**EFFECTS OF PROPELLER SCARRING ON SEAGRASS-ASSOCIATED FAUNA**

by

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ABSTRACT

Seagrasses play a critical role in the function and structure of coastal ecosystems, and they are an important habitat for a variety of marine organisms. Damage to seagrass beds from boat propellers is significant in many areas. Recognizing the need to protect this valuable habitat, three voluntary no motor zones were established in Redfish Bay, Texas. This study was designed to test the effectiveness of these protected areas and to assess the impact of propeller scarring on nekton. To examine compliance, I made visual observations of boat activity in these areas. My data showed zero boater compliance in the voluntary no motor zones. I characterized the effects of the propeller scarring on seagrass-associated fauna by: (1) comparing measures of fish and decapod crustacean at three distinct scarring intensities to unscarred sites; and (2) characterizing the functional relationships size, mortality; growth rate to scarring intensity. I selected 10 replicate (10 x 25m quadrats) sites representing three distinct scarring intensities: reference (0%), low (5% or less), moderate (5-15%), and severe (>15%). Sites were sampled in 2003-2004 for nekton during 4 seasons (summer, fall, winter, and spring) using epibenthic sleds. Growth rates of selected fauna were examined using field enclosure experiments and otolith microstructure analysis. My results suggest that even severe (>15%) propeller scarring may not affect density patterns, mean size, or mortality of the organisms collected. Otolith microstructure analysis on pinfish (*Lagodon rhomboides*) indicated no difference in growth rate at various scarring intensities; however, field growth enclosure experiments with white shrimp (*Litopenaeus setiferus*) showed significantly lower growth in highly scarred areas than reference sites. These results suggest the need for

further study at different spatial scales and at higher scarring intensities to determine at what level propeller scars affect the functionality of seagrass.

# DEDICATION

This thesis is dedicated to my family, friends, and 64,587 organisms that participated in my study.

# ACKNOWLEDGMENTS

I would like to thank Dr. Greg Stunz for the care and dedication he has provided as a mentor in the two years I have spent at Texas A&M University-Corpus Christi. He has been a wonderful and patient mentor as well as providing necessary technical, logistical, and financial support for my project. I would also like to thank my committee members for their help and insight on this project: Dr. Kirk Cammarata for his useful perspective on this project, Dr. Kim Withers for attention to detail, and Dr. Chris Onuf for his willingness to share his wealth of seagrass knowledge with me. I would also like to thank Dr. Ken Dunton of University of Texas Marine Science Institute and Dr. Richard Watson for flights over my study area. I appreciate the help of Beau Hardegree of USFWS for providing background information on the no motor zones. I also appreciate the hard work and efforts of the student workers, volunteers, and members of the Fisheries Ecology Lab at TAMUCC who assisted with the extensive field and lab work. Particularly, I would like to thank Amanda Bushon, Megan Reese, Brooke Stanford, Ryan Fikes, and Annette Cardona for their countless hours of help. I would also like to thank Sarah McBee not only for revisions on early drafts of this thesis but also for her support since the beginning of my graduate education. Finally, I would like to thank my family. My graduate career at TAMUCC would not have been possible without them.

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# CHAPTER I

# INTRODUCTION

Seagrasses, or submerged aquatic vegetation (SAV), play a critical role in the function and structure of coastal ecosystems (Hemminga & Duarte 2000). They are one of the most productive and valuable marine habitat types (Quammen & Onuf 1993, Short & Wyllie-Echeverria 1996, Levin et al. 1997) and typically support a great abundance of fish and invertebrates (Beck et al. 2001). Seagrasses function as nursery habitat for economically and recreationally important fishery species (Heck & Thoman 1981, Short & Wyllie-Echeverria 1996, Levin et al. 1997, Minello 1999, Beck et al. 2001, Heck et al. 2003). Specifically, seagrasses provide a structurally complex habitat (Attrill et al. 2000, Heck et al. 2003) providing protection from predation (Rooker et al. 1998, Stunz & Minello 2001) and increased growth rates for associated fauna (Holt et al. 1983, Heck & Thoman 1984, Orth et al. 1984, Rozas & Odum 1988, Stunz et al. 2002b, Heck et al. 2003).

Despite the importance of seagrass, this marine habitat type has gone through worldwide (Short & Wyllie-Echeverria 1996) and local (Quammen & Onuf 1993) decline in recent decades. Seagrass decline results from several anthropogenic disturbances including dredging (Quammen & Onuf 1993, Onuf 1994), nutrient enrichment (Tomasko & Lapointe 1991, Short et al. 1995), and mechanical damage (eg. propeller scarring) (Zieman 1976, Sargent et al. 1995, Dawes et al. 1997, Bell et al. 2002, Dunton & Schonberg 2002, Uhrin & Holmquist 2003).

As boating activity in seagrass meadows has increased, damage from boat propellers has become a significant problem (Dunton & Schonberg 2002). A propeller

scar is created when a boat propeller tears through the rhizomal mat of a seagrass bed (Zieman 1976, Dawes et al. 1997). This causes erosion of the surrounding area (Eleuterius 1987), leading to deterioration of seagrass bed integrity and coverage that may affect the function of the entire seagrass community (Zieman 1976).

Propeller scarring is prevalent in the shallow seagrass flats along the coast of south Texas (Dunton & Schonberg 2002). They occur for a number of reasons including: shortcuts at channel junctions, access to shallow grass beds from blind channels, and use of PVC poles to mark prop-dredged channels (Zieman 1976, Sargent et al. 1995, Dawes et al. 1997, Bell et al. 2002, Dunton & Schonberg 2002, Uhrin & Holmquist 2003). In addition, the intensity of propeller scarring increases with population density (Sargent et al. 1995, Dunton & Schonberg 2002). It is estimated that seagrasses along the Texas coast are worth 2.1 to 6.6 billion dollars, with the per acre value of seagrass on the Gulf Coast between $9000 and $28,000 (Lipton et al. 1995). Using the per acre value of seagrass (Lipton et al. 1995) and data of scarring intensity (Dunton & Schonberg 2002), the loss of seagrass from propeller scarring in Estes Flats and Redfish Bay alone can be estimated at $800,000 to 6.7 million dollars in lost recreational and commercial value.

Seagrass can recover and regrow in propeller scars, but the process is slow and species dependant. Single scars can regrow in 0.9 to 4.6 years in *Halodule wrightii* (Sargent et al. 1995) and 1.7-10 years in *Thalassia testudinum* (Dawes et al. 1997). Due to this slow growth rate seagrass may show long-term damage from propeller scarring (Dawes et al. 1997). To date, studies examining scar regrowth have focused on the recovery of a single scar. However, areas along channel edges and at channel junctions

are susceptible to repeated scarring (Sargent et al. 1995, Dunton & Schonberg 2002), and it is unknown how long, if ever, it will take for these areas to recover.

Recognizing the need to protect valuable habitats and their ecologically important function, the United States government has established several marine protected areas (MPAs). In south Texas, 3 regions of Redfish Bay were designated as "no motor zones," a type of MPA, to protect the seagrass habitat from continued damage from propeller scarring. Heavily damaged seagrass meadows were chosen for voluntary no motor zones that were temporarily established for a period of five years. At the end of this five-year period, no motor zone effectiveness will be evaluated to determine future protective measures.

There have been very few studies examining the effects of propeller scarring (see Zieman 1976, Sargent et al. 1995, Dawes et al. 1997, Bell et al. 2002, Dunton & Schonberg 2002, Uhrin and Holmquist 2003). Some studies have focused on the physical characteristics of the seagrass system and regrowth from propeller scarring (eg. Zieman 1976, Dawes et al. 1997). These studies found that propeller scarring alters the dynamics of the seagrass bed and that seagrass, specifically *Thalassia testudinum*, can take several years to regrow. Other studies used aerial surveys to classify areas according to scarring intensity and quantify overall propeller scar coverage in Florida (Sargent et al. 1995) and south Texas (Dunton & Schonberg 2002). Sargent et al. (1995) established a classification scheme to describe scarring intensities: light (< 5%), moderate (5-20%), and severe (>20%) scarring. Using that classification scheme, Dunton & Schonberg (2002) found 39% of the 5,700ha of seagrass beds surveyed in south Texas were moderately or severely scarred.

There have only been two published studies examining the faunal effects of propeller scarring (see Bell et al. 2002, Uhrin and Holmquist 2003). Uhrin and Holmquist (2003) took a small-scale approach to examining the effects of propeller scarring by measuring faunal densities within the scar and at varying distances from the scar. They found a significant decrease in shrimp and mollusks up to 5m from the scar. Bell et al. (2002) took a landscape approach to examining propeller scarring, and they did not find a difference between scarred (6-31%) and unscarred sites. Clearly, further study is needed to determine if there is a relationship between nekton and scarring intensity.

Typically, areas with greater invertebrate or fish densities are considered better nursery habitats (Minello 1999, Beck et al. 2001). Invertebrates and fish show differential selection to habitat types (Minello 1999, Stunz et al 2002a); therefore, examining habitat-specific density patterns is useful in determining relative habitat value. For example, several studies have shown greater nekton densities in vegetated versus unvegetated habitats (Zimmerman & Minello 1984, Minello & Webb 1997, Rozas & Minello 1998, Castellanos & Rozas 2001, Stunz et al. 2002a), with densities 2 to 25 times greater in seagrass (see SCPT 1999). Furthermore, a variety of commercially important estuarine taxa show a positive relationship between seagrass coverage and production (Heck et al. 2001).

High quality habitat is a key requirement for reproduction, growth, and survival of estuarine dependant species (Levin & Stunz in press). Low nekton densities can be caused by low recruitment or high mortality (Heck et al. 2001), indicating poor habitat quality (Glancy 2003). Therefore, it is important for organisms to select habitats with high refuge value (i.e., high growth and low predation). The structural complexity of the

seagrass canopy provides benefits to settlers (Rooker et al. 1998), and provides refuge from predation for invertebrates and small fish (Orth et al. 1984, Minello 1993, Sogard & Able 1994) allowing for decreased predation (Holt et al. 1983). Mortality from predation greatly impacts survival in the early life stages of fish (Houde 1987) and is an important factor determining distribution of seagrass-associated fauna (see reviews by Heck & Orth 1980, Orth et al. 1984). Rapid growth rates reduce the time juvenile fish and invertebrates spend at sizes most vulnerable to predation. Juvenile fish and invertebrates use shallow estuarine areas as nursery habitat (Heck & Thoman 1984, Levin et al. 1997, Minello 1999), and in these areas they have access to abundant food supplies to promote rapid growth (Boesch & Turner 1984, Kneib 1993). Growth rates can be used to measure habitat quality and can be used to define habitat value as an indicator of habitat degradation. Propeller scarring may impact all of these functional qualities of seagrass.

Propeller scarring removes seagrass, creating an overall decline in the amount of habitat. An area without propeller scarring provides a more structured habitat and may increase survival of juvenile organisms. For example, Rooker et al. (1998) and Stunz and Minello (2001) have shown more structurally complex habitats often increase the survival of juvenile fish. However, it has also been shown that abundance of organisms is highest at intermediate levels of habitat fragmentation. It is believed that increased edge gives a greater area for the organisms to forage for food (Holt et al. 1983). Alternately, some species use seagrass beds for shelter and forage over adjacent unvegetated habitats (Summerson & Peterson 1984). Seagrass-associated fauna may exhibit different density patterns and behavior based on their use of the edge habitat (Bell et al. 2001). The degree to which habitat fragmentation alters animal dispersal depends upon organism mobility

and the scale of fragmentation under investigation (Doak et al. 1992). It is unknown if a single scar can affect the faunal distribution (Uhrin & Holmquist 2003).

To date, there is no clear understanding of the exact effects of habitat fragmentation from propeller scarring: at what level, if any, the fragmentation is beneficial, and at what point this causes degradation of the functionality of the community. This research is the first study to examine nekton responses to varying levels of fragmentation. I examined organism density, growth rates, and survival as indicators of nursery value. By understanding the effects of propeller scarring, we can begin to evaluate this habitat degradation and use this information for protecting seagrass meadows.

My research was designed to determine the extent of compliance in designated voluntary no motor zones and to study the functionality of seagrass meadows at different levels of propeller scarring. In assessing habitat functionality in relation to propeller scarring, my objectives were first to examine density patterns of fish and decapod crustaceans and then examine the underlying functional mechanisms. To characterize the functional relationships and assess habitat value, I examined growth rate, size, and mortality for fish and decapod crustaceans. Specifically, my objectives were:

(1) Examine boater compliance in voluntary no motor zones Redfish Bay, Texas.

(2) Compare density patterns of fish and decapod crustaceans in three distinct scarring intensities to unscarred sites.

(3) Characterize functional relationships between common estuarine organisms and scarring intensity by examining:

(a) scarring intensity in relation to growth rate

(b) scarring intensity in relation to mortality

***Study Site***

Redfish Bay (27°54'27" N, 97°06'45" W) is a secondary bay in the 447-km$^2$ Aransas Bay complex (Fig. 1), located along the coast of south Texas (Britton & Morton 1997). It is a barrier-built, positive estuary with freshwater inflows from the Mission and Aransas Rivers (Britton & Morton 1997) and one open connection to the Gulf of Mexico. All five species of seagrass (*Halodule wrightii*, *Thalassia testudinum*, *Syringodium filiforme*, *Halophila engelmanii*, and *Ruppia maritime*) found in Texas occur in Redfish Bay region; however, *Halodule wrightii* is dominant in this system (SCPT 1999). The maximum water depth is 2.9m (Montagna 1998) with a mean water depth of 0.5m in the study area. The tides are mixed, primarily diurnal, with a mean daily range of 0.12 m (Rockport, Aransas Bay, National Ocean Service, NOAA). Aerial surveys (Dunton & Schonberg 2002) confirmed the wide-spread presence of propeller scarring within Redfish Bay.


Fig. 1. Redfish Bay (27°54’27” N, 97°06’45” W), a secondary bay in the 447 km$^2$ Aransas Bay complex, located along the coast of south Texas.

# CHAPTER II

# BOATER COMPLIANCE IN NO MOTOR ZONES REDFISH BAY, TEXAS, USA

## *Introduction*

Whether MPAs are beneficial is controversial, and it remains an emerging topic of research. Roberts et al. (2001) performed a study at the Merritt Island National Wildlife Refuge at Cape Canaveral Florida. This MPA is extremely well enforced due to its close proximity to the Kennedy Space Center. The study showed an overwhelming benefit with an increase in several game-fish species inside the reserve boundaries and in the surrounding areas. However, the lack of baseline data has been subject to intense criticism (Witek 2002). As is the case for many MPAs, baseline data are lacking so comparisons before implementation of the MPA cannot be compared to conditions after implementation (Botsford et al. 2003).

Despite several conflicting studies on the effectiveness of MPAs, a panel of 161 experts on marine reserves compiled a formal opinion and presented it as a *Scientific Consensus Statement on Marine Reserves and Marine Protected Areas*. This statement was designed to express a clear view of accepted facts and identify areas that need further research. A summary of this statement (Lubchenco et al. 2003) indicates that reserves often result in long lasting and rapid increases in abundance, diversity, and productivity of marine organisms. These changes are due to decreased mortality, decreased habitat destruction, and indirect ecosystem effects. In addition, reserves reduce the probability of extinction for resident marine species and increased reserve size results in increased benefits. However, even small reserves have positive effects. The hope is that with

MPAs, an exploited species will have a greater chance to reproduce, particularly due to the protection of the older, more fecund females.

One of the greatest problems facing marine protected areas is enforcement. There are very few cases like that of Merritt Island National Wildlife Refuge (Roberts et al. 2001) where there is strict enforcement resulting in high user compliance. My study focuses on this key issue of compliance. It is important to determine the biological effectiveness of the no motor zone MPAs; however, these no motor zones are voluntary, and there is no enforcement. Therefore, it is important to determine the extent of boater compliance prior to examining the ecological function of these areas.

Recognizing the need to protect valuable habitats and their ecologically important function, the United States government has established a mechanism for the establishment of marine protected areas (MPAs). Executive Order 13158 defines an MPA as "any area of the marine environment that has been reserved by Federal, State, territorial, tribal, or local laws or regulations to provide lasting protection for part or all of the natural and cultural resources therein."

In June of 2000, three regions of Redfish Bay, Texas were designated as “no motor zones,” a type of MPA. This designation was intended to protect the seagrass habitat from the effects of propeller scarring. Redfish Bay is particularly valuable because it contains approximately 5,700 hectares of seagrass beds and all five species of seagrass found along the Texas coast. These areas have been temporarily established in Texas for a period of five years to protect heavily prop scarred seagrass meadows. At the end of this five-year period, no motor zone effectiveness will be evaluated to determine

future protective measures. Specifically, this study was designed to examine boater compliance in these voluntary no motor zones.

## *Materials and Methods*

### Study Location

Observations were made in the three regions of Redfish Bay, Texas (Fig. 1) designated as no motor zones in 2000: Estes Flats, Terminal Flats and Brown and Root Flats (Fig. 2). The no motor zone in Estes Flats is in the northern most region of Redfish Bay and is located near Rockport, Texas. Terminal Flats is near Aransas Pass, Texas and is bordered by high traffic channels to the south and west. Brown and Root Flats is located near Port Aransas, Texas. It contains two run channels that allow access to a deeper run zone located within the no motor zone. Each no motor zone was marked with yellow posts and signage indicating the boundaries of the no motor zones. Sites were in shallow water and in close proximity to deep channels with high boat traffic.


Fig. 2. Map of Redfish Bay on the south Texas coast showing the three no motor zones in Estes Cove (1), Terminal Flats (2), and Brown and Root Flats (3). Note that Brown and Root Flats contains run channels to a deeper area in the center of the no motor zone that is a designated run area.

Field Observation Methods

Two observers with binoculars monitored each of the marked no motor zones from the nearest automobile-accessible shoreline with maximum visibility of the site. Surveys took place March and April 2003 during periods of peak boater traffic. Each no motor zone was observed for one hour on 6 d, with the order that sites were visited randomly assigned. Only boaters within the boundaries were included in compliance counts. Boaters that complied for the entire observation period (eg. anchored fishermen) could not be scored for compliance and were excluded from further analysis.

***Results***

We performed 18 one-hour observations and observed 225 boats (Fig. 3): 0 boats complied, 8 boats had unclear compliance, and 217 boats did not comply. The 8 boats counted as unclear compliance were excluded from analysis. There was 100% non-compliance for every sample period at each site for all observations that could be scored for compliance.

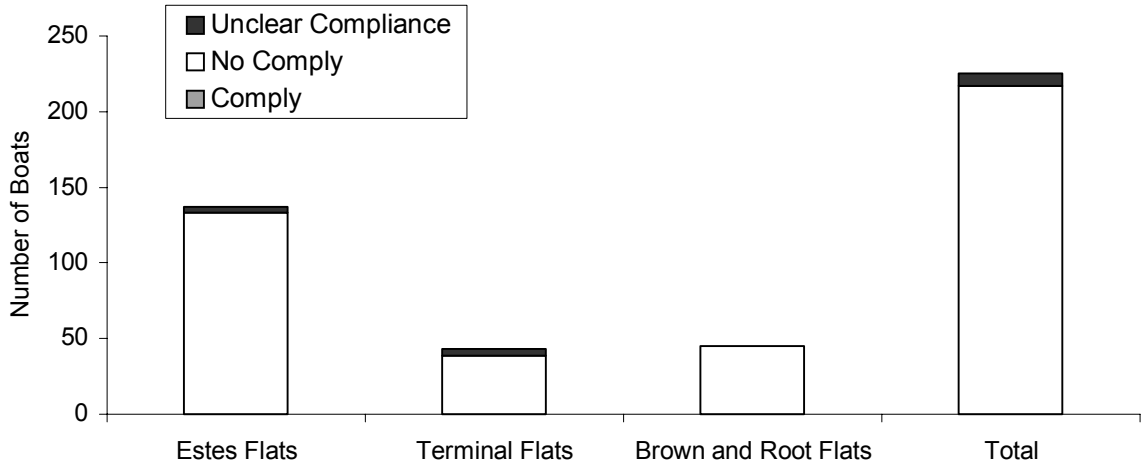



Fig. 3. Boater compliance in no motor zones in Estes Flats, Terminal Flats, Brown and Root Flats, Redfish Bay, Texas.

***Discussion***

No motor zones in Texas have been controversial since their creation in 2000. There was an initial educational campaign on the importance of seagrass to recreational fisheries and the role no motor zones play in protecting seagrass. However, three years later these results show no evidence of boater compliance.

Implementation of this program failed to overcome public resistance. Large signs are posted at several boat ramps in the area to educate the public (Fig. 4). These signs discuss, in detail, local species of seagrass, the importance of seagrass, and the locations of the no motor zones.

The sites were technically established as state scientific areas. They were set up to not only protect the valuable habitat but also as a place for scientists to study the

functionality of these communities. The implementation plan included monitoring of these sites and a full time biologist to study these areas. The current monitoring plan is to take annual aerial photographs of the protected area. Photographs have been taken each year since the implementation. These photographs may be a better method of measuring boater compliance in these areas. By examining scarring patterns over time one could determine if there has been a change in scarring intensity since the implementation of these areas or if scarring patterns have changed.


Fig. 4. Picture of sign posted at boat ramps in the Redfish Bay area. This sign alerts boaters of the locations of no motor zones and the ecological value of seagrass.

It is also important to acknowledge the bias in this observation method. My surveys did not take into account boaters that properly used run channels marked by the no motor zone signs or boaters that avoided the no motor zones. A survey that took into account these factors would generate a more accurate determination on boater compliance.

The no motor zones in Redfish Bay were established on a temporary basis until 2005, and their fate is uncertain. It is possible that this designation will be removed or converted to mandatory no motor zones, at which time tickets and fines could be issued to non-compliers. Sargent et al. (1995) suggested a four-point approach to effective seagrass management that includes: education, proper USCG-approved signage, enforcement, and limited motoring zones. Locally, each of these aspects should be examined. However, providing clearly marked run channels and fining boaters destroying seagrass may have the greatest benefit.

Although this study indicates that there is no boater compliance in voluntary no motor zones, it provides a valuable test of the limits of public education and voluntary compliance for the protection of seagrass. Further work needs to be performed to create a viable plan to effectively manage these areas. This plan should complement community outreach and education with consequences for non-compliance with enforced mandatory no motor zones.

# CHAPTER III

# EFFECTS OF PROPELLER SCARRING ON NURSERY HABITAT VALUE

## *Introduction*

In the Gulf of Mexico 98% of commercially important species are estuarine dependent (Chambers 1992), and seagrasses function as a nursery habitat for many of these species (Heck & Thoman 1981, Short & Wyllie-Echeverria 1996, Levin et al. 1997, Minello 1999, Beck et al. 2001, Heck et al. 2003). Specifically, they are a structurally complex habitat (Attrill et al. 2000, Heck et al. 2003) providing protection from predation (Rooker et al. 1998) and increased growth rates for associated fauna (Holt et al. 1983, Heck & Thoman 1984, Orth et al. 1984, Rozas & Odum 1988, Heck et al. 2003).

Typically areas with higher invertebrate or fish densities are considered better nursery habitats (Minello 1999, Beck et al. 2001). Invertebrates and fish show differential selection for habitat types (Minello 1999, Stunz et al. 2002a); therefore, examining habitat specific density patterns is useful in determining relative habitat value. For example, several studies have shown greater nekton densities in vegetated versus unvegetated habitats (Zimmerman & Minello 1984, Minello & Webb 1997, Rozas & Minello 1998, Castellanos & Rozas 2001), with densities 2 to 25 times greater in seagrass (see SCPT 1999). Furthermore, a variety of commercially important estuarine taxa show a positive relationship between SAV coverage and production (Heck et al. 2001).

Despite the importance of seagrass, this marine habitat type has gone through worldwide (Short & Wyllie-Echeverria 1996) and local (Quammen & Onuf 1993) decline in recent decades. This decline has been the result of several anthropogenic disturbances including dredging (Quammen & Onuf 1993, Onuf 1994), nutrient enrichment (Tomasko

& Lapointe 1991, Short et al. 1995), and mechanical damage (e.g. propeller scarring) (Zieman 1976, Sargent et al. 1995, Dawes et al. 1997, Bell et al. 2002, Davidson 2002, Dunton & Schonberg 2002, Uhrin & Holmquist 2003).

