Habitat Characterization, Habitat Use and Associated Growth of Juvenile Spotted Seatrout (*Cynoscion nebulosus*) in a Mississippi Bay System: Implications for Stock Enhancement Practices

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HABITAT CHARACTERIZATION, HABITAT USE AND ASSOCIATED GROWTH
OF JUVENILE SPOTTED SEATROUT (Cynoscion Nebulosus)
IN A MISSISSIPPI BAY SYSTEM: IMPLICATIONS FOR
STOCK ENHANCEMENT PRACTICES

by

Joseph Read Hendon

Abstract of a Dissertation
Submitted to the Graduate School
of The University of Southern Mississippi
in Partial Fulfillment of the Requirements
for the Degree of Doctor of Philosophy

May 2013
ABSTRACT

HABITAT CHARACTERIZATION, HABITAT USE AND ASSOCIATED GROWTH OF JUVENILE SPOTTED SEATROUT (Cynoscion nebulosus) IN A MISSISSIPPI BAY SYSTEM: IMPLICATIONS FOR STOCK ENHANCEMENT PRACTICES

by Joseph Read Hendon

May 2013

The spotted seatrout (Cynoscion nebulosus) is the most targeted recreational fishery species in Mississippi coastal waters. Consequently, the Seatrout Population Enhancement Cooperative stock enhancement program was initiated to investigate methods for supplementing this heavily exploited population. Given ecological data needs for both wild and hatchery-reared (HR) juveniles of the species, habitat mapping, directed sampling and a caging study were conducted. Submerged aquatic vegetation (SAV), oyster shell and non-vegetated substrates were mapped at water depths < 2 m in Point aux Chenes Bay, Mississippi, using a sidescan sonar, which provided more accurate estimations of habitat coverage, particularly for SAV, compared to point-based sampling. Applying habitat strata delineated from sidescan mapping, late-juvenile spotted seatrout [125 - 275 mm total length (TL)] were sampled in SAV, marsh-edge and non-vegetated habitats as part of a seasonal gill net survey conducted in 2011 and 2012. Spotted seatrout ranging from 138 to 485 mm TL were collected using a small-mesh gill net, and 65% of collected individuals fell within the targeted late-juvenile size range. Overall mean catch-per-unit-effort of C. nebulosus did not differ for abundance or biomass between years, but both metrics were significantly higher in SAV habitats.
Employing cage enclosures that excluded predators and competitors, cumulative survival of HR fish introduced into natural conditions was 69.2% over a 29 d period, and specific growth rate was negative in all cages but significantly higher in SAV and non-vegetated shorelines than in non-vegetated open water. At the time of recovery, prey items occurred in the stomachs of 49% of fish surviving the entire trial period, and major prey taxa consumed by HR fish were consistent with those found in diets of wild fish. This study supports previous findings of the importance of SAV to the early life stages of *C. nebulosus* and extends that understanding to the late-juvenile stage. However, SAV was limited in spatial coverage, and spotted seatrout utilized all habitats sampled in the study area. Study results also indicate that HR spotted seatrout can survive natural environmental conditions and transition to wild prey after release into a natural ecosystem.
HABITAT CHARACTERIZATION, HABITAT USE AND ASSOCIATED GROWTH OF JUVENILE SPOTTED SEATROUT (Cynoscion Nebulosus) IN A MISSISSIPPI BAY SYSTEM: IMPLICATIONS FOR STOCK ENHANCEMENT PRACTICES

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CHAPTER I

GENERAL INTRODUCTION

Introduction

Habitat is an essential template (Southwood 1977) for many basic functional ecological relationships of fishes and their prey. Higher quality habitats generally foster growth and survival through a combination of abundant food sources, areas of refuge from predators, and favorable environmental regimes (Meng et al. 2000). The importance of habitat to the management of fish populations is underscored in the Magnuson-Stevens Fishery Conservation and Management Act [16 U.S.C. §1853(a)(7)], which requires Essential Fish Habitat (EFH) to be designated for all federally managed species relative to, among other things, spawning, feeding, refuge and migratory needs (GMFMC 2004). Despite this critical importance, habitat requirements are often poorly understood for many important fishery species, often as a result of incomplete information relative to early life-history stages.

The spotted seatrout (Cynoscion nebulosus) is an important component of the recreational and commercial fisheries complex of the Gulf of Mexico (GoM), wherein this inshore species is associated primarily with marsh and estuarine habitats (Bortone 2003). Because of its non-migratory life history (Iverson and Tabb 1962), the spotted seatrout is managed on a state-by-state basis throughout its range. In Mississippi, it is the most heavily exploited recreational saltwater species (NMFS 2013). The most recent assessment of Mississippi’s spotted seatrout population indicated that in light of rapidly increasing trends in fishing effort more conservative management actions would be needed to maintain adequate recruitment (Fulford and Hendon 2010), a need which has
been exacerbated by historical declines in critical vegetated habitats within Mississippi coastal waters.

