New Project: YES

Project Name: Protection and Restoration of the Valuable Seagrass Ecosystem in the Big Bend and Florida Panhandle

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Project Location: *(Include a map, if possible, and the city, county, watershed, longitude/latitude dec.deg, Parcel # if Applicable)*

This study will be conducted in the seagrass meadows of the Big Bend and Florida Panhandle regions in the northeastern Gulf of Mexico (Figure 1). The primary sites are located between eastern St. George Sound and Apalachee Bay in the waters off St. James Island, Franklin County, FL (Figures 2 & 3, Table 1), sites that cross four Franklin County watersheds: (1) The Lower Ochlockonee (03120003), (2) The Apalachicola River (03130011), (3) The New River (03130013), and (4) The Apalachicola Bay (03130014) (the numbers following each watershed name are the designations appearing in the U.S. Environmental Protection Agency website, "Surf Your Watershed": http://cfpub.epa.gov/surf/locate/index.cfm).

Figure 1. Map of Florida showing distribution of seagrass ecosystems. The Big Bend seagrass ecosystem covers approximately 3000 km^2.

Sampling sites include the shallow near-shore waters from Lanark Reef to Turkey Point Shoals (including seagrass beds off Dog Island Reef) that represent a continuum of low to high quality seagrass habitat. Additional sites added in year two are within western and central St. George Sound, Apalachicola Bay, and Alligator Harbor, to develop a more comprehensive view.

Table 1. Coordinates of study area off St. James Island, Franklin County, Florida, given in decimal degrees.

latitude	**longitude**
29.69°	-85.21°
29.59°	-85.21°
29.92°	-84.5°
29.81°	-84.43°

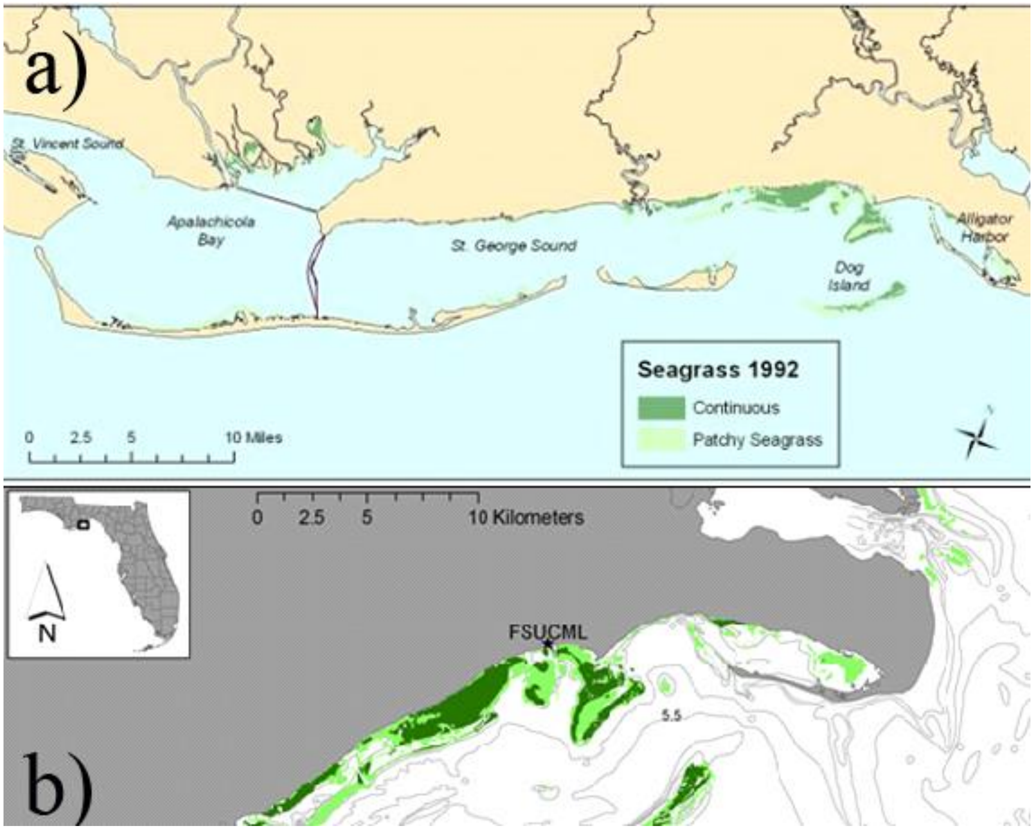


Figure 2. **a)** Seagrass ecosystem from Alligator Harbor to Apalachicola Bay. The total area of this seagrass system in Franklin County waters is approximately 60 km^2 (map from Yarbro & Carlson (2013)). **b)** Seagrass ecosystem adjacent to St. James Island, Florida. The total area of this seagrass system adjacent to St. James Island (excluding Dog Is Reef and Alligator Harbor) is 15.4 km^2. Dark green is dense seagrass and light green is sparse.

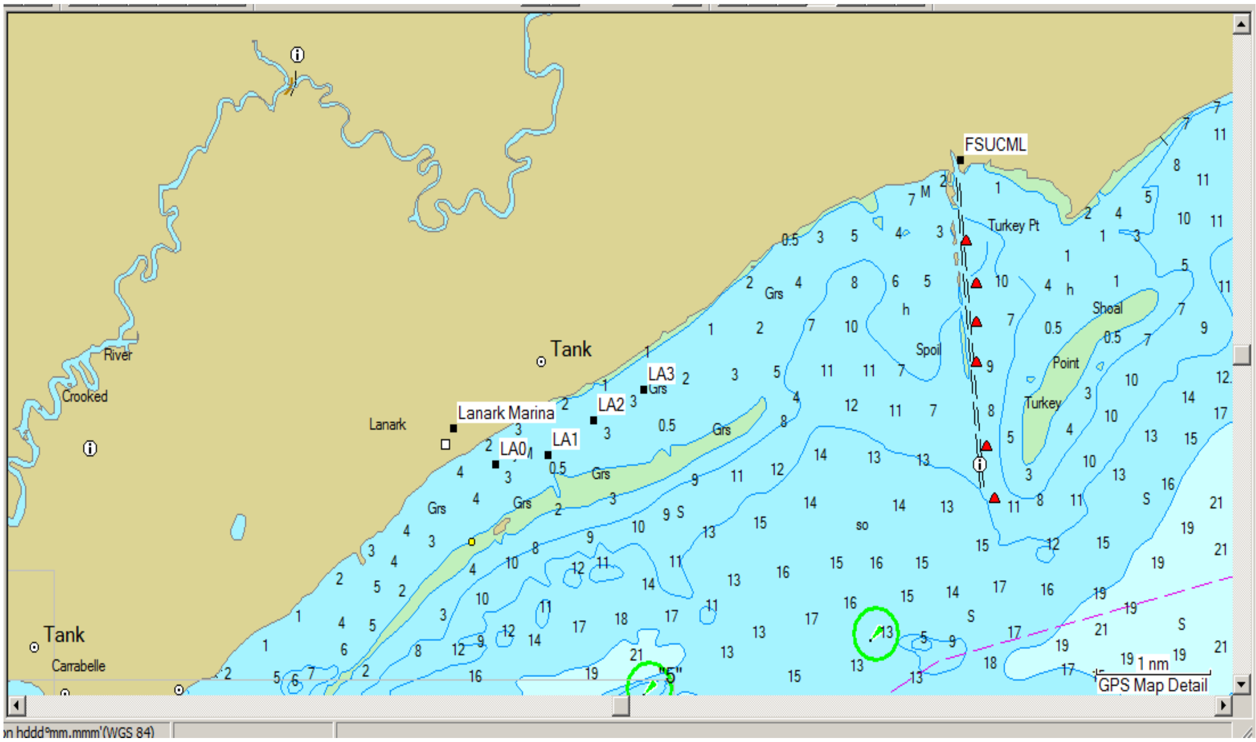


Figure 3. Sampling stations off Lanark, Franklin County, FL, following a continuum of seagrass quality from low (station LA0) to high (station LA3) seagrass beds.

Project Description: *(Describe all aspects of the project)*

Seagrass meadows are among the most valuable ecosystems in the world (Orth *et al.* 2006). The Big Bend region of North Florida contains the second largest contiguous seagrass habitat in the state and is considered to be one of North America's most pristine coastal areas (Livingston 1984; Zieman, Zieman & Pendleton 1989; Mattson *et al.* 2002). While there are many estimates of the economic value of seagrasses, the most recent puts it for fisheries alone at roughly $80,000 per acre annually (National Geographic Newswatch 2014). Estimates like these suggest that the seagrass system over the area of this study is valued in the tens of millions of dollars per year, without even touching on the essential ecosystem services that they provide, from being important food sources for organisms ranging from sea turtles and manatees to economically important fishes; stabilizing sediments, protecting coastlines from surge, enhancing water clarity, and producing enormous amounts of organic carbon. In addition, they are considered the "coastal canaries" that point to systems approaching tipping points in sustainability (Orth *et al.* 2006; NSF 2009). Studies we conducted in 1998 (Koenig & Coleman 1998) suggest that the contribution of gag (*Myteroperca microlepis*) alone is more than one million juvenile fish each year. Clearly, there are tremendous benefits provided by seagrass beds, making their protection and preservation critical for the ecological and economic resiliency of this region.

Despite their value, seagrass meadows are being lost world-wide at a rate of 2-5% per year (Duarte *et al.* 2008). Similar losses are occurring throughout the Big Bend and Florida

Panhandle regions in the northeastern Gulf of Mexico, including our proposed study sites. Yarbro & Carlson (2013), for instance, report seagrass declines in Franklin County of almost 50% since 1992. There is strong evidence for decline around Lanark Reef. Indeed, we have detected a gradient in seagrass quality that declines from east (Turkey Point) to west (off Lanark Reef) (Figure 4). Certain portions of the seagrass beds off Lanark have disappeared entirely, with concurrent loss of biodiversity and nursery habitat, increased turbidity, and a build-up of macroalgae, which is a known indicator of habitat decline (our unpublished data).

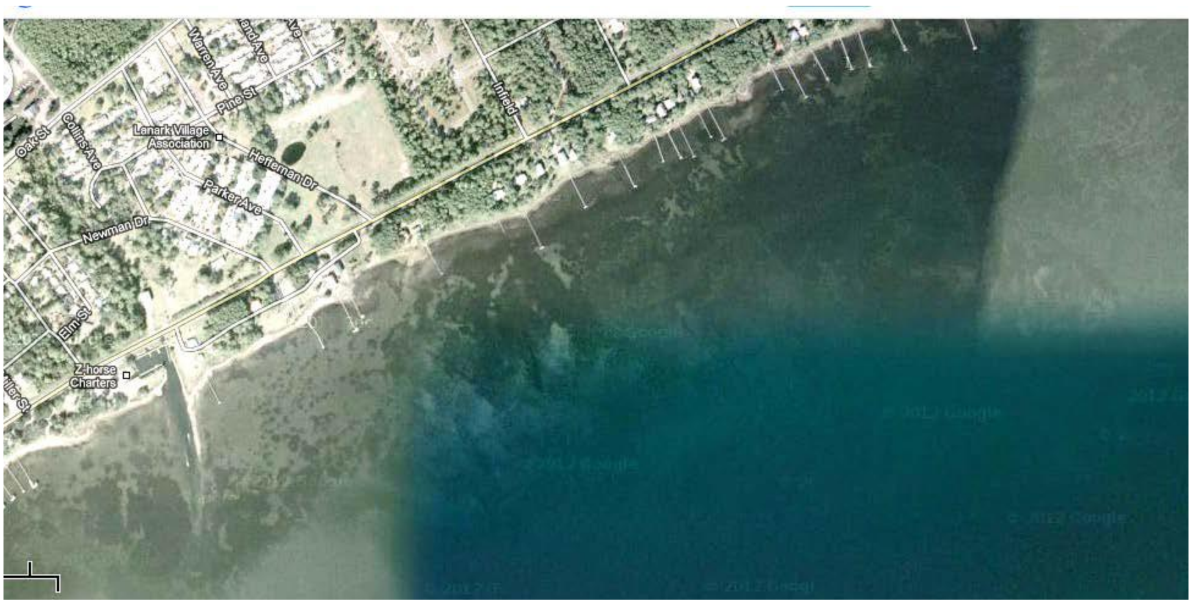


Figure 4. Aerial photo showing seagrass condition off the city of Lanark in eastern St George Sound. Near shore light areas are lacking seagrass, whereas dark areas are seagrass beds.

Many factors likely contribute to this decline, including groundwater concentrations of nutrients and sediment contaminations of toxicants. Many additional stressors are likely to occur in the future, including large-scale development proposed for St. James Island. Preliminary studies (our unpublished data) have suggested that groundwater nutrient contamination, including nitrogen and phosphorous, is a major player in this observed seagrass decline pattern. Given the national recognition of Lanark Reef as essential habitat to some of the Gulf of Mexico's most imperiled seabird and shorebird species—Brown Pelicans, American Oystercatchers, Black Skimmers, and Willets, among others—and importance as wintering habitat for rare species—Red Knots, Piping and Snowy Plovers—it is critical that we understand the causes of the decline and find means of restoring the resiliency and sustainability of this system.

