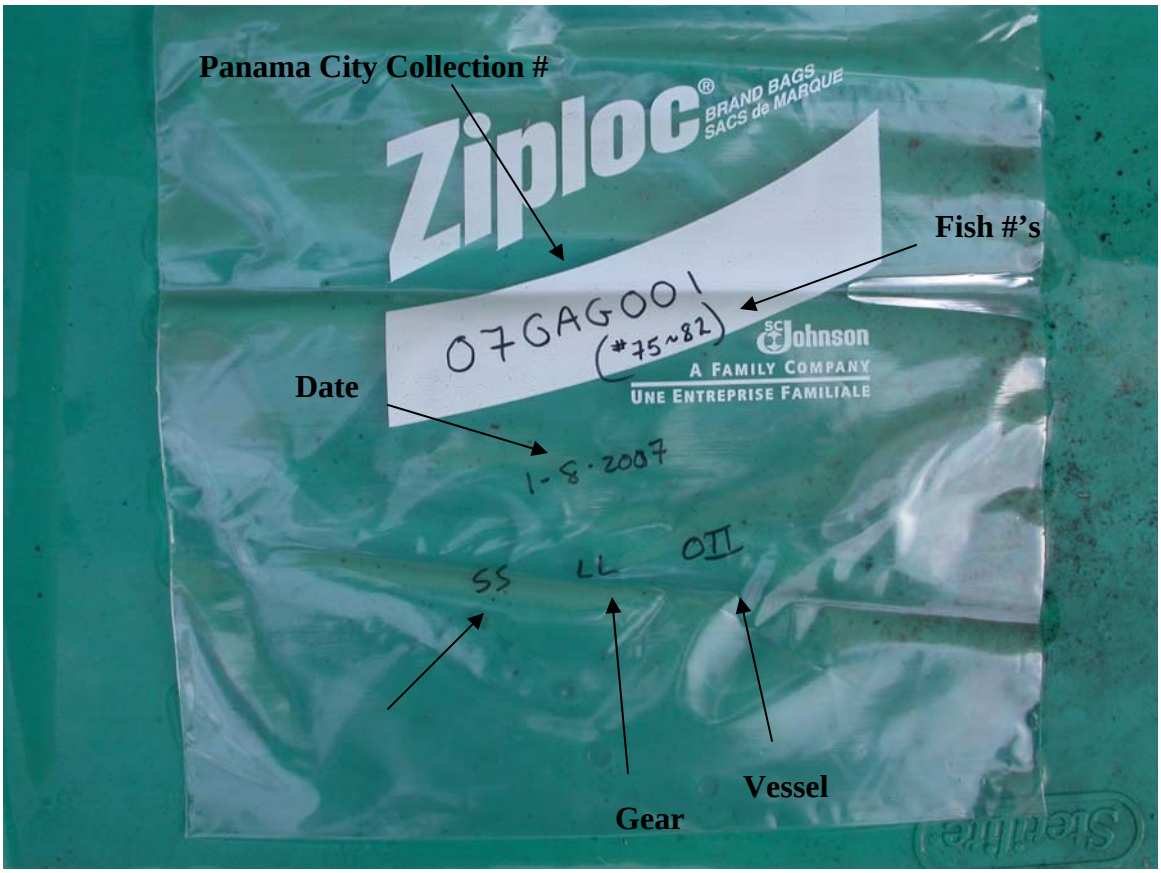


Figure 7.5.2 The appropriate labeling of a Ziploc bag for gonad storage

Step 8 The bucket's contents should be clearly marked on the outside in <u>black</u> Sharpie Marker with the species and associated PC collection number. The bucket should also be marked and cataloged.

Buckets checked in and inventoried are then taken to the Haz-mat storage building for storage and eventual blocking.

Step 9 Once all gonads have been checked in place the Gonad Processing Data Sheets into the appropriate folder for data entry.

7.6 Historical Reproductive Codes (no longer in use as of 2008)

Micro Stage

Microscopic staging of ovaries formerly (prior 2007) used numeric codes for each stage. There were 6 classifications based on the diameter of the largest oocytes.

1. Primary growth
Diameter up to ~4 omu, slightly opaque or clear, no nucleus or yolk present

2. Cortical Alveolar
Diameter up to ~ 8 omu, oocyte more opaque, with nucleus and/or yolk.

3. Vitellogenesis
Diameter up to ~20 omu, oocytes more opaque, mostly filled with yolk globules, may have thin, clear oocyte perimeter.

4. Hydration or Spawning
Diameter ~ 33 omu, oocytes much less opaque, may have oil globules, may be partially collapsed.

5. Spent or Regenerating after spawning
Diameter up to ~20 omu, 'ragged', much debris in ovary.

6. Unknown.

Gonad Conditions

Below is a list of gonad conditions, and currently (2008 -) are no longer in use. There has been some confusion on the appropriate description of a gonad regarding its condition and/or gonad remark. These remarks are listed here for the historical presence of such data.

Presently, any unusual or unknown conditions should be written on field sheets, gonad processing datasheets and recorded in the database in the gonad comments field.

Variations in the physical appearance of gonad

1) Gonad intact and in good condition
2) Gonad cut but intact (at least 90% present)
3) Gonad cut and not intact (<10% missing)
4) Gonad with parasites; intact
5) Gonad with parasites; cut and not intact
6) Gonad with green spot; intact
7) Gonad with parasites and green spot; intact

8) No code
9) Gonad with possible preservation problem, intact
10) Same as 6
11) Gonad with parasites and green spot cut and not intact
12) Same as 7
13) Gonad with parasites and possible tumor intact
14) Gonad with parasites and possible tumor intact
15) Gonad with parasites, brown spots; intact
16) Possible tumor inside gonad; intact
17) Gonad with parasites and possible preservation problem; not intact
18) White mass in gonad
19) Brown spot(s), parasites, intact
20) Possible preservation problem, no parasites; intact
21) Possible preservation problem and green spots; intact
22) Possible preservation problem and green sports; not intact
23) Brown spots; intact
24) Brown spots; cut but intact
25) Brown spots; not intact
26) Yellow spots; intact
27) Black spots; intact
28) Yellow spots; not intact
29) Gonad with parasites and yellow spots; not intact
30) Gonad with clear spots; intact (MALE)
31) Gonad with clear spots; not intact (MALE)
32) Gonad with parasites; cut but intact
33) Gonad with purple spots (use either 34 or 35)
34) Gonad with purple sports, intact
35) Gonad with purple spots; not intact
36) Gonad with purple sports; cut but intact
37) Black spots; cut but intact
38) Black spots; not intact
39) Yellow spot; cut but intact
40) Routine sample, possible preservation problem; not intact
41) Routine sample; not intact
42) Rotten gonad (soup like) tossed

Remarks

These are rarely used presently (2008 -) and some descriptions are similar to the gonad conditions list above. These remarks are listed here for the historical presence of such data.

Presently, any unusual or unknown remark should be written on field sheets, gonad processing datasheets and recorded in the database in the gonad comments field.

1. Routine sample
2. “Specks”, Large (~1 mm) spherical objects in gonad
3. No gonad
4. Includes decomposing tissue
5. Milky fluid in gonad
6. Flaccid
7. Copperbelly (Gag- actually refers to fish pigment—not gonad)
8. Red tinge on face
9. Abnormal gonad
10. Duplicated sample number
11. Possible copperbelly – “Beginner” (refers to fish pigment)
12. Majority of oocytes small (1-4 OMU) with a few odd oocytes that are larger
13. Collected during full moon phase and discarded
14. Gonad discarded [Kept: N]
15. Gonad preserved in 10% formalin, soaked in water before examination
16. To be blocked and sent to LSU [Kept: Y]
11. Running ripe, hydrated oocytes coming out of anus, or pouring out of ovary if cut
12. Plug of “hard packed” hydrated oocytes
13. Abdominal cavity lined with “compressed” hydrated oocytes
14. Abdominal cavity filled with fat (unusual for groupers)
15. Full moon phase
16. Possible transitional sample
17. Sample was in bad condition, internal organs “pulverized” and gonad is bloody
18. Possible parasite eggs
19. Gonad appears black (bloody?)
20. Specks and possible parasite
21. Destroyed gonad, eggs loose in bag
22. Sub-sample taken from gonad after total gonad weight was taken
23. Possible POFs candidate
24. Tumor
25. Routine sample, DNA taken

Chapter 8: Gonad Blocking and Storage Updated June 2008

This chapter provides guidelines for the blocking and storage of all gonad samples. Gonads are blocked (cross-sectioned in 4 mm slices) and tissue is put into small cassettes and shipped to the Histology Lab of the College of Veterinary Medicine at Louisiana State University where the tissue is made into histological slides (thin sections). Gonads are stored in 5-gallon buckets temporarily in the Age, Growth and Reproduction Lab and then permanently in the Hazardous Materials (Formalin) Storage Building located near dockside on the Panama City Laboratory grounds. All blocking must be conducted under the hood located in the Age, Growth and Reproduction Lab, due to the fact that all gonads have been preserved in 10% formalin. It is also very important to wear the proper protective clothing, nitrile gloves, and protective eyewear when working around 10% formalin.