Numerous ecological studies of spotted seatrout have been conducted throughout GoM and state coastal waters. The ecology and early life history of spotted seatrout has been well-documented in Florida (Hettler 1989; McMichael and Peters 1989; Rutherford et al. 1989; Chester and Thayer 1990; Powell 2003; Powell et al. 2004), Louisiana (Laska 1973; Sabins 1973; Arnoldi 1984; Herke et al. 1984; Peterson 1986; Rakocinski et al. 1992; Helser et al. 1993; Baltz et al. 1998) and Texas (Pearson 1929; King 1971; Vetter 1977; Tolon et al. 1997; Rooker et al. 1998). Early life history characteristics have generally been described for larval and early juvenile spotted seatrout in Mississippi (Benson 1982; Warren et al. 2001), where other studies have evaluated migratory patterns (Hendon et al. 2002), age and growth (Hendon et al. 2012), reproduction (Brown-Peterson and Warren 2001), and otolith microchemistry (Comyns et al. 2008) of adults. The ecology of late-juvenile spotted seatrout in Mississippi waters is entirely lacking, and knowledge of this stage is sparse in other regions.

In 2004, the Seatrout Population Enhancement Cooperative (SPEC) was developed as a state-collaborative stock enhancement program to develop procedures to augment Mississippi’s heavily exploited spotted seatrout population by rearing and releasing cultured fish into the wild. For an enhancement program to succeed, hatchery-reared (HR) fish must be able to survive and grow in the natural environment without displacing wild individuals (Leber et al. 1995; Huntingford 2004). Survival of HR fish requires acclimation to natural conditions, transition to feeding on wild prey, and predator avoidance, each of which should be evaluated through post-release monitoring.
and directed field experiments (Blankenship and Leber 1995; Leber et al. 1995; Walters and Martell 2004; Lorenzen 2006; Hervas et al. 2010). Subsequent to those assessments, post-release survival can be maximized as needed through pre-release conditioning of fish and by implementation of release strategies (Munroe and Bell 1997; Mahnken et al. 2004), that include identification and utilization of beneficial release habitats (Stunz and Minello 2001; Andersen et al. 2005). As a management tool, stock enhancement is supplementary to the more common practices of fishing effort control and habitat conservation and restoration (Lorenzen 2008), but a comprehensive approach involving all three management strategies can be effective for ameliorating the over-exploitation of heavily targeted populations.

Given the high level of exploitation of spotted seatrout in Mississippi coastal waters, data are needed to evaluate habitat use and assess stage-specific ecological requirements of the species. This is especially true for late-juveniles, about which little is known. Assessments of the ability of HR fish to acclimate to natural conditions are also needed to gauge post-release responses of fish in order to develop release strategies to maximize survival. Accordingly, the objectives of this study were to (1) delineate benthic habitats available to late-juvenile spotted seatrout in a Mississippi bay system using remote sensing (sidescan sonar); (2) determine habitat use by late-juvenile spotted seatrout among delineated habitats; and (3) evaluate growth and feeding of late-juvenile HR fish subjected to restricted natural conditions (i.e., enclosures) within delineated habitats.
LITERATURE CITED


CHAPTER II
REMOTE SENSING OF BENTHIC HABITATS IN THE GRAND BAY
NATIONAL ESTUARINE RESEARCH RESERVE

Introduction

Habitat provides many assets that support basic functional relationships of fishes and their prey (Southwood 1977). Optimal habitats can foster rapid growth, serve as refuge from predators, and provide favorable physio-chemical conditions (Knieb 1987; Meng et al. 2000). For fished stocks, the Magnuson-Stevens Fishery Conservation and Management Act [16 U.S.C. §1853(a)(7)] requires that Essential Fish Habitat (EFH) be designated for all federally managed species to identify important spawning, nursery, feeding, refuge, and migratory environments (GMFMC 2004). Identification of marine and estuarine habitats and evaluation of their relative ecological benefits to fishes are therefore a vital component for fisheries management.

Delineation of benthic habitats in nearshore coastal waters has historically been accomplished through a variety of techniques depending on the habitat of interest. Aerial photography and satellite imagery have been widely used to determine submerged aquatic vegetation (SAV) coverage (Kenny et al. 2003; Peneva et al. 2008), whereas single-point transect sampling via poling has been a common method for mapping shellfish reefs (Smith et al. 2001; Allen et al. 2005). Although widely accepted, there are limitations to the acquisition of accurate, spatially-explicit data associated with common survey techniques. Limiting factors include water clarity and spatial precision issues (Rivera et al. 2006). Suitable water clarity is critical to the application of aerial surveys to delineate habitats. In areas of high turbidity, the ability to visually detect submerged
vegetated habitats or shellfish reefs may be severely limited due to the low penetration of light into the water column (Allen et al. 2005; Grizzle et al. 2008). Also problematic is the accuracy of single-point transect sampling and aerial/satellite imagery, particularly when a relatively high degree of precision is needed. Transect poling lacks the precision necessary for fine-scale habitat mapping due to its dependence on a small number of data points relative to the large, often patchy, environment being sampled (Allen et al. 2005).