We propose to elucidate the causative agents driving seagrass degradation using a combination of experimental research and monitoring. Our first step will be to determine the historical conditions of the grass beds on a spatial scale using geo-referenced aerial photographs and satellite imagery to measure temporal changes over the last few decades. We will evaluate the impact of recent events that may have caused or played part in significant declines in seagrass habitat (e.g. hypoxia, red tides, DWH oil spill). We will capitalize on the seagrass habitat quality gradient by experimentally quantifying the progression of seagrass loss and the mechanisms involved. Such an approach provides early warning signs of continuing seagrass deterioration, so that authorities could be alerted and the causes of loss could be abated.

We will monitor the system repeatedly over time for biologically (quantitative estimates of fish and invertebrate abundance, condition of foundation species, and species interactions within communities), chemically (nutrients in surface and groundwater, toxicants such as oil or pesticides, salinity, dissolved oxygen, and PAH levels in fishes) and physically (seepage rates, rainfall, turbidity, and temperature fluctuations) important factors over a range of seagrass conditions to evaluate cause and effect. The presence of oil and high PAH levels will indicate any direct effects that DWH oil and dispersants had on this pattern of declining seagrass beds. Such monitoring will provide estimates of natural variation which could also be used to compare with catastrophic events such as future oil spills and hurricanes, thus strengthening our confidence in the impacts of such events.

The most effective approach for evaluating environmental effects is the **BACI (Before, After, Control, Impact) design** (Underwood 1991; Underwood 1994). The modern form of this experimental design, which we will use, has multiple controls (sites away from the potential impact) before and after the environmental impact to account for environmental variability. Thus, 'before' data would be collected on seagrass conditions now to be compared with any future disturbances, and also to be compared with any positive trends that occur over time as a result of lessoning exposure to oil (from DWH oil spill) through microbial degradation activity of the oil.

One of the biggest limitations to BACI studies is the lack of 'before' impact data, given that disturbances most often occur unplanned, before data has been collected. Such is the case with areas and ecosystems impacted by the oil spill. It is critical to begin our studies now, prior to coastal urbanization (including anthropogenic impacts such as oil, municipal waste, toxic inputs, or natural impacts such as hurricanes), so that we can measure any changes that occur during and after development. Our proposed approach, then, is to evaluate the seagrass system off Lanark, an area that has undergone regressive changes, to understand chemical and biological early warning signs of seagrass loss. At the same time, we will seasonally monitor the seagrass beds in St. George Sound, The Apalachicola Bay, The Apalachee Bay, and Alligator Harbor systems both chemically and biologically so as to build a 'before' baseline. In a medical analogy, we propose to describe the symptoms of a disease, say a type of cancer, prior to metastases and the death of the patient so that effective treatment may be applied leading to the recovery of the patient. In the case of the seagrass ecosystem, elevated contamination of groundwater or runoff and the altered state of the seagrass community constitutes the symptoms, and the treatment would be to alert authorities and the land owners to the regressive condition so that proper controls may be put into place.

***<u>Preliminary Seagrass Data</u>*:** The dominant cause of seagrass degradation and loss on a global scale is not direct damage by dredging, propeller scars or intentional destruction, but instead by indirect input of nitrogen (N) and phosphorous (P) that enter the system either through rivers and streams or through groundwater seepage (Figure 5). These N inputs are invariably anthropogenic, associated with coastal development, urbanization, and agriculture. The seagrass beds off St. James Island are no exception. We are finding loss or degradation of seagrass beds around the Lanark area and a gradient of improvement to the east of the Lanark Marina (sites from west to east: LA0, LA1, LA2, LA3; Figure 3). Preliminary data and observation indicate that turtle grass (*Thalassia testudinum*) density has abruptly declined off Lanark Village (Figure 6), and that N and P concentrations are extraordinarily high near Lanark at LA1 (Figures 7 & 8). **What is needed now is the rigorous research approach proposed here to investigate the causes of these nutrient peaks and seagrass losses, and to investigate other potential causes of the regressive pattern in these seagrass systems.**

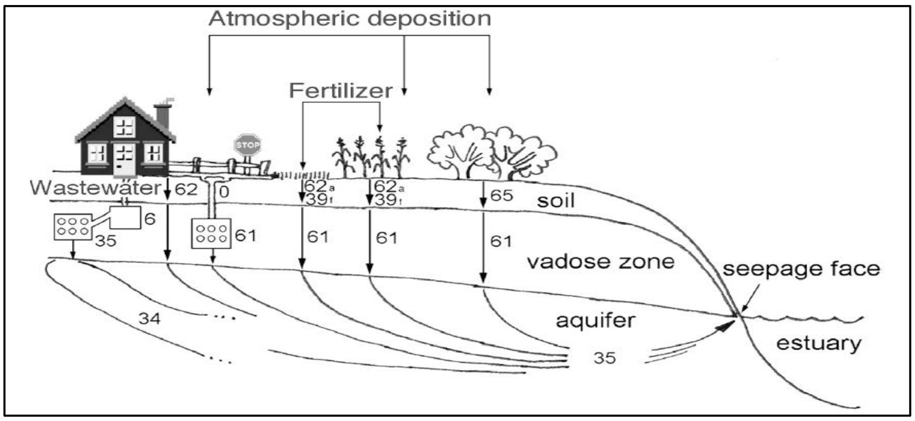


Figure 5. Processes leading to the loss of seagrass habitat. Deposition of nitrogenous compounds in upland areas results in the movement into the estuary and seagrass system via groundwater. Soil in most coastal zones is composed of sand. Water drains through the sand until it reaches the impermeable limestone layer, then it moves laterally to the estuary. It then enters the surface waters in the shallow coastal zone as submarine seepage water, carrying with it any nutrients or other pollutants picked up along the way. If seagrass beds are in this zone, then seagrass beds are impacted. Figure from Bowen and Valiela (2001).

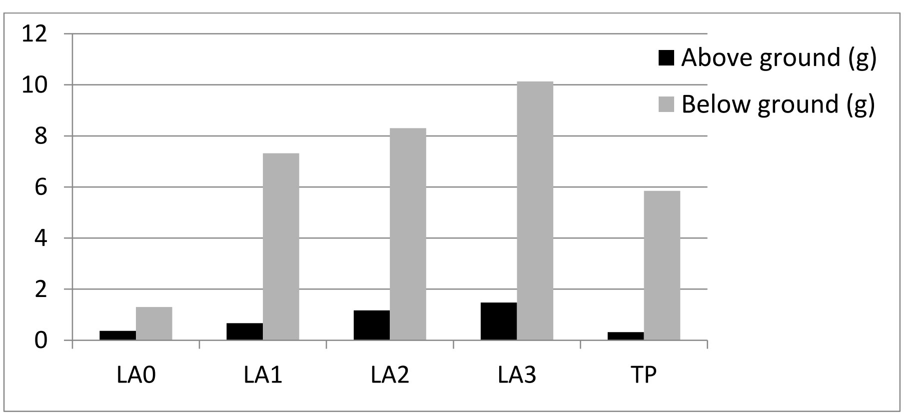
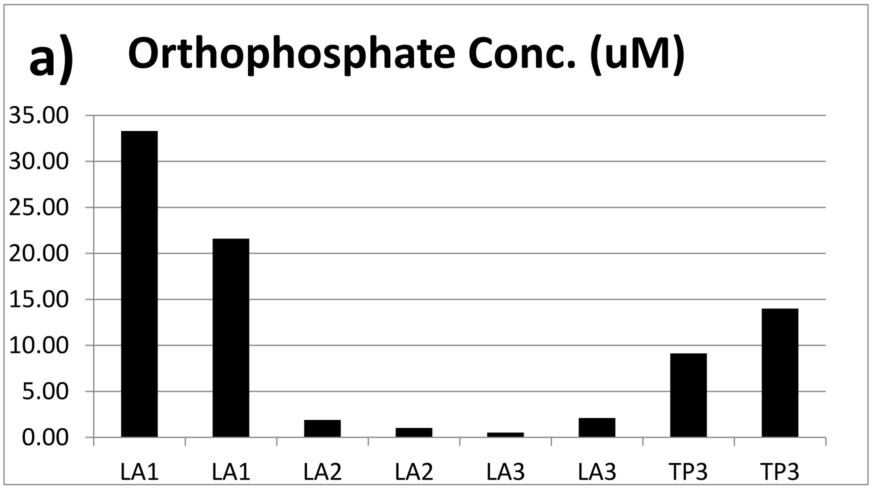


Figure 6. Seagrass biomass density (grams per core [7 in^2]), below and above ground, in the area near Lanark. Stations LA0 to LA3 are at 1/2 nautical mile intervals. TP is a reference station near Turkey Point, but it consists mostly of manatee grass (*Syringodium filiforme*) which has thin leaves and rhizomes,

rather than Turtle grass (*Thalassia testudinum*) which has wide leaves and thick rhizomes, so is not comparable in this context.



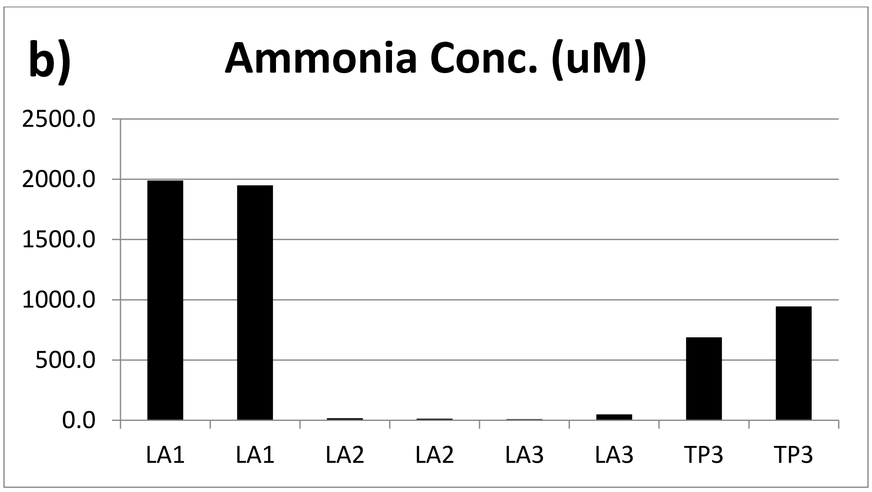


Figure 8. Relative levels of (a) orthophosphate (micromoles per liter), and (b) ammonia (micromoles per liter) in submarine seepage water (groundwater source) off Lanark (LA) and Turkey Point (TP). Note that duplicate water samples were taken at each site.

<u>Nutrient influence on seagrass health:</u>

<u>Seagrass sampling</u>:

For this work, we will sample multiple impacted and multiple control sites (observably non-impacted reference sites) of similar characteristics: depth, physical-chemical, distance from shore. Sampling will be done seasonally to evaluate seagrass community changes and seagrass die-back during the winter. Seagrass will be collected at each site using core tubes to (1) sample both above- and below-ground biomass, blade and rhizome characteristics, (2) measure productivity at field levels of N and P, (3) measure the proportional abundance of the three common seagrass species found in these seagrass beds—turtle grass (*Thalassia testudinum*), manatee grass (*Syringodium filiforme*), and shoal grass (*Halodule beaudettei*), and (4) measure stable isotope patterns (^{15}N and ^{13}C) of seagrass along the gradient of habitat regression.

Historic photographs and satellite imagery will be used to measure temporal patterns of change in the seagrass system. We will look for correlations between measured nutrient input and patterns of seagrass decline.

Macroalgae sampling:

Macroalgae is a known indicator of habitat decline, and overabundance is often caused by eutrophication from anthropogenic sources (Figure 9). If abundant, macroalgae may reduce seagrass productivity by out-competing the seagrass for both nutrients and light. The abundance of macroalgae will be determined by measuring the volume of wet algae collected during standardized 2-minute trawls. Stable isotope patterns will be determined for macroalgae over the area of study. High ^{15}N is associated with human sewage pollution; very low ^{15}N levels are associated with inorganic fertilizer contamination.

Figure 8. Macroalgae from a recent sample (June 2012) taken from LA0 seagrass station near Lanark Marina. Such large quantities of macroalgae are characteristic of eutrophic (hyper-nitrified) systems. There were very few fishes and invertebrates associated with the macroalgae

Water sampling:

In order to collect the full spectrum of major nutrient sources to the seagrass beds, three types of water samples will be collected at each sampling station: (1) **surface waters** from the top and bottom of the water column, (2) **seepage water** (collected using seepage meters that are set into the seafloor), and (3) **groundwater** (collected using wells driven into the seafloor). Seepage flow rate will also be measured to determine the relationship between rate of discharge and distribution of seagrass beds.

Water samples will be sent to the analytical lab at UC Santa Barbara Marine Science Institute (http://www.msi.ucsb.edu/services/analytical-lab/seawater-nutrients-fia) for analysis of N and P compounds and any toxic compounds we suspect from upland sources. A flow injection analysis (FIA) system is used to measure various N and P compound concentrations of interest (e.g. nitrates, nitrites, ammonia, orthophosphate).