Previous studies on propeller scarring have focused on seagrass recovery (Dawes et al. 1997), classifying scarring patterns and intensities (Sargent et al. 1995, Dunton & Schonberg 2002), and nekton density patterns (Bell et al. 2002, Uhrin & Holmquist 2003). However, there have been no studies that have examining the effects of propeller scarring as it relates to habitat value. Specifically, it is important to determine if propeller scarring affects an organism's density, mortality, and growth rate.

Size is an important aspect of an organism's life history and increased size provides many ecological advantages. Rapid growth early in life is advantageous for nekton because it reduces the time juvenile fish and invertebrates spend at sizes most vulnerable to predation. Juvenile fish and invertebrates use shallow estuarine areas as nursery habitat (Heck & Thoman 1984, Levin et al. 1997, Minello 1999), and in these areas they have access to abundant food supplies to promote rapid growth (Boesch & Turner 1984, Kneib 1993). Additionally, nurseries allow for decreased predation (Holt et al. 1983). Site-related mortality has great impact on survival in the early life stages of fish (Houde 1987) and is thought to be an important factor in determining the distribution of seagrass-associated fauna (reviews by Heck & Orth 1980, Orth et al. 1984). Measuring growth rates can be used as an indicator of habitat quality and is important for defining habitat value and understanding the consequences of habitat degradation.

Examining differences in size distribution provides an indirect measure of site-specific mortality. In sites of low mortality, nekton would be recruiting and surviving,

resulting in the presence of larger size classes. However, in areas of high mortality recruits would not be surviving, and one would expect to find a lower mean size of fish. The distribution in age should be lower in a habitat with higher mortality, since fewer fish survive to a larger size. Conversely, a wide distribution of fish size would suggest the juveniles are recruiting to these areas and surviving. Therefore, areas containing fish with a greater average age and higher variation in age may potentially be a better habitat.

We can further characterize habitat value by examining growth rates of seagrass-dependent organisms. Growth data provides valuable information on the consequences of habitat loss and degradation. Field enclosure experiments are an effective method to measure growth over a short period of time in the field (Stunz et al. 2002b). Enclosures restrict organisms to a given scarring intensity but allow access to the bottom substrate for foraging. By enclosing an organism in a given scarring level, one can examine habitat structure as it relates to growth potential.

Otolith microstructure can also be used to examine differences in growth rates of fish. There is a strong correlation between otolith growth and somatic growth (Pannella 1971), and daily patterns recorded in otoliths can be used to estimate recent growth by measuring incremental widths near the otolith margin (Stunz et al. 2002b).

To date, there is no information on the effects of propeller scarring on the nursery value of seagrass meadows. This research is a key preliminary step in understanding the effect of propeller scarring on habitat value by examining faunal responses to varying levels of fragmentation. Specifically, my objectives were to determine if propeller scarring affects density patterns, size, mortality or growth rate of fish or crustaceans in field growth enclosures and otolith microstructure analysis.

***Materials and Methods***

Delineation quadrats for scarring intensity

Maps of scarring intensity by Dunton & Schonberg (2002), aerial surveys, and intensive ground truthing were used to locate sites. Since the majority of propeller scarring occurs in waters < 1m (Zieman 1976), sites were selected with a relatively uniform mean water depth of 0.5m (Dunton & Schonberg 2002) making them susceptible to propeller scarring.

Ten replicate 10 m x 25 m quadrats (Fig. 5) of three distinct scarring intensities: low (1-5% scarring), moderate (5-15% scarring), severe (>15% scarring), and reference (0%) sites (see Sargent et al. 1995) were identified and sampled in summer, fall, winter, and spring of 2003-2004 (Fig. 6). To characterize the quadrat, the length and width of each scar, measured every 5 m and averaged, were used to calculate the percentage of scarring within each quadrat. Reference sites were in areas without propeller scarring and within 100 m of scarred sites.

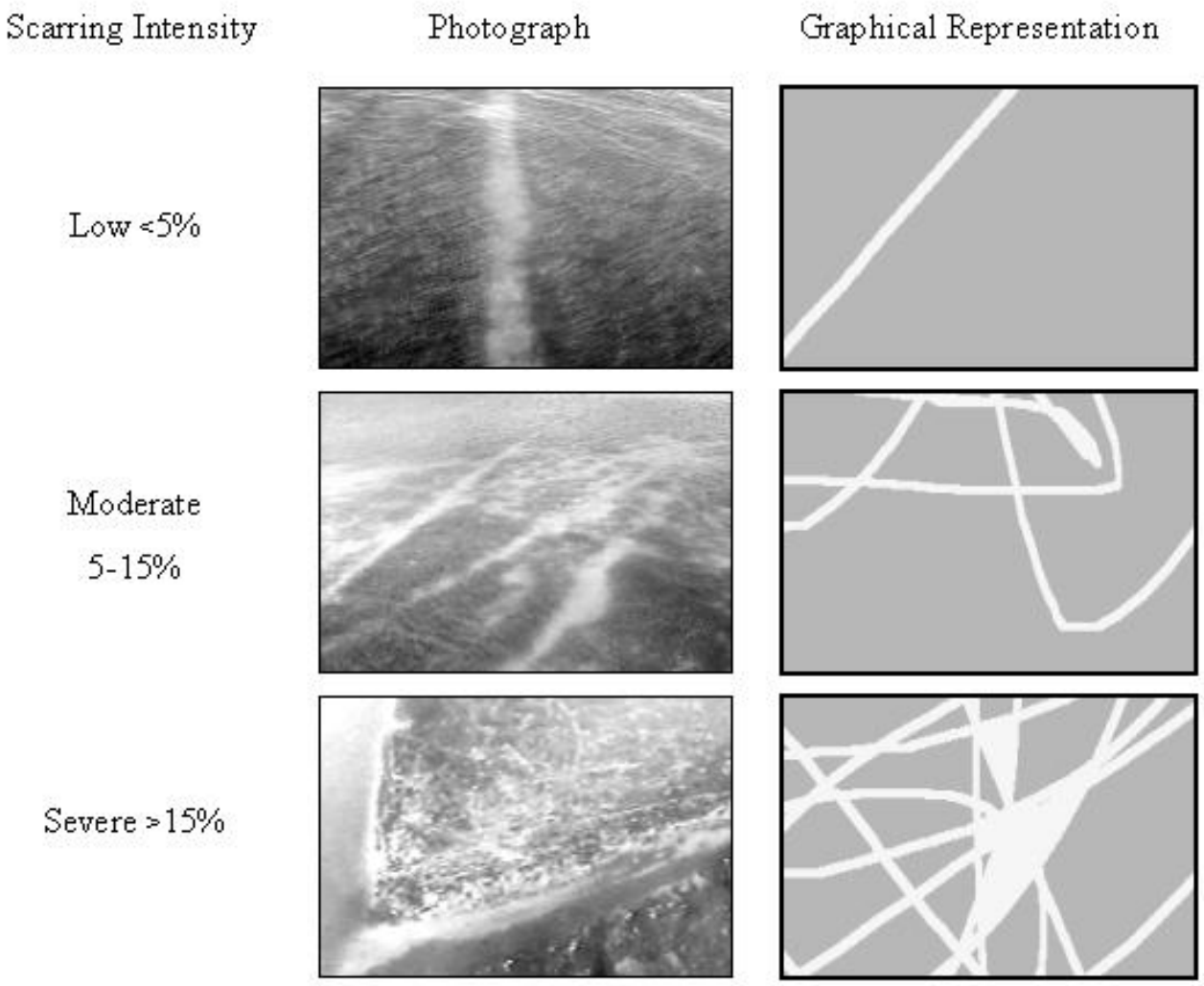



Fig. 5. Sites were classified into three distinct scarring intensities (modified from Sargent et al. 1995): low (1-5%), moderate (5-15%), and severe (>15%). This figure shows pictures taken in Redfish Bay, Texas of these scarring intensities and graphical representation. Note bare patches in severely scarred areas created by erosion of patches created by several scars.

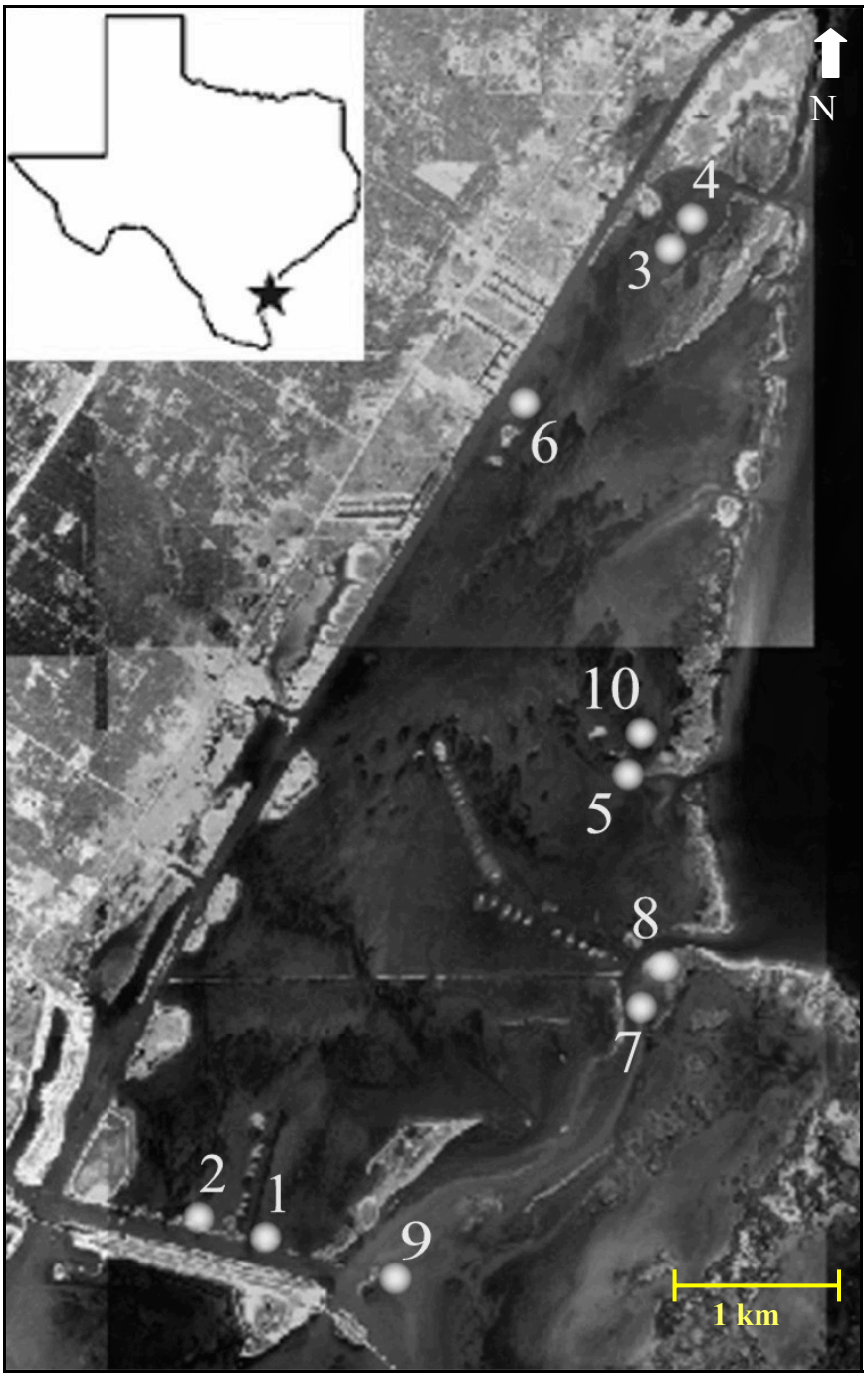



Fig. 6. Map representing the location of sample sites within Redfish Bay, Texas. Each site contains one high (>15% scarred), medium (5-15% scarred), low (1-5% scarred), and reference (0%) site.

Physical Parameters

Water depth, salinity, dissolved oxygen, and temperature were measured at each site to assess similarity among sites. Temperature and dissolved oxygen were measured using a YSI model DO 200, and water depth was recorded as the average of four depths taken in each quadrat.

To compare seagrass characteristics at each site, samples were taken in monotypic stands of *Halodule wrightii* and seagrass shoot density and above and below ground biomass were measured. Shoot density and above and below ground biomass were measured between April 18th, 2004 and May 6th, 2004. Three 10.05 cm diameter cores were taken at each site and averaged to calculate the mean shoot density and biomass per site. Cores were taken haphazardly within areas of solid seagrass within the quadrat, and prop scars, when present, were avoided. Biomass samples were processed by separating above and below ground material and placing samples in aluminum trays in an oven for 120 h at 90°C. Desiccated samples were weighed to the nearest 0.001 g and converted to g seagrass $m^{-1}$.

Nekton Collection

I sampled during four seasons: summer (30 July-10 August 2003), fall (18-24 October 2003), winter (7-8 January 2004) and spring (17-18 March 2004) using epibenthic sleds (see Stunz et al. 2002a). Briefly, epibenthic sleds can estimate small-scale density patterns and have been shown to provide accurate quantitative sampling in seagrass (Rozas & Minello 1997). Specifically, I sampled each site by simultaneously placing two sleds at the top of each quadrat and walking a semicircular route around the sampling area to avoid disturbance. Sleds were pulled at the same time with one person

towing each sled. Sleds were pulled by hand the length of the rope (16.7m) to cover 10 $m^2$ of bottom. Organisms were sorted from seagrass and detritus and preserved in 70% ethanol. Density from duplicate sled tows were averaged between tows.

Organisms were identified to species or the lowest possible taxon. Total length (TL) of penaeid shrimp, standard length (SL) of fish, and carapace width (CW) of other decapod crustaceans were measured to nearest mm.

Nekton Analysis

I selected the 8 most numerically abundant taxa to analyze individually by season. Pinfish (*Lagodon rhomboides*), pipefish (*Syngnathus* spp.), code goby (*Gobionellus robustum*), darter goby (*Gobionellus boleosoma*), killifish (Fundulidae), blue crab (*Callinectes sapidus*), Atlantic mud crab (*Panopeus herbstii*), and grass shrimp (*Palaemonetes* spp.) were dominant in all seasons. I selected an additional 5 species (red drum (*Sciaenops ocellatus*), bay whiff (*Citharichthys spilopterus*), spot (*Leiostomus xanthurus*), brown shrimp (*Farfantepenaeus aztecus*), and white shrimp (*Litopenaeus setiferus*) that were not dominant in all seasons but were analyzed in the season(s) when they were most abundant. For the purpose of analysis all species of killifish (Fundulidae) and pipefish (*Syngnathus* spp.) were grouped together by family and genus, respectively. All organisms collected were included in the overall density analysis.

I converted the number of organisms collected in each quadrat to density (organisms/$m^2$) and a log (x+1) transformation was used to minimize heteroscedasticity. I used analysis of variance (ANOVA) to assess the differences in abundance of nekton among sites and at different levels of scarring intensity ($\alpha$ =0.05). I compared the

percentage of scarring in each site with the density of organisms present using a linear regression ($\alpha$ =0.05).

Size Analysis

I selected the 5 most numerically abundant taxa to analyze individually by season. Pinfish, code goby, darter goby, blue crab, and Atlantic mud crab were dominant in all seasons. For the additional 5 species (red drum, bay whiff, spot, brown shrimp, and white shrimp), I used analysis of variance (ANOVA) to assess the differences in size among sites and at different levels of scarring intensity ($\alpha$ =0.05).

Length Frequency Analysis

I used length frequency analysis of the 5 most abundant species as a measure of age structure in varying levels of propeller scarring. I measured SL of pinfish and spot, TL of brown shrimp and white shrimp, and CW of blue crab. I separated them into (5 mm) size classes and compared age structure among scarring intensities.

Growth

*Field Growth Experiment*

To assess field-based growth I conducted a field growth enclosure experiment. Using 24 field enclosures made from polypropylene barrels (0.283 $m^2$; 60 cm diameter x 1 m deep) with the ends removed to create a cylindrical enclosure, I placed 6 barrels in replicate sites of each scarring intensity (low, moderate, severe) and six reference sites. Enclosures were pushed 15 cm into the substrate and anchored from the outside with three wooden stakes. I swept enclosures with dip nets (1mm mesh) removing only

predators and covered enclosure tops with 1mm mesh nylon netting. Prior studies have shown (Stunz et al. 2002b) no significant difference in water quality parameters inside versus outside of the enclosures. Therefore, water quality conditions were only measured inside the enclosures at three times during the growth trial at 4 d, 7 d, and 11 d.

The field growth enclosure experiment began on November 5, 2003 by stocking each enclosure with 3 white shrimp (mean = 43.6 mm, SE = 1.45). Shrimp were collected in adjacent seagrass meadows, measured to the nearest 1 mm, and marked with an orange elastomer tag. The shrimp remained in the enclosures for 11 d and were recovered by using dip nets. I determined the growth rate for each shrimp by identifying the individual shrimp, measuring them to the nearest 1 mm, and subtracting the original length measurements. I used analysis of variance (ANOVA) to assess the differences in growth at different levels of scarring intensity ($\alpha$ =0.05). Tukey's post hoc test was used for pair wise comparison of mean growth.

*Otolith Microstructure Analysis*

I estimated recent growth among varying treatment levels by measuring increment width near the otolith margin corresponding to recent somatic growth and used that as a proxy for fish growth during this time period (Levin et al. 1997). Twenty-five fish were collected from 10 replicate sites of different scarring intensities. Pinfish were collected in spring (17-18 March 2004) and preserved in 70% ethanol. I measured the fish to the nearest 0.1 mm SL and did not adjust for shrinking during preservation.

I removed the left lapilar otolith from 25 pinfish (SL=28.5mm, SE=0.236) from each scarring intensity following the procedures of Secor et al. (1991). Otoliths were

placed in immersion oil and read after 48 h. Daily growth ring increments were readily apparent and the otoliths did not need further processing. I did not experimentally verify the existence of daily growth increments; however, daily growth rings are known to occur in this family (see Francis et al. 1993). Otolith microstructure analysis has been used to measure daily growth in pinfish without age validation (Levin et al. 1997). There was a significant relationship between otolith diameter and fish length (Fig. 32) allowing me to use otolith-based growth as a proxy for fish somatic growth. I determined growth rate by identifying and counting the daily growth rings using a digital image enhancing system (Motic Images 2000 1.3) I measured from the otolith margin in 10 daily growth rings representing the last 10 days of growth. Two observers measured each otolith. If the two measurements were not identical the otolith was measured for a third time. If there were not two identical length measurements then the otolith was removed from analysis. Mean increment width was compared using analysis of variance (ANOVA).

***Results***

Physical Parameters

All scars within sites were measured to calculate scarring intensity (Fig. 7). Mean scarring intensities were: 1.93%, SE= 0.192 (low), 8.86%, SE= 0.975 (Moderate), and 20.27%, SE= 1.209 (Severe). Scarring intensities were significantly different in each scarring level ($F = 105.841$, df = 10, $p < 0.001$). At each site, dissolved oxygen (Fig. 8), salinity (Fig. 9), temperature (Fig. 10), depth (Fig. 11), seagrass shoot density (Fig. 12), and seagrass above (Fig. 13) and below (Fig. 14) ground biomass were measured. All physical parameters were not different among sites.

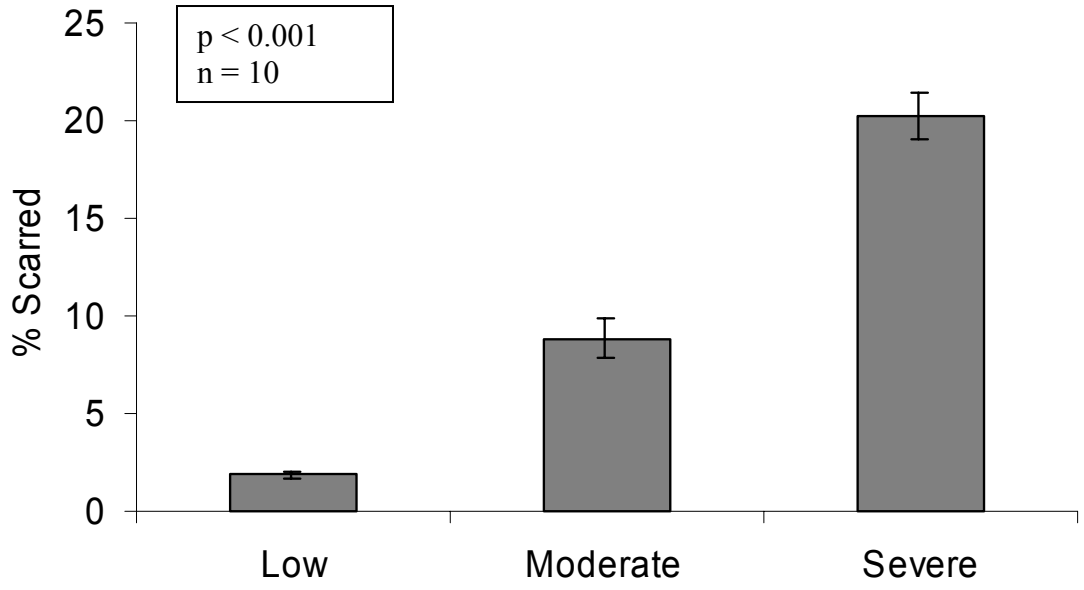



Fig. 7. Mean measured scarring intensities at sites in Redfish Bay, Texas were: 1.93%, SE= 0.192 (low), 8.86%, SE= 0.975 (Moderate), and 20.27%, SE= 1.209 (Severe). The P-value is from an ANOVA comparing measured scarring intensities within each scarring level.

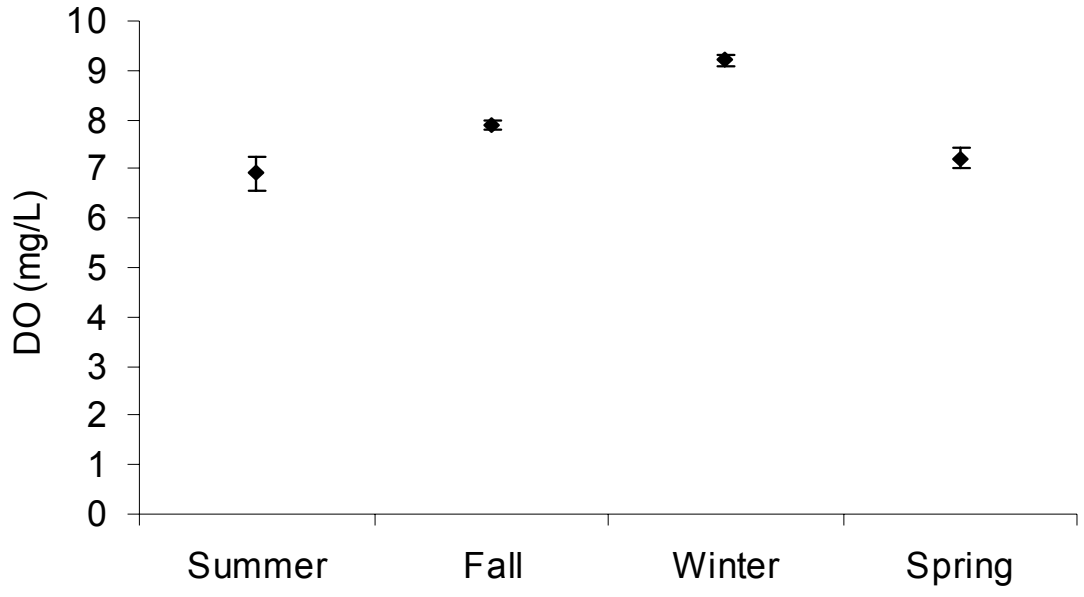



Fig. 8. Mean dissolved oxygen (± SE) for all sites in Redfish Bay, Texas in each season.