In this chapter, the following is described:

8.1 Guidelines for Blocking Gonads

8.2 Guidelines for Storage of Gonads
    8.2.1 Blocked Gonads Long Term Storage

Contributors: Alan Collins, Bill Walling, Carrie Fioramonti, Melissa Cook

8.1 Guidelines for Blocking Gonads

Gather the following materials:

- Green Histological Notebook, dated annually: use sample year
- Gonad Processing Datasheet (found in RFRSLAC)

Gonads that are to be blocked (cross-sectioned for providing an adequate tissue sample for histological slides) must be preserved in 10% formalin for at least 2 weeks. This preservation is considered to be part of Primary Storage and will be discussed in Guidelines for Gonad Storage. Sections to be blocked require a **"PCL"** number that is assigned sequentially for the LSU histopathology laboratory. This is the histological tracking number and is different than the PC Coll # that tracks the specific fish at the Panama City Lab.

Step 1 Put on protective nitrile gloves, apron and goggles. Do not use latex gloves as formalin will leach through the latex gloves.

Step 2 Turn hood exhaust-fan and light on, open hood curtain and place bucket of gonads that are ready to be blocked inside the hood.

Step 3 Remove a Ziploc bag containing a gonad from bucket and place it on the porcelain tray under the hood. Place gonad and waterproof tag on paper towel to blot dry.

Step 4 In the green lab Histo-book (labeled by year), record the PC Coll # next to the sequential PCL #. Label the cassette that will hold the gonad section with the sequential PCL # (in Green Lab Histo-book) using a special histological marking pencil. Write the PCL # on the gonad tag.

Write PCL# on the front of cassette and on both sides of cassette. This is to ensure that the numbers can be read at LSU.

Figure 8.1.1 An example of a histology cassette labeled correctly.

Step 5 Check the tag with the gonad sample (Figure 8.1.2). If PC Coll # is not on the tag inside the, locate the PC Coll # that corresponds to the gonad and write the PC Coll # on the tag. If there is no tag, make a tag 1” x 1” using waterproof paper with the PC Coll#, PCL # (cassette #), and the fish # on it, and place it in the bag with the gonads. Be sure to write the PCL # on the tag.

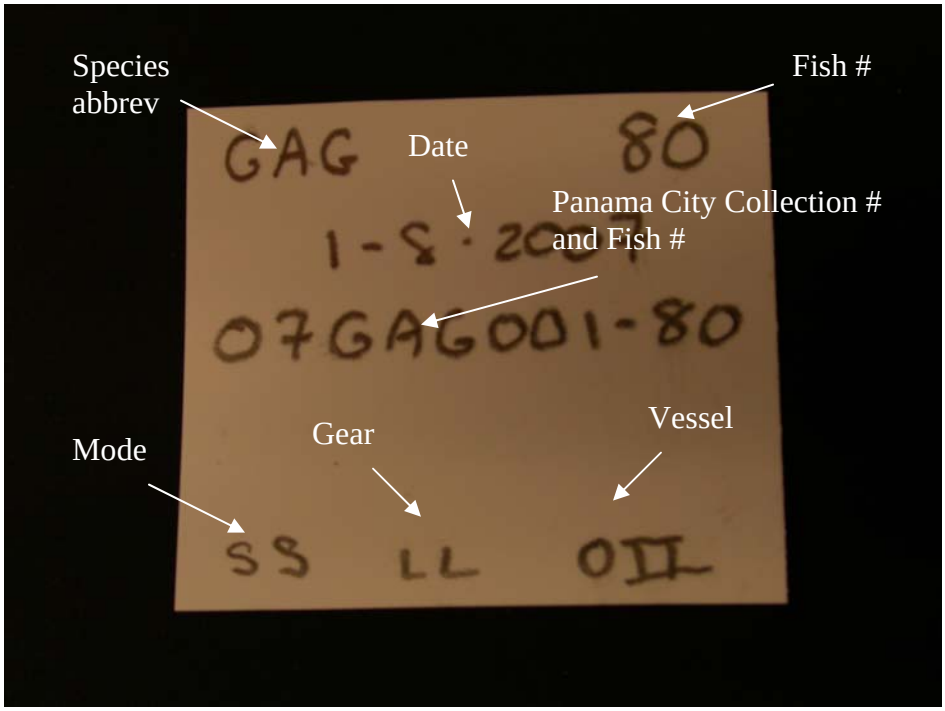


Figure 8.1.2 Gonad sample tag written on waterproof paper with the appropriate data

Step 6 Ensure that gonad is properly blotted to allow an accurate gonad weight in formalin. Trim off any excess connective tissue, fat, and parasites, and re-weigh gonad. This is to ensure an accurate fixed gonad weight.

As you process the gonad, record the following in the Green Histological Blocking Book:

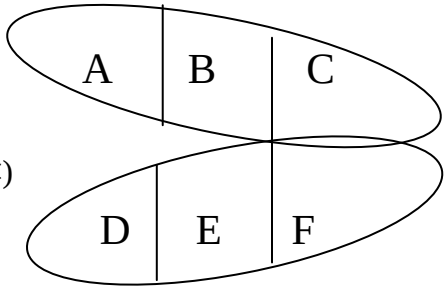
PCL # or Histo #
PC Coll #
Fish #
Catch Date
State & Location
Region of Gonad Sampled A, B, C, D, E, F
% Gonad Compare with RFRSLAC
Gonad Wt (g) in formalin
Remarks

Remarks

Step 7 For all gonochoristic (separately sexed) species such as the snappers (Lutjanidae), roll a six-sided die to randomly select the gonad region in which the block will be made. See sketch below. The locations are A-F (1-6), with locations A and D marking the right and left anterior ends. Locations C and F, the right and left posterior ends, usually contain the connection between the lobes. The top of the gonad will usually have connective tissue attached and the bottom of the gonad will not have any connective tissue. Record the gonad region and approximate percent of gonad in sample in the Green PCL# Lab Histo-Book.

A	1
B	2
C	3
D	4
E	5
F	6

(Fish's head<<<)



(>>>Fish's tail)

Figure 8.1.3. Diagram of a gonad labeled A – F, sections labeled in relation to the position of the gonad in the fish's body cavity

For all hermaphrodites (fish that change sex) such as the groupers (Serranidae), block the gonad at section C or F, or both C and F if they will fit into a cassette. The reason for this different sectioning area for hermaphrodites is that the posterior is where they have been found to first show signs of sex change.

Step 8 Use the long-handled double-edged razor blade to cut a cross section no thicker than 4 millimeters. Be sure to take a sample that includes the outer covering (or tunica) on gonads that are large. If gonad is small enough and sections from both sections lobes fit into the cassette, take a sample (cross-section) from both corresponding locations to get the most tissue in the cassette and the histo-slide.

Step 9 If gonad lobes are too large to both fit into the cassette, section only one lobe and mark the corresponding gonad section in the gonad area sectioned of the histo slide blocking log book. Section large lobes so that the sectioned area reaches from the center of the gonad to the tunica, this may require several sections and several cassettes on very large gonads (over 500gms). Label each of the cassettes with a section location letter (A, B, etc.) along with the PCL number.

Step 10 Place the section in the tissue cassette and snap the lid on. Place cassette in quart-size Ziploc bag, which will hold 40-45 cassettes per bag.

Step 11 Place remainder of gonad back in Ziploc bag with its tag and close. Place this bag back in white bucket. Be sure to replace lid back on bucket tightly and promptly because of formalin fumes.

Step 12 Once the Ziploc bag is full of cassettes, label it with the PCL #s that are contained in the bag. Add a little bit of formalin to the Ziploc bag, and place it under the hood in the bucket marked histo cassettes, until shipment. After a sufficient number of cassettes are made about (200-250), ship bags to LSU.

8.2 Guidelines for Storage of Gonads

Archival Storage – after gonad blocked:

Step 1 After gonad samples have been blocked, return gonad to appropriate Ziploc, according to PC Coll #.