Epiphytes (e.g. microalgae):

Epiphytes attach directly to seagrass blades and form the base of many seagrass community food webs. Like seagrass, epiphytes are influenced by many biotic and abiotic factors, including light intensity, season, nutrients, and pollutants. Although they serve a positive role in the community at normal abundance levels, when elevated by excess nutrient input they can block sunlight from reaching the seagrass. In part, increased abundance under these conditions can be caused by loss of grazers (small shrimp, crabs and other crustaceans) from the system. Therefore, epiphyte biomass and grazer abundance may be important measures of ecosystem health. Epiphyte load will be measured by calculating seagrass weight before and after epiphyte removal. Epiphytes are bound to the grass blades by calcium carbonate—application of a dilute phosphoric acid solution dissolves the carbonate and releases the epiphytes.

***Finding the source of nutrient pollution*:** Stable isotopes such as nitrogen isotope ratios, $\delta^{15}N$, are good indicators of sewage stress and can be used to discriminate among anthropogenic N inputs to marine ecosystems, such as sewage contamination (Kendall, Elliott & Wankel 2007; Risk *et al.* 2009). Using isotope ratio mass spectrometry (IRMS) at the Florida State University National High Magnetic Field Laboratory, we will analyze submarine groundwater, sediment, seagrass, and macroalgae, collected throughout our sampling region from Apalachicola Bay to Apalachee Bay, to determine potential sources of pollution. Wastewater nutrients (e.g. from septic systems, sewage treatment plants) are typically enriched with $\delta^{15}N$, while synthetic fertilizer wastes are depleted in $\delta^{15}N$ values—indicative of golf course or lawn fertilizer. $\delta^{15}N$ signatures from seagrass and macroalgae, for example, have been used as excellent indicators of sewage input, and results have led to enhanced efforts to reduce N input and restore ecosystems (Costanzo *et al.* 2001; Savage & Elmgren 2004). If we detect high levels of pollution and determine the sources early enough, we can work with the community to reduce contamination, prevent further destruction of our seagrass beds and loss of local resources provided by these valuable habitats, and effectively **restore Franklin County seagrass beds**.

***Oil and dispersant effects on seagrass*:** Oil and dispersant effects are known to have independently high deleterious effects on vegetation such as seagrass, and together they can have an even greater impact on benthic communities than the oil alone (DeLaune *et al.* 1984). Dispersants can actually enhance the uptake of oil 1.5 times by seagrasses, and contribute additional toxic effects to that of the spilled oil (Baca & Getter 1984). Although it was determined that most of the oil from the DWH event that reached the coast was extensively weathered (Reddy *et al.* 2012), meaning that there was a lessened opportunity for damage compared to the initial crude oil, toxic effects of the oil may still have caused reductions in relative abundance of benthic species (i.e. vegetation, invertebrates, fishes). Acute reduction in benthic fauna (i.e. mortality or avoidance) can be caused by direct effects of the oil, or by indirect effects, including reduced oxygen availability (Sanders *et al.* 1980; Mendelssohn *et al.* 2012).

The amount and condition of oil that reaches vegetated areas, including seagrass beds and wetlands, depends primarily on the specific make-up of oil compounds, microbial activity, and environmental factors such as oxygen, nutrients including nitrogen and phosphorus, salinity, and pH (Mendelssohn et al. 2012). Although direct effects of crude oil contamination are difficult to measure, we can correlate differences found across seagrass gradients with the presence/absence of oil contamination. Sublethal leaf discoloration is one observable symptom that has been linked to these toxic effects, thus providing a reference for where to sample. Baca and Getter (1984) showed that yellowing and browning of turtle grass leaves could be symptomatic of crude oil and dispersant toxicity exposure, and that percentage of green leaves could be a valid measure of healthy conditions useful in detecting possible damage from such toxicants.

Sediment samples will be collected using sediment cores— these samples will be used to test for the presence of oil. The presence of oil will be analytically tested using a mass spectrometer, connected to a gas chromatograph, to provide oil composition data. If oil is present in any form, it should show up in these data. Oil composition data will also provide a baseline for continued research and monitoring in the case of future oil spills.

***Combined nutrients and oil effects*:** Biogeochemically complex processes often occur as a result of oil exposure and nutrient availability. Oil degradation is strongly affected by nutrient availability, particularly of nitrogen. The addition of crude oil can stimulate nitrogen fixation and drive a system towards phosphorous limitation; therefore, the addition of nitrogen can actually increase microbially mediated hydrocarbon breakdown from the oil. However, long-term effects of this can be negative through the alteration of nutrient cycling and subsequent seagrass composition, eutrophication, and oxygen depletion. Testing for the presence of oil, as described above, is important for understanding how oil exposure may effect microbial processes of coupled nitrification-denitrification and sulfate reduction. **Because of the effects that increased nutrient levels and oil contamination can have on seagrass communities, and complexities that arise between their interactions, it is imperative that we have knowledge of oil components and nutrient levels so that remediation efforts can be made in impacted areas.**

Fish and invertebrate communities: One of the most direct ecological and economic benefits of protecting and restoring seagrass beds is the habitat it provides for faunal organisms, from small grazing invertebrates up to large groupers and other economically important fishes. For example, it has been estimated that these local seagrass beds have the capacity to produce up to 1 million gag grouper (*Mycteroperca microlepis*) per year (Koenig & Coleman 1998). We will investigate the impact that seagrass health has on community structure by quantifying abundance and diversity across the same gradient of seagrass density.

Figure 10. Rotating drop-camera with GoPro housing mount.

To quantify fish and invertebrate abundance and diversity, we will use a multi-faceted approach of: (1) surveying visually with **rotator drop cameras** (GoPro video camera continuously surveys 360° at 2 RPM with no diver present) (Figure 10), (2) trapping larger, more mobile, fish on the bottom using both large and medium **chevron traps**, and (3) 2-minute **otter trawl** tows to collect benthic fishes and large invertebrates. Fish and invertebrates caught in the traps and trawls will be identified, enumerated, measured for length and weight, and released alive in the same sites they were collected. Tissues will be sampled from select species and analyzed for toxic constituents of oil such as PAHs (polycyclic aromatic hydrocarbons).

An important component of the seagrass community is the grazers – organisms that feed on epiphytes, and provide food for larger organisms. Grazers are smaller macroinvertebrates that include caridean shrimp, amphipods, isopods, hermit crabs, and gastropods. To more accurately quantify this group of smaller macroinvertebrates (grazers) living in intimate association with the seagrass, we will sample using a suction device over an area of the seafloor approximating 80 cm^2. We will use a weighted screened-in quadrat to enclose the defined area of the bottom. To release these organisms from the substrate, an anesthetic, quinaldine, will be used prior to suctioning them from the quadrat for identification and quantification.

Estimated Project Costs: *(Describe the estimated costs of the project, including any assumptions for contingency and ongoing operations/maintenance. Identify other secured funding sources such as matching funds, in-kind contributions or state/federal dollars. In addition, if possible, complete and submit the Cost Appendix Sheet associated with this Form)*

The first year of the study will be defining the study site, hiring personnel, and identifying promising students (both high school and undergraduate) to participate in the study. In the first year, we will also test for the presence of oil. All experiments will be set up and the monitoring that will continue for all 4 years will begin. As soon as possible (by the end of the 2^{nd} year), we will have a good handle on the source of the problem in the shallow near-shore waters from Lanark to Turkey Point Shoal and will initiate communications with state agencies responsible for oversight on habitat quality. During the second year, we will initiate an expansion of the study area to include Alligator Harbor, all of St. George Sound, The Apalachicola Bay, and The Apalachee Bay. At the end of 4 years, we will have sufficient spatial and temporal coverage to understand not only the status of the seagrass beds, but the rate of change.

This seagrass experimental study will run for 4 years, but sampling installations, such as groundwater wells and sampling sites, will be permanent so that monitoring can continue into the future. After the 4-year study, seasonal monitoring will continue for years as part of the undergraduate **Field Marine Science** course taught in the spring-summer each year at the FSUCML and will involve the participation of middle school students participating in the **Florida State-funded Saturday-at-the-Sea (SATS) Program**, as detailed below.

See the attached Cost Appendix Sheet (Table 2) for details on funding needed to complete this research and restoration project.

Other Funding: *(Indicate if the project is submitted for any potential funding or if it may be used to leverage additional funding, if so, please describe the funding source [e.g. State/Federal Grants])*

This project currently has no other funding, nor has it been submitted for other potential funding.

Technical Feasibility: *(Describe the technologies involved and any relevant past experience or proven success with similar projects)*

The authors and their colleagues have expertise in all of the technologies and methods described in this proposal. We also have proven success, evident through publications and preliminary results, which have formed the basis for this proposed project. C. Koenig and F. Coleman have worked extensively in seagrass systems and have published a paper on ecosystem-level disturbances occurring in Florida Bay using stable isotope analysis (Chasar *et al.* 2005). Most of the stable isotope analysis was done in house at Florida State University. C. Koenig has experience sampling seagrass beds from past work, and during the Field Marine Science course that he leads at FSUCML annually he brings undergraduate students to local seagrass beds and teaches these techniques. Other techniques taught during this course include: submarine seepage-water sampling (we also work with colleagues in the Department of Earth, Ocean, and Atmospheric Science (EOS) at FSU who commonly use similar techniques) and fish and macro invertebrate sampling (C. Koenig and F. Coleman have completed other lab projects related to this type of sampling over many years). We send samples off for much of the chemical analyses to the University of California, Santa Barbara (UCSB). Marcus Huettal, a colleague in the EOS Department at FSU has projects currently funded by Deep-C looking at oil contamination in sediments. Another colleague, Jim Gelsleichter at University of North Florida (UNF), is currently working with us on a Deep-C project looking at PAH levels in fishes sampled post-DWH oil spill. We will utilize our colleagues' expertise in the use of techniques for analyzing oil components in sediment and PAH levels in tissues from fishes.

Conflicts or Complements to Existing Efforts: *(Describe any ongoing activities in the project implementation area, if the project is part of another plan, and why the project does or does not interfere with that work. Please consider how the project may complement existing local, regional, and state efforts/plans/objective)*

The proposed project has a very high probability of success in terms of identifying the mechanism causing seagrass decline and providing the scientific rationale for informing policy to ameliorate the problem. It will augment and complement ongoing studies conducted by the Florida Fish and Wildlife Conservation Commission (FWC) by adding greater depth and breadth of analysis. The current work conducted by FWC results in a snapshot every year or of the presence or absence of seagrass. This approach does not allow them to evaluate natural temporal and spatial changes in the quality of seagrass systems, nor does it provide opportunities to identify early warning signs in these systems or discern between the various impacts that lead to the loss. Our more detailed approach includes integrating the biological, chemical, and physical aspects to have an integrated understanding of the local ecological community and possible ecosystem-level effects of seagrass losses. The results of this work will provide the basis necessary to develop early warning for chronic impacts and will allow identification of acute impacts such as oil contamination.

Our proposed project does not conflict with any funded Phase I Early Restoration Plan projects. A project was submitted for Restore Act funding by Linda Fitzhugh at St. Andrew Bay Resource Management Association titled "Regional Community Resilience Foundation for the Eight NWFL Panhandle Counties" to build an index of bay health, but it is unclear how the author proposes to monitor water quality, submerged habitat, and species diversity. This would complement our monitoring and restoration efforts, and it is possible that we could work with the NGOs involved in this community resilience program to assist in their development of long-term monitoring techniques. We have extensive knowledge in this area of research and monitoring and would like to see similar methods extended throughout other coastal regions. **Restoration efforts cannot be proposed or enacted without knowing the source of ecosystem health decline, and our proposed research and monitoring will indicate the source/s of such decline.**

Complies with Federal, State, Local, and Tribal Laws/Regulations:

(Describe any concerns or potential conflicts)

The are no conflicts. We have complied with all regulations throughout decades of research.

Readiness for Implementation: *(Describe if the project has had any design or permitting work started or completed [attach permits or design work]. Please address any issues that may delay start to finish of the project)*

We will obtain the necessary special activity permits from the Florida Fish and Wildlife Conservation Commission, a type of permit that we have routinely had to support our research in the coastal waters throughout Florida. The FSUCML maintains an FWC permit for trawling and collecting marine organisms for educational purposes from the seagrass areas we are describing. All the proposed work here would be covered under this permit. The work will be conducted under the auspices of the Florida State Animal Care and Use Committee (ACUC) following protocols already approved for our research in the region.

Environmental Benefits: *(Describe the nature, magnitude, and timing of any environmental benefits attributable to the project. If possible, describe potential environmental performance measures [e.g. pollutant reduction]. Please address any potential environmental impacts associated with implementing or maintaining the project [e.g. loss of a habitat or conversion of habitat from one type to another during implementation])*

Ultimately, success would be measured in the protection of the productive seagrass habitat of Franklin County. Providing the necessary restoration and protection requires two elements: (1) understanding the mechanism by which seagrass beds have declined; and (2) providing oversight of restoration and protection through ongoing monitoring. Human activities can be very destructive to these sensitive habitats—seagrass loss is common in areas of urbanization—but if we understand the effects of our actions, we are much more likely to change our behavior.