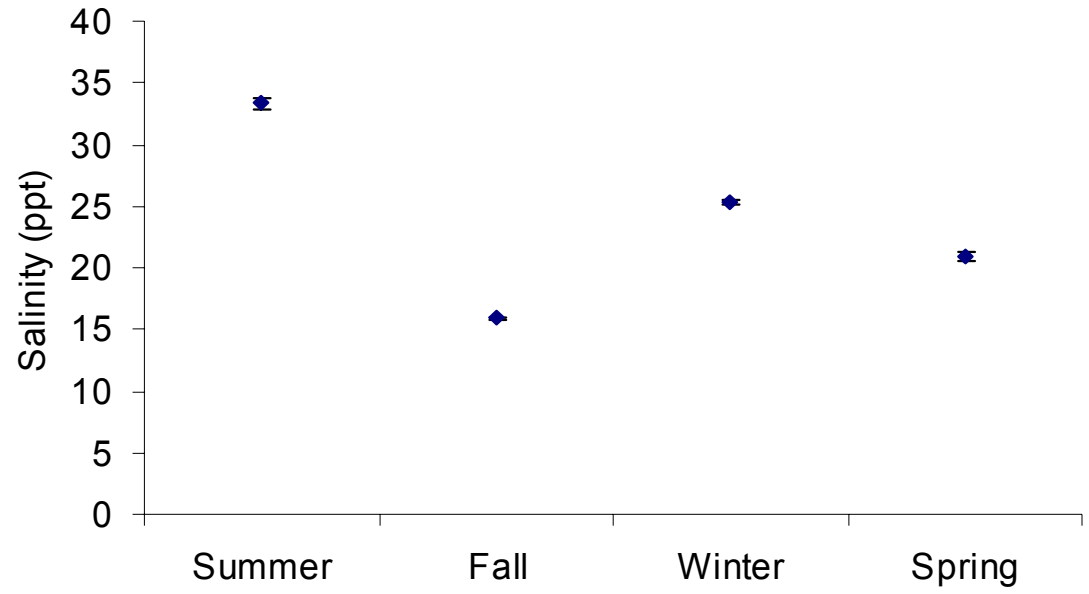



Fig. 9. Mean salinity (± SE) for all sites in Redfish Bay, Texas in each season.

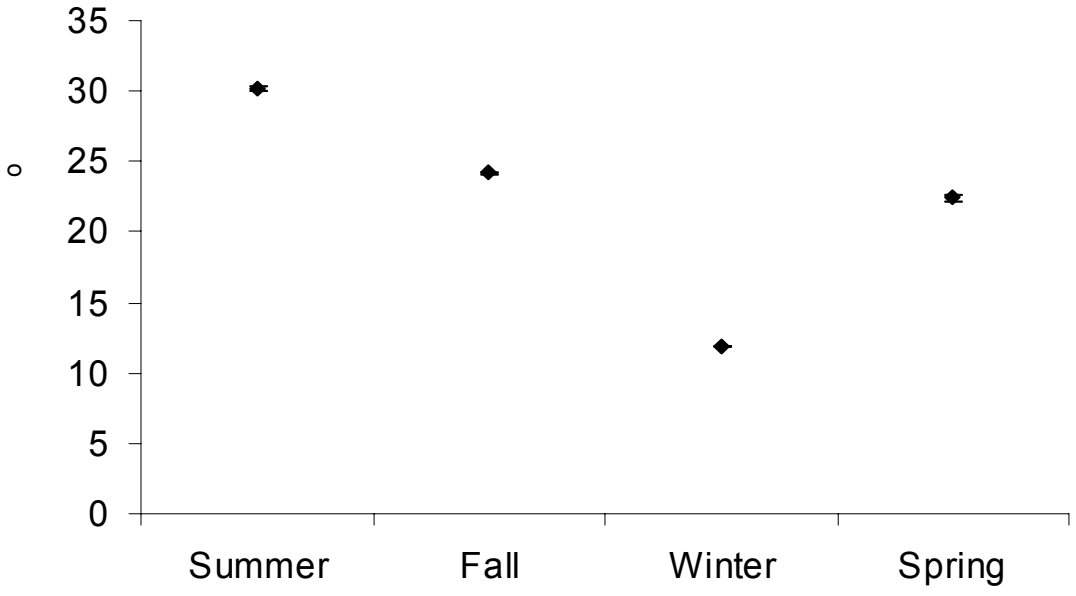



Fig. 10. Mean water temperature (± SE) for all sites in Redfish Bay, Texas in each season.

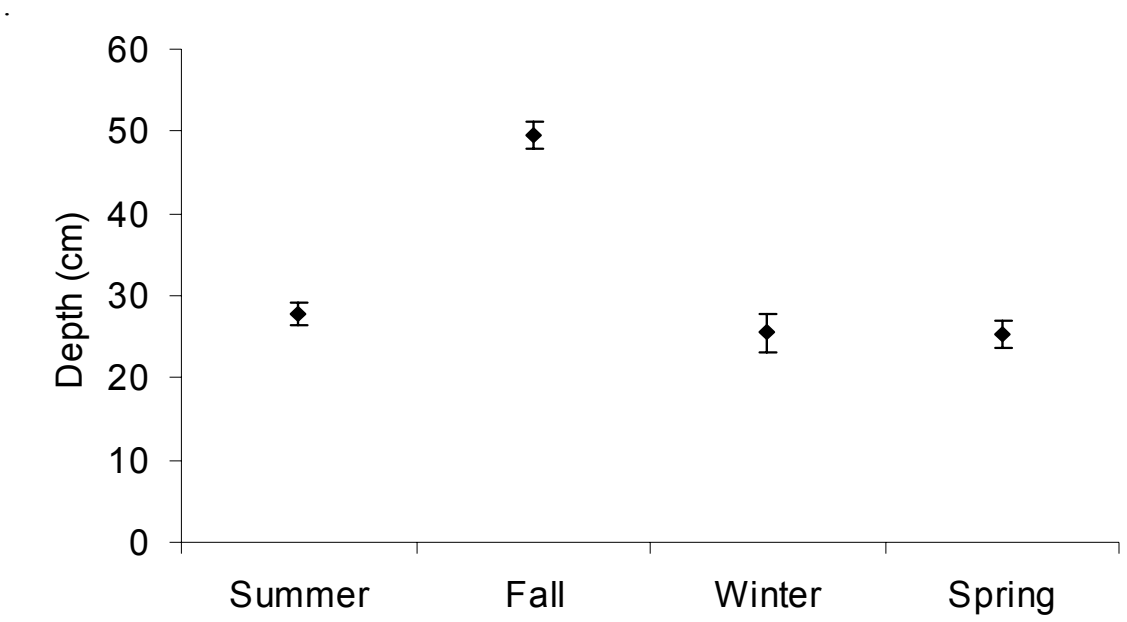



Fig. 11. Mean water depth (± SE) for all sites in Redfish Bay, Texas in each season.

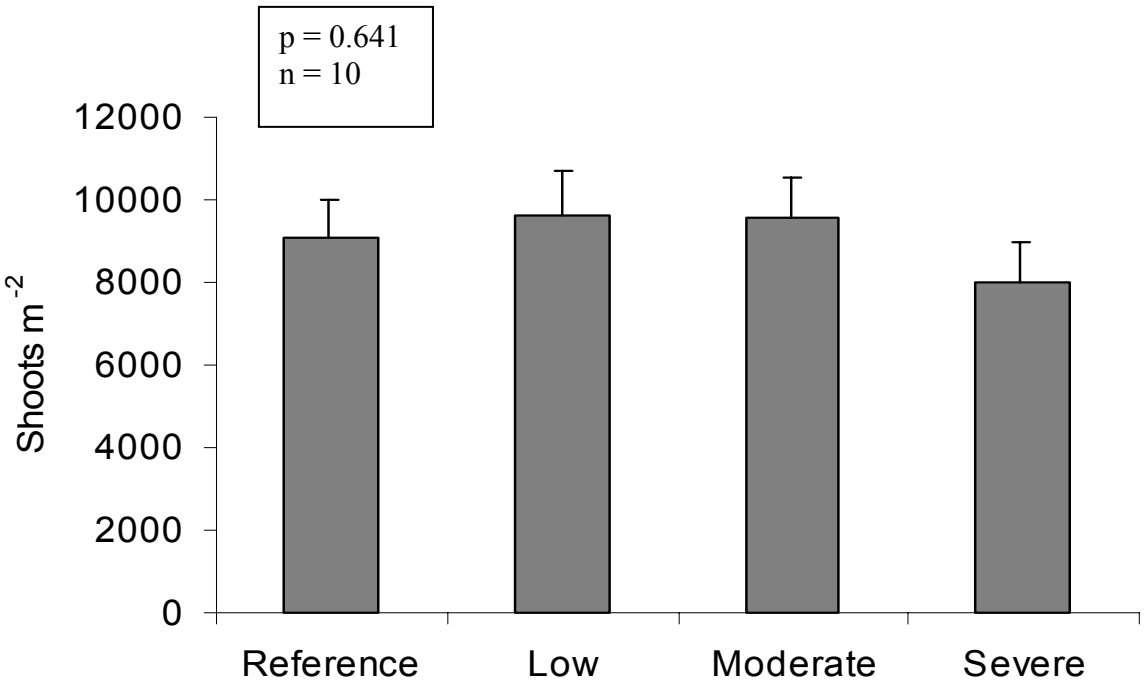



Fig. 12. Shoot density of *Halodule wrightii* measured in spring of 2004, Redfish Bay, Texas. The P-value is from an ANOVA comparing shoot density within each scarring level.

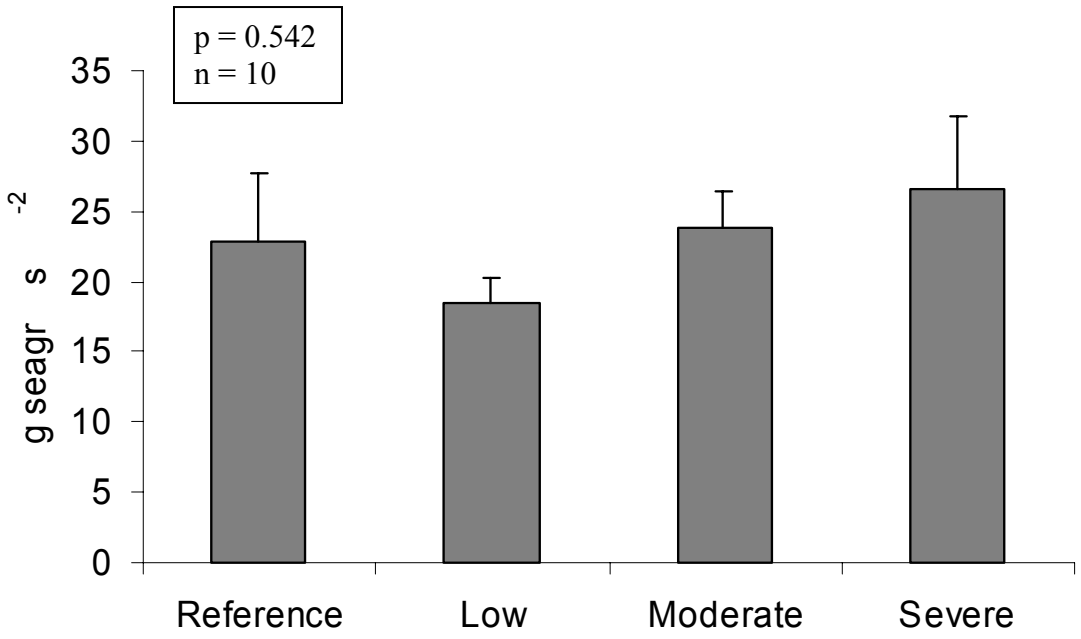



Fig. 13. Above ground biomass of *Halodule wrightii* measured in spring of 2004, Redfish Bay, Texas. The P-value is from an ANOVA comparing aboveground biomass within each scarring level.

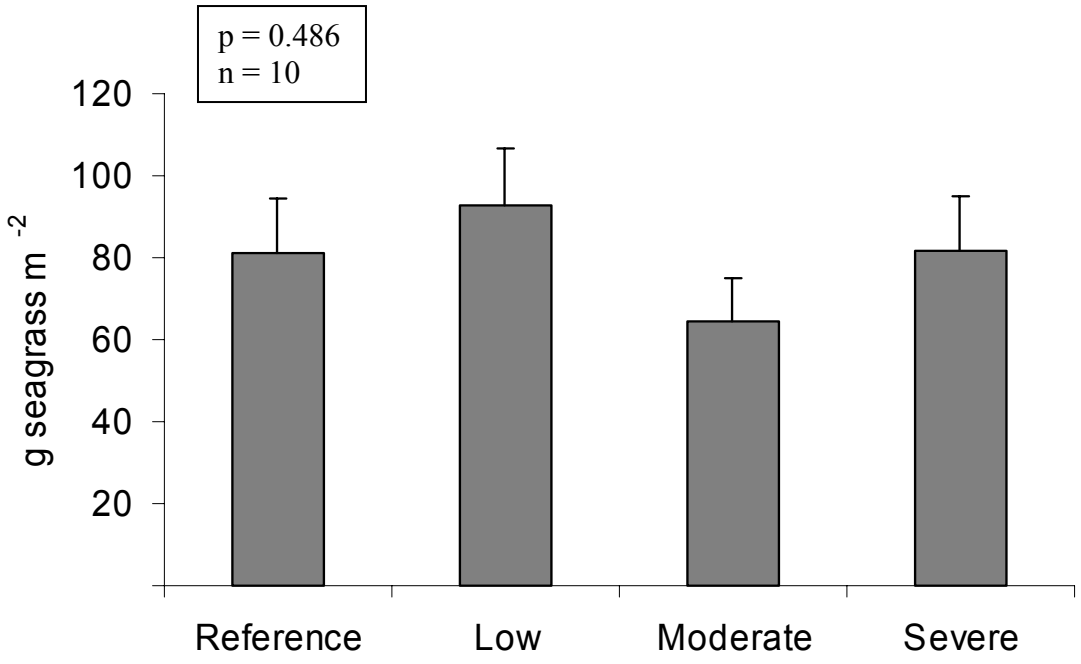



Fig. 14. Below ground biomass of *Halodule wrightii* measured in spring of 2004, Redfish Bay, Texas. The P-value is from an ANOVA comparing belowground biomass within each scarring level.

Nekton Density

I collected a total of 24 species of fish and 6 species of crustaceans in the summer, 23 species of fish and 6 species of crustaceans in the fall, 20 species of fish and 6 species of crustaceans in the winter, and 18 species of fish and 4 species of crustaceans in the spring (Table 1). Crustaceans numerically dominated the catch and accounted for 84% in summer, 92% in fall, 87% in winter, and 77% in spring of the total fauna. As would be expected, there were differences in nekton densities and composition in relation to season. However, there were 8 taxa numerically dominant in all seasons: pinfish, pipefish, code goby, darter goby, killifish, blue crab, Atlantic mud crab, and grass shrimp (Fig. 16). I found (all species included) no significant differences across varying levels of scarring intensity (Fig. 15, Table 2).

Table 1. Mean density (± SE) of 8 numerically dominant taxa and 5 seasonally abundant taxa collected in different scarring intensities. Samples were collected in summer and fall 2003 and winter and spring 2004 in Redfish bay, Texas, USA using an epibenthic sled.

| | Reference | | Low | | Moderate | | Severe | |
|---|---|---|---|---|---|---|---|---|
| **Summer** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| Overall | 18.07 | 3.36 | 16.11 | 2.96 | 13.00 | 2.62 | 14.16 | 4.25 |
| *Lagodon rhomboides* pinfish | 0.17 | 0.04 | 0.17 | 0.04 | 0.24 | 0.08 | 0.24 | 0.08 |
| *Gobionellus robustum* code goby | 0.25 | 0.07 | 0.44 | 0.15 | 0.46 | 0.21 | 0.50 | 0.26 |
| *Gobionellus boleosoma* darter goby | 2.05 | 0.55 | 0.91 | 0.30 | 1.32 | 0.45 | 0.86 | 0.37 |
| *Syngnathus* spp. pipefish | 0.52 | 0.27 | 0.25 | 0.13 | 0.39 | 0.19 | 0.40 | 0.22 |
| Fundulidae killifish | 0.33 | 0.27 | 0.20 | 0.10 | 0.88 | 0.42 | 0.04 | 0.02 |
| *Callinectes sapidus* blue crab | 0.58 | 0.12 | 0.63 | 0.18 | 0.57 | 0.08 | 0.54 | 0.15 |
| *Panopeus herbstii* Atlantic mud crab | 0.16 | 0.07 | 0.07 | 0.03 | 0.24 | 0.18 | 0.20 | 0.07 |
| Penaeid Shrimp | 1.00 | 0.35 | 0.86 | 0.22 | 1.02 | 0.32 | 0.74 | 0.32 |
| *Farfantepenaeus aztecus* brown shrimp | 0.68 | 0.31 | 0.66 | 0.19 | 0.73 | 0.24 | 0.34 | 0.13 |
| *Palaemonetes* spp. grass shrimp | 14.73 | 3.08 | 12.45 | 2.71 | 9.99 | 2.30 | 10.78 | 3.66 |
| | | | | | | | | |
| **Fall** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| Overall | 32.75 | 6.79 | 33.09 | 8.51 | 40.20 | 8.01 | 29.17 | 6.52 |
| *Lagodon rhomboides* pinfish | 0.61 | 0.29 | 0.35 | 0.11 | 0.39 | 0.16 | 0.45 | 0.31 |
| *Gobionellus robustum* code goby | 0.54 | 0.20 | 0.72 | 0.28 | 0.71 | 0.30 | 0.83 | 0.40 |
| *Gobionellus boleosoma* darter goby | 1.00 | 0.36 | 1.11 | 0.30 | 1.30 | 0.53 | 1.27 | 0.54 |
| *Syngnathus* spp. pipefish | 0.97 | 0.50 | 0.64 | 0.13 | 1.18 | 0.55 | 0.95 | 0.44 |
| Fundulidae killifish | 0.12 | 0.10 | 0.03 | 0.02 | 0.03 | 0.02 | 0.21 | 0.21 |
| *Sciaenops ocellatus* red drum | 0.10 | 0.05 | 0.12 | 0.06 | 0.11 | 0.05 | 0.11 | 0.05 |
| *Callinectes sapidus* blue crab | 0.95 | 0.22 | 1.09 | 0.22 | 0.87 | 0.18 | 0.57 | 0.17 |
| *Panopeus herbstii* Atlantic mud crab | 0.43 | 0.36 | 0.05 | 0.02 | 0.07 | 0.03 | 0.06 | 0.03 |
| Penaeid Shrimp | 1.57 | 0.31 | 1.89 | 0.39 | 1.90 | 0.25 | 1.74 | 0.32 |
| *Farfantepenaeus aztecus* brown shrimp | 1.34 | 0.26 | 1.54 | 0.35 | 1.83 | 0.22 | 1.67 | 0.30 |
| *Palaemonetes* spp. grass shrimp | 27.17 | 6.64 | 27.42 | 8.14 | 34.31 | 7.13 | 23.73 | 5.67 |
| | | | | | | | | |
| **Winter** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| Overall | 13.10 | 6.14 | 7.41 | 2.63 | 9.02 | 4.46 | 8.20 | 3.20 |
| *Lagodon rhomboides* pinfish | 0.08 | 0.03 | 0.07 | 0.02 | 0.09 | 0.02 | 0.03 | 0.02 |
| *Gobionellus robustum* code goby | 0.13 | 0.09 | 0.12 | 0.06 | 0.04 | 0.03 | 0.05 | 0.03 |
| *Gobionellus boleosoma* darter goby | 0.37 | 0.13 | 0.58 | 0.29 | 0.40 | 0.23 | 0.25 | 0.11 |
| *Syngnathus* spp. pipefish | 0.16 | 0.09 | 0.04 | 0.02 | 0.12 | 0.07 | 0.02 | 0.01 |
| Fundulidae killifish | 0.04 | 0.02 | 0.04 | 0.04 | 0.02 | 0.01 | 0.05 | 0.03 |
| *Callinectes sapidus* blue crab | 0.91 | 0.26 | 0.90 | 0.34 | 1.02 | 0.54 | 1.16 | 0.53 |
| *Panopeus herbstii* Atlantic mud crab | 0.06 | 0.03 | 0.06 | 0.03 | 0.02 | 0.01 | 0.02 | 0.01 |
| Penaeid Shrimp | 0.17 | 0.09 | 0.25 | 0.20 | 0.18 | 0.09 | 0.13 | 0.05 |
| *Palaemonetes* spp. grass shrimp | 11.78 | 6.20 | 5.25 | 2.44 | 6.58 | 3.65 | 5.90 | 2.80 |
| | | | | | | | | |
| **Spring** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| Overall | 19.97 | 5.87 | 19.05 | 5.15 | 27.98 | 7.12 | 23.84 | 6.21 |
| *Lagodon rhomboides* pinfish | 1.56 | 0.91 | 2.00 | 0.98 | 2.46 | 1.12 | 2.95 | 1.27 |
| *Gobionellus robustum* code goby | 0.54 | 0.20 | 0.72 | 0.28 | 0.71 | 0.30 | 0.83 | 0.40 |
| *Gobionellus boleosoma* darter goby | 1.37 | 0.77 | 1.41 | 0.64 | 2.36 | 0.78 | 1.55 | 0.45 |
| *Syngnathus* spp. pipefish | 0.19 | 0.09 | 0.19 | 0.10 | 0.26 | 0.13 | 0.38 | 0.19 |
| Fundulidae killifish | 0.01 | 0.01 | 0.01 | 0.01 | 0.01 | 0.01 | 0.01 | 0.01 |
| *Leiostomus xanthurus* spot | 0.34 | 0.16 | 0.30 | 0.09 | 0.81 | 0.41 | 0.78 | 0.28 |
| *Citharichthys spilopterus* bay whiff | 0.33 | 0.10 | 0.19 | 0.04 | 0.24 | 0.06 | 0.21 | 0.07 |
| *Callinectes sapidus* blue crab | 2.61 | 0.63 | 2.21 | 0.61 | 4.11 | 0.90 | 2.30 | 0.47 |
| *Panopeus herbstii* Atlantic mud crab | 0.20 | 0.11 | 0.07 | 0.03 | 0.32 | 0.14 | 0.34 | 0.17 |
| Penaeid Shrimp | 3.58 | 1.07 | 5.27 | 1.99 | 9.12 | 3.28 | 7.83 | 2.87 |
| *Litopenaeus setiferus* white shrimp | 2.80 | 0.85 | 4.88 | 1.86 | 7.82 | 2.98 | 6.61 | 2.58 |
| *Palaemonetes* spp. grass shrimp | 9.65 | 4.34 | 7.18 | 2.75 | 8.90 | 2.84 | 8.08 | 2.30 |

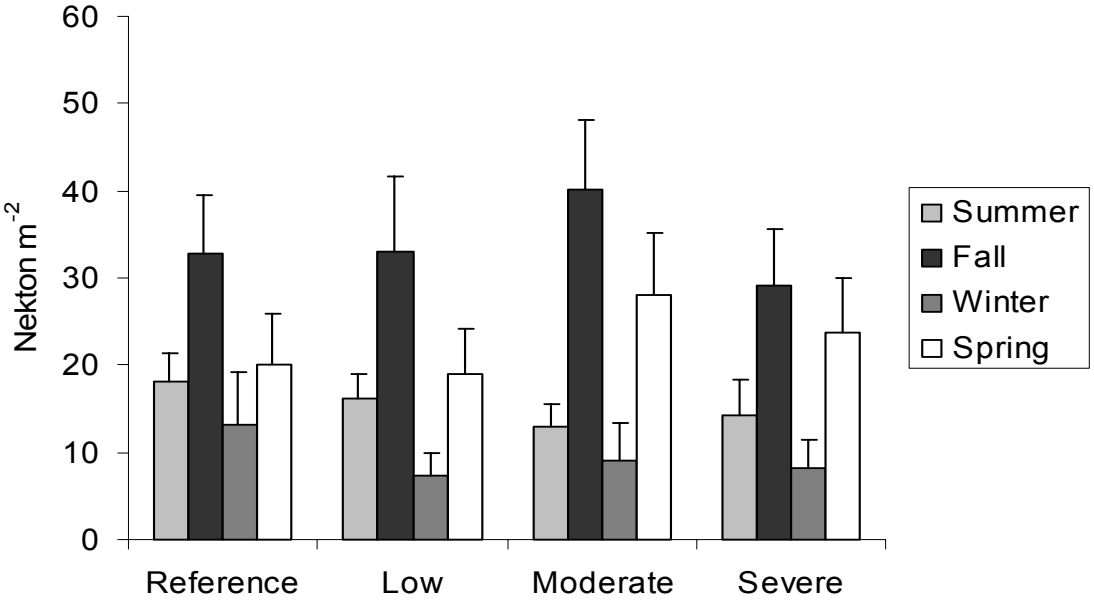


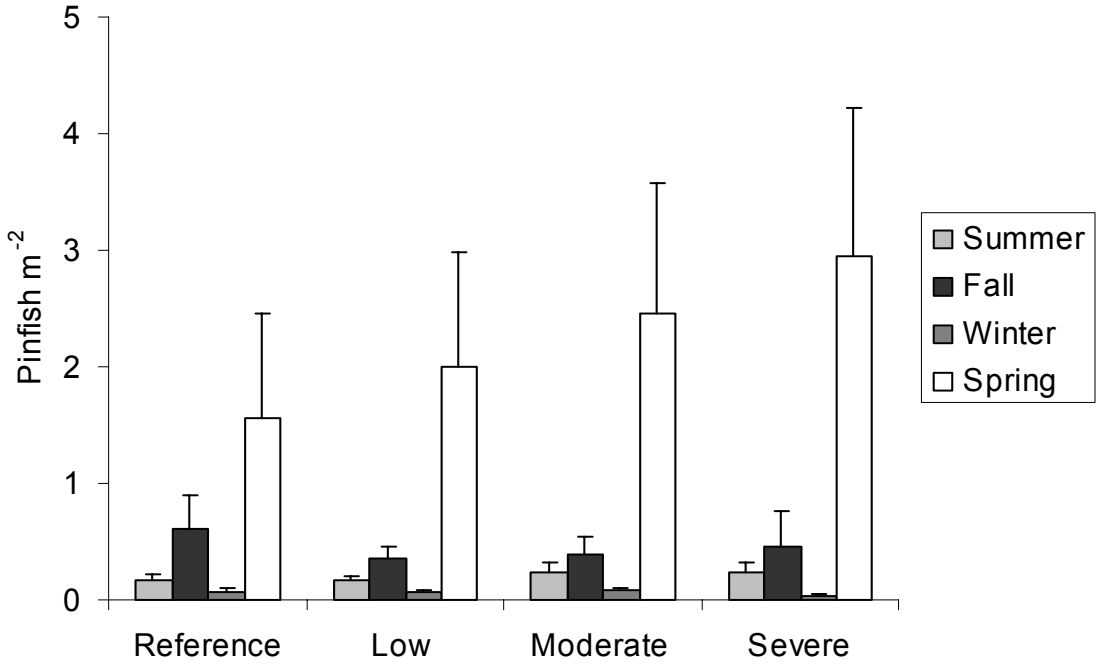

Fig. 15. Mean nekton density (all species combined) in each season. Differences in means were analyzed with an ANOVA.