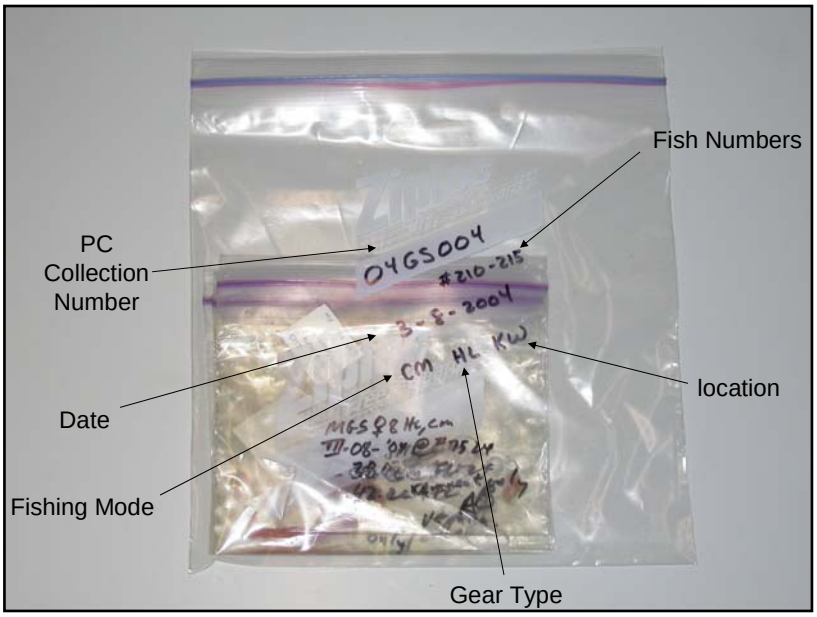


Figure 8.2.1 Example of labeled Ziploc bag for gonad long-term gonad storage

Step 2 When bucket is full, close and secure lid.

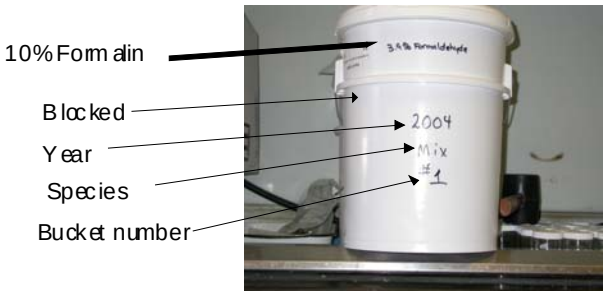


Figure 8.2.2 Appropriate labeling of 5-gallon bucket for the long-term storage of blocked gonads

Step 3 Remove "To Be Blocked" with isopropyl alcohol. With a black Sharpie pen, label bucket lid and side "Blocked", the current date, and the corresponding PCL #s (example: 04PCL0390-0480). If the gonads in the bucket are of a particular species or come from a particular sea cruise, include that on the label.

Step 4 Return the bucket to the gonad storage building. (Hazardous Material Storage Building).

8.2.1 Blocked Gonads Long Term Storage

Step 1 Move "Blocked" buckets from the Age, Growth and Reproduction Lab, to the Hazardous Materials Gonad Storage Building. The key to this building is located on a key chain (with a large faded orange float) in the top right cabinet drawer in the very back (near window) of Robert Allman's office.

Step 2 Before entering the Hazardous Materials (Formalin) Storage Building, turn on the exhaust fan. Open the door and run the exhaust fan for at least 90 seconds before entering the building. Door must remain open and exhaust fan must remain on while occupied.

Step 3 There are two methods of archiving gonads.

The old system is by numbering the buckets (samples collected 1970s – 1998). These buckets start from bucket #1 and run up to bucket #175.

The new method separates out major species (RS, VS, GAG, KM, SCA, RDG, YTS, RDP and GTL) by year, and the rest of the species are in Mix Species buckets. As buckets are filled, species and year specific buckets are numbered.

Chapter 9: Histological Classification for Gonads of Gonochoristic and Hermaphroditic Fishes

Updated June 2008

Histological slides of cross sectioned gonads were examined microscopically at 40x – 100x magnification and staged accordingly using the oocyte developmental characteristics described by Wallace and Selman (1981), Hunter and Macewicz (1985) and Tyler and Sumpter (1996). We have also incorporated ideas and language from recent workshop compilations on histological staging (Barbieri et al. 2006, Brown-Peterson et al. 2007). We deviate somewhat from the workshop classifications in allowing for the possibility of "skipped" annual reproduction among mature females. Therefore, we give more attention to indicators of prior spawning and distinguish to the degree possible, current from prior season's development. References for identifying atresia, brown bodies (melanomacrophages) and indicators of prior spawning include (but are not limited to: Rhodes and Sadovy 2002, Agius and Roberts 2003, Rideout et al. 2003). The most advanced stage of oocyte or sperm development was recorded as the leading gamete stage for the entire gonad. We use the terms "class" to describe gonadal development and "stage" for gamete development following Taylor et al. (1998). Histological slides were prepared by Louisiana State University School of Veterinary Medicine, Department of Pathology.

In this chapter, the following is described:

9.1 Sex Determination

9.2 Leading Gamete Stage
- 9.2.1 Female
- 9.2.2 Male

9.3 Atresia
- 9.3.1 Atresia and Brown Body Stages
- 9.3.2 Short-Term Atresia: leading oocyte stage

9.4 Postovulatory follicle

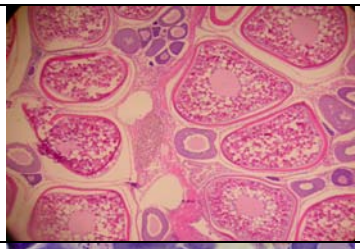
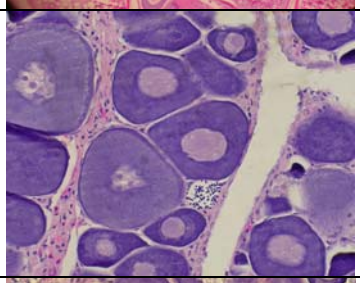
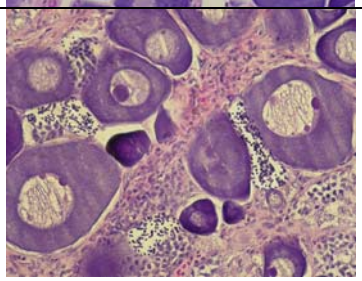
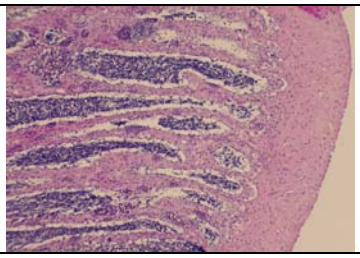
9.5 Indicators of prior spawning

9.6 Histological Gonad Class

9.7 Appendices
- A. Female (F) Gonad Histological Classification Categories
- B. Male (M) and Transitional (T) Gonad Histological Classification Categories
- C. Histology Slide Reading Sheet
- D. Commonly Used Comment Section Abbreviations

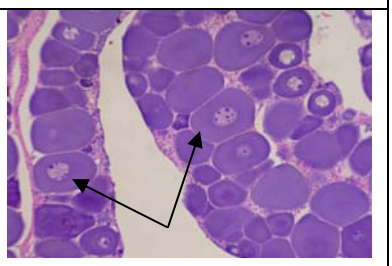
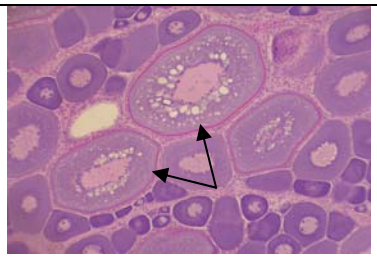
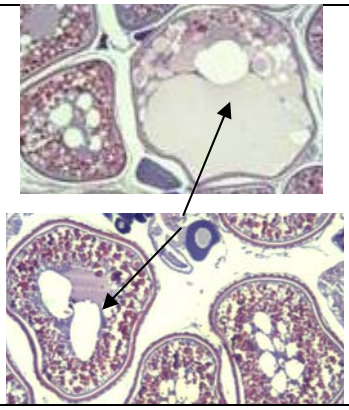
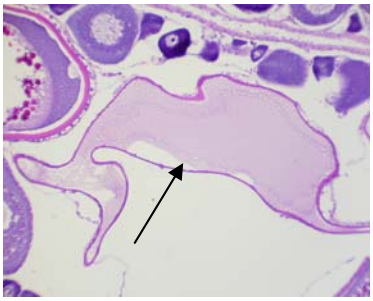
Contributors: Hope Lyon, Michelle Duncan, Alan Collins, Melissa Cook, Gary Fitzhugh, Carrie Fioramonti