Economic and Social Benefits: *(Describe the economic and social benefits including those related to the project's improved ecosystem services and any estimates on jobs created or preserved)*

The main beneficiaries of this work would be local commercial and recreational fishermen, coastal land owners, and the general public. Healthy seagrass beds result in productive fisheries, and productive fisheries means improved revenues and job preservation. The grass beds in this region function as major nursery areas for gag grouper, pink shrimp, gray snapper, black seabass, and spotted trout, and provide habitat for red drum and Gulf flounder. These species bring the highest revenues into our state. Seagrass ecosystems also provide oxygen to the water and sediment, sequester carbon, export food and nutrients to adjacent food webs (trophic subsidies), protect against erosion by stabilizing sediment, improve water clarity, dampen waves and protect shorelines, trap and recycle nutrients, and they support a rich biodiversity. This extensive list of services cannot be matched by any other ecosystem, provided the seagrass bed health is maintained through protection from destructive anthropogenic practices.

Also benefiting will be students ranging from middle school to undergraduate students who will participate in the research, and a graduate student who will form his doctorate dissertation around this project. With undergraduates for instance, students enrolled in the **Field Marine Science course** taught in the spring-summer each year at the FSUCML will help with monitoring every year. The focus of the course is teaching quantitative sampling techniques that allow the statistical determination of environmental changes, and the field sites are the seagrass beds detailed in this study. Monitoring can also be accomplished through the **Florida State-funded Saturday-at-the-Sea (SATS) Program**, a class designed to introduce middle-school children to the marine environment, especially to the high productivity and diversity of seagrass systems. These provide **STEM (Science, Technology, Engineering, and Math)-related experiences** for students early in their educational training which can have lasting effects on their interest in the conservation and management of living marine resources. Increasing student awareness ensures critical long-term success in protection of these important resources to benefit both the environment and local economy.

We will be active participants in working with local government to accomplish all of our research goals. Our approach is simple logistically, relatively inexpensive, and repeatable across sea grass habitats throughout Florida, especially in the near-pristine Big Bend regions in the northeastern Gulf of Mexico seagrass system. Ongoing monitoring is sustainable through the educational programs indicated here so that once the habitat is restored, monitoring may continue at very little cost.

Community Resilience: *(Describe if the project assists Florida's ability to anticipate, withstand, or recover from hazards or threats [e.g. hurricane preparedness, establishing living shorelines])*

Living shorelines are defined as shoreline stabilization techniques that use natural habitat elements like salt marsh and seagrass systems to enable communities to become more resilient to coastal flooding and erosion. They provide an effective alternative to "hard armoring", the common use of concrete and other man-made barricades, which is intended to protect property but can often divert flooding to another coastal area and increase erosion (http://www.georgetownclimate.org/tags/living-shorelines). Seagrass beds protect shorelines from erosion and sequester carbon and nutrients (e.g. nitrogen, phosphorous), all increasing the productivity of the beds. **Restoration efforts through mitigation of hyper-eutrophication and pollution sources will enhance the function of seagrass beds for such community resilience.**

Public Acceptance: *(Describe any known or potential public approval or opposition to the project)*

The public, especially the fishing community and the coastal land owners, would be strongly in favor of this project because they stand to benefit the most from a healthy seagrass ecosystem in terms of all the human services provided.

Additional Information you wish to provide: *(Please include any maps, designs, drawings, photos or background resources that may assist in completely and accurately understanding the project. If these are too large to attach here, please email them to: Restoration.Projects@dep.state.fl.us)*

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Absolute Abundance and Survival of Juvenile Gags in Sea Grass Beds of the Northeastern Gulf of Mexico

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Abstract.—The absolute number of individuals and survival of juvenile gags *Mycteroperca microlepis* in a 15.4-km^2 shallow sea grass meadow in St. George Sound, Florida, were estimated by the Jolly–Seber mark–recapture method. Abundance was estimated within three sampling stations (150 m × 150 m) by means of standard (150 m, 1.8 km/h) tows with a 5-m otter trawl. The average densities (95% confidence limits), based on three stations sampled every 1–2 weeks from June to mid-September 1991, were 496 (251–744), 424 (210–670), and 549 (165–1,773) juveniles/ha. Trawl capture efficiencies and the effects of sea grass habitat conditions and size of juvenile gags were estimated so that absolute numbers could be determined outside the sampling stations. We estimated absolute abundance outside the sampling stations by trawling and dividing gag captures per standard tow by trawl capture efficiency estimates. The estimated absolute abundance of juvenile gags for the entire 15.4-km^2 area was 943,615. Survival of juvenile gags in sea grass was near 100%, as estimated from trawl capture rates over the sampling period. Survival estimates were generally variable but were confounded by emigration from the sampling areas. The small-scale spatial pattern of juvenile gags was random. The results of this work provide the basis for establishing a juvenile recruitment index by providing quantitative estimates of juvenile abundance.

Fisheries managers have recently turned their attention to habitat conservation because of the strong correlation between habitat degradation and fishery declines (Dayton et al. 1995; Arthur et al. 1996; Langton et al. 1996). Habitats in estuarine environments are of particular interest because of their proximity to human activity and their clear importance as nursery grounds for a number of economically important species (Butler et al. 1995).

Gag *Mycteroperca microlepis* is one of several warm–temperate reef species (including gray snapper *Lutjanus griseus*, lane snapper *Lutjanus synagris*, hogfish *Lachnolaimus maximus*, and black sea bass *Centropristis striata*) for which the estuarine environment plays an integral part in the early life history. A major portion of the Gulf of Mexico population of gag spawns on deepwater reefs of the West Florida Shelf (WFS) in winter (Coleman et al. 1996). Their pelagic larvae are transported to coastal sea grass beds (McErlean 1963; Gilmore 1977; Mullaney and Gale 1996), where they settle as juveniles and remain until autumn, when they move to nearshore reefs, a migration referred to as egress (Keener et al. 1988; Ross and Moser 1995; this study).

The objective of this study was to quantify total recruitment by estimating the absolute abundance (number of individuals), survival, spatial pattern, and temporal patterns of settlement and egress of juvenile gags in sea grass beds of northwest Florida. Our results have important implications for developing alternative methods of stock assessment that incorporate fishery-independent (forecasting) methodologies rather than traditional fishery-dependent (hindcasting) methods.

Study Site

The sea grass beds of the northwest Florida coast form the only structurally complex habitat in the area. They are surrounded primarily by mud and sand, and there are no natural inshore reefs. Three sampling sites were set up within two adjacent sea grass beds located just off the Florida State University Marine Laboratory (FSUML) in St. George Sound in the northeastern Gulf of Mexico (Figure 1): stations 1 and 2 in the eastern sea grass bed and station 3 in the western sea grass bed. The criteria for selecting the sampling sites were (1) that they were located in shallow (1–2-m), uniform (not patchy) sea grass beds; (2) that they had different sea grass compositions and characteristics; and (3) that juvenile gags were present. Each of the three sampling sites was marked by buoys anchored at the corners of a square area (150 m on a side) and subdivided into six lanes (each 25 m × 150 m; Figure 1). The middle two lanes (lanes 3 and 4) were designated the mark–recapture (M–R) lanes, and the outer lanes (lanes 1, 2, 5, and 6) were designated reference (RF) lanes.

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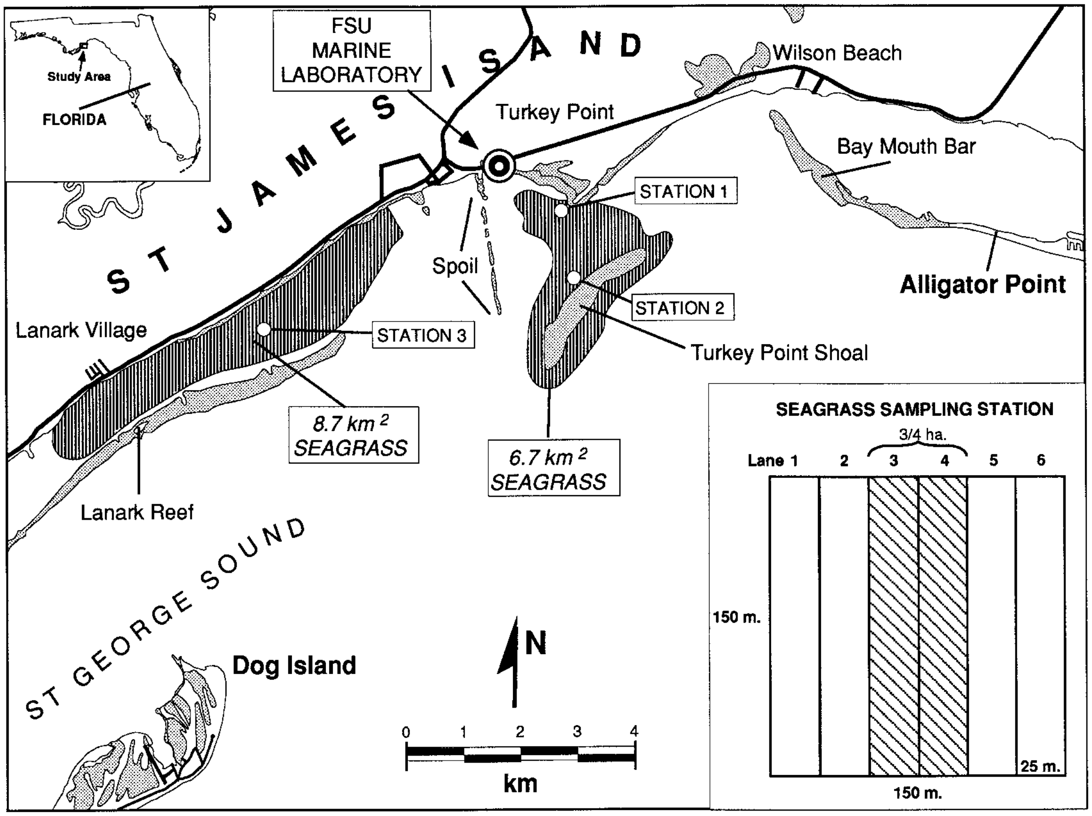

FIGURE 1.—Sea grass sampling areas (hatched area) and experimental design. Station locations are located off the Florida State University (FSU) Marine Laboratory in St. George Sound, on the northwest coast of Florida. The eastern sea grass bed was 6.7 km^2; the western sea grass bed was 8.7 km^2; lightly shaded areas are sand bars.

Methods

Absolute Abundance Determination

We used the Jolly–Seber mark–recapture method (Krebs 1989) to determine the densities of juvenile gags within the sampling sites. This method deals specifically with open populations, provides information on survival rates, and provides the opportunity to examine growth rates, movement patterns, and small-scale spatial patterns of individuals. We then used site abundance estimates to calculate capture efficiencies, which were then applied to the total sea grass area to develop abundance estimates of juvenile gags in two sea grass beds in St. George Sound.

Sampling protocol.—Juveniles were collected by standardized tow (1.8 km/h for 150 m) completed in 5 min with a 5-m otter trawl outfitted with a 3-mm-mesh liner in the bag end. The most efficient tow speed was determined by direct underwater observation; faster tows caused the net to ride off the bottom, and slower tows allowed fish to escape. We determined the length of tow by balancing the collection of a sufficient number of gags for the mark–recapture study with sample size; very large sample sizes affect the functions of the trawl. Tows within lanes were randomized by the designation of 10 2.5-m-wide tracks (unmarked) within each 25-m-wide lane and the use of a random numbers table to select tracks. It was nearly impossible to tow precisely within tracks, but this procedure minimized unconscious sampling bias.

During each sampling event, trawls were deployed 16 times in the M–R lanes (eight tows per lane) and a single time in each of the four RF lanes. The RF lanes served as controls for the effects of trawling on the abundance estimates; trawling in those lanes was kept to a minimum. At each sampling time, water temperature and salinity were measured. Dissolved oxygen was measured several times during the sampling period just above the sediment surface and just before dawn, the place and time at which minimum dissolved oxygen concentrations would be expected. Sampling was not scheduled at extreme low tides (when sampling depth was <0.7 m), because the propeller wash from the sampling boat could potentially disrupt sea grass and benthic organisms and probably influence juvenile gag catch rates.

Juvenile gags captured from the M–R lanes were immediately placed in aerated coolers, transported to the FSUML, and transferred to labeled plastic mesh baskets immersed in a 5,000-L tank supplied with running seawater. All RF-lane captures were returned immediately to their respective lanes. All first-time-captured fish from each sampling time in the M–R lanes were measured for standard lengths (mm, SL) and marked on the left side with a three-digit number specific to the individual and the sampling time. Marks were made by freeze-branding with devices made of 8–10-gauge copper wire that were cooled by liquid nitrogen. We avoided freeze damage to muscle tissues by limiting branding time to 3 or 4 s. First-time captures were branded, and their brand numbers and standard lengths recorded. Brand numbers of previously branded fish were recorded. All juveniles captured in the M–R lanes were released within 24 h in the lanes from which they were captured. The release procedure involved driving the boat in a zigzag fashion across the lanes and releasing fish haphazardly along the way. No fish died or were injured during any of these procedures.