Table 2. Analysis of variance table for nekton density patterns in Redfish Bay, Texas.

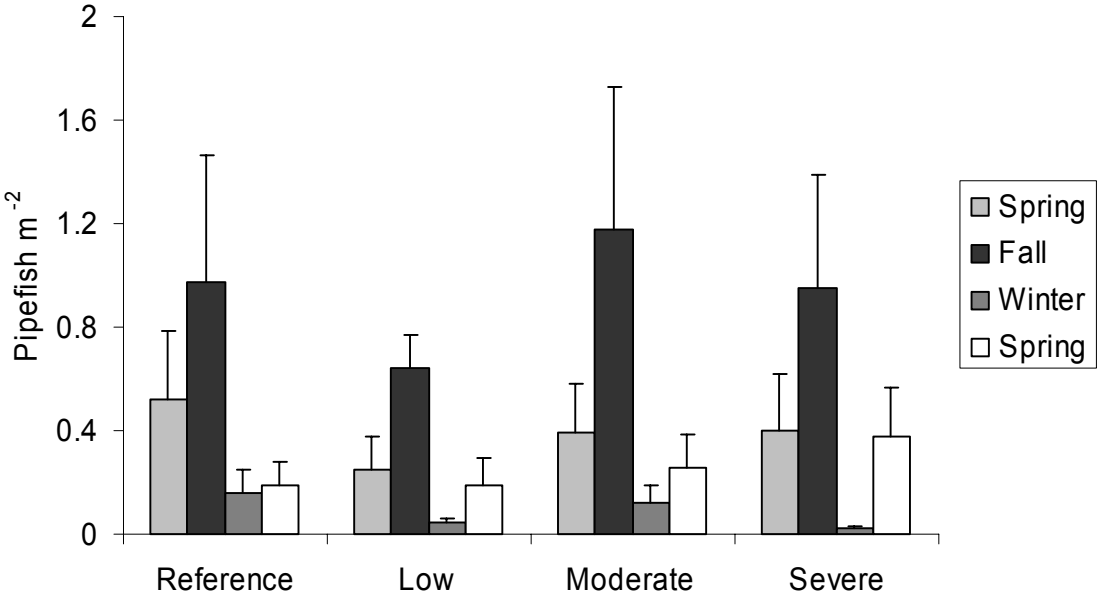
| | Summer | | | | Fall | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | SS | *F* | p | n | SS | *F* | p |
| Overall | 10 | 0.215 | 0.586 | 0.628 | 10 | 0.158 | 0.474 | 0.702 |
| *Lagodon rhomboides* pinfish | 10 | 0.004 | 0.260 | 0.854 | 10 | 0.003 | 1.419 | 0.253 |
| *Gobionellus robustum* code goby | 10 | 0.022 | 0.345 | 0.793 | 10 | 0.008 | 0.065 | 0.978 |
| *Gobionellus boleosoma* darter goby | 10 | 0.207 | 1.119 | 0.355 | 10 | 0.006 | 0.038 | 0.990 |
| *Syngnathus* spp. pipefish | 10 | 0.016 | 0.212 | 0.888 | 10 | 0.016 | 0.114 | 0.951 |
| Fundulidae killifish | 10 | 0.025 | 0.726 | 0.543 | 10 | 0.011 | 0.437 | 0.728 |
| *Sciaenops ocellatus* red drum | | | | | 10 | 0.000 | 0.010 | 0.999 |
| *Callinectes sapidus* blue crab | 10 | 0.008 | 0.223 | 0.880 | 10 | 0.078 | 1.351 | 0.273 |
| *Panopeus herbstii* Atlantic mud crab | 10 | 0.011 | 0.443 | 0.723 | 10 | 0.033 | 0.963 | 0.421 |
| Penaeid Shrimp | 10 | 0.020 | 0.210 | 0.888 | 10 | 0.027 | 0.312 | 0.816 |
| *Farfantepenaeus aztecus* brown shrimp | 10 | 0.055 | 0.790 | 0.508 | 10 | 0.053 | 0.628 | 0.602 |
| *Palaemonetes* spp. grass shrimp | 10 | 0.332 | 0.813 | 0.496 | 10 | 0.230 | 0.459 | 0.712 |

| | Winter | | | | Spring | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | SS | *F* | p | n | SS | *F* | p |
| Overall | 10 | 0.072 | 0.113 | 0.952 | 10 | 0.198 | 0.479 | 0.699 |
| *Lagodon rhomboides* pinfish | 10 | 0.013 | 0.153 | 0.927 | 10 | 0.124 | 0.284 | 0.836 |
| *Gobionellus robustum* code goby | 10 | 0.005 | 0.467 | 0.707 | 10 | 0.009 | 0.103 | 0.958 |
| *Gobionellus boleosoma* darter goby | 10 | 0.016 | 0.235 | 0.872 | 10 | 0.184 | 0.928 | 0.437 |
| *Syngnathus* spp. pipefish | 10 | 0.005 | 0.249 | 0.862 | 10 | 0.013 | 0.258 | 0.855 |
| Fundulidae killifish | 10 | 0.002 | 0.493 | 0.689 | 10 | 0.000 | 0.133 | 0.940 |
| *Leiostomus xanthurus* spot | | | | | 10 | 0.094 | 1.083 | 0.369 |
| *Citharichthys spilopterus* bay whiff | | | | | 10 | 0.007 | 0.655 | 0.587 |
| *Callinectes sapidus* blue crab | 10 | 0.001 | 0.007 | 0.999 | 10 | 0.273 | 1.905 | 0.146 |
| *Panopeus herbstii* Atlantic mud crab | 10 | 0.002 | 0.917 | 0.443 | 10 | 0.039 | 1.064 | 0.377 |
| Penaeid Shrimp | 10 | 0.002 | 0.218 | 0.882 | 10 | 0.351 | 0.557 | 0.647 |
| *Litopenaeus setiferus* white shrimp | | | | | 10 | 0.224 | 0.334 | 0.801 |
| *Palaemonetes* spp. grass shrimp | 10 | 0.238 | 0.312 | 0.817 | 10 | 0.100 | 0.151 | 0.928 |

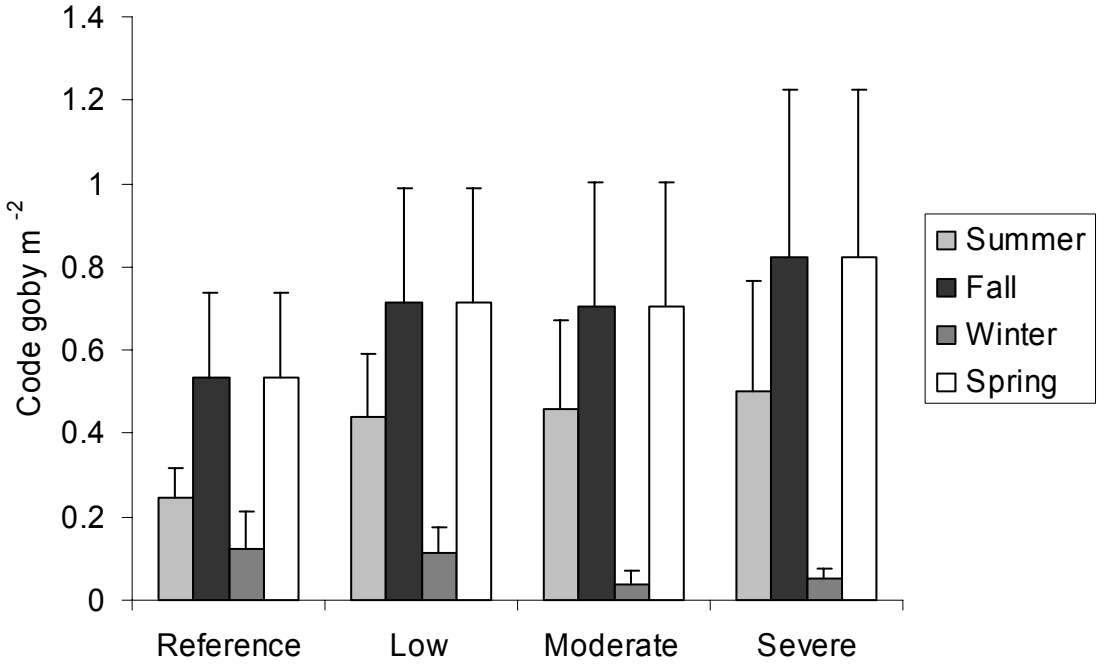
(A) Pinfish



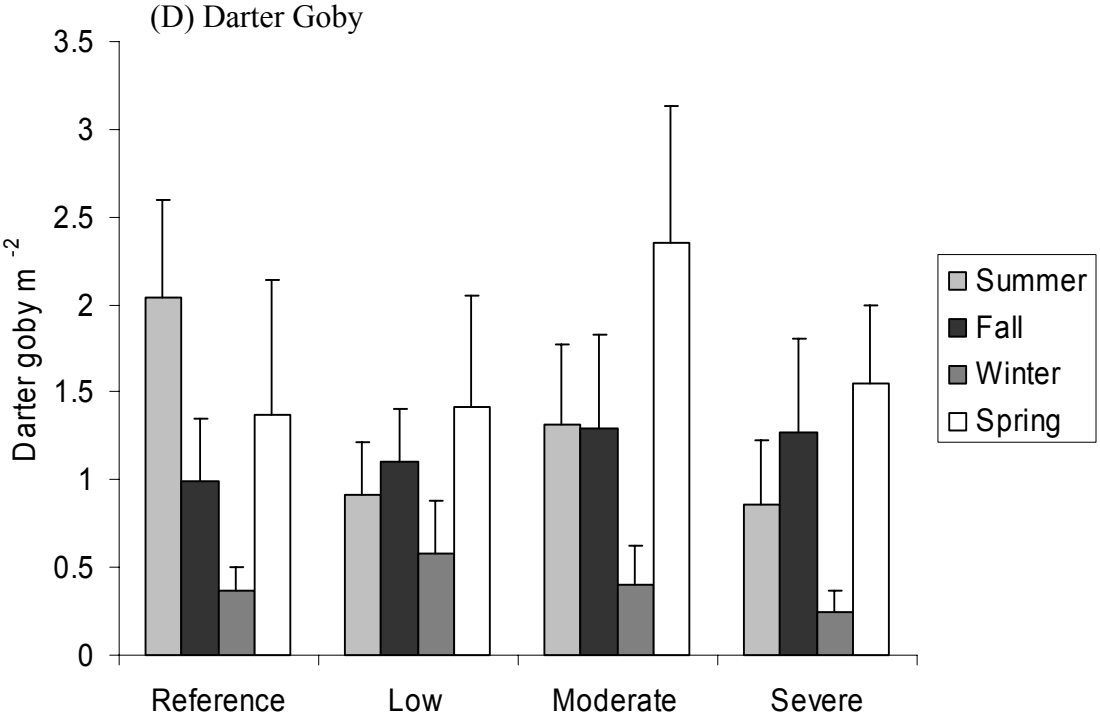

(B) Pipefish



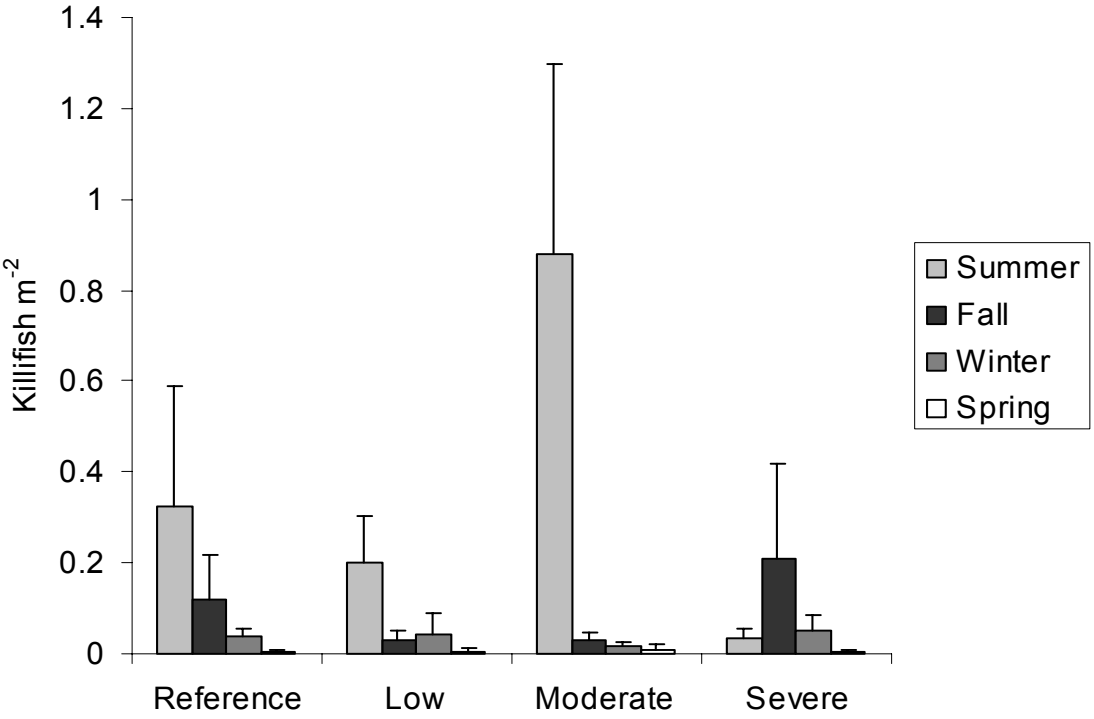
(C) Code Goby



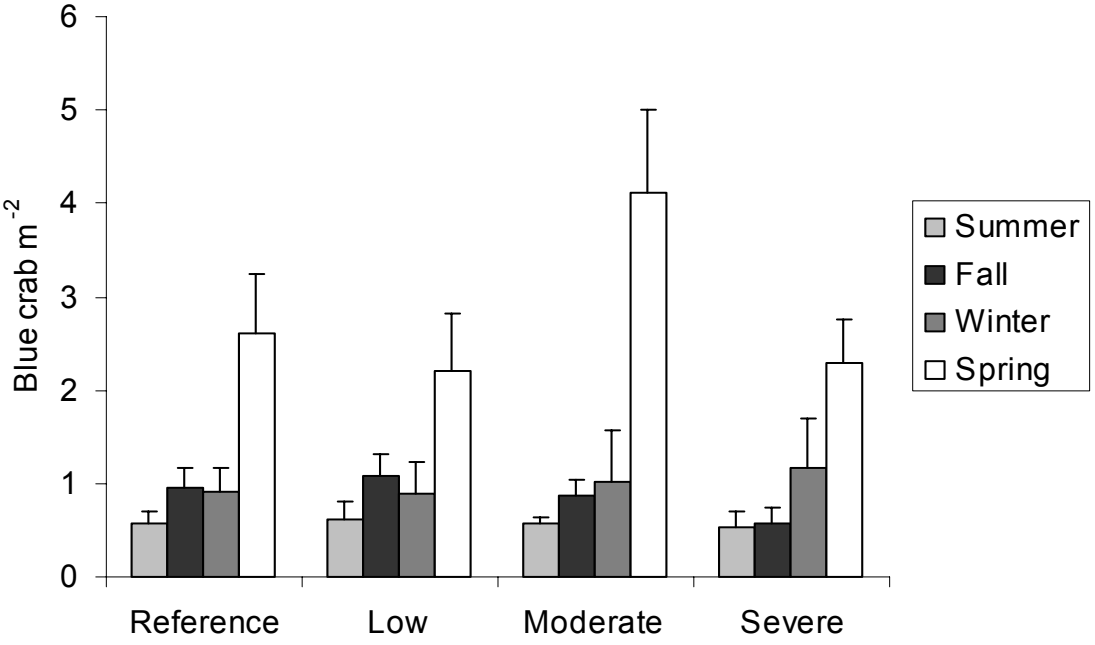

(D) Darter Goby



(E) Killifish




(F) Blue Crab



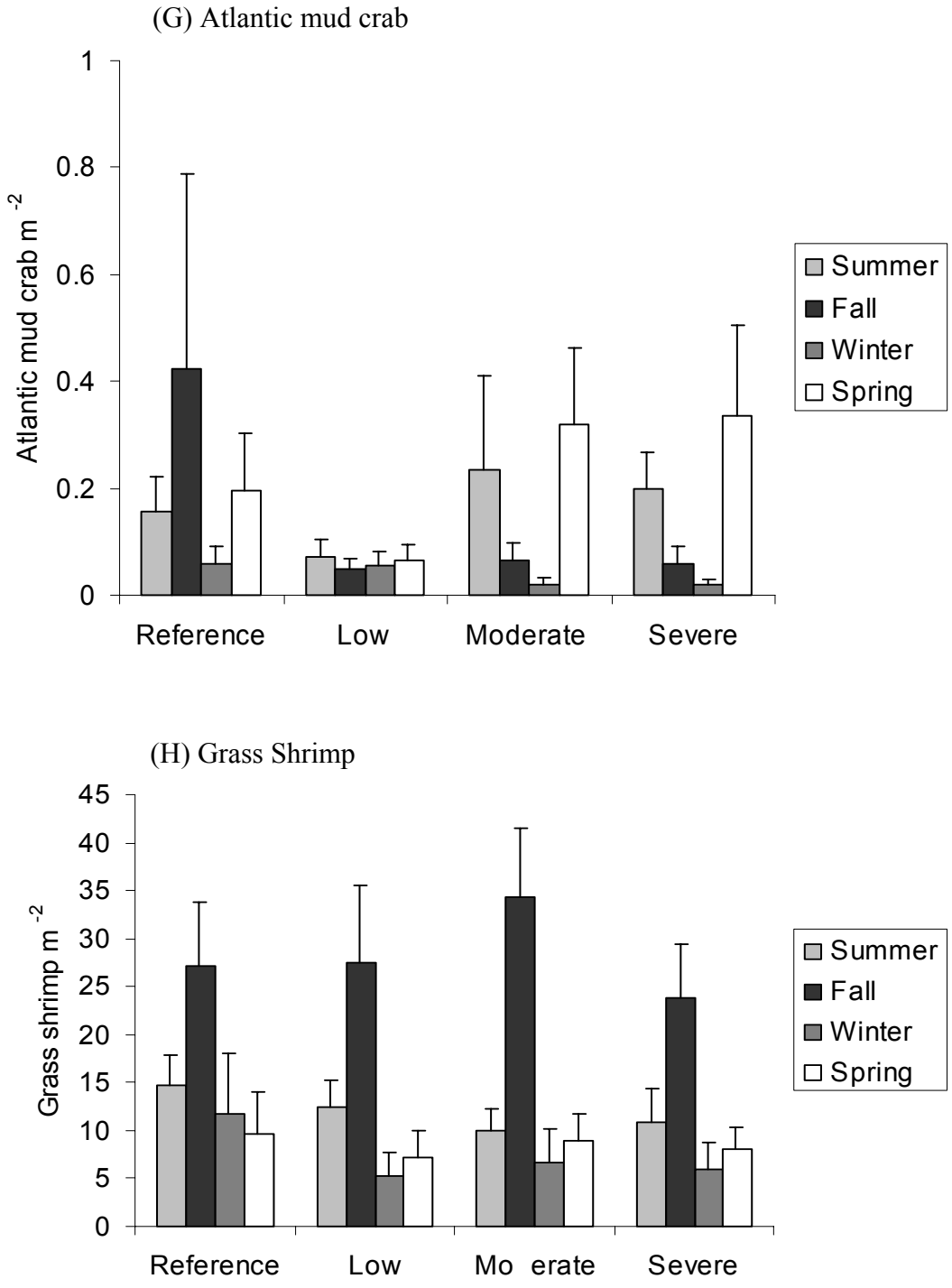



Fig. 16. Mean density (± SE) of target organism collected with epibenthic sleds in Redfish Bay, Texas in summer and fall 2003 and winter and spring 2004. Each organism in each season was analyzed separately (N=10) with analysis of variance. Graph A represents pinfish density. Graph B represents pipefish density. Graph C represents code goby density. Graph D represents darter goby density. Graph E represents killifish density. Graph F represents blue crab density. Graph G represents mud crab density. Graph H represents grass shrimp density.

In summer, the 8 dominant taxa accounted for 94% of the total catch. Brown shrimp accounted for an additional 2% of the total catch in summer (Fig. 17). Densities of the 9 most abundant taxa in spring were not significantly different in relation to scarring intensity (Table 2).

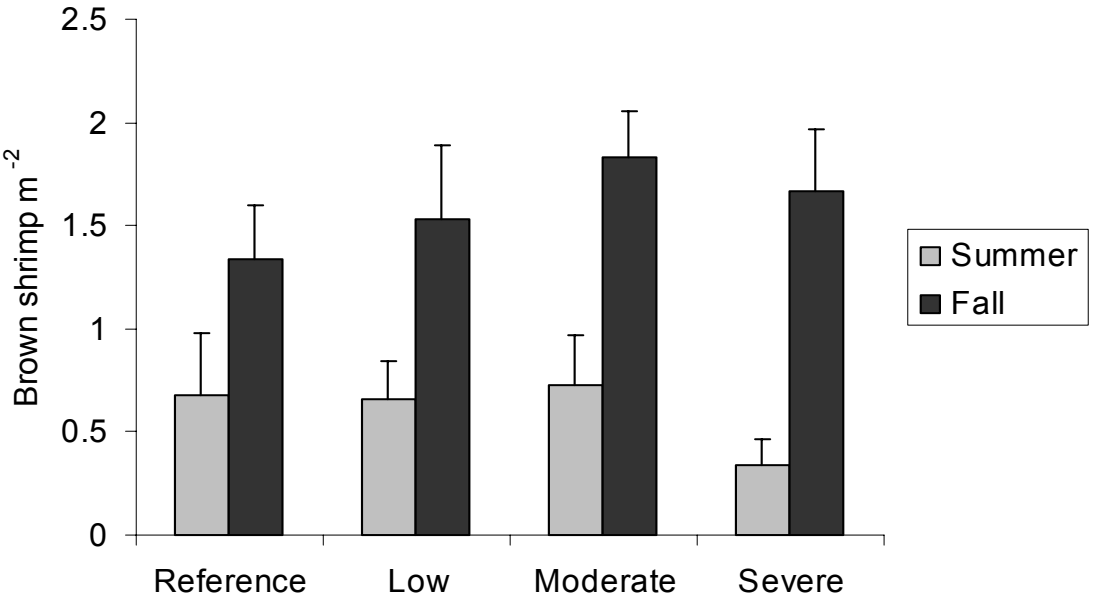



Fig. 17. Mean density (± SE) of brown shrimp during summer and fall 2003 with an epibenthic sled in Redfish Bay, Texas. Seasons were analyzed individually with analysis of variance.