9.1 Sex Determination

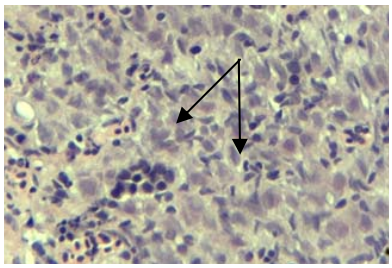
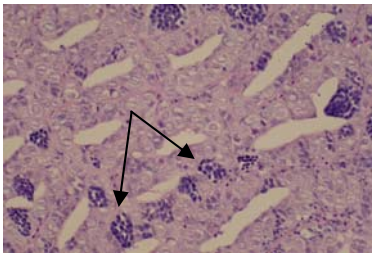
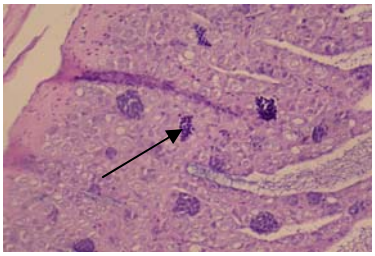
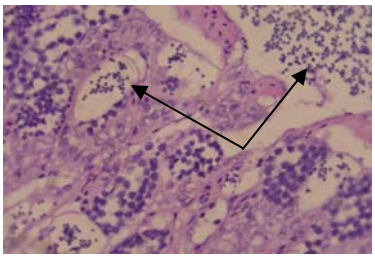
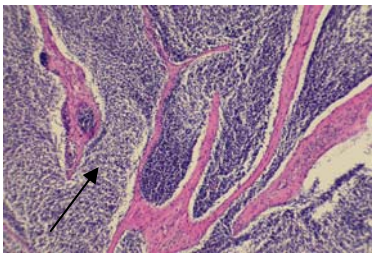
Sex	Description	Example Image
F— Female Gonochorists	Female oocyte stages are present (from primary growth through hydration). No evidence of male gametes.	
F— Female Protogynous hermaphrodites	Female oocyte stages are present (from primary growth through hydration). Spermatocytes and sperm crypts are present in small amounts; typically detected along the ovarian lamellae. Individual appears to remain a functional female.	
T— Transitional Protogynous hermaphrodites	Sperm crypts are present and proliferating throughout the section. Gamete stages from primary spermatocyte through spermatid should be present (at least 3 male stages). Oocytes can be described as remnant and possibly undergoing alpha atresia; particularly in all large (>0.2 mm) oocytes and/or there may be evidence of recent spawning as a female.	
M— Male Gonochorists and hermaphrodites	Gamete stages from primary spermatocyte through spermatozoa may be present. Sinuses should be present for male hermaphrodites.	

9.2 Leading Gamete Stage

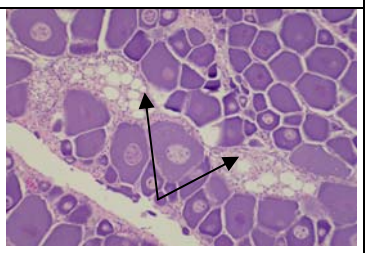
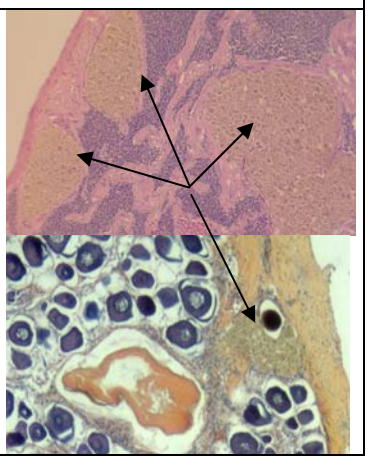
9.2.1 Female

Female Gamete Stage	Description	Example Image
PG—Primary Growth	Only primary growth oocytes are present (immature or resting female) or predominant (post-spawning female).	
CA—Cortical Alveolar	Primary growth and larger cortical alveolar oocytes are present. A red ring (zona radiata) may be apparent in late stage CA oocytes.	
V—Vitellogenic	Yolked oocytes are mostly spherical and less than ~0.450 mm in diameter. Yolk is first seen as small, red, spherical granules around the nucleus.	
H—Early Hydrated	Yolked oocytes ~0.450 mm in diameter or greater and less spherical in shape in conjunction with lipids coalescing into one or two clear spheres. Some attributes of the maturation phase may have begun including migration of the nucleus. Start of final oocyte maturation.	
LH—Late Hydrated	Oocytes reach their maximum volume during the maturation phase and are amoeboid in shape. Yolk-plate formation is clearly underway or complete.	

9.2 Leading Gamete Stage
9.2.2 Male

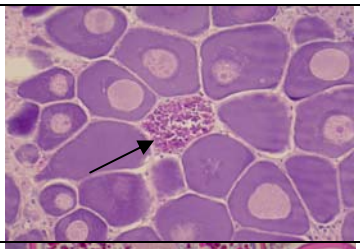
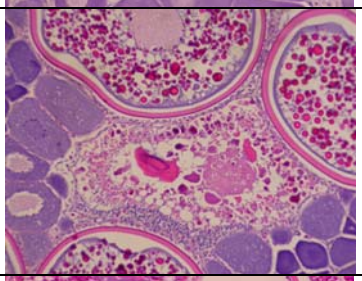
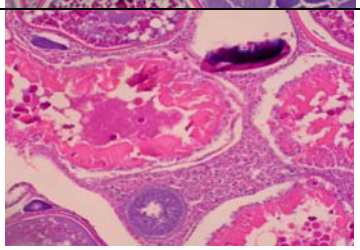
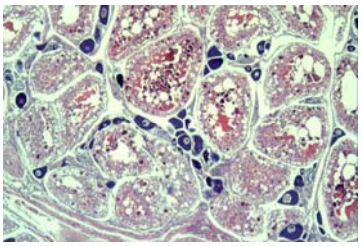
Male Gamete Stage	Description	Example Image
SG—Spermatogonium	Stained light purple, large in diameter (≈ 10 microns), fuzzy in appearance, not located in spermatocysts.	
PS—Primary Spermatocyte	Darkly stained cells, mostly non-spherical in shape, relatively large diameter (≈ 5 microns) and present in spermatocysts.	
SS—Secondary Spermatocyte	Cells are more spherical and darker than primary spermatocytes, smaller in diameter (≈ 3 microns) and present in spermatocysts.	
ST—Spermatid	Black staining, spherical and smaller than previous stages (≈ 1.5 microns) in spermatocysts. Tails not present.	
SZ—Spermatozoa	Black staining, pink tails present, spherical and similar in size to spermatids (≈ 1.5 microns). Spermatocysts appear to merge together (due to extensive discontinuous germinal epithelium) forming large pools.	

9.3.1 Atresia and Brown Body Stages (reference for following Tables)

Atresia Stage	Description	Example Image
Alpha	Initial atresia of oocytes. The structure is still identified as an oocyte as resorption proceeds, leaving only the follicular layers (see arrow). In yolked oocytes, the end of this stage is signaled by total yolk resorption.	
Beta	The beta-stage of atresia leaves a much reduced oocyte. In this stage the major degeneration and resorption of the follicle occurs. The presence of disorganized granulosa cells with pycnotic nuclei and intracellular vacuoles are characteristic traits. We note that beta stage atresia typically is identified or detectable in oocytes of later development stage (e.g., V through ovulation). Little is known about atresia during earlier stages of development (Tyler and Sumpter 1996).	
Brown Body (melanomacrophage)	Small to large brown mass in appearance which is a phagocytic response to tissue atresia. Since these structures occur in tissues other than gonads, their occurrence is not a direct indication of past spawning (as are post-ovulatory follicles) but rather associated with post-spawning metabolic recycling within the ovary. Often numerous throughout the gonads of hermaphrodites.	

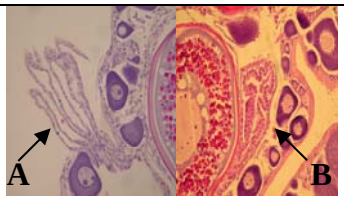
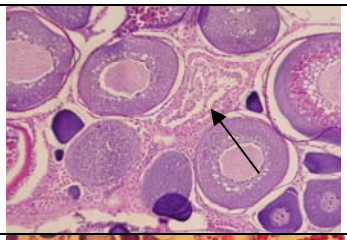
9.3.2 Short-Term Atresia: leading oocyte stage

This table pertains to the leading oocyte stage of the current spawning season for females. Note that some atresia may be persistent and detected in male and transitional protogynous hermaphrodites and females from a prior spawning season (see 9.5 for characteristics of prior spawning).

State Number	Description	Example Image
0	No detectable alpha atresia of unyolked or yolked oocytes.	
1	Alpha atresia of unyolked oocytes is noted. Yolked oocytes are not present in gonad.	
2	State in which less than 50% of the yolked (vitellogenic) oocytes are undergoing the alpha stage of atresia. Some beta atresia may also be present.	
3	State in which 50% or greater of the yolked oocytes are undergoing the alpha stage of atresia. Beta stage atresia may be present. Typically this occurs near the end of the spawning season where an ovary is either transitioning from spawning to spent or is spent (spawning complete). However this might be noted in a female that has prematurely ended reproductive development.	 