All associated sea grass fauna, primarily decapods and fishes, were returned to the sampling lane from which they were captured. Many of the smaller species (e.g., caridean shrimp) did not survive the capture and release process.

The Jolly–Seber method assumes that (1) every individual in the sampling area, whether marked or unmarked, has the same probability of being caught in every sample; (2) every marked individual has the same probability of surviving from one sampling time to the next; (3) marks are permanent and legible throughout the sampling period; and (4) sampling time is small relative to intervals between samples.

A drawback of this method is that assumptions 1, 2, and 3 cannot be tested until the study is completed. Using the Leslie, Chitty, and Chitty test (Krebs 1989), we were able to test and support the assumption of equal catchability (assumption 1) within the marked population of station 2. Within the marked population, this test compares the estimated number of fish joining the population with the known number joining. For station 2 the difference was only 2%, so we assumed equal catchability. We tested the assumption of equal survival (assumption 2) by holding 40 marked fish in a 2,000-L laboratory tank for over 6 months. None died, and neither laboratory-held fish nor field-recaptured fish showed any evidence of irritation or infection around the brand marks. Marks were typically legible throughout the sampling period of June to early October 1991 (assumption 3); recaptured fish with fading brands were remarked with the same number. The fourth assumption was easily met because sampling time in this study was 1–2 d, and the interval between samples was 1–2 weeks.

Capture efficiency.—We calculated trawl capture efficiencies (E) for each station by dividing the mean number of juveniles captured per standard tow by their density, as determined by the Jolly–Seber method. We then determined juvenile density in sea grass areas outside of the sampling stations from the following relationship: density $= (10{,}000/S)\ (C/E)$, where $10{,}000 =$ the conversion factor for meters to hectares, $S =$ sample area (m^2) per standard tow, $C =$ the mean number of juveniles captured per standard tow; and $E =$ capture efficiency estimate.

Juvenile density was then multiplied by total sea grass area of interest (ha) to yield the absolute abundance or total number of juveniles in the area of interest (Figure 1).

To evaluate the effect of sea grass characteristics on capture efficiency, we examined sea grass species composition, density (number of blades per unit area), and blade length (mm) at the three sampling sites in August when sea grass productivity was at a maximum. We chose 18 625-cm^2 quadrats from each site by picking three random coordinate sets within each lane to maximize dispersion and still retain randomness. Square quadrat frames were placed against the sediment, and all grass within the quadrat area was cut off at sediment level. The effect of sea grass density on capture efficiency was determined by examination of the area sampled during a standard tow.

The influence of juvenile gag size on capture efficiency over the size range used in this study (approximately 50–200 mm SL) was determined by comparison of the density estimates in the M–R lanes with those in the RF lanes over the sampling period. If both remained relatively constant over the sampling period, the influence of size on capture efficiency was considered negligible.

Spatial Patterns

We evaluated the spatial patterns (i.e., random, clumped, or regular) of juvenile gags in the sea grass beds by means of the variance-to-mean ratio of captures per standard tow and comparison of the sampling distribution with the Poisson distribution (Krebs 1989).

Survival.—Probability of survival from sample time t to $t + 1$ was equal to the size of the marked population at the beginning of sample $t + 1$ divided by the size of the marked population at the end of sample t (Krebs 1989). Survival as used here entails both remaining alive and remaining in the study area. That is, marked fish that leave the study area are counted as deaths. Thus, high survival estimates indicated both low mortality and low emigration.

Survival was also determined by regression of $\log_e$-transformed data for catch per unit effort (CPUE) on time (e.g., see Iles and Beverton 1991), the slope of which estimates mortality (Z); the power (β) of this analysis was estimated (Peterman 1990).

Times of Settlement and Egress

We determined settlement times for juvenile gags by counting presumed daily growth rings of the lapillus otolith following procedures in Brothers and McFarland (1981) and Keener et al. (1988). We did not directly validate daily incremental growth or settlement marks but instead relied upon the rationale of Keener et al. (1988) and upon the distribution of sizes over time, which indicated a single discrete cohort. There were three independent readers on all otoliths, and E. Brothers (EFS Consultants, personal communication) confirmed our counts on a sample of 20 otoliths. We determined egress times by tracking changes in density of juveniles over the estuary-dependent phase.

Results

Environmental conditions in the sea grass beds during this study remained relatively constant. Dissolved oxygen concentrations never fell below 5.0 mg/L during the sampling period. At all stations, salinity ranged from 26 to 32‰, and water temperature ranged from 28 to 31.5°C before early October. The first major cold front of the season, which occurred 4–6 October 1991, decreased water temperatures from 29 to 19–20°C at all stations.

Absolute Abundance

Repeated tows within a sampling time caused diminished catch per tow in the M–R lanes (Figure 2). Because of this decline, only the first three tows in each M–R lane were used to estimate relative abundance (captures per tow) in those lanes.

Mean capture of juvenile gags per standard tow in the M–R lanes was uniform early in the sampling period but declined during later sampling events at all three stations (Figure 3). Mean captures per tow in the RF lanes showed no such trend ($\alpha = 0.05$; Table 1). The significant declines in the M–R lanes apparently resulted from the intensity of the sampling effort in those lanes, even though all captured gags were returned to capture lanes. Thus, estimates based on M–R lanes underestimate the gag density in the general area, increasingly so from about mid-July to the end of the sampling period.

Although the mechanism for the progressive decline in the M–R lanes is unknown, our concern was that the apparent decline may have been caused by a movement of gags out of the heavily trawled M–R lanes and into the lightly trawled RF lanes. This occurrence, however, would be reflected in the progressive increase in catch in the RF lanes adjacent to the M–R lanes (lanes 2 and 5) over that in the RF lanes far from the M–R lanes (lanes 1 and 6). Such an increase would be reflected in greater slopes of regressions of CPUEs over the sampling period for lanes 2 and 5 than for lanes 1 and 6. The slopes for adjacent and nonadjacent RF-lane regressions are as follows: station 1, -0.0218 and -0.0199; station 2, -0.094 and -0.0229; and station 3, 0.0061 and 0.0103. The patterns of decline (at stations 1 and 2) and increase (at station 3) indicate no consistent increase in RF lanes adjacent to M–R lanes, so refugees from M–R lanes are unlikely to have inflated catches in RF lanes. Therefore, we corrected density estimates for July, August, and September by dividing the RF-lane mean captures per tow (averaged over the sampling period) by the M–R-lane mean captures per tow (estimated from M–R-lane regression equations at the specific sampling time). The resulting factor was then multiplied by the density estimate of the same sampling time to yield a corrected density estimate (Table 2).

There were no significant differences in the relative abundances of gags among lanes at any station sampled ($\alpha = 0.05$). Homogeneity of juvenile distribution within sampling sites was determined a posteriori. Interlane differences for each station were compared for the first part of the sampling period only (i.e., before the divergence occurred).

Because captures per standard tow were constant in the RF lanes over the sampling period, we

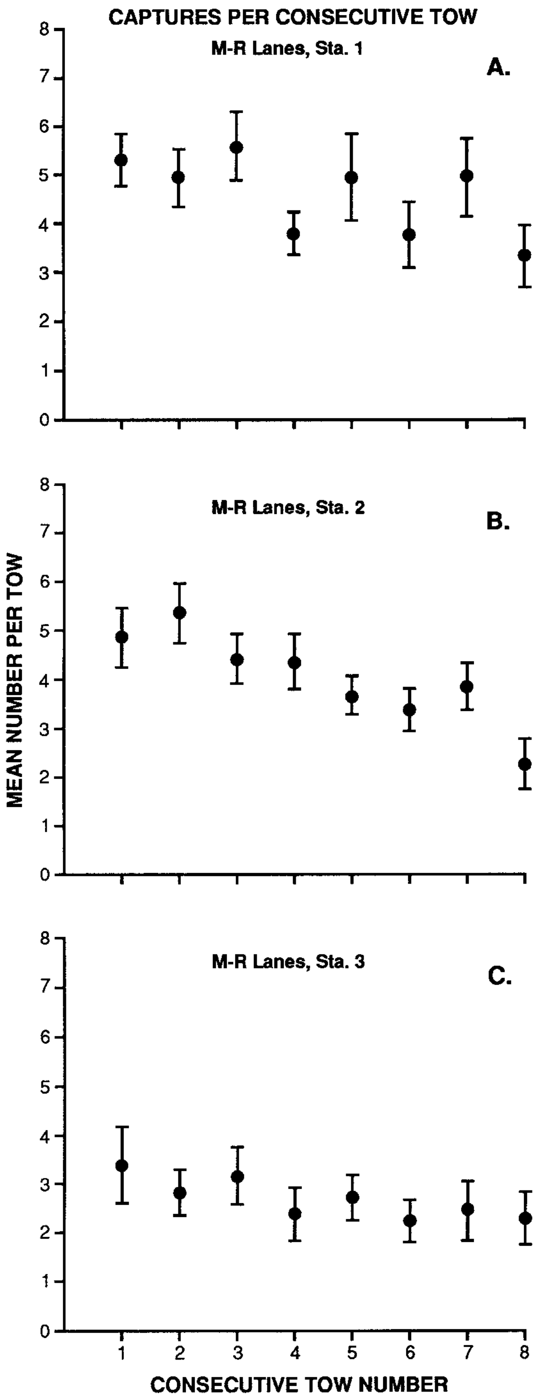

FIGURE 2.—Number of juvenile gags captured in mark–recapture (M–R) lanes in consecutive tows by station (Sta.), averaged over the sampling period. Vertical lines represent ±SE.

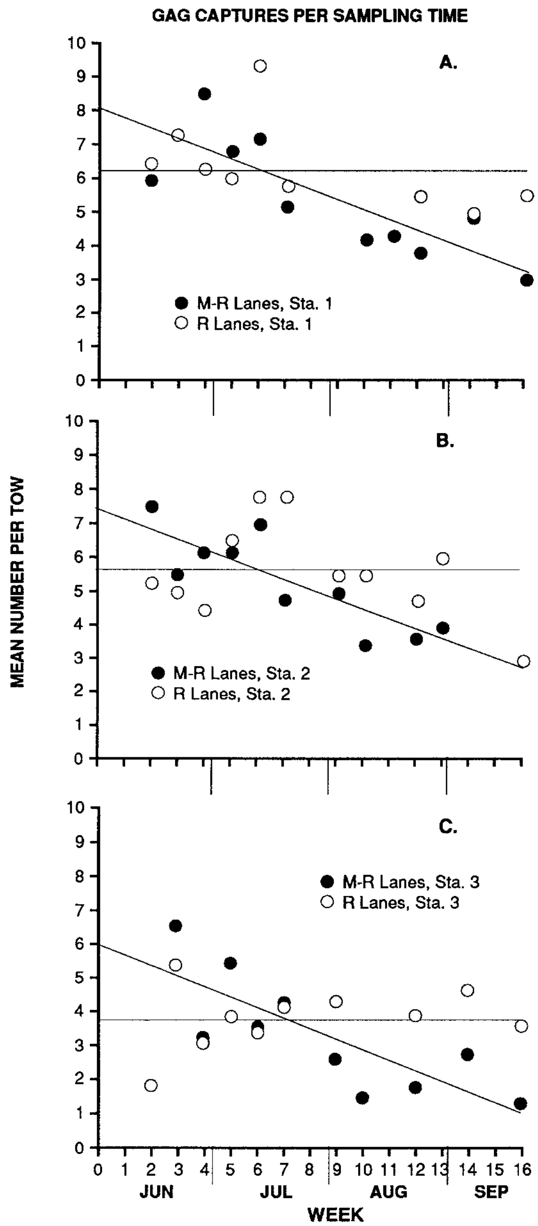

FIGURE 3.—Mean number of gags captured per standard tow in mark–recapture (M–R) lanes (filled circles) and reference lanes (R, open circles) by station (sta.), over the sampling period. Regression equations and significance levels are given in Table 1.

assumed low natural mortality and constant density in the general area. Therefore, we averaged the most precise estimates of density (i.e., those with the smallest confidence intervals) to estimate the overall density at each station (5, 9, and 20 August for station 1; 15 July and 1 August for

TABLE 1.—Linear regressions of juvenile gag captures per standard tow in St. George Sound, Florida, over the sampling period June–September 1991. Each of three sampling stations contained mark–recapture (M–R) and reference (RF) lanes; b = slope, a = intercept.