In fall, the 8 dominant taxa again accounted for 94% of the total catch. Brown shrimp (Fig. 17) and red drum (Fig 18) accounted for 5% and 0.32% total catch respectively. Densities of the 10 most abundant taxa in fall were not significantly different in relation to scarring intensity (Table 2).

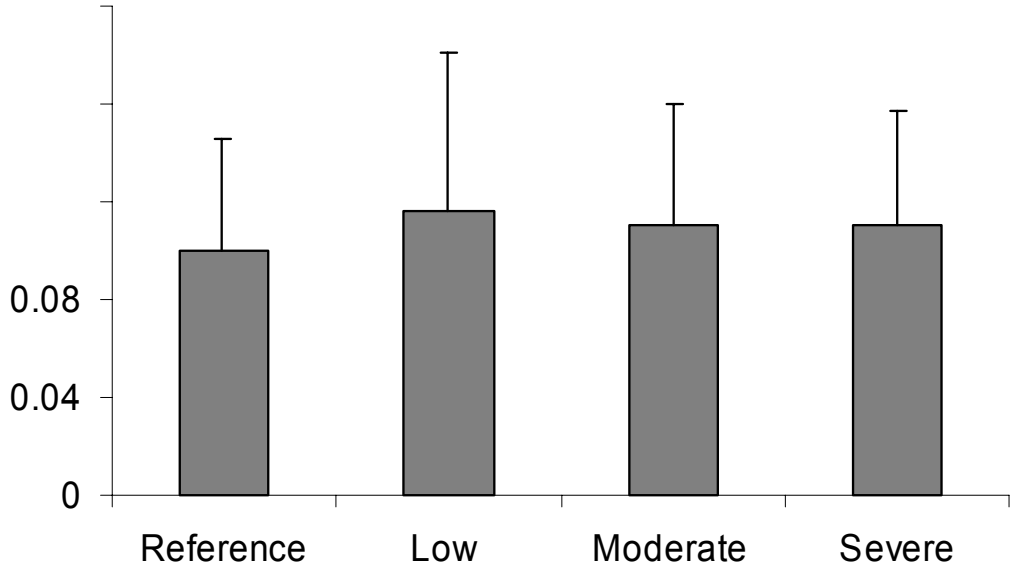



Fig. 18. Mean density (± SE) of red drum collected in fall with an epibenthic sled in Redfish Bay, Texas. Densities were analyzed with analysis of variance.

In winter, the 8 most abundant taxa accounted for 97% of the total catch. As evident in figure (Fig. 15) winter had the lowest organism density in relation to the other seasons. However, there were no differences in the density of the 8 most abundant taxa in relation to scarring intensity (Table 2).

In spring, the 8 most abundant taxa accounted for 71% of the total catch. White shrimp (Fig. 19), spot (Fig. 20), and bay whiff (Fig. 21) accounted for an additional 24%, 2%, and 1% of the total catch respectively. The greatest numbers of organisms were collected in the spring. However, there were no significant differences in density of the 11 most abundant species in relation to scarring intensity (Table 2).

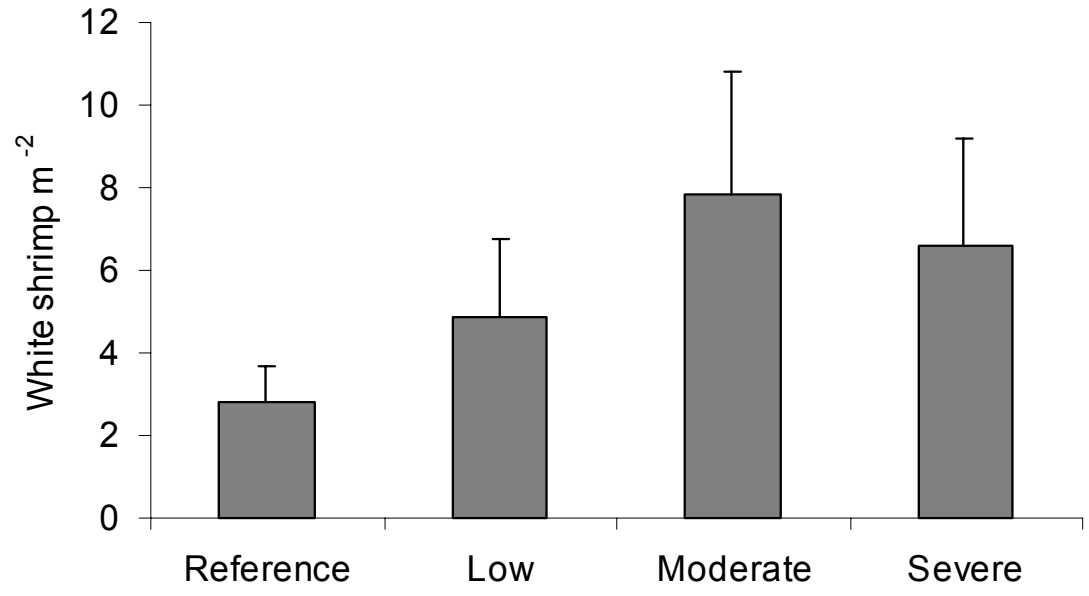



Fig. 19. Mean density (± SE) of white shrimp collected in spring with an epibenthic sled in Redfish Bay, Texas. Densities were analyzed with analysis of variance.

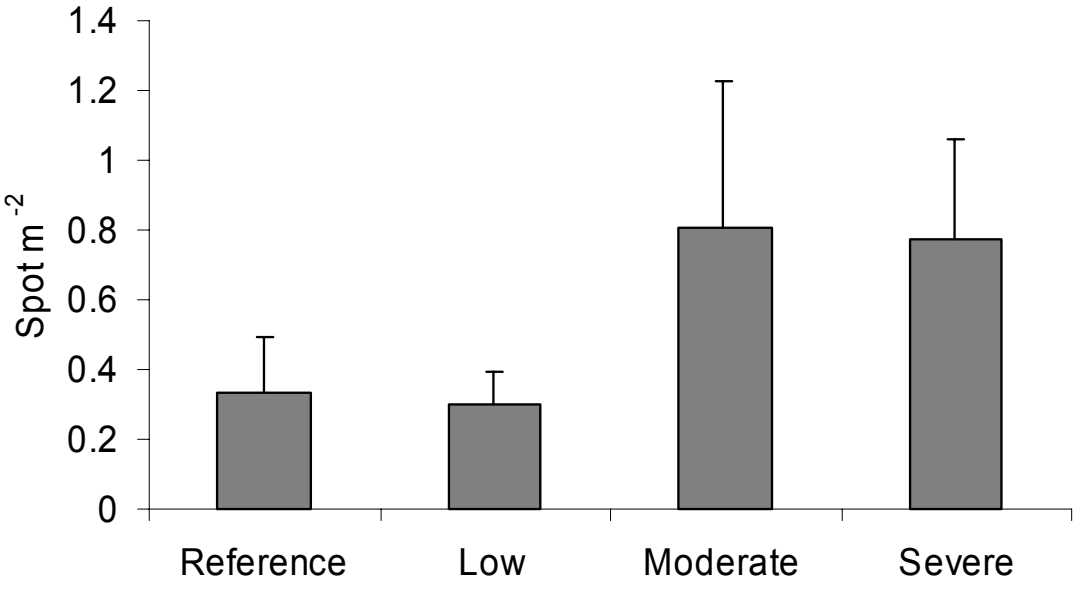



Fig. 20. Mean density (± SE) of spot collected in spring with an epibenthic sled in Redfish Bay, Texas. Densities were analyzed with analysis of variance.

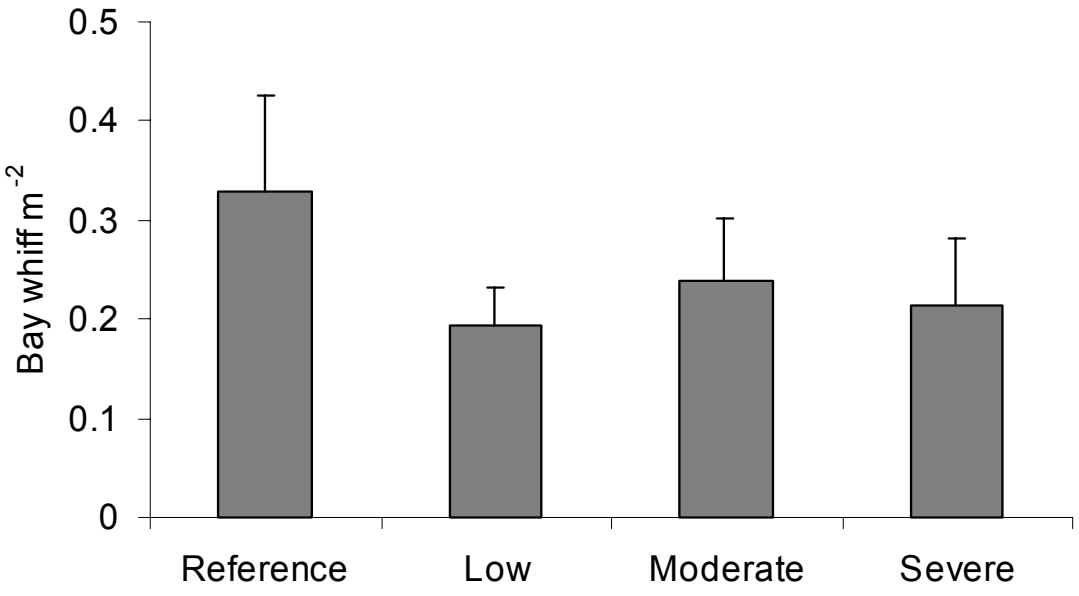
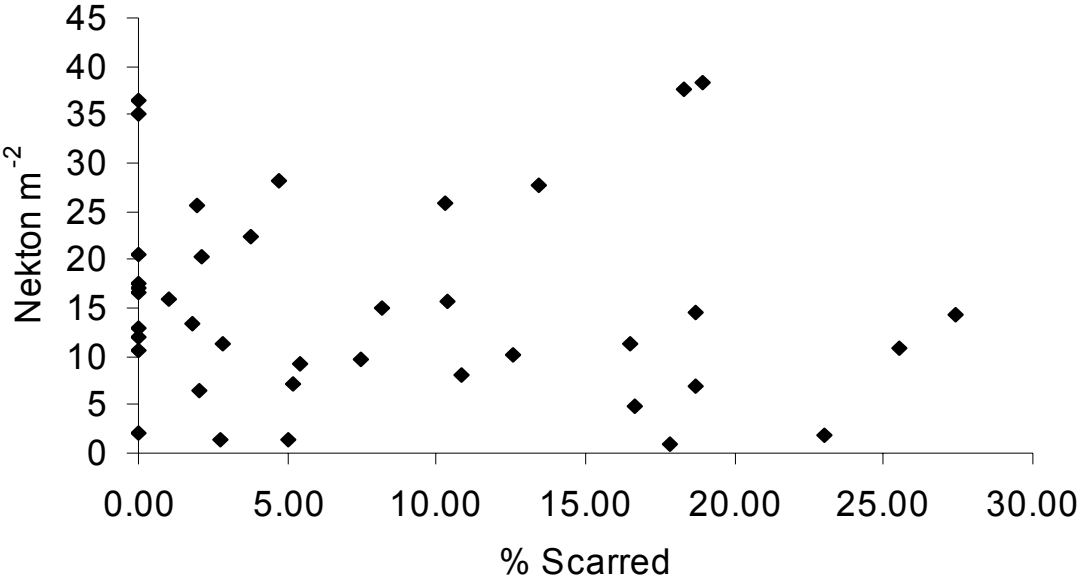


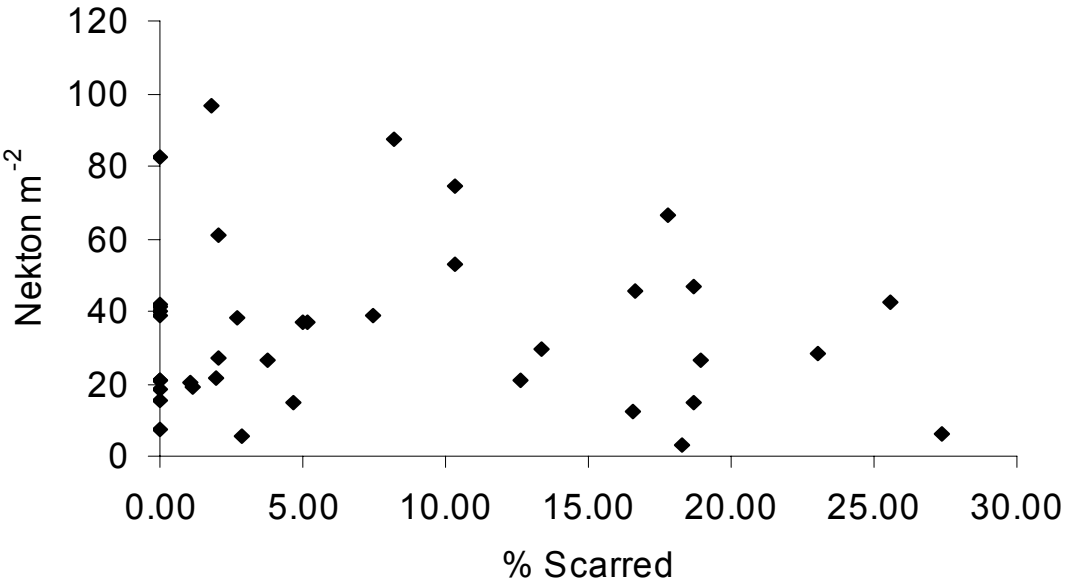

Fig. 21. Mean density (± SE) of bay whiff collected in spring with an epibenthic sled in Redfish Bay, Texas. Densities were analyzed with analysis of variance.

Each site was measured for total scarring intensity. A plot of scarring intensity versus nekton density (all species included; Fig. 22) showed no relationship (Table 3). This analysis was run each season for the dominant species of pinfish, code goby, darter goby, red drum, bay whiff, spot, blue crab, Atlantic mud crab, penaeid shrimp, white shrimp, brown shrimp, and grass shrimp. Results were not significant for any species in all seasons (Table 3).

(A) Summer




(B) Fall



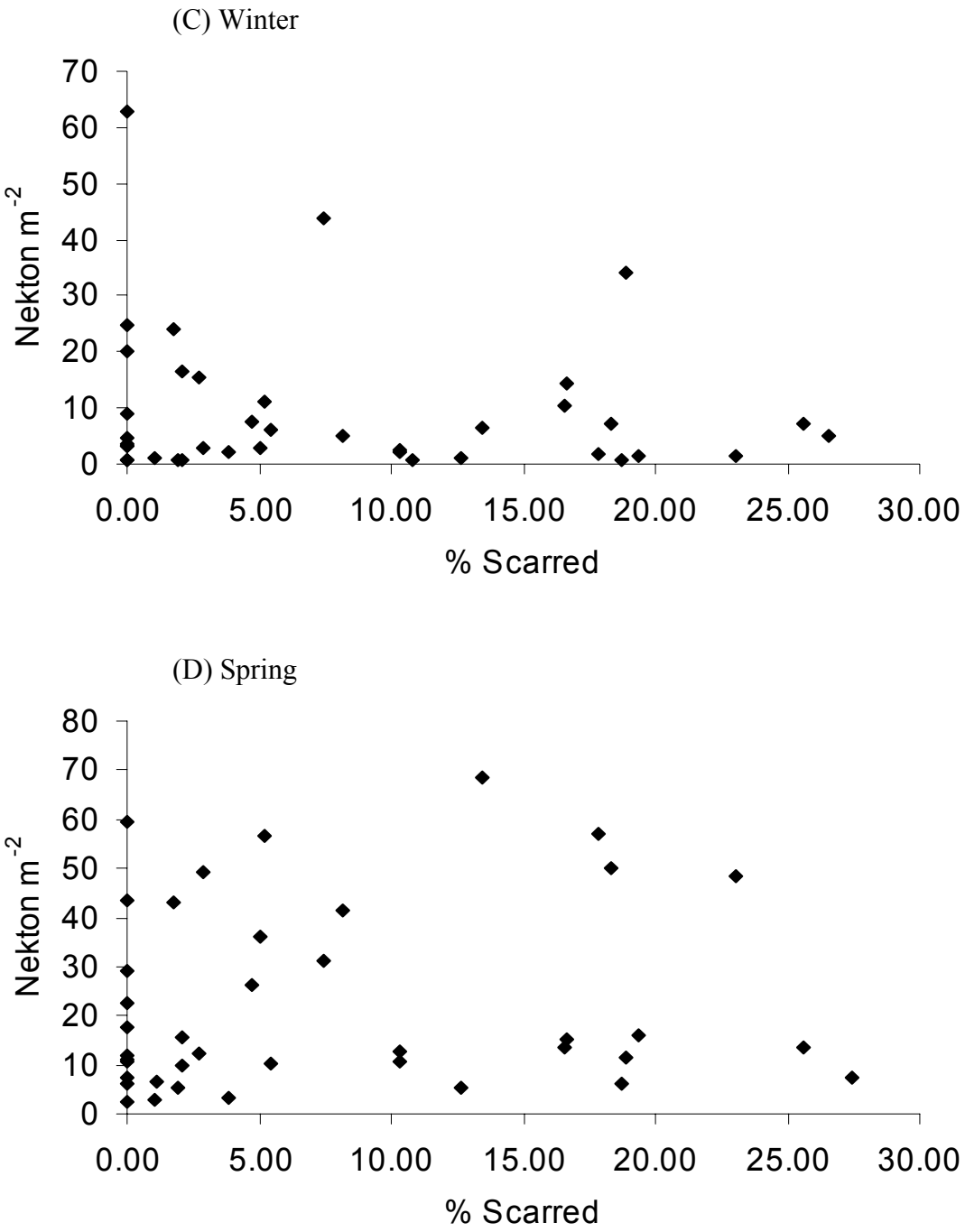



Fig. 22. Linear regression of scarring intensity versus nekton density (all species included). Graph A represents scarring intensity versus summer nekton density. Graph B represents scarring intensity versus fall nekton density. Graph C represents scarring intensity versus winter nekton density. Graph D represents scarring intensity versus spring nekton density.

Table 3. Summary of linear regressions to determine the relationship between scarring intensity and organism density.

| | Summer | | | | Fall | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | *F* | p | $r^2$ | n | *F* | p | $r^2$ |
| Overall | 40 | 0.278 | 0.601 | 0.007 | 40 | 0.240 | 0.627 | 0.007 |
| *Lagodon rhomboides* pinfish | 40 | 0.899 | 0.349 | 0.024 | 40 | 0.015 | 0.902 | 0.000 |
| *Gobionellus robustum* code goby | 40 | 0.822 | 0.370 | 0.022 | 40 | 0.533 | 0.470 | 0.014 |
| *Gobionellus boleosoma* darter goby | 40 | 0.492 | 0.487 | 0.013 | 40 | 1.233 | 0.274 | 0.031 |
| *Syngnathus* spp. pipefish | 40 | 0.070 | 0.793 | 0.002 | 40 | 0.082 | 0.776 | 0.002 |
| *Sciaenops ocellatus* red drum | | | | | 40 | 0.096 | 0.759 | 0.003 |
| *Callinectes sapidus* blue crab | 40 | 0.130 | 0.720 | 0.004 | 40 | 1.111 | 0.298 | 0.028 |
| *Panopeus herbstii* Atlantic mud crab | 40 | 0.456 | 0.504 | 0.012 | 40 | 0.698 | 0.409 | 0.018 |
| Penaeid Shrimp | 40 | 0.007 | 0.935 | 0.000 | 40 | 0.108 | 0.745 | 0.003 |
| *Farfantepenaeus aztecus* brown shrimp | 40 | 0.745 | 0.394 | 0.021 | 40 | 0.055 | 0.817 | 0.002 |
| *Palaemonetes* spp. grass shrimp | 40 | 0.847 | 0.364 | 0.024 | 40 | 0.045 | 0.833 | 0.001 |

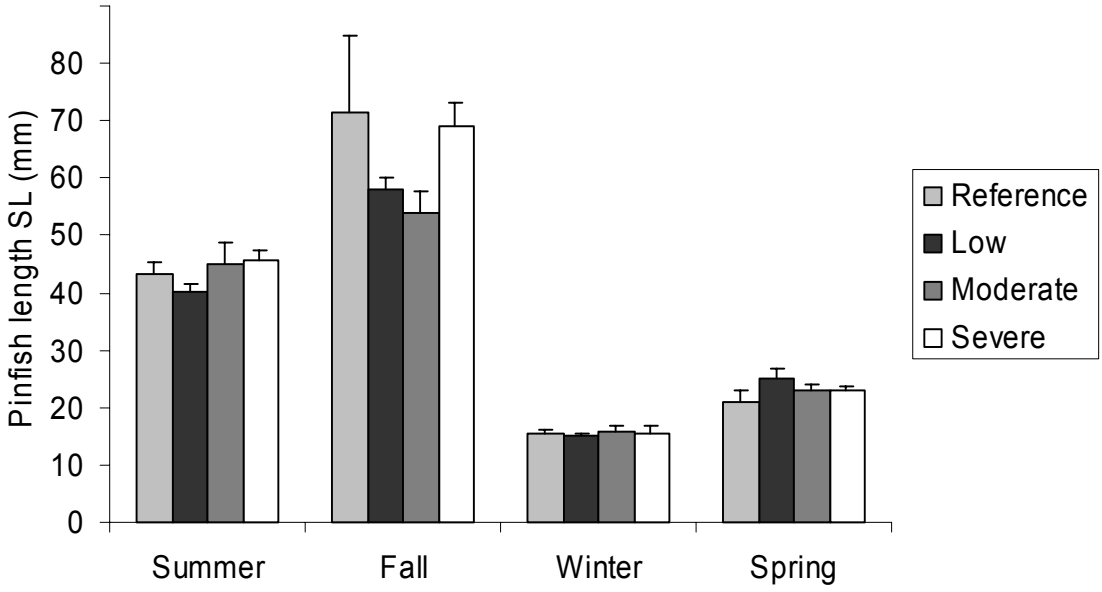
| | Winter | | | | Spring | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | *F* | p | $r^2$ | n | *F* | p | $r^2$ |
| Overall | 40 | 0.702 | 0.407 | 0.019 | 40 | 0.343 | 0.561 | 0.009 |
| *Lagodon rhomboides* pinfish | 40 | 5.180 | 0.029 | 0.120 | 40 | 0.478 | 0.494 | 0.012 |
| *Gobionellus robustum* code goby | 40 | 1.130 | 0.295 | 0.030 | 40 | 0.345 | 0.560 | 0.009 |
| *Gobionellus boleosoma* darter goby | 40 | 0.807 | 0.375 | 0.021 | 40 | 0.008 | 0.928 | 0.000 |
| *Syngnathus* spp. pipefish | 40 | 0.431 | 0.516 | 0.012 | 40 | 0.001 | 0.974 | 0.000 |
| *Leiostomus xanthurus* spot | | | | | 40 | 2.462 | 0.125 | 0.061 |
| *Citharichthys spilopterus* bay whiff | | | | | 40 | 0.325 | 0.573 | 0.011 |
| *Callinectes sapidus* blue crab | 40 | 0.327 | 0.571 | 0.009 | 40 | 0.142 | 0.708 | 0.004 |
| *Panopeus herbstii* Atlantic mud crab | 40 | 0.603 | 0.442 | 0.016 | 40 | 2.721 | 0.107 | 0.067 |
| Penaeid Shrimp | 40 | 0.421 | 0.528 | 0.034 | 40 | 1.684 | 0.203 | 0.045 |
| *Litopenaeus setiferus* white shrimp | | | | | 40 | 1.061 | 0.310 | 0.030 |
| *Palaemonetes* spp. grass shrimp | 40 | 0.325 | 0.572 | 0.009 | 40 | 0.073 | 0.789 | 0.002 |

Nekton Size

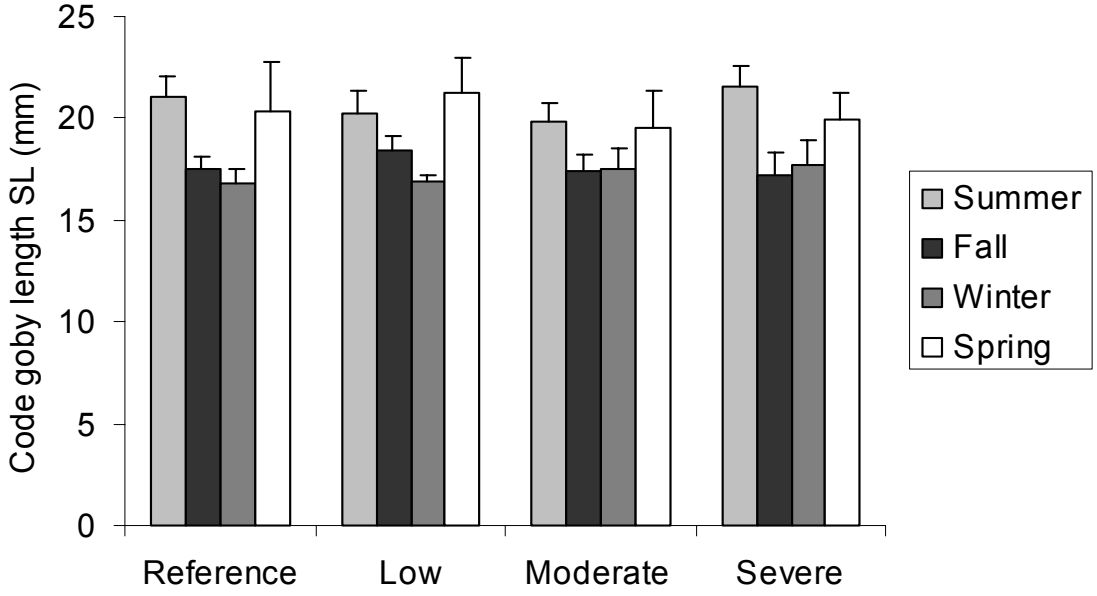
I collected a total of 24 species of fish and 6 species of crustaceans in the summer, 23 species of fish and 6 species of crustaceans in the fall, 20 species of fish and 6 species of crustaceans in the winter, and 18 species of fish and 4 species of crustaceans in the spring. The numerically dominant species in all seasons were: pinfish, code goby, darter goby, blue crab, and Atlantic mud crab. An additional 5 species (red drum, bay whiff, spot, brown shrimp, and white shrimp) were not dominant in all seasons. These species were analyzed in the season(s) where they were most abundant. Mean organism sizes (Table 4, Fig. 22) were analyzed with analysis of variance and there were no significant differences in any species during any season (Table 5).