9.4 Postovulatory follicle

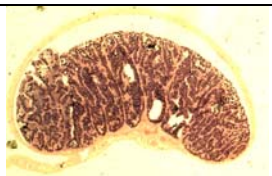
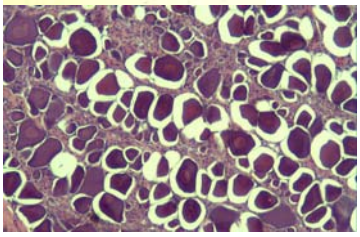
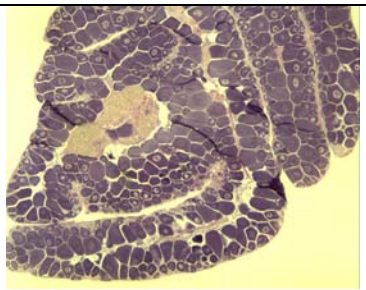
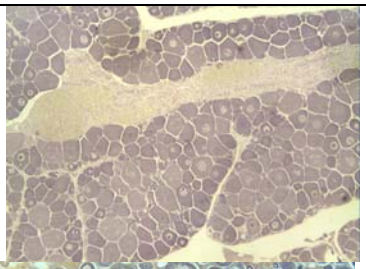
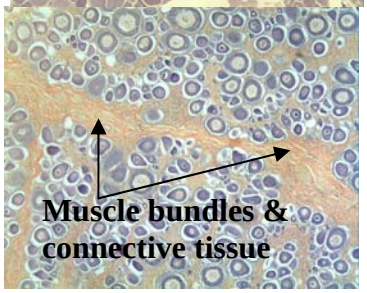
Postovulatory follicles are typically ephemeral; lasting only a day or two, and their appearance indicate that fish have recently spawned (thus pertaining to the current spawning season).

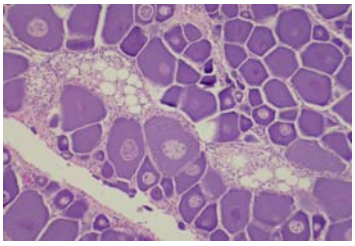
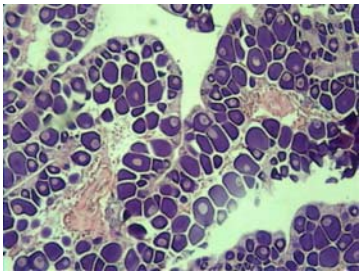
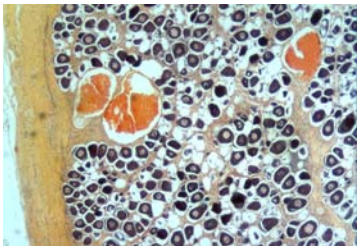
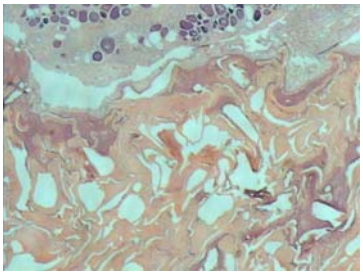
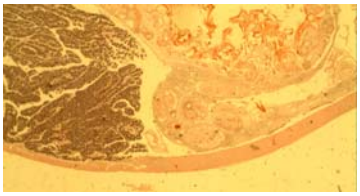
State Number	Description	Example Image
P0	None are detected or none can be distinguished from later stage atresia of intact oocytes.	
P1	**A**. Brand new postovulatory follicles. Resemble long strings folded upon themselves. **B**. Postovulatory follicles are new. Lumen is clearly evident and granulosa cell nuclei appear to be "lined up".	
P2	Postovulatory follicles are old. Lumen is filled partially or completely. Granulosa cells have broken down or ragged and no longer appear to have a linear pattern of nuclei.	
P3	Both new (**P1**) and old (**P2**) postovulatory follicles are present, presumably indicating high spawning frequency.	

A fish with <u>any</u> type of postovulatory follicles (P1, P2, P3) present has a histological gonad class of 'Spawning'.

9.5 Indicators of prior spawning

This table includes characteristics of prior spawning that are persistent for months to years (e.g. not ephemeral structures like post-ovulatory follicles). These traits are most useful in distinguishing immature from regressed females. Additionally these traits may aid in distinguishing mature females who may have skipped a spawning season.

State Number	Description	Example Image
0	*No evidence of prior spawning*. No brown bodies are evident or they are few and small. No old atresia of advanced oocytes, no elongated lamellae, few to zero muscle bundles. Unless there is evidence of development for the current season, a female scored as zero for this trait would be immature.	 
1	*Some evidence of prior spawning*. One to a few brown bodies and/or muscle bundles are evident. Cross sectional area of the gonad is larger than in state zero. Unless there is evidence of development for the current season, a female scored as one for this trait might be scored as "uncertain maturity".	
2	*Likely to have spawned before*. More than 5 brown bodies are evident throughout the section. Muscle bundles and large amounts of connective tissue are quite apparent. Lamellae are elongated. Cross sectional area is notably larger. If the leading oocyte stage is PG, this state would distinguish regressed (or resting) females from immature females. <u>Further</u>, if the leading oocyte stage is PG (or CA) and it is near the peak of the spawning season ("peak"-- based on observations of other females of the same species), this female could be a potential skipped spawner.	 

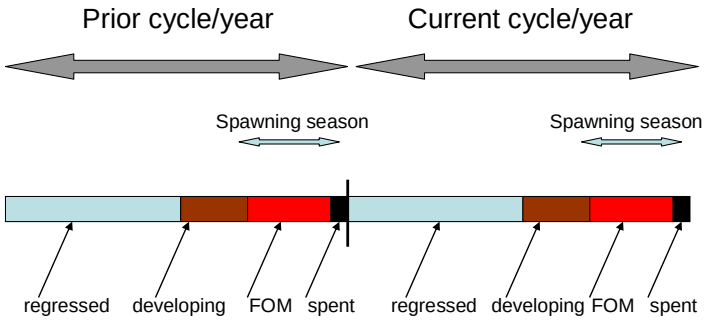
State Number	Description	Example Image
3	*Certain to have spawned before*. Persistent states that may last years: a hardened mass of old hydrated oocytes present (either within the lamellae or encapsulated within the lumen), evident muscle bundles, elongated lamellae, cross section area is notably larger. Other indicators of having spawned within the year (months) are extensive beta stage atresia of advanced oocytes, extensive vascularization, apparent brown bodies and a thick or folded tunica. If the leading oocyte stage is PG, this state would distinguish regressed (or resting) females from immature females. <u>Further</u>, if the leading oocyte stage is PG (or CA) and it is near the peak of the spawning season (based on observations of other females), and the appearance is distinctly different than a recently spent female (which are expected to be detected at the end of the season) this female would be a potential skipped spawner. The reader should be conscious of making a decision whether the fish spawned or can spawn in the current season (or year) as opposed to a prior season (or year). See gonad classification R versus Rs.	 Beta atresia  Muscle bundles  Remnant hydrated oocytes within the lamellae.   Remnant hydrated oocytes in lumen.

9.6 Histological Gonad Class

Gonad Class	Female	Male
I—Immature, inactive (was INI) *Corresponds to Macroscopic class 1*	"Immature": Not capable of spawning within the distant future (weeks to months). Includes females with primary growth oocytes and no yolked oocytes, atresia or other indicators of prior spawning.	Difficult to distinguish from regressed class, except immature males do not have residual spermatozoa. Lobules small or non-existent. Includes males with spermatogonia and no evidence of spermatogenesis.
U—Inactive, uncertain maturity (was INU) *Corresponds to Macroscopic class 1*	"Uncertain": Not capable of spawning within the distant future (weeks to months) and any evidence of past spawning is unclear. Includes females without vitellogenic oocytes but with some atresia of unyolked oocytes. Any atresia present cannot be clearly determined to have been derived from vitellogenic or more advanced oocytes. (**Caution**; in some early notations at Panama City, "U" was used to indicate that a sample was missing or not taken).	
D—Developing, <u>with</u> clear indicators of prior spawning. - - - - - - - - - - - - - - - - - - - Dv—Developing virgin or <u>without</u> clear indicators of prior spawning. *Corresponds to Female Macroscopic class 2 and Male class 2 & 3*	Cortical alveolar (CA) stage oocytes are present. Indicators of prior spawning are used to confirm or support that the female is mature. - Cortical alveolar (CA) stage oocytes are present and there are <u>no</u> indicators of prior spawning to support that the fish spawned in the past. Some individuals may reach this stage but not advance to final oocyte maturation (premature development). These fish would not be considered truly mature and are classified as "Dv".	Initiation of spermatogenesis and formation of spermatocysts. All stages of spermatogenesis (spermatogonia, spermatocytes, spermatids) may be present including spermatozoa. Little or no spermatozoa present in lumen of lobules or sperm ducts. Spermatocysts line lobule walls as continuous germinal epithelium. - Spermatogenesis begins; spermatocytes present & <u>no</u> prior indicators of maturity.
A—Active, mature, non-spawning (was AMN) *Corresponds to Macroscopic class 3*	"Active or developing": Capable of spawning within the near future (days to weeks). Includes females with vitellogenic oocytes but no hydrated oocytes or postovulatory follicles. May have some atresia (state 2).	**This phase is not used for males since it is essentially the same as developing, except discontinuous germinal epithelium is present at either the periphery or ducts.*