Station	Lane	b	a	r	df	F	P	Mean captures/tow[a] Total	Jun	Jul	Aug	Sep
1	M–R	−0.286	7.811	0.51	56	19.7	<0.001	5.28	6.34	5.95	4.69	3.45
	RF	−0.142	7.451	0.25	33	2.14	0.153	6.34	6.34	6.34	6.34	6.34
2	M–R	−0.299	7.521	0.52	56	20.6	<0.001	4.93	5.59	5.58	4.26	2.96
	RF	−0.110	6.462	0.19	40	1.50	0.228	5.59	5.59	5.59	5.59	5.59
3	M–R	−0.240	5.079	0.45	55	14.0	<0.001	2.98	3.69	3.52	2.46	1.42
	RF	−0.049	3.297	0.09	43	0.37	0.547	3.69	3.69	3.69	3.69	3.69

[a] The M–R values for July, August, and September were calculated from regression equations as midpoints of months. June M–R values were made equivalent to those of reference lanes.

station 2; and 31 July and 2 August for station 3). On the basis of these selections, the average density estimates for stations 1, 2, and 3 were very similar: 496, 424, and 549 fish/ha, respectively (Table 3).

We then determined absolute abundance in the sea grass beds surrounding the sampling sites—the eastern bed, containing stations 1 and 2, and the western bed, containing station 3. The total number of juvenile gags in the eastern sea grass bed was 335,485 (95% confidence limits = 156,780 and 478,380) (Table 4). The absolute

TABLE 2.—Mark–recapture estimates of density and survival for juvenile gags from stations 1–3, St. George Sound, Florida, in 1991, according to the Jolly–Seber method. Estimates of the number of gags/ha and 95% confidence limits are adjusted to compensate for the trawling effect on the mark–recapture lanes.

Sample date	Proportion of population marked	Number of gags/ha	Probability of survival	95% confidence limits[a] for: Gags/ha Lower	Gags/ha Upper	Survival Lower	Survival Upper
Station 1							
26 Jun			1.000			0.545	1.000
1 Jul	0.075	1,132	0.338	398	2,814	0.154	0.794
8 Jul	0.153	497	0.466	218	1,081	0.226	1.000
20 Jul	0.086	1,146	0.309	393	2,817	0.180	0.553
5 Aug	0.169	378[b]	1.000	186	613	0.660	1.000
9 Aug	0.258	587[b]	0.818	292	860	0.490	1.000
20 Aug	0.406	524[b]	1.000	274	758	0.506	1.000
4 Sep	0.458	1,109		340	3,324		
16 Sep	0.533						
Station 2							
25 Jun			0.587			0.275	1.000
1 Jul	0.065	449	0.767	184	1,166	0.437	1.000
9 Jul	0.138	799	0.409	341	1,458	0.240	0.727
15 Jul	0.242	364[b]	0.598	186	561	0.376	0.979
1 Aug	0.218	484[b]	1.000	233	780	0.838	1.000
5 Aug	0.256	1,052	0.578	483	1,675	0.294	1.000
19 Aug	0.216	1,168	0.871	515	2,004	0.317	1.000
27 Aug	0.348	1,038		340	2,730		
16 Sep	0.346						
Station 3							
24 Jun			0.421			0.141	1.000
2 Jul	0.062	593	0.702	164	2,506	0.251	1.000
10 Jul	0.086	959	0.617	262	3,374	0.210	1.000
18 Jul	0.078	1,319	0.361	381	4,266	0.127	1.000
31 Jul	0.182	475[b]	0.693	147	1,474	0.219	1.000
2 Aug	0.146	623[b]	0.841	183	2,073	0.180	1.000
22 Aug	0.214	783		169	4,205		
4 Sep	0.269						

[a] Determined by the method of Manly (1984).
[b] Best estimates averaged for overall density estimates.

TABLE 3.—Summary of abundance and capture efficiency estimates for juvenile gags captured from June to mid-September 1991 in sea grass beds in St. George Sound, Florida, corrected as necessary.

Location	Area sampled per tow (m^2)	Mean number of gags: Per hectare[a]	Area sampled/tow	Captured/tow[b]	Capture efficiency (%)
Station 1	300	496	14.9	6.3	42.6
Station 2	300	424	12.7	5.6	43.9
Station 3	225	549	12.4	3.7	29.9

[a] Average of selected estimates from mark–recapture lanes.
[b] Determined from reference lanes only.

abundance of juvenile gags in the western sea grass bed was 608,130 (95% confidence limits = 143,550 and 1,542,510). The combined total was therefore 943,615 (95% confidence limits = 150,565 and 1,010,445). In general, juvenile gag densities in the shallow sea grass beds in St. George Sound in 1991 were about 500 fish/ha or about 50,000 fish/km^2. The high variability in abundances in the western sea grass bed was attributed to sea grass characteristics; longer sea grass blades at that site caused lower trawl sample size and lower capture efficiencies.

Movement and Survival Patterns

The mobility of juvenile gags in the grass beds during the sampling period was low. Marked juveniles were caught repeatedly over the sampling period in the same relatively small M–R sampling area (0.75 ha) even though sampling intervals were from 1 to 2 weeks. Low mobility is also evident from high survival estimates, which specifically indicate low rates of emigration of marked fish.

Survival of juvenile gags was assumed to be high because of their highly cryptic behavior and coloration. The assumption of high survival was supported statistically and by the fact that very few (0.1%; 2 of 1,500 juveniles) were found in poor or injured condition during the sampling period. Even though survival was typically variable among sampling times in M–R lanes (Table 2), a high probability of survival, because it was confounded with emigration, indicated low mortality and low emigration rates. Regression analyses of CPUE over the sampling period suggest low mortality in RF lanes. The daily *Z*s (including emigration) for stations 1, 2, and 3 were −0.00324, −0.00321, and 0.00272, respectively, but to define the limits of the mortality, we performed a power test on the regression of $\log_e$ CPUE on date of capture (≈age of cohort). With a power of 95%, mortality was less than 1%/d at all stations.

Spatial Distribution

The spatial pattern of juvenile gags was not significantly different from random within any sea grass locality sampled. The index of dispersion (I = variance/mean; Krebs 1989) for RF-lane captures per tow over the sampling period indicated no significant departure from the assumption of randomness ($\alpha = 0.05$). The indices of dispersion at stations 1, 2, and 3 were 1.20, 1.18, and 1.48, respectively. Sample frequency distributions of reference lanes were also compared with theoretical Poisson distributions by means of the χ^2 test statistic and found not to differ significantly (station 1, $\chi^2 = 15.1$, df = 14; station 2, $\chi^2 = 12.2$, df = 11; station 3, $\chi^2 = 13.4$, df = 8). Juvenile gags also showed random spatial patterns ($\alpha = 0.05$) in other sea grass areas, such as along the northwestern end of Turkey Point Shoal ($I = 1.13$), just north of station 2 ($I = 1.64$), and just south of station 1 ($I = 1.31$). Significant departure from randomness was not observed at any sampling locality.

Capture Efficiencies

Grass beds contained three species of sea grass—shoalgrass *Halodule wrightii*, manatee

TABLE 4.—Estimates of abundance of juvenile gags in selected sea grass beds in St. George Sound, Florida, based on random sample tows taken outside designated sampling sites.

Sea grass location	Number of tows	Area sampled per tow (m^2)	Capture efficiency (%)	Gags captured/tow	Gags/ha	Sea grass area (ha)	Gags in sea grass area
Eastern beds							
Station 1: SE, SW	30	300	42.6	3.7	290	286	82,940
Station 2: SW	25	225	29.9	8.0	1,185	50	59,250
Station 3: NE	21	300	43.9	7.6	577	335	193,295
Total							335,485
Western beds							
Station 3: N, E, S, W	7	225	29.9	4.7	699	870	608,130

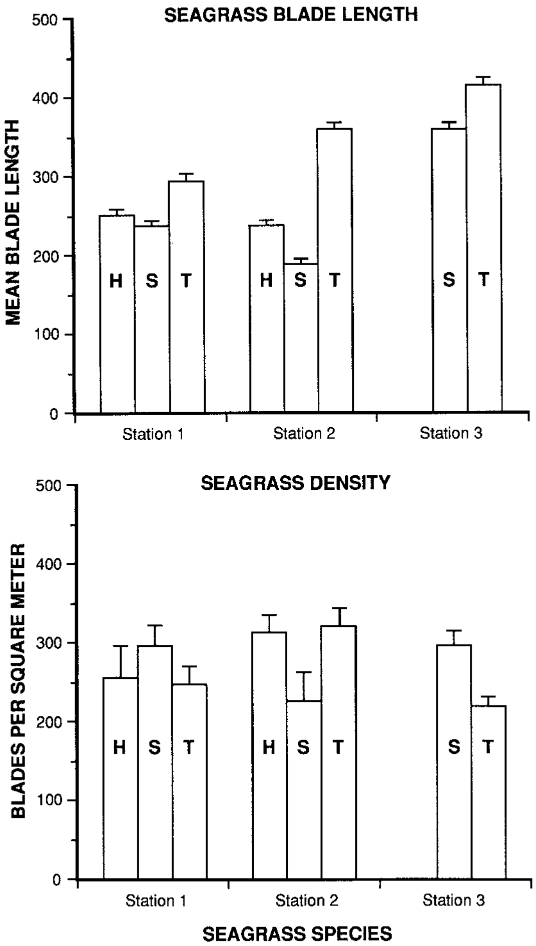

FIGURE 4.—Mean (±SE) sea grass blade length (mm) and density (blades/m^2) for three species of sea grass at three stations in St. George Sound, Florida. Sea grass species are shoalgrass *Halodule wrightii* (H), manatee grass *Syringodium filiforme* (S), and turtle grass *Thalassia testudinum* (T).

grass *Syringodium filiforme,* and turtle grass *Thalassia testudinum.* The area of the sea grass bed sampled, as determined by trawl width, was influenced by biomass captured and grass density. For example, the opening of the trawl was wide at the beginning of a tow but narrowed as the tow progressed and the tail bag filled. The trawl mouth also narrowed and the trawl rode higher off the bottom in grass beds with longer grass blades because the blades produced a greater drag on the lead line of the towed net. The average trawl mouth width at stations 1 and 2 was 2 m and at station 3, 1.5 m. Thus, a smaller area was sampled per standard tow at station 3 because of the significantly longer sea grass blades (Figure 4). The effect of sea grass density on sample size and net efficiency could not be determined because densities were similar at all three stations (Figure 4). The areas sampled at stations 1 and 2 were 300 m^2 (2-m mean trawl mouth width × 150-m tow length); the area sampled at station 3 was 225 m^2 (1.5 m × 150 m).

Timing of Settlement and Egress

Pelagic juvenile gags recruited to the sea grass habitat as a cohort in April and May after a 40–43 d planktonic stage (Figure 5). A major egress of juvenile gags to offshore reefs took place concurrent with the October cold front (Figure 6). This conclusion is supported by the appearance of first-year juveniles on offshore reefs and in offshore crab traps within several weeks after this event (our own personal observation).

Discussion

Attempts to predict stock size and recruitment variability form the core of fisheries research (Hilborn and Walters 1992). The vast majority of these attempts rely on hindcasting models based primarily on historical catch records, but the unpredictable nature of recruitment makes historical records largely unreliable for predicting future fishery abundances. Because year-class strength is probably established in juvenile stages for many species (Sissenwine 1984), quantitative juvenile abundance estimates provide promising alternatives to traditional management models that could vastly improve our current ability to assess stocks. De Lafontaine et al. (1992) provide examples and a rationale for the use of juvenile forecasting models.

Three general areas must be considered in the development of a juvenile-based forecasting model for fishery recruitment: (1) juvenile habitat, (2) juvenile sampling, and (3) offshore movement and survival. Juvenile habitat includes aspects of habitat preference and relationship (physical, chemical, and biological), seasonal habitat distribution and areal coverage, and habitat quality effects on juvenile growth and survival. Sampling considerations are directly related to habitat characteristics that can affect sampling design and capture efficiency. Offshore movement and survival patterns can directly influence the accuracy of the forecasting model by affecting the regional connection between abundances of gags in sea grass and subsequent abundances in the offshore fishery. In the following discussions, each of these three components is addressed relative to juvenile gags.