Table 4. Mean size (± SE) of nekton numerically abundant in season(s) of dominance. Nekton were collected in summer & fall 2003 and winter & spring 2004 in Redfish Bay, Texas using an epibenthic sled.

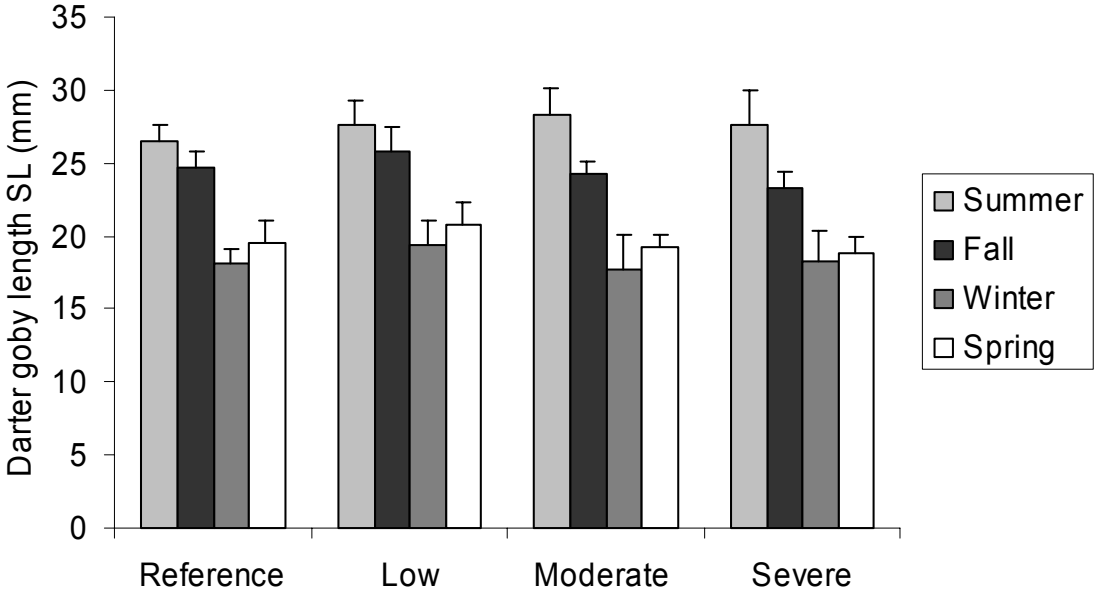
| | Reference | | Low | | Moderate | | Severe | |
|---|---|---|---|---|---|---|---|---|
| **Summer** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| *Lagodon rhomboides* pinfish | | | | | | | | 1) |
| *Gobionellus robustum* code goby | 21.09 | (1.02) | 20.28 | (1.06) | 19.85 | (.94) | 21.54 | (1.) |
| *Gobionellus boleosoma* darter goby | | | | | | | | |
| *Callinectes sapidus* blue crab | 15.30 | (1.64) | 17.77 | (1.96) | 16.72 | (1.82) | 15.15 | (2.02) |
| *Panopeus herbstii* Atlantic mud crab | | | | | | | | 3) |
| *Sciaenops ocellatus* red drum | 50.22 | (3.08) | 41.29 | (3.17) | 46.86 | (1.45) | 48.29 | (1.67) |
| | | | | | | | | |
| **Fall** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| *Lagodon rhomboides* pinfish | | | | | | | | |
| *Gobionellus robustum* code goby | 17.52 | (.58) | 18.38 | (.76) | 17.39 | (.87) | 17.20 | (1.16) |
| *Gobionellus boleosoma* darter goby | | | | | | | | |
| *Sciaenops ocellatus* red drum | 17.24 | (2.31) | 20.72 | (1.62) | 16.53 | (.94) | 17.97 | (1.79) |
| *Callinectes sapidus* blue crab | | | | (.98) | 15.30 | (1.91) | 13.31 | (1.82) |
| *Panopeus herbstii* Atlantic mud crab | 9.34 | (.78) | 7.75 | (1.14) | 8.15 | (1.52) | 8.08 | (1.4) |
| *Farfantepenaeus aztecus* brown shrimp | 41.53 | (1.23) | 39.93 | (1.02) | 41.28 | (2.18) | 46.67 | 1.63 |
| | | | | | | | | |
| **Winter** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| *Lagodon rhomboides* pinfish | 15.49 | (.67) | 14.99 | (.48) | 15.71 | (1.24) | 15.54 | (1.18) |
| *Gobionellus robustum* code goby | 16.79 | (.73) | 16.95 | (.29) | 18.08 | (1.42) | 17.76 | (1.12) |
| *Gobionellus boleosoma* darter goby | 18.18 | (.89) | 19.38 | (1.63) | 17.65 | (2.45) | 18.25 | (2.14) |
| *Callinectes sapidus* blue crab | 10.59 | (.73) | 10.68 | (.55) | 10.97 | (1.02) | 10.82 | (.53) |
| *Panopeus herbstii* Atlantic mud crab | 8.20 | (.52) | 9.54 | (2.22) | 8.00 | (.58) | 10.25 | (2.18) |
| | | | | | | | | |
| **Spring** | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* | *Mean* | *SE* |
| *Lagodon rhomboides* pinfish | 20.98 | (2.05) | 25.00 | (1.85) | 22.97 | (1.) | 22.89 | (.73) |
| *Gobionellus robustum* code goby | 20.32 | (2.49) | 21.29 | (1.72) | 19.49 | (1.86) | 19.99 | (1.29) |
| *Gobionellus boleosoma* darter goby | 19.52 | (1.6) | 20.78 | (1.52) | 19.18 | (.97) | 18.89 | (1.05) |
| *Leiostomus xanthurus* spot | 30.37 | (.95) | 29.12 | (1.18) | 28.30 | (1.85) | 30.19 | (1.83) |
| *Citharichthys spilopterus* bay whiff | 19.50 | (1.75) | 21.79 | (2.45) | 21.19 | (1.59) | 20.18 | (1.39) |
| *Callinectes sapidus* blue crab | 13.08 | (1.15) | 12.47 | (.77) | 11.90 | (.71) | 11.79 | (.74) |
| *Panopeus herbstii* Atlantic mud crab | 7.60 | (.95) | 5.79 | (1.14) | 8.13 | (1.62) | 8.63 | (1.27) |
| *Litopenaeus setiferus* white shrimp | 19.41 | (1.12) | 20.26 | (1.66) | 20.68 | (1.42) | 19.91 | (1.56) |


Pinfish length SL (mm)
0
10
20
30
40
50
60
70
80
Summer
Fall
Winter
Spring
Reference
Low
Moderate
Severe

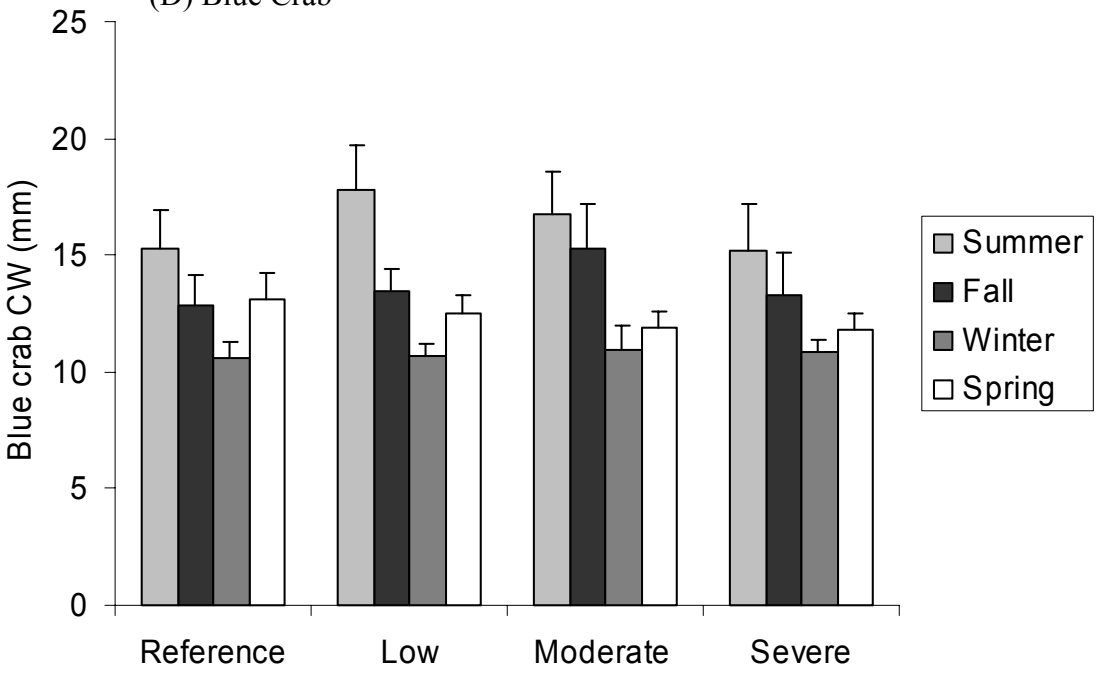
(B) Code Goby


Code goby length SL (mm)
0
5
10
15
20
25
Reference
Low
Moderate
Severe
Summer
Fall
Winter
Spring

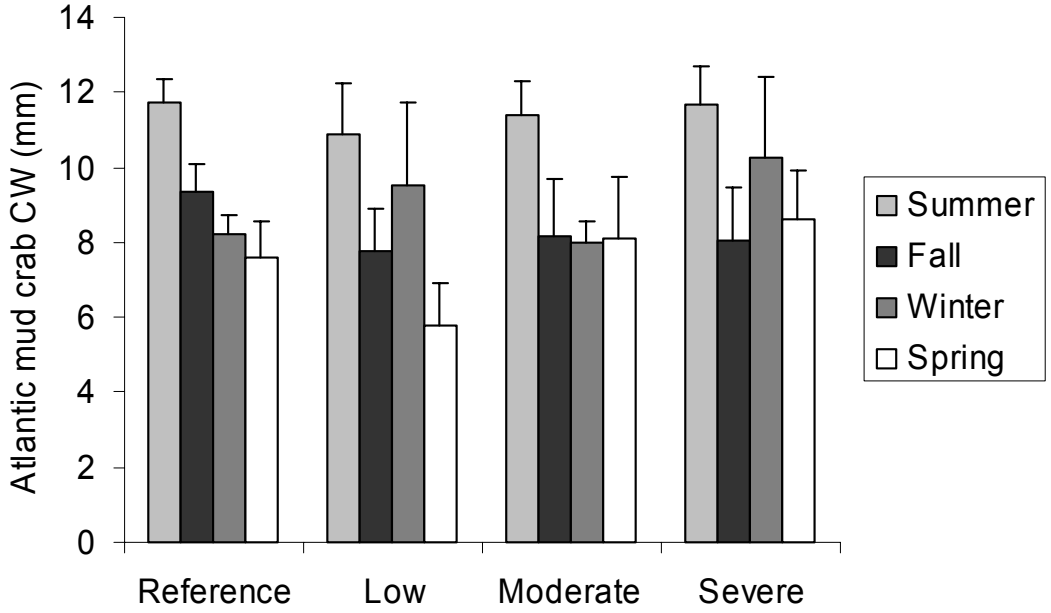
(C) Darter Goby



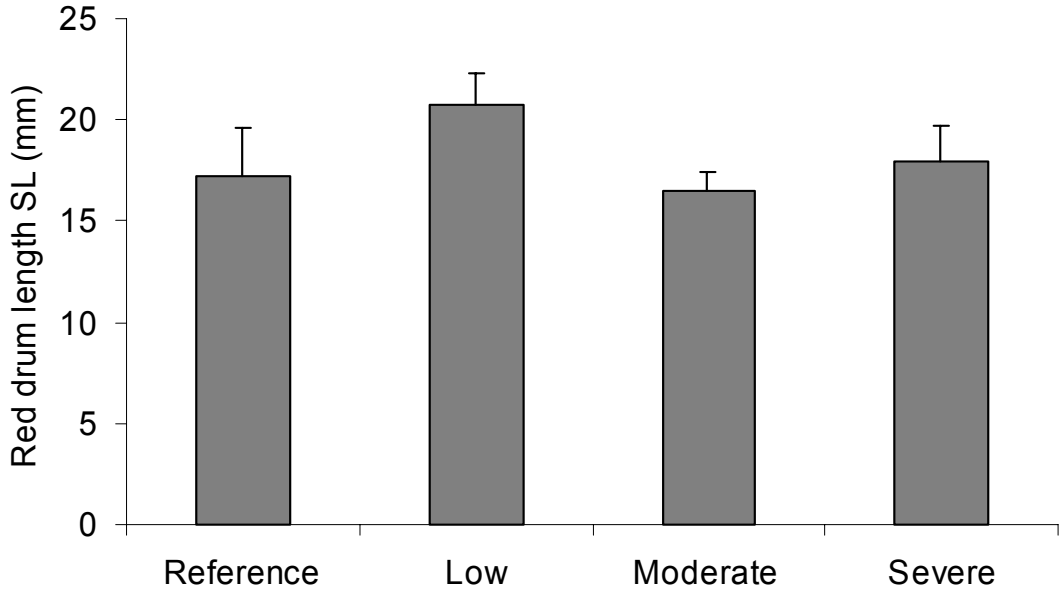

(D) Blue Crab



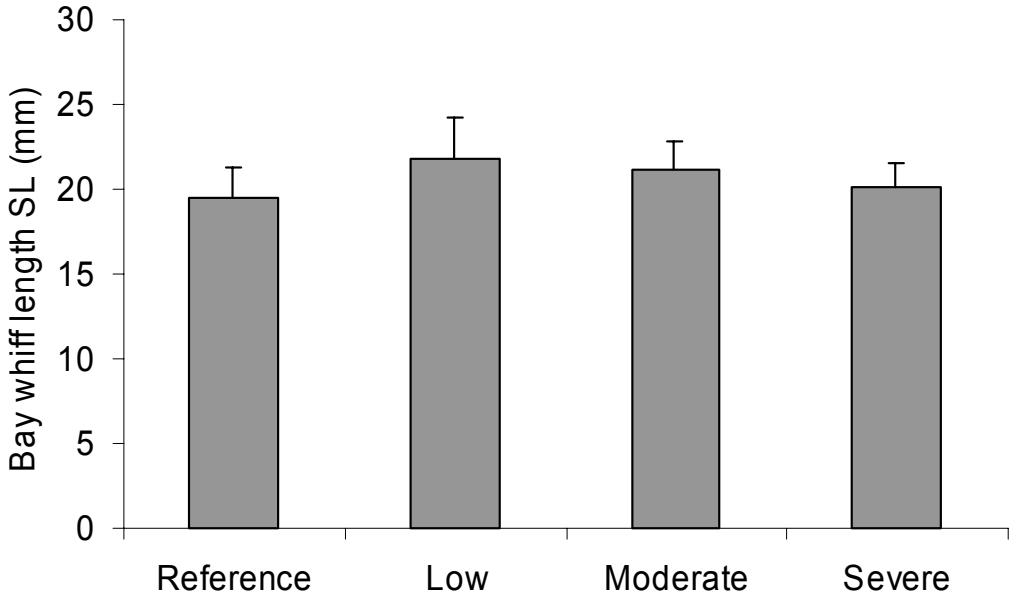
(E) Atlantic Mud Crab



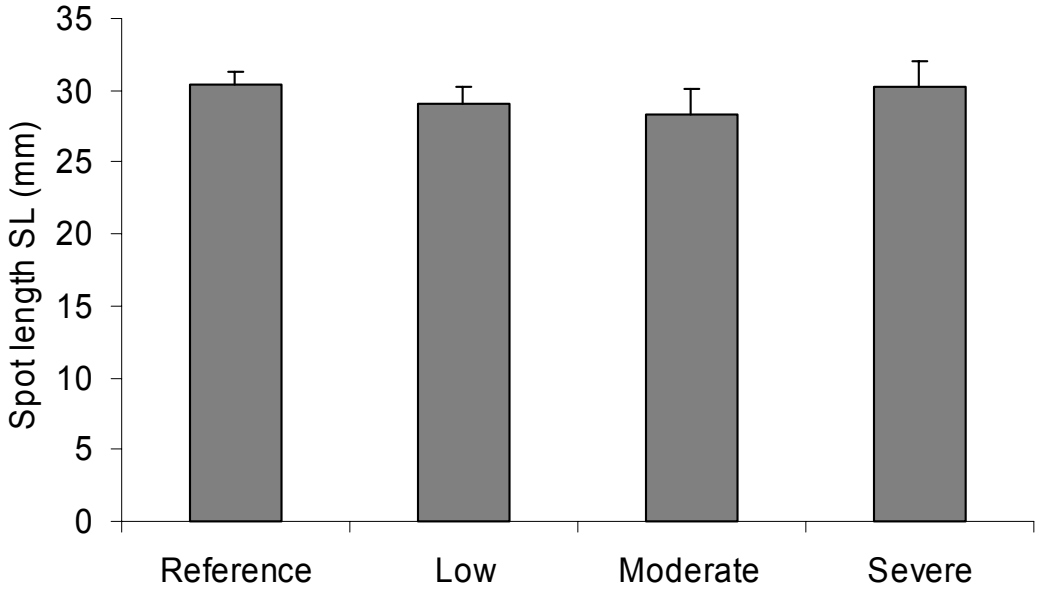

(F) Red drum



(G) Bay Whiff




(H) Spot



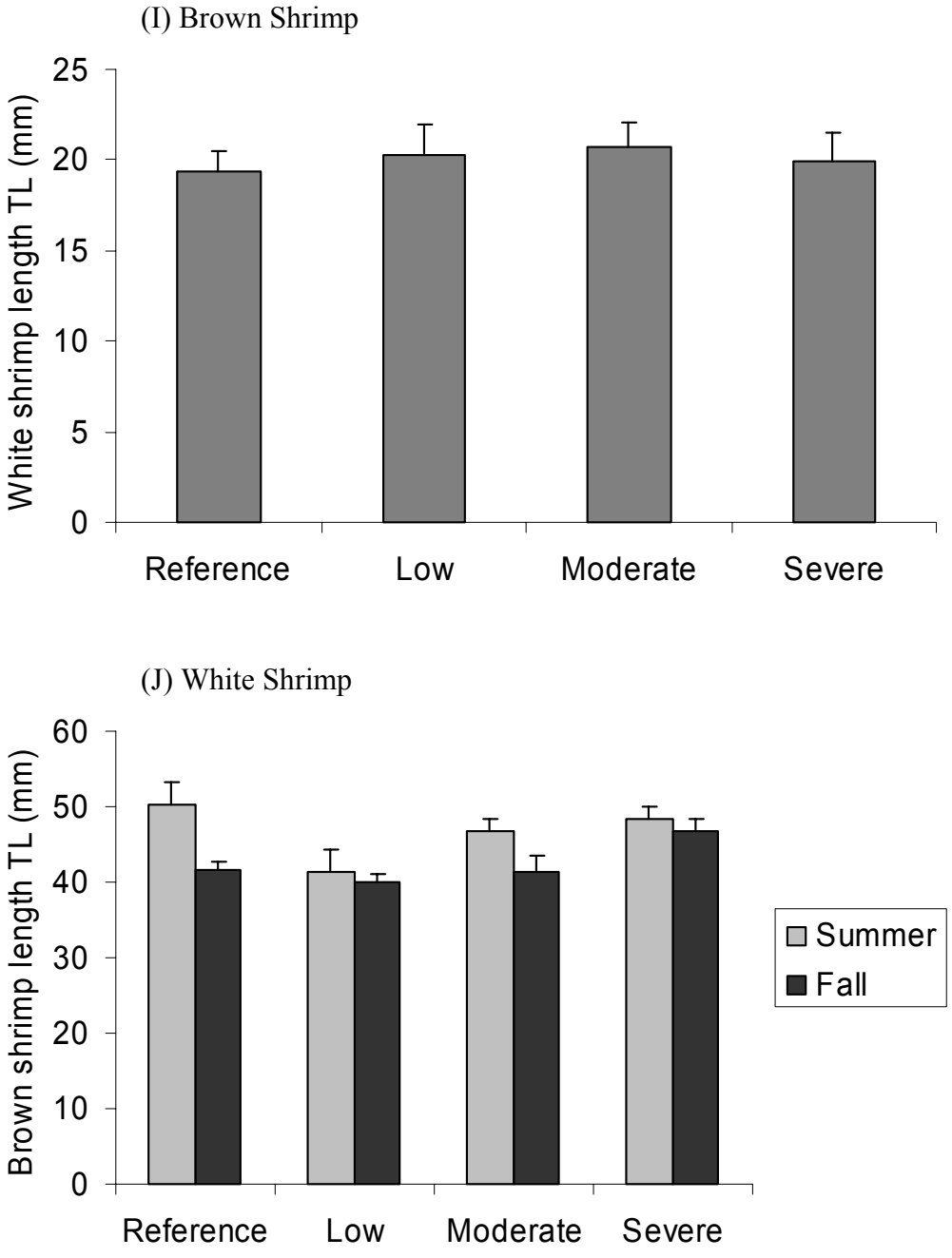



Fig. 23. Mean size (± SE) of organism in each season sampled. Each organism in each season was analyzed separately (N=10) with analysis of variance. Graph A represents pinfish length (SL). Graph B represents code goby length (SL). Graph C represents darter goby length (SL). Graph D represents blue crab carapace width (CW). Graph E represents mud crab carapace width (CW). Graph F represents red drum length (SL). Graph G represents bay whiff length (SL). Graph H represents spot length (SL). Graph I represents brown shrimp length (TL). Graph J represents bay white shrimp (TL).