Gonad Class	Female	Male
Females: S—Spawning, hydrated (was AMS) Males: S—Spawning capable *Corresponds to Macroscopic class 4*	"Spawning, ripe or recently spawned": Capable of spawning now or in the immediate future (hours). Includes females with early and late hydrated oocytes (POFs may be present). Also includes females with vitellogenic oocytes as the leading gamete stage AND <u>any</u> type of postovulatory follicles (old and/or new). May have some atresia (state 2).	Fish is reproductively active and capable of spawning. All stages of spermatogenesis may be present. Spermatocysts scattered throughout testis, germinal epithelium primarily discontinuous. Spermatozoa are evident in lumen of lobules and filling sperm ducts. Eventually, testes become predominately filled with large pools of spermatozoa, resembling a "sack of sperm".
P—Post-ovulatory, spent *Corresponds to Macroscopic class 5*	"Post-ovulatory or Spent": Denotes the end of a fishes spawning season and is a transition from spawning to regressed. Females exhibit evidence of recent spawning with greater than 50% of advanced stage oocytes experiencing atresia (state 3).	Spermatogenesis is ceasing. Some residual spermatozoa present in shrunken lobules and sperm ducts. Lumen of central collecting duct with few spermatozoa. Spermatogonia proliferation and regeneration of germinal epithelium common in periphery of testis. Testes reduced in size.

Gonad Class	Female	Male
R—Regressed, inactive, mature, but spawned in the prior season or year (was INM) Rs—Regressed, skipped, inactive, mature during the spawning period (was INM) (females only) *Corresponds to Macroscopic class 1*	"Resting or regressed" from previous spawn and thus are not capable of spawning within the distant future (weeks to months) but there is clear evidence of spawning sometime in the past. The leading oocyte stage is PG. Some alpha or beta atresia may still be present. Rs includes females detected during the peak of the spawning season that exhibit past indicators of maturity and spawning but are not developing in the current season. The leading oocyte stage is PG or CA (if well into spawning period and no further development is underway). Also includes females whose current season development has ended prematurely, prior to spawning. In the interval between spawning periods it may be very difficult to discern females that spawned months ago (R) from females that skipped the last season, rather having spawned a year or more previously (Rs).	Spermatogonia dominate testes, no active spermatogenesis. Lobules and ducts primarily empty but some residual spermatozoa may be present. Some recrudescence possible. Continuous germinal epithelium throughout. Testes small but larger in diameter than immature males. * Transitional males are also classified as regressed (R).



Generalized time-scale distinguishing a prior spawning cycle from a current spawning cycle or year

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9.7 Appendices

A. Female (F) Gonad Histological Classification Categories

B. Male (M) and Transitional (T) Gonad Histological Classification Categories

C. Histology Slide Reading Sheet

D. Commonly Used Comment Section Abbreviations

Appendix A. Female (F) Gonad Histological Classification Categories

Leading Gamete Stage	**Indicators of Prior Spawning**	**Short-Term Atresia: Leading oocyte stage**	**Post-ovulatory Follicles (POFs)**	**Histological Gonad Class and (Corresponding Macroscopic Class)**
PG – Primary growth	**0** – *None.* No old atretic oocytes, brown bodies, muscle bundles or elongated lamellae. Immature fish.	**0** – No alpha atresia of yolked or unyolked oocytes.	**P0** – None	**I** – *Immature, inactive.* (Class 1) PG oocytes only, no evidence of prior spawning. **U** – *Inactive, uncertain.* (Class 1) Not capable of spawning in distant future & prior spawning unclear.
CA – Cortical alveolar	**1** – *Some evidence.* One to few brown bodies and/or, muscle bundles, larger cross section. May describe "uncertain maturity".	**1** – Alpha atresia of unyolked oocytes. Yolked oocytes not present.	**P1** – New A). Brand new, string like B). New, lumen clearly evident, granulosa cell nuclei "lined up"	**D** – *Developing*. (Class 2) CA oocytes present & prior spawning indicators confirm female is mature. **Dv** – *Developing virgin.* (Class 2) CA oocytes present, no prior indicators of maturity.
V - Vitellogenic	**2** – *Likely.* 5+ brown bodies, muscle or vascular bundles apparent, elongated lamellae.	**2** – Alpha atresia < 50% of yolked oocytes.	**P2** – Old, lumen filled, granulosa cells ragged	**A** – *Active, mature.* (Class 3) Vitellogenic oocytes, will spawn w/in days/wks. **S** – *Spawning, hydrated.* (Class 4) Spawning now or w/in hours (H or LH oocytes). Also includes gonads with any stage of POFs present.
H – Early hydrated (coalescence of lipids, yolk &/or nuclear migration) **LH** – Late hydrated (oocyte hydration)	**3** – *Certain.* Old hardened hydrated oocytes present, evident muscle bundles & vascularization, elongated lamellae, large cross section, numerous brown bodies.	**3** – Alpha atresia > 50% of yolked oocytes. Usually at end of spawning season.	**P3** – Both new (**P1**) and old (**P2**)	**P** – *Post-ovulatory, spent*. (Class 5) Majority of oocytes experiencing atresia. **R** – *Regressed, inactive, mature*. (Class 1) PG present, evidence of sexual maturity & recent spawn, will not spawn in future weeks to months. **Rs** – *Regressed, skipped, mature*. (Class 1) PG or CA present, evidence of maturity, will <u>not</u> spawn in current season; no development or ended prematurely. PG or CA atresia may be present.

Appendix B. Male (M) and Transitional (T) Gonad Histological Classification Categories

Leading Gamete Stage	**Histological Gonad Class and (Corresponding Macroscopic Class)**
SG – Spermatogonium **PS** – Primary Spermatocyte **SS** – Secondary spermatocyte **ST** – Spermatid **SZ** – Spermatozoa	**I** – *Immature, inactive.* (Class 1) Includes males with spermatogonia and no evidence of spermatogenesis. Difficult to distinguish from regressed, except immature males do not have residual spermatozoa. **Dv** – Developing virgin. (Class 2 & 3) Spermatogenesis begins; spermatocytes present & no prior indicators of maturity. **D** – *Developing*. (Class 2 & 3) Initiation of spermatogenesis and formation of spermatocysts. All stages of spermatogenesis may be present. Little or no spermatozoa present in lumen of lobules or sperm ducts. Spermatocysts line lobule walls as continuous germinal epithelium. **A** – *Active, mature.* (Class 3) **This phase is not used for males since it is essentially the same as developing, except discontinuous germinal epithelium is present at either the periphery or ducts.* **S** – *Spawning capable.* (Class 4) Fish is reproductively active and capable of spawning. All stages of spermatogenesis may be present. Spermatocysts scattered throughout testis, germinal epithelium primarily discontinuous. Spermatozoa evident and filling sperm ducts and lobules. Eventually, testes become predominately filled with large pools of spermatozoa, resembling a "sack of sperm". **P** – *Spent*. (Class 5) Spermatogenesis is ceasing. Some residual spermatozoa present in shrunken lobules, sperm ducts and lumen of central collecting duct. Spermatogonia proliferation and regeneration of germinal epithelium common in periphery of testis. Testes reduced in size. **R** – *Regressed, inactive, mature*. Also includes <u>transitional males</u>. (Class 1) Spermatogonia dominate; no active spermatogenesis. Lobules and ducts primarily empty but some residual spermatozoa may be present. Some recrudescence possible. Continuous germinal epithelium throughout. Testes small but larger in diameter than immature males.

Appendix C. Histology Slide Reading Sheet

Histo #	Date	PC Coll #	Fish #	Previous Gonad/Histo Comments	Histo Sex	Leading Gamete Stage	Indicators of Prior Spawning	Short-term Atresia	POFs	Histo Class	Histo Comments
											.

Appendix D. Commonly Used Comment Section Abbreviations

BB Brown body (melanomacrophage aggregate)

BBS Brown bodies (melanomacrophage aggregate)

BB+ More than 5 Brown bodies (melanomacrophage aggregate)

BP Bad preservation

CGE Continuous germinal epithelium

DGE Discontinuous germinal epithelium

Good Ex. or **Good example** If good example of something is present in slide

Lipid coa Lipid coalescence is occurring

MB Muscle bundles

First-year male or **New Male** Used for hermaphrodites, fish is functioning as a male (i.e. spawning capable), however, the gonad still contained isolated remnant oocytes.

Old HO or **Old HOs** Old hydrated oocytes present in the lamellae or lumen

Plug Large mass of old hydrated oocytes usually present in the lumen

Picture If picture has been or should be taken of the slide

PAR Parasites

Vasc Vascularization

^ Insert a ^ in front of an important comment so it can be searched for and easily found in the data base.