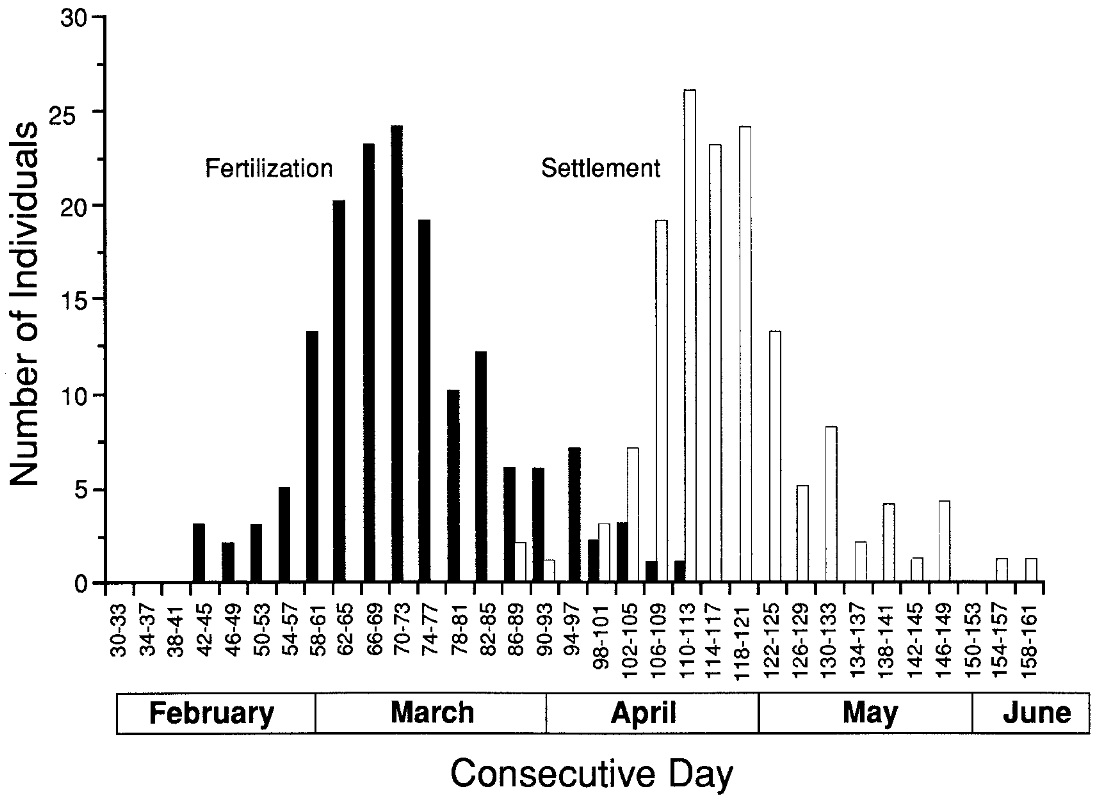

FIGURE 5.—Juvenile gags fertilization and settlement times in 1991, back-calculated from juvenile otolith incremental analysis. Numbers are day of the year (e.g., 1 = 1 January).

Juvenile Habitat

Many authors (McErlean 1963; Gilmore 1977; Heck and Thoman 1984; Mullaney and Gale 1996) have noted the presence of juvenile gags in estuaries during summer. Keener et al. (1988) and Ross and Moser (1995) considered the estuarine environment to be a consistent and integral requirement of early juvenile stages, primarily on the basis of their observation that such juveniles were abundant in high-salinity estuaries and virtually absent offshore. Numerous reef surveys in the north Florida area confirm that early juveniles do not recruit to shallow reefs (W. Lindberg, University of Florida, personal communication), with a few exceptions that occurred under unusual conditions (L. Kellogg, University of Florida, personal communication). Within estuaries, sea grass appears to be the preferred nursery habitat for this species, although the relative suitability of other estuarine habitats is unknown. In South Carolina, for example—where sea grass is absent—oyster reef is the dominant habitat for juvenile gags (Mullaney and Gale 1996). This study is the first to document the high absolute abundances of juvenile gags in the sea grass habitat, which suggests that sea grass is a preferred habitat and supports the contention that estuaries are integral to the life cycle of gags.

The sea grass habitat of the northwest coast of Florida persists from mid-spring through early autumn (Zieman and Zieman 1989; Koenig, personal observation) and dies back over the fall and winter. Adult gags spawn primarily in February and March (Coleman et al. 1996), and juveniles recruit to sea grass beds in April and May (this study), when the sea grass habitat has recovered sufficiently from winter dieback to provide adequate protection for newly settled fish. Juvenile gags become susceptible to trawling in mid to late June, so abundance can be estimated from that time through early October.

In the present study, we found juvenile gag densities of about 50,000 fish/km^2 in two shallow sea grass beds in St. George Sound. Although all sea grass habitat is not equally suitable throughout its range, extensive juvenile habitat is available else-

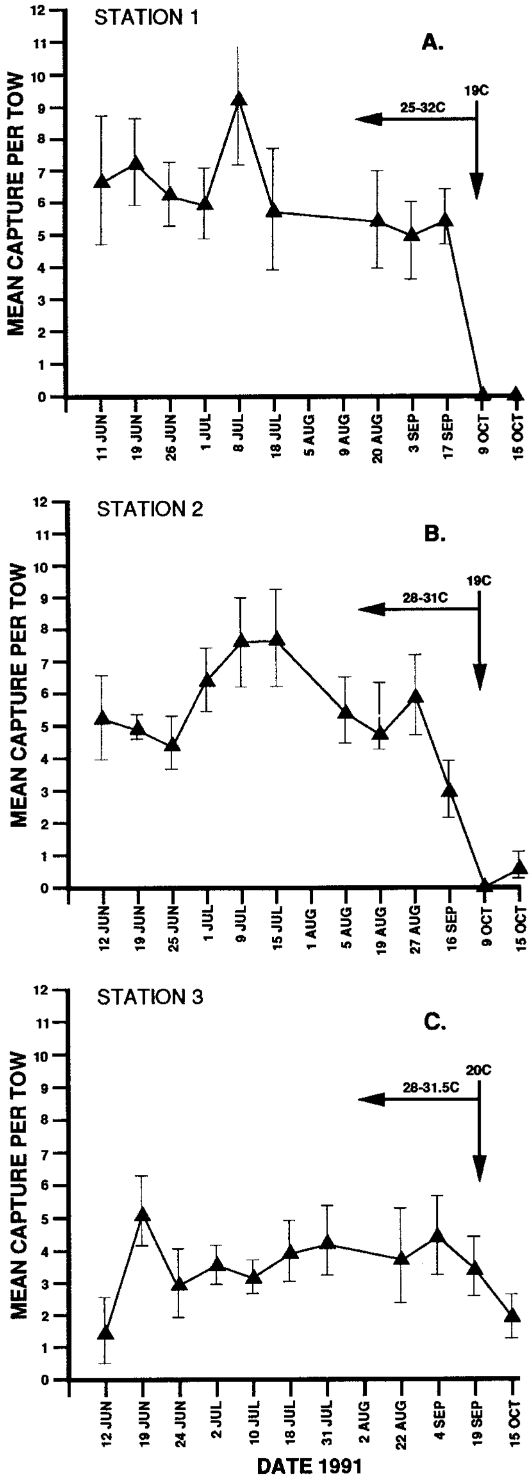

FIGURE 6.—Variation in average captures of juvenile gags per tow for three stations in sea grass beds of St. George Sound, Florida, from early June through October. Left-pointing arrows indicate water temperature ranges of 25–32°C (station 1), 28–31°C (station 2), 28–31.5°C (station 3); down-pointing arrow indicates a drop in water temperature to 19°C (stations 1 and 2) or 20°C (station 3).

where along the west coast of Florida. In the Big Bend alone (from St. Marks south to Tarpon Springs, Florida), a 3,000-km^2 seagrass meadow occurs as a coastal band 11 to 35 km wide (Iverson and Bittaker 1986). Such an area has the potential to support 150 million juvenile gags. Other extensive, relatively pristine sea grass habitats occur along the west Florida coast in St. Andrews Bay, St. Joseph Bay, and Pine Island Sound.

Because critical sea grass habitats are generally close to shore, they are susceptible to anthropogenic disturbance, including storm water runoff (and its wide variety of dissolved contaminants), siltation through shoreline erosion, damage by recreational boats, additions of wastewater from pulp mills, and oil spills (Livingston 1987; Zieman and Zieman 1989). For example, the once extensive sea grass beds of Tampa Bay have declined by 81% as a result of anthropogenic impacts (Zieman and Zieman 1989), and extensive sea grass meadows south of Cape Sable in Florida Bay have undergone losses sufficiently radical to call into question their value as nursery habitat (Butler et al. 1995). Associated economic losses through the loss of fishery production have not yet been estimated accurately, but quantitative estimates of potential production of economically important species such as gag support adoption of measures to protect these habitats.

Juvenile Sampling

Many characteristics exhibited by juvenile gags in the sea grass habitat simplify the estimation of year-class strength. Because juveniles recruit as a single cohort over a short period of time (approximately 6 weeks) in April and May, all recruits are within a narrow size range at any sampling time during the summer months (McErlean 1963; Keener et al. 1988; Hood and Schlieder 1992; Ross and Moser 1995). In addition, juvenile survival rates are high, relative movement rates are low, and small-scale spatial patterns are random. Because the sampling efficiency of an otter trawl is similar over the summer months, juveniles can be sampled by otter trawl across localities, and the resulting estimates of abundance can be combined without

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introducing errors related to survival or size-related avoidance of the trawl.

Juvenile gags in north Florida sea grass beds represent an open population; that is, immigration and emigration of individuals occur continuously. However, given that juveniles exhibit very low rates of movement and tend to maintain uniform densities over the spatial scales represented by our study area, we can treat them like a closed population and apply more cost-effective methods to determine absolute abundance (e.g., catch–effort methods outlined in Ricker 1975; Krebs 1989). The small-scale random spatial patterns exhibited by juvenile gags also simplify sample-size determination and sampling-site selection.

Offshore Movement and Survival

Several studies suggest that the mass emigration of juvenile gags from estuaries is coordinated with weather patterns or rapid declines in water temperature (Keener et al. 1988; Ross and Moser 1995; Mullaney and Gale 1996). Our observations strongly support this contention, but other factors may be involved; some juveniles move out of the grass beds before the mass emigration that coincides with declining water temperatures (Hastings 1979; Ross and Moser 1995; our personal observations), and some juveniles remain to overwinter in the estuary (our personal observations).

Accurate fishery recruitment forecasting requires the types of information on the estuary-dependent juvenile stage that we have presented here. In addition, it requires a knowledge of juvenile survival and movement after egress from the sea grass environment. Survival of late juveniles in the offshore reef environment is unknown, but we have good evidence that large juvenile year-classes from north Florida estuaries correspond to significant fishery production on a regional level (Johnson and Koenig, in press). The contribution of strong year-classes, however, may be dampened by density-dependent factors, including cannibalism of juveniles by adults on offshore reefs (W. Fable, National Marine Fisheries Service, personal communication) and forced movement of juveniles from shallow reefs to deep reefs (>50 m), which increases their susceptibility to fishing-induced mortality.

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References

Arthur, S. M., B. F. J. Manly, L. L. McDonald, and G. W. Garner. 1996. Assessing habitat selection when availability changes. Ecology 77:215–227.

Brothers, E. B., and W. N. McFarland. 1981. Correlations between otolith microstructure, growth, and life history transitions in newly recruited French grunts *Haemulon flavolineatum* (Desmarest) (Haemulidae). Rapports et Procès-Verbaux des Réunions, Conseil International pour l'Exploration de la Mer 178:369–374.

Butler, M. J., IV, and eight coauthors. 1995. Cascading disturbances in Florida Bay, USA: cyanobacteria blooms, sponge mortality, and implications for juvenile spiny lobsters *Panulirus argus.* Marine Ecology Progress Series 129:119–125.

Coleman, F. C., C. C. Koenig, and L. A. Collins. 1996. Reproductive styles of shallow water groupers (Pisces: Serranidae) in the eastern Gulf of Mexico and the consequences of fishing spawning aggregations. Environmental Biology of Fishes 47:129–141.

Dayton, P. K., S. F. Thrush, M. T. Agardy, and R. J. Hofman. 1995. Environmental effects of marine fishing. Aquatic Conservation: Marine and Freshwater Ecosystems 5:205–232.

de Lafontaine, Y., T. Lambert, G. R. Lilly, W. D. McKone, and R. J. Miller. 1992. Juvenile stages: the missing link in fisheries research. Canadian Technical Report of Fisheries and Aquatic Sciences 1890.

Gilmore, R. G. 1977. Fishes of the Indian River Lagoon and adjacent waters, Florida. Bulletin of the Florida State Museum Biological Sciences 22:101–147.

Hastings, R. W. 1979. The origin and seasonality of the fish fauna on a new jetty in the northeastern Gulf of Mexico. Bulletin of the Florida State Museum Biological Sciences 24:1–122.

Heck, K. L., Jr., and T. A. Thoman. 1984. The nursery role of seagrass meadows in the upper and lower reaches of the Chesapeake Bay. Estuaries 7:70–92.

Hilborn, R., and C. Walters. 1992. Quantitative fisheries stock assessment: choice, dynamics and uncertainty. Chapman and Hall, New York.

Hood, P. B., and R. A. Schlieder. 1992. Age, growth and reproduction of gag *Mycteroperca microlepis* (Pisces: Serranidae), in the eastern Gulf of Mexico. Bulletin of Marine Science 51:337–352.

Iles, T. C., and R. J. H. Beverton. 1991. Mortality rates of 0-group plaice (*Pleuronectes platessa* L.), dab

(*Limanda limanda* L.), and turbot (*Scophthalmus maximus* L.) in European waters. Netherlands Journal of Sea Research 27:217–235.

Iverson, R. L., and H. F. Bittaker. 1986. Seagrass distribution in the eastern Gulf of Mexico. Estuarine, Coastal and Shelf Science 22:577–602.

Johnson, A. G., and C. C. Koenig. In press. Age and size structure of the fishery and juvenile abundance of gag, *Mycteroperca microlepis*, from the northeastern Gulf of Mexico. Proceedings of the Gulf and Caribbean Fisheries Institute 47.