Table 5. Analysis of variance summary table for comparison of mean size of organism in season(s) numerically dominant.

| | Summer | | | | Fall | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | SS | *F* | p | n | SS | *F* | p |
| *Lagodon rhomboides* pinfish | 10 | 121.363 | 1.037 | 0.393 | 10 | 1410.737 | 1.588 | 0.220 |
| *Gobionellus robustum* code goby | 10 | 12.340 | 0.548 | 0.654 | 10 | 6.701 | 0.422 | 0.739 |
| *Gobionellus boleosoma* darter goby | 10 | 15.888 | 0.173 | 0.914 | 10 | 30.414 | 0.760 | 0.524 |
| *Sciaenops ocellatus* red drum | | | | | 10 | 52.742 | 1.123 | 0.366 |
| *Callinectes sapidus* blue crab | 10 | 42.668 | 0.436 | 0.728 | 10 | 34.462 | 0.480 | 0.698 |
| *Panopeus herbstii* Atlantic mud crab | 10 | 2.332 | 0.129 | 0.942 | 10 | 8.509 | 0.381 | 0.768 |
| *Farfantepenaeus aztecus* brown shrimp | 10 | 354.178 | 2.424 | 0.087 | 10 | 257.435 | 3.424 | 0.028 |

| | Winter | | | | Spring | | | |
|---|---|---|---|---|---|---|---|---|
| *Species* | n | SS | *F* | p | n | SS | *F* | p |
| *Lagodon rhomboides* pinfish | 10 | 2.146 | 0.124 | 0.945 | 10 | 64.696 | 1.208 | 0.323 |
| *Gobionellus robustum* code goby | 10 | 3.133 | 0.530 | 0.674 | 10 | 6.158 | 0.179 | 0.908 |
| *Gobionellus boleosoma* darter goby | 10 | 11.742 | 0.146 | 0.931 | 10 | 17.797 | 0.371 | 0.774 |
| *Leiostomus xanthurus* spot | | | | | 10 | 20.437 | 0.354 | 0.787 |
| *Citharichthys spilopterus* bay whiff | | | | | 10 | 24.611 | 0.279 | 0.840 |
| *Callinectes sapidus* blue crab | 10 | 0.763 | 0.055 | 0.983 | 10 | 10.573 | 0.473 | 0.703 |
| *Panopeus herbstii* Atlantic mud crab | 10 | 13.622 | 0.443 | 0.726 | 10 | 21.841 | 0.726 | 0.550 |
| *Litopenaeus setiferus* white shrimp | | | | | 10 | 7.896 | 0.138 | 0.936 |

Length frequency distributions were performed on 5 species in all seasons where they were numerically dominant. Size classes of spot (Fig. 24), pinfish (Fig. 25), blue crab (Fig. 26), brown shrimp (Fig. 27), and white shrimp (Fig. 28) were similar across all scarring intensities in each season examined.

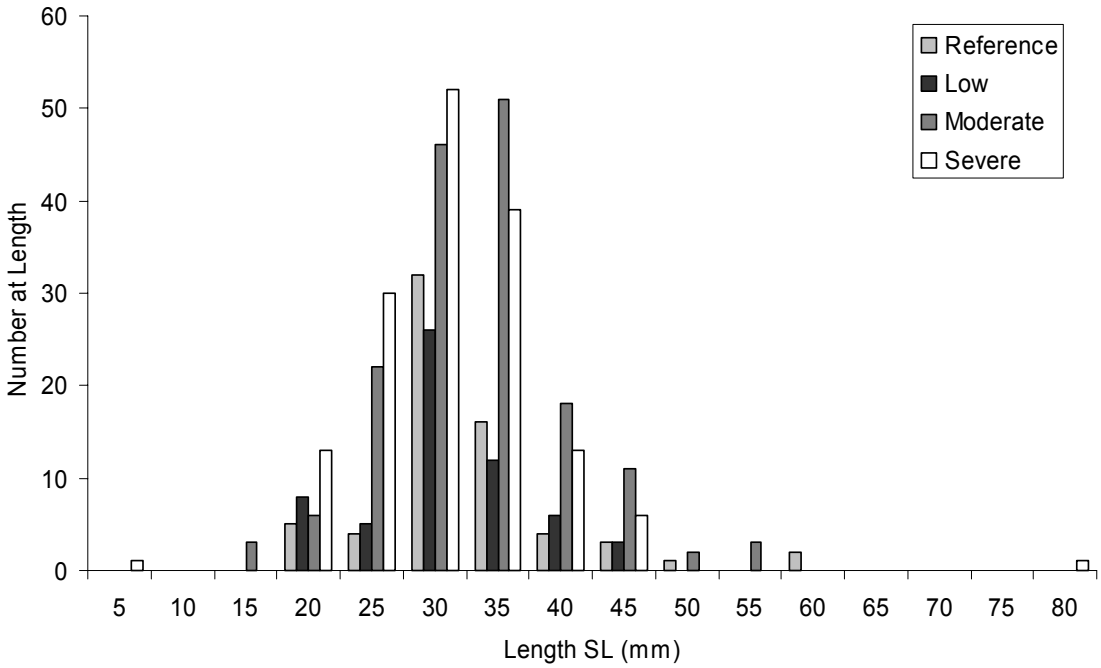



Fig. 24. Length-frequency distribution of spot in spring (N=444) collected in Redfish Bay, Texas with an epibenthic sled.

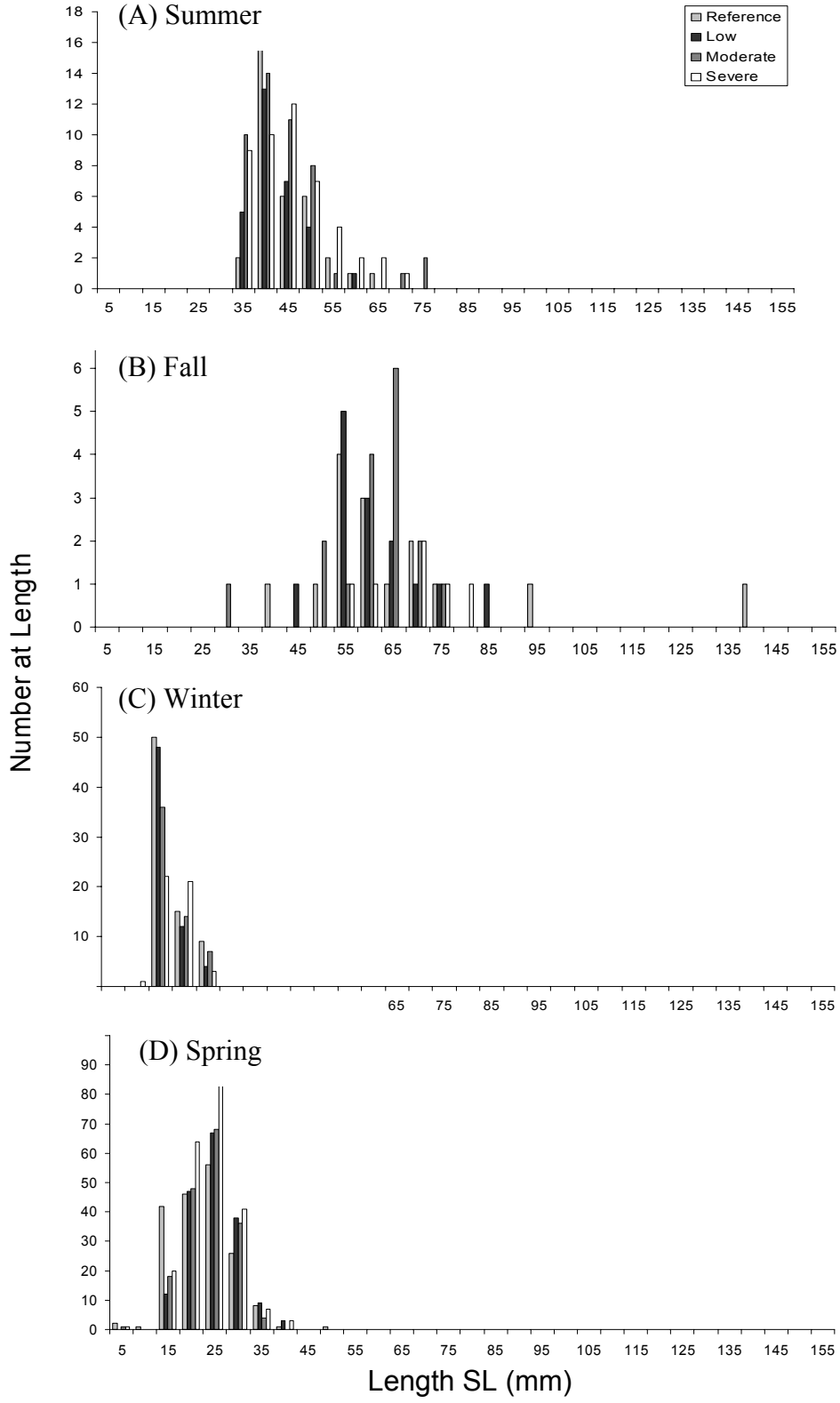



Fig. 25. Pinfish length frequency distribution in summer (N=158), fall (N=52), winter (N=242), and spring (N=761) collected in Redfish Bay, Texas with an epibenthic sled.

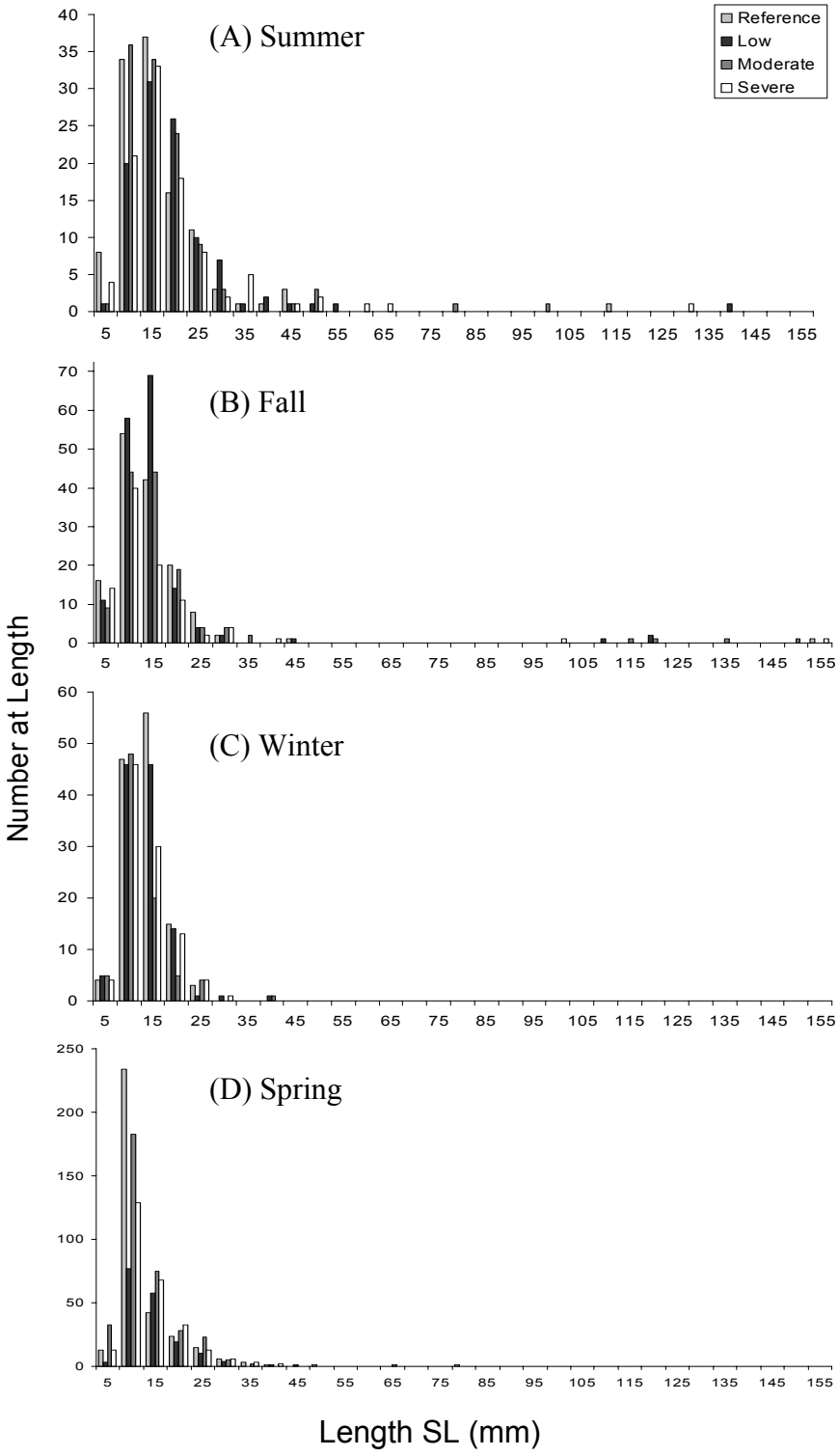



Fig. 26. Blue crab length frequency distribution in summer (N=427), fall (N=530), winter (N=420), and spring (N=1130) collected in Redfish Bay, Texas with an epibenthic sled.

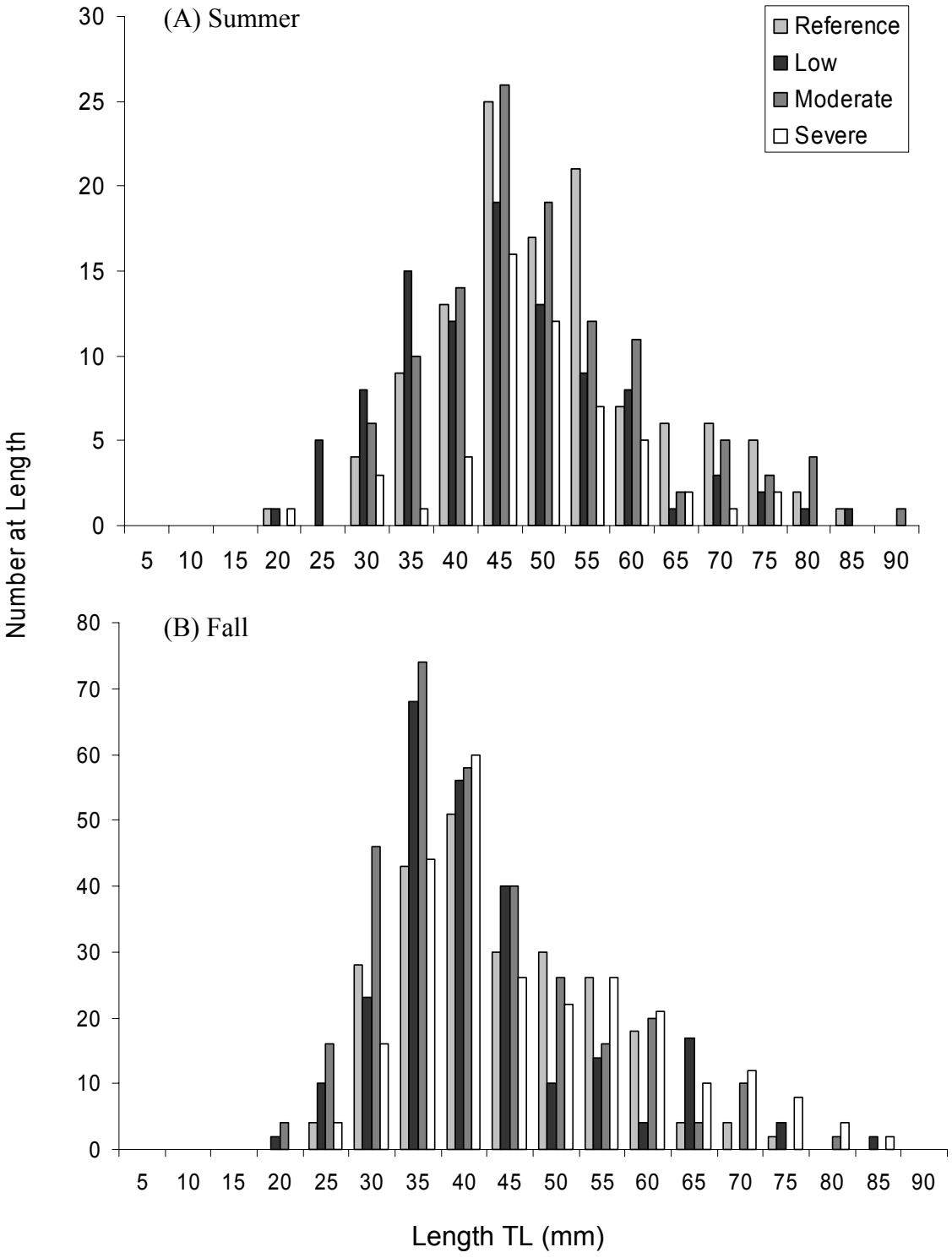



Fig. 27. Brown shrimp length frequency distribution in summer (N=382) and fall (N=1061) collected in Redfish Bay, Texas with an epibenthic sled.

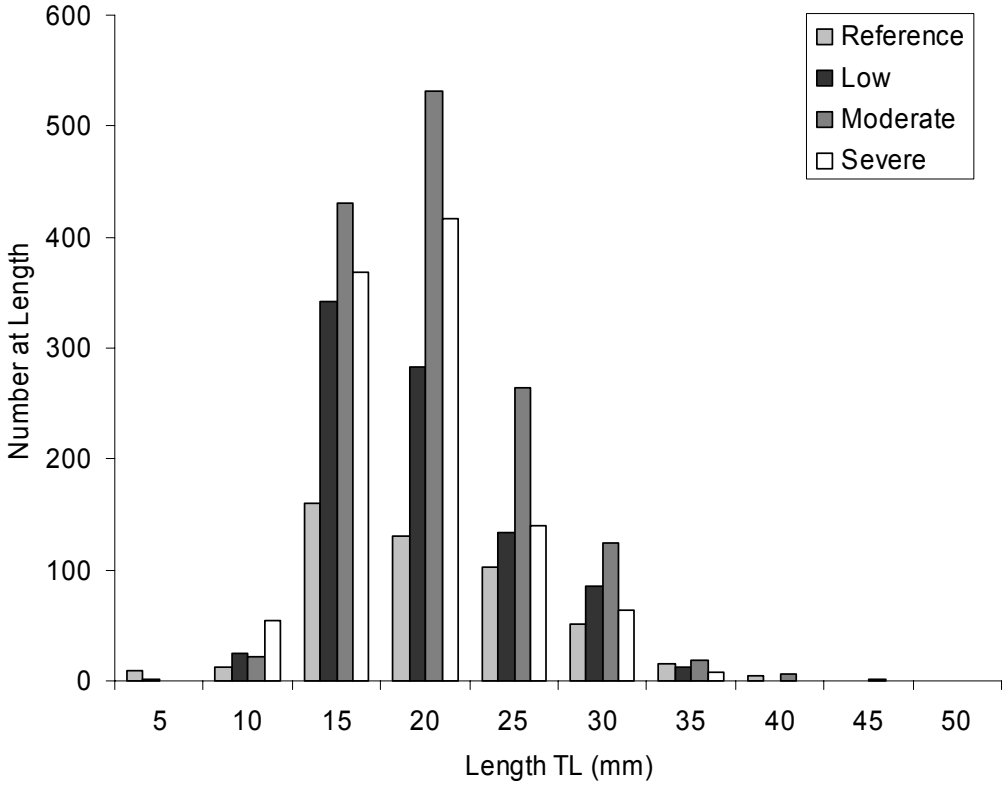



Fig. 28. White shrimp length frequency distribution in spring (N=3817) collected in Redfish Bay, Texas with an epibenthic sled.

Salinity (Fig. 29), temperature (Fig. 30), and dissolved oxygen (Fig. 31) were measured three times over the course of the enclosure experiment and were similar between scarring intensities. Mean white shrimp growth in the enclosure experiment was 10.46 (SE = 0.76) in the reference sites, 6.38 (SE = 1.76) in the lightly scarred sites, 8.72 (SE = 0.57) in moderately scarred sites, and 3.92 (SE = 0.762) in severely scarred sites (Fig. 32). Analysis of variance indicated a significant difference in white shrimp growth ($p = 0.016$, $df = 3$, $F = 4.827$). Among scarring levels a Tukey post-hoc test indicated that white shrimp growth was significantly lower in highly scarred sites than reference sites, and all other sites were similar. Overall, there was 76% recovery of all shrimp and 100% recovery of at least one shrimp per intact enclosure.

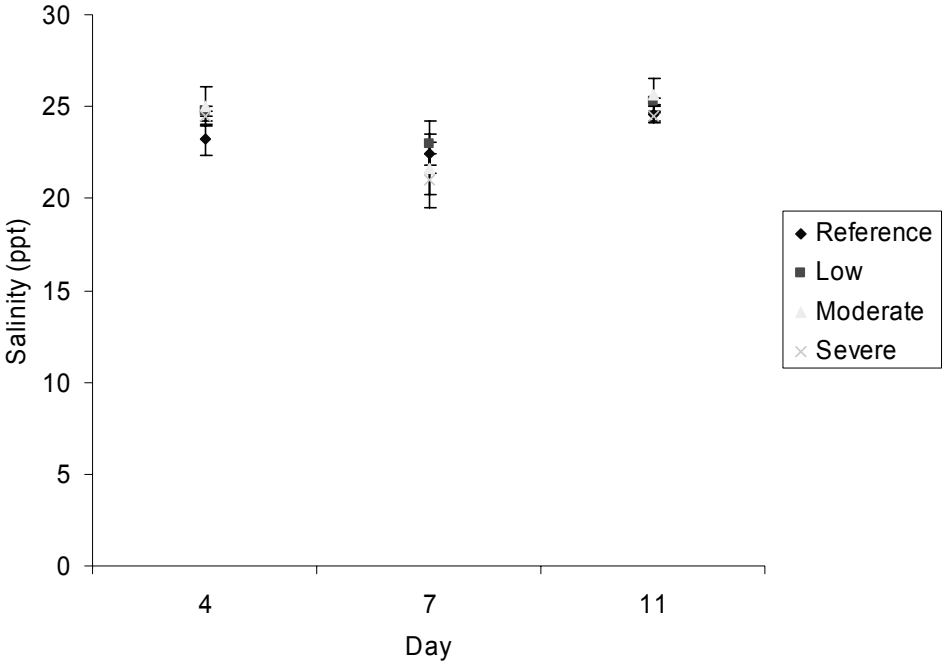



Fig. 29. Salinity (± SE) inside enclosures taken at 4d, 7d, and 11d into the growth study, Redfish Bay, Texas.

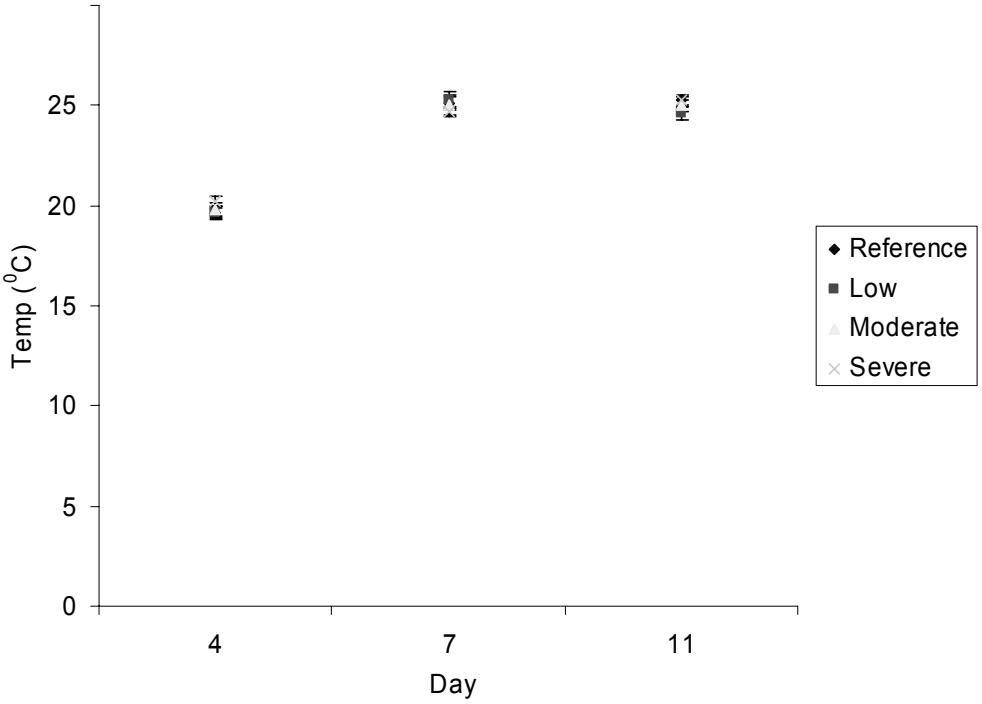



Fig. 30. Temperature (± SE) inside enclosures taken at 4d, 7d, and 11d into the growth study, Redfish Bay, Texas.