Chapter 10: Guidelines for estimating fecundity Updated January 2008

Estimates of reproductive potential (the number of oocytes spawned annually), or fecundity, are needed for accurate stock assessments of commercially and/or recreationally important species in the Gulf of Mexico. Since individuals of most of these species spawn many times each year and annual fecundity is indeterminate, estimates of batch fecundity, spawning fraction and duration of the spawning season must be made in order to estimate annual fecundity (Murua et al. 2003). In general, the overall timeline of fecundity processing is first macroscopic/microscopic examination of ovaries wherein candidate females are identified for having hydrated oocytes, followed by histological examination (tissue cassettes are sent to LSU vet school, stained slide preparations are returned and read/confirmed), then tissue sub-samples are extracted from confirmed ovaries and oocytes counted.

This chapter includes the following sections:

10.1 Estimating Batch Fecundity
- 10.1.1 Steps for Preparing Samples
- 10.1.2 Steps for Counting Samples

10.2 Estimating Spawning Frequency

10.3 Estimating the Spawning Season Duration

10.4 Estimating Annual Fecundity

Contributors: Alan Collins, Gary Fitzhugh, Bill Walling, Carrie Fioramonti

10.1 Estimating Batch Fecundity

Estimating batch fecundity can be difficult if you don't know exactly where and when the target species spawns (and this, of course, is usually the case). It is necessary to sample ovaries of fishes in the last few hours before spawning to obtain accurate estimates of batch fecundity by using hydrated oocytes (Harris et al. 2002). Fully hydrated oocytes are by far the best to use because they are easy to separate from vitellogenic oocytes. If the most advanced oocytes are not at least in an early stage of hydration, it is difficult for the counter to separate the last batch of oocytes from the next batch coming up (Hunter et al. 1985). Finally, many batch fecundity estimates (n > 50 females) are usually required in an attempt to adequately cover spatial and temporal variables as well as the ranges of female length, weight, and age.

Knowledge of the distribution of hydrated oocytes in ovaries for each species is also important for estimating batch fecundity since a smaller number of samples from each hydrated oocytes can be used if the distribution is homogeneous. We use a two-way analysis of variance (Hunter et al. 985) to test for homogeneity of variance in batch fecundity estimates among ovarian regions and lobes. So far, all of our snappers and groupers, as well as king mackerel, have not exhibited significantly different densities of hydrated oocytes among gonad regions, but it is still necessary to perform this test for each new species.

Like most recent fecundity studies in the Gulf of Mexico, we use the hydrated oocyte method of Hunter et al. (1985) to estimate batch fecundity. As soon as possible after ovaries arrive at the lab and preliminary processing is completed (see Chapter 5), all intact ovaries that contained hydrated oocytes are placed in 10% buffered formalin for at least two weeks. Histological slides are then made and an examination of each slide used to confirm the presence of hydrated oocytes and to check for the presence of new postovulatory follicles, or POFs (Hunter et al. 1985). If new POFs are present in the ovary with hydrated oocytes, then the fish had just started to spawn before capture and the batch fecundity count is incomplete. In this case, the ovary is removed from consideration for batch fecundity estimation.

After finding the distribution of hydrated oocytes (HO) to be homogeneous among regions within the ovary, we take two fixed ~ 0.075 g samples per ovary and then determine the average HO count per unit of ovary weight. The locations of the two samples in each ovary are from the periphery and center portion of the cross-section respectively. The two samples are taken from the same region of each ovary, randomly selected from 6 possible regions (the anterior, middle or posterior region of the left or right ovarian lobe). This method was adopted as an improved technique for dealing with fishes having large gonads. Before 2002, we used a single wedge-shaped sample of varying weight (to the nearest .0001 g) taken from a randomly-sampled cross-section of each ovary. The wedge-shape of this sample was supposed to account for any differences in hydrated oocyte numbers from the periphery to the center of the ovary in cross-section (Hunter et al. 1985) but sampling the ovaries in this way usually gives us a much greater sample weight (up to ~ 0.70 g), and counts became difficult to replicate in larger ovaries.

Batch fecundity was estimated as (total number of hydrated oocytes in the two fixed samples * ovary weight in g) / total weight of the two fixed samples in g. We used the weight of the ovary in formalin instead of the fresh weight of the ovary because the sample (from which the count of hydrated oocytes is made) is also in formalin. Often, however, only fresh gonad weight was recorded. A conversion for estimating preserved from fresh weight can usually be made if deemed necessary after examination of data (plots) where both fresh and preserved weights were recorded from the same ovary.

10.1.1 Steps for Preparing Batch Fecundity Samples

Step 1 Once fish have been identified macroscopically, microscopically, and histologically as batch fecundity candidates, gonads are retrieved from storage.

Step 2 Begin filling out the Batch Fecundity Estimate worksheet with the following information:

- Species
- PC Coll #
- Fish #
- Histo #
- Date of capture
- Time of capture (if available)
- Fresh Wt. (g) of gonad

This information can be taken from the original data sheet found in the RFRSLAC/Reproduction binders.

Fixed 10% formalin Wt. of gonad – Record from green histological log book (provides most accurate weight of whole gonad, prior to processing

Sample # – assigned consecutively to each fish in the study; each fish will have a sample A and B (i.e. 05-043, 05-044)

Step 3 While working under the exhaust hood in the Age, Growth, and Reproduction laboratory, randomly (roll a 6 sixed die) take a slice of the gonad approximately 3-4 mm thick. Record the region of the gonad sampled for BFE.

Step 4 Take two samples from the slice using the appropriate utensil. Sample A should be from the “core” of the gonad. Sample B should be from the “periphery” of the gonad. See the example Batch Fecundity Worksheet at the end of this section. Weigh the samples to the nearest .001g (it should be ~.075 g)

Step 5 Place the sample in a labeled, small glass vial and cover with 33% glycerin. Place in appropriate vial containers.

Step 6 Allow the BFE samples to set for at least 48 hours prior to counting.

10.1.2 Steps for Counting Batch Fecundity Estimates

You will need the following: Heerbrugg WILD Model # M5-46824 dissecting microscope with micrometer in 10X ocular and microscope magnification set at 25X, glass Petri dish with grid or counting chamber, disposable pipettes, probes, hand held counter, extra vial (labeled "counted"), BFE samples, and BFE data sheets. Be sure that the counting dish, extra vial, and probes are sanitary before handling the samples, to prevent contamination of samples with mold, etc.

Step 1 Gently pipette a small portion of the sample into the counting dish. Be sure to have adequate coverage of glycerin, but not so much that the oocytes slosh around as you move the dish.

Step 2 Gently tease apart the oocytes using the probe.

Step 3 Spread the oocytes out within the counting grid.

Step 4 Randomly measure the diameters of 2-3 oocytes from this portion using the micrometer within the ocular of the microscope. In total, the entire sample should have 10 oocytes diameters.

Step 5 Starting on one side of the sample, begin to count oocytes within each grid. Use the counter to keep an accurate count of the oocytes. Examples of countable and not-countable hydrated oocytes can be seen in Figure 10.1.2.1 & 10.1.2.2.

Note: Some oocytes may be "on the line" of a grid. Make the decision as to what grid the egg belongs to, and consistently use this rule throughout your counts. Otherwise, you run the risk of over- or under- counting these oocytes.

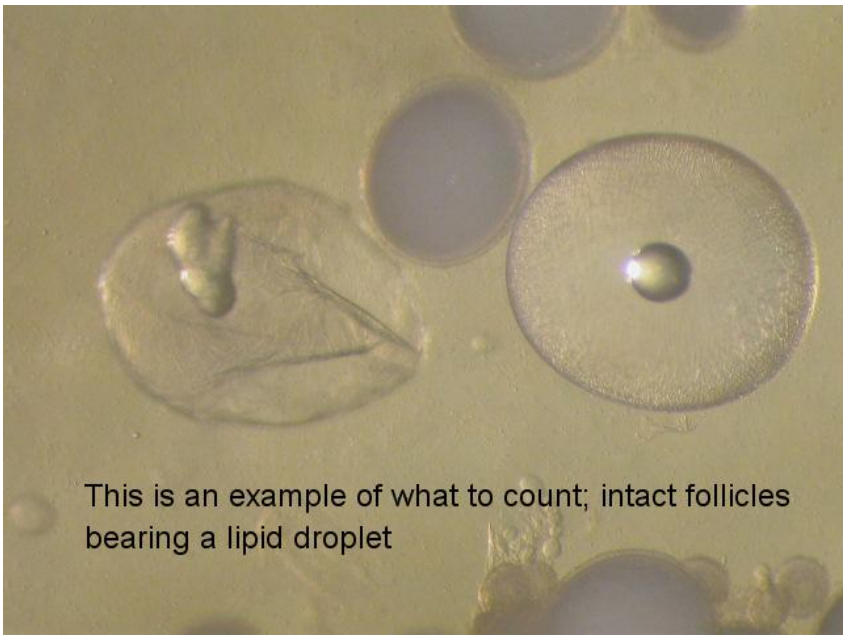


Figure 10.1.2.1 Countable hydrated oocytes for batch fecundity estimates; both of these oocytes should be included in the count.