Keener, P., G. D. Johnson, B. W. Stender, E. B. Brothers, and H. R. Beatty. 1988. Ingress of postlarval gag, *Mycteroperca microlepis* (Pisces: Serranidae) through a South Carolina barrier island inlet. Bulletin of Marine Science 42:376–396.

Krebs, C. J. 1989. Ecological methodology. Harper and Row, New York.

Langton, R. W., R. S. Steneck, V. Gotceitas, F. Juanes, and P. Lawton. 1996. The interface between fisheries research and habitat management. North American Journal of Fisheries Management 16:1–7.

Livingston, R. J. 1987. Historic trends of human impacts on seagrass meadows in Florida. Florida Marine Research Publications 42:139–151.

Manly, B. F. J. 1984. Obtaining confidence limit on parameters of the Jolly–Seber model for capture–recapture data. Biometrics 40:749–758.

McErlean, A. J. 1963. A study of the age and growth of gag, *Mycteroperca microlepis* (Pisces: Serranidae), on the west coast of Florida. Florida Board of Conservation, Marine Research Laboratory Technical Series 41.

Mullaney, M. D., Jr., and L. D. Gale. 1996. Ecomorphological relationships in ontogeny: anatomy and diet in gag, *Mycteroperca microlepis* (Pisces: Serranidae). Copeia 1996:167–180.

Peterman, R. M. 1990. Statistical power analysis can improve fisheries research and management. Canadian Journal of Fisheries and Aquatic Sciences 47:2–15.

Ricker, W. E. 1975. Computation and interpretation of biological statistics of fish populations. Fisheries Research Board of Canada Bulletin 191.

Ross, S. W., and M. L. Moser. 1995. Life history of juvenile gag, *Mycteroperca microlepis*, in North Carolina estuaries. Bulletin of Marine Science 56: 222–237.

Sissenwine, M. P. 1984. Why do fish populations vary? Pages 59–94 *in* R. M. May, editor. Exploitation of marine communities. Springer-Verlag, Berlin.

Zieman, J. C., and R. T. Zieman. 1989. The ecology of the seagrass meadows of the west coast of Florida: a community profile. U.S. Fish and Wildlife Service Biological Report 85(7.25).

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Cost Appendix Sheet:

Table 2. Components of the proposed budget. The upper panel provides a complete overview of the budget. The lower panel provides details of the requested funds for field and laboratory supplies.

Budget Period: 4 Years			**05/01/2014-6/01/2018**		**Year 1**	**Year 2**	**Year 3**	**Year 4**	**Total YRS 1-4**
PERSONNEL									
Salaries		**hourly**	**months**	**rate**					
	C. Koenig		1.0	$104,920	$17,487	$9,181	$9,640	$10,122	$46,428
	F. Coleman (PI)		0.5	$100,000	$4,167	$4,375	$4,594	$4,823	$17,959
1	Technician (OPS)		4	$25,056	$8,352	$8,770	$9,208	$9,668	$35,998
1	Grad student (Co-PI, C. Malinowski)		12	$21,581	$22,228	$22,895	$23,582	$24,290	$92,995
2	undergraduate, high school school :	10	3	$9,600	$9,600	$9,600	$9,600	$9,600	$38,400
TOTAL PERSONNEL				$261,157	$61,834	$54,820	$56,623	$58,503	$231,781
Fringe Benefits		**Health/y**	**Health/mo**	**%**					
	C. Koenig	8,575	$715	17.59%	$3,790	$1,972	$2,053	$2,138	$9,953
	F. Coleman	8,576	$715	17.59%	$1,090	$1,127	$1,165	$1,206	$4,588
	Tech (OPS)	7,098	$592	2.25%	$2,554	$2,563	$2,573	$2,584	$10,274
	Grad students			0.80%	$1,478	$1,483	$1,489	$194	$4,644
	undergraduate, high school school student			0.80%	$77	$77	$77	$1,377	$1,607
TOTAL FRINGE					$8,989	$7,222	$7,357	$7,498	$31,067
Supplies	Supplies				$55,395	$36,145	$36,145	$36,145	$163,830
	Transportation (includes vessel)				7,031	7,031	7,031	7,031	$28,125
Other Costs	Tuition				$11,712	$12,590	$13,535	$14,550	$52,387
	Publication						$1,000	$1,000	$2,000
Total Direct Costs					$144,961	$117,809	$121,692	$124,727	$509,189
Indirect Costs	FSU rate	52%	(MINUS tuition, % subcontracts)		$69,290	$54,714	$56,241	$57,292	$237,537
Totals:					**$206,651**	**$164,923**	**$170,333**	**$174,420**	**$716,326**

Field Supplies	***Unit***	***Cost***	***Year 1***	***Unit***	***Cost***	***Year 2***	***Unit***	***Cost***	***Year 3***	***Unit***	***Cost***	***Year4***
Traps	10	$550	$5,500	0	$550	$0	0	$550	$0	0	$550	$0
Seepage meters	20	$50	$1,000	10	$50	$500	10	$50	$500	10	$50	$500
sample jars	200	$5	$1,000	200	$5	$1,000	200	$5	$1,000	200	$5	$1,000
well materials	1	$1,000	$1,000	0	$1,000	$0	0	$1,000	$0	0	$1,000	$0
dissection kits	3	$20	$60	0	$20	$0	0	$20	$0	0	$20	$0
identification books	3	$60	$180	0	$60	$0	0	$60	$0	0	$60	$0
floats	20	$10	$200	0	$10	$0	0	$10	$0	0	$10	$0
Shackles, swivels, clips	14	$15	$210	0	$15	$0	0	$15	$0	0	$15	$0
Disposabals (bags, etc)	20	$5	$100	20	$5	$100	20	$5	$100	20	$5	$100
Air fills (per tank)	240	$8	$1,920	240	$8	$1,920	240	$8	$1,920	240	$8	$1,920
Bait	1	$500	$500	1	$500	$500	1	$500	$500	1	$500	$500
Other	1	$1,000	$1,000	1	$1,000	$1,000	1	$1,000	$1,000	1	$1,000	$1,000
Subtotal	Total		$12,670	Total		$5,020	Total		$5,020	Total		$5,020
Laboratory supplies	***Unit***	***Cost***	***Year 1***	***Unit***	***Cost***	***Year 2***	***Unit***	***Cost***	***Year 3***	***Unit***	***Cost***	***Year4***
Formalin and isopropyl	7	$25	$175	7	$25	$175	7	$25	$175	7	$25	$175
Shipping for identifications	8	$50	$400	8	$50	$400	8	$50	$400	8	$50	$400
Cleaning supplies	1	$50	$50	1	$50	$50	1	$50	$50	1	$50	$50
Chemical, isotopic analysis	500	$20	$10,000	500	$20	$10,000	500	$20	$10,000	500	$20	$10,000
chemical, analyte analysis (e.g. NO2, PO4)	500	$30	$15,000	500	$30	$15,000	500	$30	$15,000	500	$30	$15,000
PAH	100	$80	$8,000	0	$80	$0	0	$80	$0	0	$80	$0
Oil analysis	50	$50	$2,500	0	$50	$0	0	$50	$0	0	$50	$0
Miscellaneous lab supplies	1	$500	$500	1	$500	$500	1	$500	$500	1	$500	$500
Computer software	1	$500	$500	0	$500	$0	0	$500	$0	0	$500	$0
Computer harddrive	2	$300	$600	0	$300	$0	0	$300	$0	0	$300	$0
Subtotal			$37,725			$26,125			$26,125			$26,125
Y1 TOTAL SUPPLIES			$50,395			$31,145			$31,145			$31,145
Transportation	No. Days (miles)	Price		No. Days	Price		No. Days	Price		No. Days	Price	
Vessel use (daily rate)	40	$87	$3,480	40	$87	$3,480	40	$87	$3,480	40	$87	$3,480
Fuel for vessel	480	$4	$1,920	480	$4	$1,920	480	$4	$1,920	480	$4	$1,920
Boat Trailering Trip	40	$10	$400	40	$10	$400	40	$10	$400	40	$10	$400
Vehicle use	2400	$0.513	$1,231	2400	$0.513	$1,231	2400	$0.513	$1,231	2400	$0.513	$1,231
Subtotal			$7,031			$7,031			$7,031			$7,031
TOTAL			$57,426			$38,176			$38,176			$38,176

Literature Cited

Baca, B.J. & Getter, C.D. (1984) The toxicity of oil and chemically dispersed oil to the seagrass Thalassia testudinum. *Tom E. Allen***,** 314-323.

Bowen, J.L. & Valiela, I. (2001) The ecological effects of urbanization of coastal watersheds: historical increases in nitrogen loads and eutrophication of Waquoit Bay estuaries. *Canadian Journal of Fisheries and Aquatic Sciences,* **58,** 1489-1500.

Chasar, L.C., Chanton, J.P., Koenig, C.C. & Coleman, F.C. (2005) Evaluating the effect of environmental disturbance on the trophic structure of Florida Bay, USA: multiple stable isotope analyses of contemporary and historical specimens. *Limnology and Oceanography,* **50,** 1059.

Costanzo, S., O'donohue, M., Dennison, W., Loneragan, N. & Thomas, M. (2001) A new approach for detecting and mapping sewage impacts. *Marine pollution bulletin,* **42,** 149-156.

DeLaune, R., Smith, C., Patrick Jr, W., Fleeger, J. & Tolley, M. (1984) Effect of oil on salt marsh biota: methods for restoration. *Environmental Pollution Series A, Ecological and Biological,* **36,** 207-227.

Duarte, C.M., Dennison, W.C., Orth, R.J. & Carruthers, T.J. (2008) The charisma of coastal ecosystems: addressing the imbalance. *Estuaries and Coasts,* **31,** 233-238.

Kendall, C., Elliott, E.M. & Wankel, S.D. (2007) Tracing anthropogenic inputs of nitrogen to ecosystems. *Stable isotopes in ecology and environmental science,* **2,** 375-449.

Koenig, C.C. & Coleman, F.C. (1998) Absolute abundance and survival of juvenile gags in sea grass beds of the northeastern Gulf of Mexico. *Transactions of the American Fisheries Society,* **127,** 44-55.

Livingston, R.J. (1984) The relationship of physical factors and biological response in coastal seagrass meadows. *Estuaries,* **7,** 377-390.

Mattson, R.A., Frazer, T.K., Hale, J., Blitch, S. & Ahijevych, L. (2002) Florida Big Bend. *IN: L. Handley, D. Altsman, and R. DeMay (eds.), Seagrass Status and Trends in the Northern Gulf of Mexico,* **2002,** 171-188.

Mendelssohn, I.A., Andersen, G.L., Baltz, D.M., Caffey, R.H., Carman, K.R., Fleeger, J.W., Joye, S.B., Lin, Q., Maltby, E. & Overton, E.B. (2012) Oil impacts on coastal wetlands: implications for the Mississippi River Delta ecosystem after the Deepwater Horizon oil spill. *BioScience,* **62,** 562-574.

NSF (2009) Transitions and Tipping Points in Complex Environmental Systems. *National Science Foundation Advisory Committee for Environmental Research and Education, Washington, DC*

Orth, R.J., Carruthers, T.J., Dennison, W.C., Duarte, C.M., Fourqurean, J.W., Heck, K.L., Hughes, A.R., Kendrick, G.A., Kenworthy, W.J. & Olyarnik, S. (2006) A global crisis for seagrass ecosystems. *BioScience,* **56,** 987-996.

Reddy, C.M., Arey, J.S., Seewald, J.S., Sylva, S.P., Lemkau, K.L., Nelson, R.K., Carmichael, C.A., McIntyre, C.P., Fenwick, J. & Ventura, G.T. (2012) Composition and fate of gas and oil released to the water column during the Deepwater Horizon oil spill. *Proceedings of the National Academy of Sciences,* **109,** 20229-20234.

Risk, M.J., Lapointe, B.E., Sherwood, O.A. & Bedford, B.J. (2009) The use of δ15N in assessing sewage stress on coral reefs. *Marine pollution bulletin,* **58,** 793-802.

Sanders, H.L., Grassle, J.F., Hampson, G.R., Morse, L.S., Garner-Price, S. & Jones, C.C. (1980) Anatomy of an oil spill: long-term effects from the grounding of the barge Florida off West Falmouth, Massachusetts.

Savage, C. & Elmgren, R. (2004) Macroalgal (Fucus vesiculosus) $\delta^{15}N$ values trace decrease in sewage influence. *Ecological applications,* **14,** 517-526.

Underwood, A. (1991) The logic of ecological experiments: a case history from studies of the distribution of macro-algae on rocky intertidal shores. *Journal of the Marine Biological Association of the United Kingdom,* **71,** 841-866.

Underwood, A. (1994) On beyond BACI: sampling designs that might reliably detect environmental disturbances. *Ecological applications,* **4,** 3-15.

Yarbro, L.A. & Carlson Jr, P.R. (2013) Summary Report for Franklin County Coastal Waters. SIMM Program Report No.1. *Fish and Wildlife Research Institute*.

Zieman, J.C., Zieman, R.T. & Pendleton, E.C. (1989) The ecology of the seagrass meadows of the west coast of Florida: a community profile. *Biological report*.