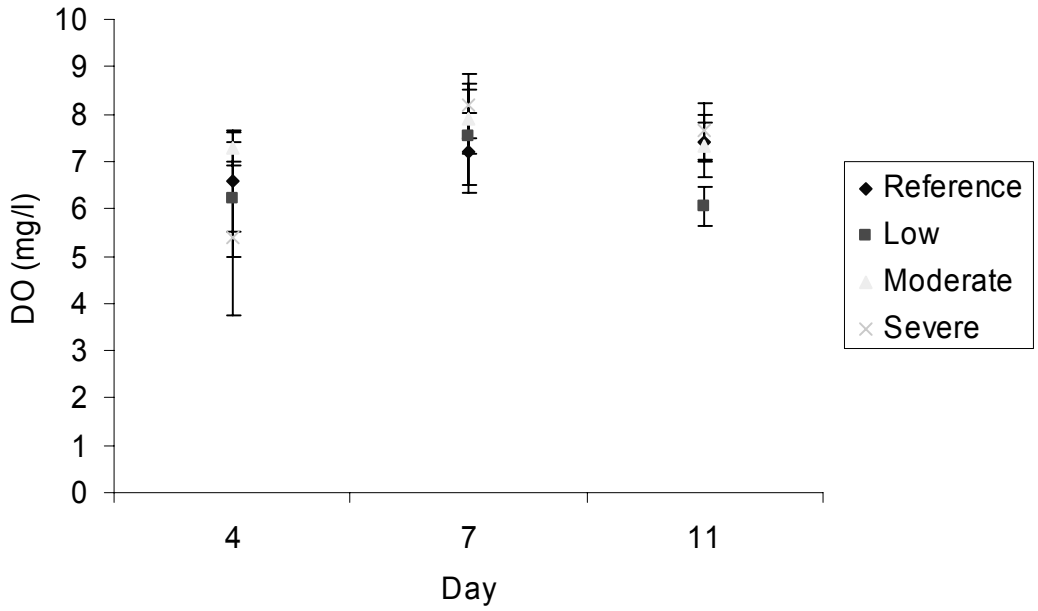



Fig. 31. Dissolved oxygen (± SE) inside enclosures taken at 4d, 7d, and 11d into the growth study, Redfish Bay, Texas.

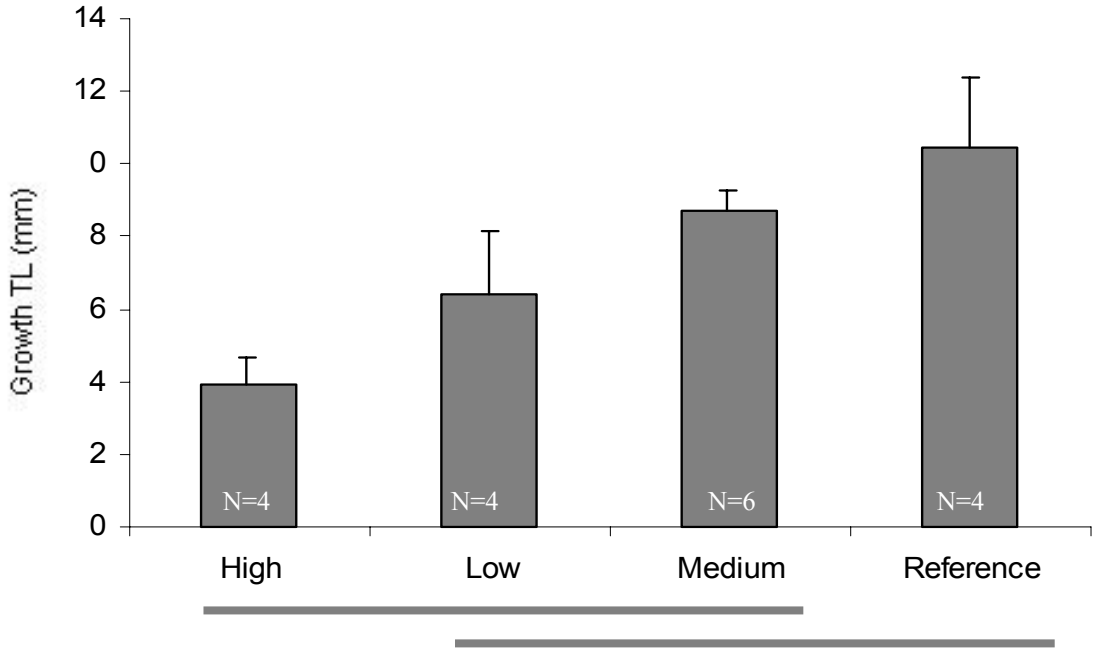



Fig. 32. Mean growth (mm over the 10 d experiment ± SE) for white shrimp in field enclosures among in distinct scarring intensities. There were six replicate enclosures of each scarring intensity. The P-value is from an ANOVA comparing mean growth of white shrimp in each scarring intensity. Horizontal bars below the x-axis indicate results of Tukey's test, and scarring intensities sharing the same bar are not significantly different.

Because I found a significant relationship between pinfish length and otolith diameter ($r^2 = 0.46$, n = 100, $p < 0.001$), I was able to use otolith increment measurements as a proxy for somatic growth (Fig. 32.). Increment widths were 55.5 μm (SE = 1.87) for reference sites, 56.3 μm (SE = 1.14) for low scarring, 55.34 μm (SE = 1.25) for moderate scarring, and 59.3 μm (SE = 1.67) for severe scarring (Fig. 31.). The mean increment width for the last 10d indicated that growth was not significantly different between scarring intensities (n = 25, $F = 318.54$, p = 0.135).

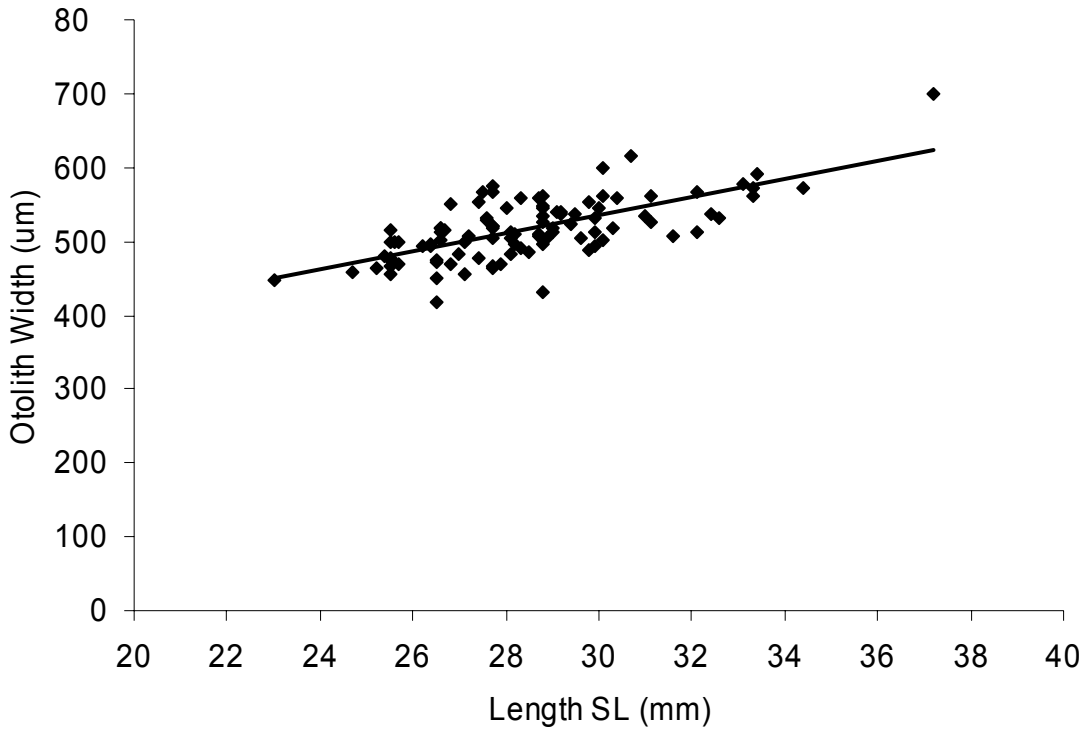



Fig. 33. The relationship between the SL (mm) and the diameter (μm) of the lapillar otolith of pinfish collected from reference, low, moderate, and severe scarring intensities in Redfish Bay, Texas. Regression model: Diameter = 12.287 (SL) + 166.64; N = 100, $r^2 = 0.4575$, $P < 0.001$

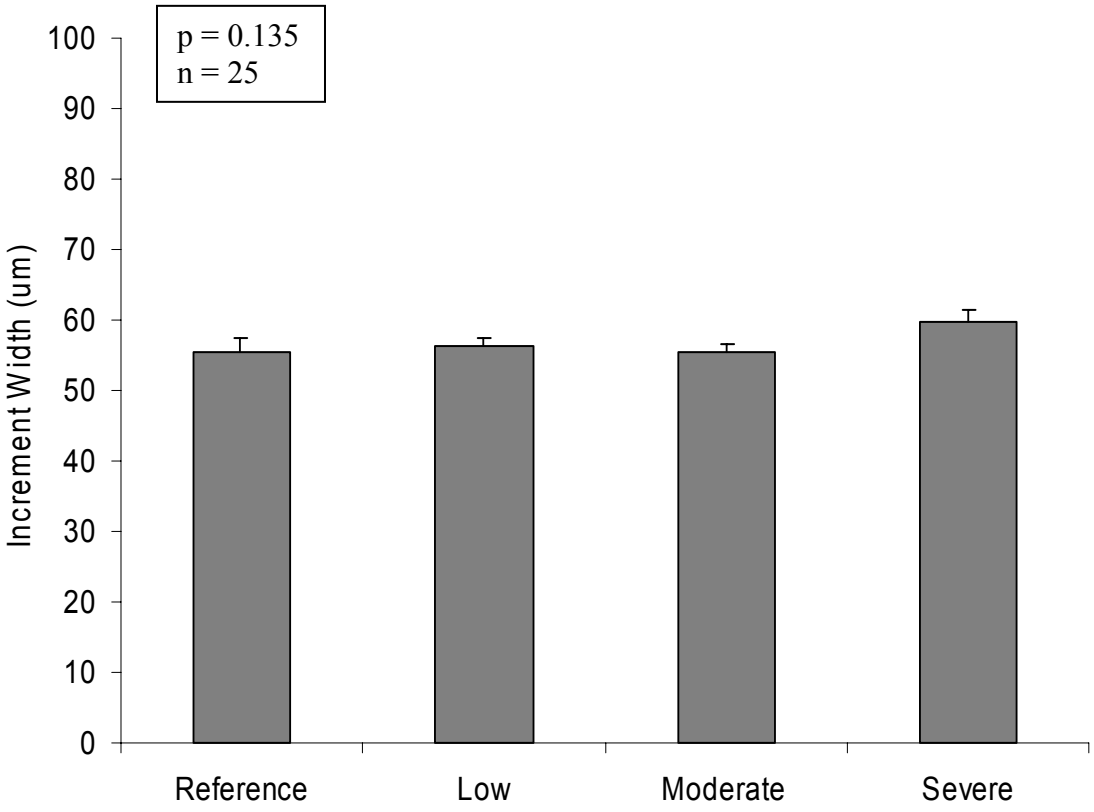



Fig. 34. Mean otolith increment widths for the last 10 d of growth for pinfish collected from reference, low, moderate and severe scarring intensities in Redfish Bay (25 fish from each scarring intensity). The P-value is from an ANOVA comparing mean increment width among the scarring intensities.

***Discussion***

This study evaluated the effects of propeller scarring on nekton abundance. Density patterns of juvenile organisms can serve as an indicator of habitat quality since it reflects recruitment, mortality and emigration (Minello 1999). It has been suggested that propeller scarring decreases the faunal densities (Zieman 1976). This study indicates that different levels of propeller scarring do not affect faunal densities in any season. Furthermore, there does not appear to be a relationship between nekton density and the percent scarring in a site for scarring intensities between 1% and 27%. However, propeller scarring does impact white shrimp growth.

Mortality from predation can be a major factor controlling variability in survival of juvenile marine organisms (Houde 1987). Predation on juvenile organisms is often thought to be related to the structural complexity of available habitat. To characterize habitat value, I used length frequency analysis as an instantaneous measure of mortality, and I did not detect a difference in size or mortality of nekton examined. In my study up to 27% of the seagrass structure was removed; however, in terrestrial systems as little as 20% of the original habitat may be necessary to maintain population survival (Fahrig 1997).

The similarity across all scarring intensities may be the result of the nearby presence of large unscarred areas where fauna can either move to find resources or from where new individuals can immigrate (Bell et al. 2002). Since there is no difference in density, it may imply that organisms must aggregate in greater densities in the vegetation in scarred areas versus unscarred areas (Bell et al. 2002). When there is a high scarring intensity there is proportionately less seagrass coverage and more edge habitat (Uhrin &

Holmquist 2003). This small-scale patchiness or habitat heterogeneity may be beneficial. Holt et al. (1983) found that habitat patchiness was the greatest landscape factor in affecting density and that there were more red drum at the seagrass sand ecotone than in homogeneous seagrass. Holt et al. (1983) found high densities of red drum in patchy seagrass meadows, suggesting movement between unvegetated feeding areas and predation refuge in the seagrass. Though patchy habitats temporarily can support higher faunal densities, they have decreased ability to endure physical disturbance (Holt et al. 1983) and are highly variable based on wind generated waves and tidal currents (Robbins & Bell 1994).

To further examine the functionality of the areas, I used two methods to examine nekton growth rates. Field enclosure experiments using white shrimp showed significantly lower growth rates in severely scarred areas than reference sites. However, an examination of otolith-based growth in pinfish showed no difference in growth rate between scarring intensities.

In field growth enclosure experiments white shrimp grew between 0.951 mm $day^{-1}$ in reference areas and 0.356 mm $day^{-1}$ in severely scarred areas. Growth rates were significantly lower in severely scarred areas than reference areas. Growth rates in reference areas are comparable to prior studies with growth rates between 0.833 and 1.33 mm $day^{-1}$ (Gunter 1950). Brown shrimp are known to prefer benthic infauna, but white shrimp show no preference in food type, and the composition of their diet is largely unknown (McTigue & Zimmerman 1998). White shrimp have been known to feed on plants (Hunter 1984), and it is possible that seagrass, epiphytes, and benthic diatoms are an important part of their diet. Consequently, they may show a greater impact from

vegetation removal. However, laboratory studies by McTigue & Zimmerman (1998) showed little to no growth in white shrimp on a plant-based diet, suggesting that a combination of food sources may be necessary (Kneib 1997). This sensitivity to habitat degradation is of particular concern because loss of high quality nursery habitat is thought be the most serious potential threat to the white shrimp fishery (Webb & Kneib 2002).

Once humans cause a change in an area it is difficult to determine the exact consequences of that change since the environment is constantly undergoing natural change (Robbins & Bell 1994, Haila 2002). This may contribute to this study in that the age of the propeller scars may have been in different stages of recovery. However, this is a necessary cost since manipulative experiments that destroy habitat may be ethically undesirable (Nagelkerken et al. 2001).

Habitat fragmentation has become a key theme considered when examining anthropogenic degradation of the environment (Haila 2002) and it is particularly important to consider the structure of the landscape (Fahrig & Merriam 1994). Not much attention has been given to fragmentation in marine systems (McNeill & Fairweather 1993); however, seagrass systems possess several ecological characteristics suited to the application of terrestrially developed techniques (Robbins & Bell 1994). A landscape ecology approach may provide better insight to the importance on patch location, heterogeneity, and size and shape (Robbins & Bell 1994, Bell et al. 1999). It is important to remember that water moving through seagrasses connects the system (Robbins and Bell 1994) resulting in a demographically open population (Levin et al. 1997, Carr et al. 2003) is a constraint in using terrestrial landscape techniques in marine systems.

Variation in growth rates of juvenile fishes are also important for individual survival, and may influence successful recruitment into adult populations (Houde 1987, Connell and Jones 1991, Hixon 1991). Variation in food availability has been suggested as an important factor in regulating variation in growth rates (Sogard & Able 1992, Levin et al. 1997). Pinfish play significant ecological roles in coastal systems (Potthoff & Allen 2003); therefore, they served as an ideal model species to examine otholith-based growth in varying levels of propeller scarring. In pinfish there was a significant relationship between otolith diameter and length indicating a direct relationship between otolith and somatic growth. This allowed me to use otoliths for estimations of recent growth (Secor & Dean 1989, Sogard & Able 1992). My examination of otolith microstructure did not show differences in recent growth between scarring levels. Levin et al. 1997 found growth rates of 0.40 mm SL $day^{-1}$, which are similar to my growth rate of 0.24 mm SL $day^{-1}$. Pinfish have relatively high sight fidelity; however, they do move over relatively large areas (Potthoff & Allen 2003). Scarred sites were adjacent to large unscarred areas and some species use seagrass beds for shelter, but forage in adjacent unvegetated habitats (Summerson & Peterson 1984).

This difference brings about two possible explanations. There may be a greater effect of propeller scarring on white shrimp growth than pinfish growth. However, field caught pinfish were not restricted to a specific habitat type prior to capture. Therefore, this movement can affect the utility in using growth rates determined by free ranging fish (Stunz 2002b).

The highest scarring intensities I examined were ca. 27%. This was because small scale severely scarred areas (>15%) were rare. I may find differences at those levels, but

due to the low aerial extent, they may not be a significant factor in overall abundance. However, high scarring intensities may cause a loss in seagrass bed stability, and it would be useful to focus future research on finding limits to bed stability (Bell et al. 2002). Scarring intensities of 50% would be a good way to examine the effects of propeller scarring (Fonseca & Bell 1998) and would be useful in addressing the possibility of a threshold point. Looking at areas of 50% or greater propeller scarring was considered in this study; however, it was not possible to find enough areas <50% scarred to properly replicate a treatment at the level. As well as looking at higher scarring intensities and different spatial scales, modeling may serve as a useful tool in estimating a threshold point in propeller scarring for both faunal responses and limits of seagrass bed stability.

This study indicates the propeller scarring may not affect density, size and, mortality of nekton at a maximum scarring intensity of 27%. However, shrimp growth rates may be impacted by propeller scarring, suggesting a decrease in habitat quality. Until the impacts of propeller scarring are fully understood, it is important to protect the remaining seagrass habitat from degradation.

# CHAPTER IV

## SUMMARY AND CONCLUSIONS

The impacts of the decline in seagrass have been the focus of several studies (Quamman & Onuf 1993, Minello 1999, Levin & Stunz in press). Much of this loss may be attributed to anthropogenic degradation of the environment from increased nutrient input, pollution, dredging, and mechanical damage (Short & Wyllie-Echeverria 1996). Recently, studies focused on quantifying propeller scar coverage (Sargent et al. 1995, Dunton & Schonberg 2002) and the faunal impacts of propeller scarring (Bell et al. 2002, Davidson 2002, Uhrin & Holmquist 2003).

This increased interest in understanding the effects of propeller scarring has generated several plans for seagrass conservation. One such plan was the installation of voluntary no motor zones along the coast of south Texas. Though my preliminary study indicates that there is no boater compliance in the voluntary no motor zones, they remain a valuable preliminary step toward public awareness and protection of seagrass. Since there is no compliance in these areas, it is important to understand the ecological effects of propellers scarring to have a better understanding of how to conserve these resources.

To examine the faunal impact of propeller scarring, I evaluated the effects of propeller scarring on nekton abundance and size among three distinct scarring intensities. Nekton abundances were not significantly different between scarring levels for any species in all seasons. In examining mean size of organisms I found clear differences in size between seasons but sizes were similar between scarring intensities in each season.

To further assess habitat quality I examined length frequency as a measure of mortality and nekton growth rates. By plotting the length frequencies, I looked at size

structure as a measure of mortality for 5 numerically dominant species. I was able to determine that individuals are recruiting to all sites and subsequently surviving. Examining pinfish growth using otolith microstructure analysis there was no difference in growth rates between scarring intensities; however, there was a significant difference in the field growth enclosure experiment. Growth measured in enclosures indicated that white shrimp in severely scarred seagrass beds grew significantly less than those in unscarred areas. My results are consistent with Bell et al. (2002) who did not find a difference in density, mean size, or size structure. This study indicates that propeller scarring does not affect density, size, and age structure but may affect growth of white shrimp. There are several key elements that need to be considered to truly understand other potential interactions occurring in these areas.

This study took a landscape approach to studying propeller scarring by looking at a large area of scarring and did not determine a difference between scarred and unscarred sites at any scarring intensity. Bell et al. (2002) took a similar approach and also did not find any meaningful differences between scarred and unscarred sites. Others, Uhrin and Holmquist (2003) took a small-scale approach by examining a single scar. They found that shrimp and mollusc abundances were lower in the scars and up to 5m from the scars. However, it is unknown how, if at all, these effects will scale up (Uhrin & Holmquist 2003). Several spatial and temporal scales need to be considered when studying fragmentation and these parameters may vary by species, location, and habitat type (Haila 2002). Scale is important in examining distribution and abundance patterns and may greatly affect the interpretation of results (see review by Eggleston et al. 1999). It is difficult to determine the scale at which habitat structure affects survival and abundance

of organisms (Hovel & Lipcius 2002). The appropriate scale to examine a population depends upon the size and dispersal capability of the organism (Fahrig & Merriam 1994). In this study we examined highly mobile species, and therefore it may be more appropriate (Robbins & Bell 1994) to look at higher scarring intensities of propeller scarring over a large area (bay) rather than small regions within the system (Bell et al. 2002).

Though there is inherently habitat loss associated with habitat fragmentation they are separate entities (McGarigal & Cushman 2002); consequently, it may be more important to focus on the habitat loss versus the underlying habitat mosaic. Single scar regrowth can take 0.9 to 4.6 year in *Halodule wrightii* (Sargent et al. 1995) and 1.7-10 years in *Thalassia testudinum* (Dawes et al. 1997). Due to this slow growth rate seagrass may show long-term damage from propeller scarring (Dawes et al. 1997). Additionally, propeller scars can fill with sediment creating an environment that inhibits rhizome growth (Zieman 1976). To date, studies examining scar regrowth have focused on the recovery of a single scar. However, areas along channel edges and at channel junctions are susceptible to repeated scarring (Sargent et al. 1995, Dunton & Schonberg 2002), and there have not been any studies looking at the effects of scarring aggregation. How long (if ever) will it take for severely scarred areas to regrow? The physical disturbance of seagrass through propeller scaring often creates a clear habitat loss (SCPT 1999) resulting in a cumulative reduction of productive habitat (FDEP 1998). This landscape fragmentation can generate changes in the physical forces across the landscape, which may have important effects on the remaining vegetation (Saunders et al. 1991).

Specifically, wave energy in propeller scars could lead to erosion (Zieman 1976) and deepening of the disturbed area (Eleuterius 1987).

This study suggests that propeller scarring may not impact density patterns, mean size, and age structure of the organisms that we collected on the spatial scale that we studied. This does not necessarily imply that propeller scarring does not at some level potentially have an effect since we have seen that propeller scarring may impact growth rates. Obviously, at some point increased propeller scarring will degrade habitat and reduced functionality. However, up 27% scarring there does not appear to be widespread impact. As well as looking at higher scarring intensities and different spatial scales, modeling may serve as a useful tool in estimating a threshold point in propeller scarring. It will be difficult to stop the occurrence of propeller scars; however, two actions need to be taken (1) clearly mark channels and (2) follow the models used in Florida by penalizing people that carelessly and intentionally create propeller scars.

More information is needed to characterize the effects of propeller scarring on both the seagrass and associated fauna. Future research looking at higher scarring levels and different spatial scales as they relate to both seagrass bed stability and faunal impact would aid in understanding the net impact of propeller scarring.

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