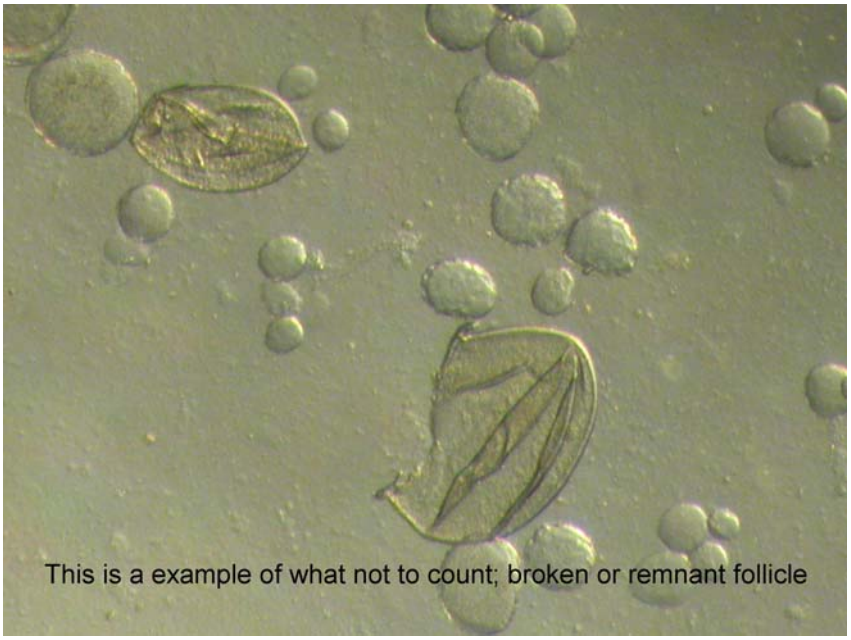


Figure 10.1.2 .2 These oocytes can not be counted for batch fecundity estimates; these remnants cannot be included in the count, nor can the smaller oocytes as they are not hydrated.

Step 6 Record the number of oocytes in that portion of the sample.

Step 7 Record any remarks or observations about the sample on the data sheet in the space provided.

Step 8 Carefully pipette that portion of the sample into the "counted" vial. Be sure to transfer ALL of the oocytes. Also take care not to splatter oocytes all over the vial, as it makes it difficult to retrieve them after counting.

Step 9 Repeat Steps 1-7 with the remaining portions of the sample.

Step 10 Pipette the sample back into the original vial. BE SURE to get all of the oocytes. This is important, as there will be a small percentage of BFE's that will have 2nd and 3rd counts to estimate reader/counter error. The samples need to contain the exact number of oocytes in order for this to be effective.

Step 11 Calculate BFE using the formula:

$$\text{BFE} = \frac{\text{Total \# of HOs in both samples * whole ovary weight (g)}}{\text{Total weight (g) of two samples}}$$

HOs = hydrated oocytes

Figure 10.1.2.3 Datasheet used to record Batch Fecundity Estimates

Batch Fecundity Estimates (BFE)

Species: _____ PC #:_____ Fish #:_____

Histo #: _____ Date of capture:_____ Time of capture:_____

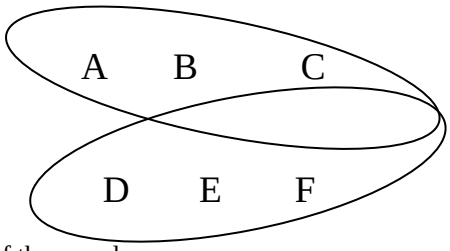
Counted by:_____ On:_____

I. Record the weight of the gonad (found on data sheet):

Fresh wt._____ g Fixed 10% formalin wt._____ g

II. Circle the gonad region sampled:

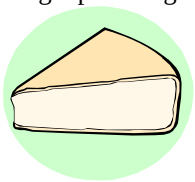
(Fish's head<<<)



(>>>Fish's tail)

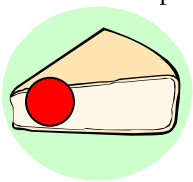
III. Take the weight of the sample:

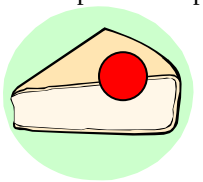
Little gonads
Single pie-wedge **OR**



_____ g

Big gonads
A. Core sample **AND** **B**. Peripheral sample





A_____ g **B**_____ g

Total Sample Weight=_____ g (combined)

IV. Put the sample in a jar filled with 30% glycerol.

V. Under a microscope, tease apart all the hydrated oocytes (HOs). Count the # of HOs found in the entire pie wedge (or combined core and peripheral sample) by looking at several sub-samples. Always fill the jar with 30% glycerol.

A._____ _____ _____ _____ _____ _____ Total # oocytes sample A_____
B._____ _____ _____ _____ _____ _____ Total # oocytes sample B_____
Total # oocytes A+B _____

VI. Remarks and maximum oocyte-diameter (OMUs at 10 x ocular, 25 x obj.) on 10 HOs:
A.____ ____ ____ ____ ____ ____ ____ ____ ____ ____ Remarks_____
B.____ ____ ____ ____ ____ ____ ____ ____ ____ ____ Remarks_____

Estimate of BFE: = $\frac{\text{Total \# of HOs * whole ovary wt. (g)}}{\text{Total sample wt. (g)}}$ (fixed ovary wt. if available)

BFE = _____HOs in whole ovary

10.2 Estimating Spawning Frequency

Although there has been some inconsistency with terms in the literature, we follow Murua et al. (2003) in defining the spawning fraction – as the fraction of mature females spawning per day observed from random samples of mature females. The inverse of this is the spawning frequency (or interval) which is the average expected interval in days between spawning events. The overall spawning season duration in days divided by the average interval yields the expected number of spawns (egg batches) per female per annual reproductive season.

Accurately estimating spawning frequency is usually much more difficult than estimating batch fecundity. It helps to know where and when the fish are spawning, but you cannot just rely on hydrated oocytes to indicate spawning females unless your sampling times coincide with when those structures are likely to occur (Harris et al. 2002). In addition, our experience dictates that the number of active females required for accurate spawning frequency estimates (preferably n > 200 for each year and area from collections well distributed over the spawning season) should be much greater than the number of hydrated females needed for batch fecundity estimates.

Records that show exact times of sampling for each female are helpful in estimating spawning frequency. Spawning frequency estimates using hydrated oocytes will not be accurate if sampling occurs after spawning just ended or too many hours before spawning starts (before the formation of hydrated oocytes, which usually can begin in a few to several hours before spawning, depending upon species, temperature, etc.

It may be useful to compare spawning frequency estimates based upon POFs and HOs. Any difference between the two estimates should be resolved by some understanding of how the detection of POFs or HOs act as indicators of spawning. For example, POFs may degenerate faster than HOs if preservation is delayed. If POFs are not properly (quickly) preserved, then new POFs (from the last spawn) may appear old enough to be from a previous spawn. Relatively high water temperatures in the Gulf of Mexico (at least for summer spawners) can also accelerate the rate of decay of POFs and decrease the appearance of POFs relative to hydrated oocytes. The assumption is that identification of POFs and HOs are reliably made within a day (+/- 12 hours) of spawning. Adjustments may have to be made if detectable durations of POFs or HOs are much longer or shorter (see Fitzhugh et al. 1993).

10.3 Estimating the Spawning Season Duration

Spawning season is defined by Murua et al. (2003) as the duration elapsed between the occurrence of 50% of mature females in advanced but pre-spawning stages (late stage vitellogenesis) to 50% of females in post-spawning stages (spent). Murua et al. 2003 also point out than an important assumption is that the distribution of the spawning stock remains relatively fixed in that fish do not migrate in or out of the spawning area. The expected number of spawns per female per year is then estimated as the spawning season duration divided by the average spawning interval in days.

10.4 Estimating Annual Fecundity

Annual fecundity, the total number of hydrated oocytes spawned by an individual during the year, is estimated by simply multiplying the batch fecundity estimate by the expected number of spawns per female per year (Hunter et al. 1985).

Ovary weight has also been used as a proxy for fecundity. Ovary weight is often available from more females than are batch fecundity samples. Selection criteria for ovary weight as a fecundity proxy can include vitellogenic and hydrated ovaries during the peak spawning months. However, it needs to be recognized that the process of hydration can considerably increase the ovary weight in a short amount of time. Therefore, combining ovary weights of vitellogenic and hydrated females (e.g., sorted by age or size) possibly adds noise to the fecundity proxy relationship and potentially introduces bias if some age classes, for example, are only represented by vitellogenic or only hydrated ovary weights. Of course selection of only hydrated ovary weights would be ideal, but then the sample size advantage would diminish and one might as well just estimate batch fecundity.

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