Charting of seabed characteristics using acoustic (echo-sounding) devices began in the 1940s with the development of sidescan sonar sonographs that produced low-resolution images of relatively large objects on the seafloor in a hard copy format (Fish and Carr 1990). Recent technological advances have made the use of field-based, remote sensing a viable and practical alternative to traditional point-based mapping methods. Sidescan data are collected via transducers on the sonar unit which emit a series of acoustic pulses, and topographic data are generated based on the intensity of the return of those pulses. Acoustic mapping has become a common application for delineating marine benthic habitats (Newton and Stefanon 1975; Kenny et al. 2003). Technological advances have led to a reduction in the size (and price) of sidescan sonar units, along with increased image resolution and better data-acquisition capabilities in a digital format. Although typically used along the continental shelf, where water depths range between 20 and 180 m in the Gulf of Mexico (GoM), sonar-based mapping has recently been used to delineate shellfish and SAV habitat in relatively shallow bay systems of less than four and 15 m depths, respectively (Allen et al. 2005; Parnum and Gavrilov
2009) and to validate aerial imagery through precise delineation of SAV boundaries (Sagawa et al. 2008).

Ecological studies can greatly benefit from spatially-explicit maps that provide delineations of relevant habitat types. In Mississippi, little is known about the late-juvenile stage of spotted seatrout, a species which is heavily exploited in coastal waters (NMFS 2013). Knowledge of habitat availability and spatial coverage can inform directed sampling to identify habitat preferences of this poorly understood stage, thus allowing for an appropriate distribution of effort when defining ecological characteristics. Comprehensive habitat mapping can therefore provide a sound basis not only for habitat conservation efforts but also for habitat assessments for the proper management of exploited fished stocks.

The purpose of this study was to map benthic habitats potentially used by late-juvenile spotted seatrout, including SAV, shellfish bottom and non-vegetated water bottom, in a shallow estuarine bay system identified as being an important nursery area (Comyns et al. 2008). Sidescan sonar and traditional point-based sampling methodologies were used to compare the relative efficiencies of both mapping methods and to identify major habitat types of importance to late-juvenile spotted seatrout and other coastal fishes. The resulting maps provided a baseline for subsequent targeted habitat-based sampling of late-juvenile spotted seatrout as part of a larger study (Chapter III). Results will contribute to a resource management protocol for identifying and quantifying important ecological habitats within shallow coastal waters.
Materials & Methods

Habitat mapping was conducted in Point aux Chenes Bay, Mississippi (Figure 1), part of the Grand Bay National Estuarine Research Reserve (GBNERR), in July 2011 and July 2012 using an Imagenex SportScan dual-channel, digital sidescan sonar unit integrated with a laptop PC and Garmin GPS-17N WAAS-differential global positioning system (dGPS). The sonar unit can acquire data at either 330 kHz or 330/800 kHz frequencies simultaneously on the port and starboard transducers, each of which is tilted downward at a 20° angle. Data collection and real-time, geo-referenced visualization of the sonar data being collected were accomplished using SonarWiz.Map version 4 software (Chesapeake Technology, Inc. 2009).

Figure 1. Map of survey area (dashed polygon) in Point aux Chenes Bay, southeastern Mississippi, USA. Inset shows general survey location (star) relative to the northern Gulf of Mexico.
Because of the shallow water depths in the survey area (generally 0.75 – 2.0 m), the sonar was hard-mounted to the bow of the vessel using a custom frame to allow it to operate just below the water surface with minimal acoustical interference from the vessel’s wake (Allen et al. 2005). After test trials to determine the most effective acoustic options, data were ultimately acquired at the 330/800 kHz (high) frequency at a 30 m range (dual beam) and a gain of 26 db. For data collection, the vessel navigated 36 pre-plotted transects spaced at 55 m intervals, allowing for sufficient overlap (5 m) of acoustic imagery between adjacent transects. Transects sampled in 2011 were steered along a line of latitude (east-west) at a speed of between 5.2 and 6.1 km/hour, depending on sea conditions; longitudinal (north-south) transects were applied in 2012 only in the general area of SAV identified in 2011 (northeast section of survey area).

Upon completion of field-based mapping, acoustic data were processed using the SonarWiz.Map software to produce image mosaics. Post-processing tools, including bottom tracking and signal processing, were utilized to produce the highest quality imagery for habitat delineation. Overlapping data between adjacent transects were averaged by the software. Processed data were converted to geo-referenced TIFF files (‘geoTIFF’) and plotted in ArcGIS, version 9.3 (ESRI 2005), for final map production. Initial habitat classification was based on visual interpretation of the acoustic data (Rivera et al. 2006). Strong acoustic returns (high backscatter) at the level of the benthos indicated the presence of oyster shell (or other hardened features), but strong returns were also uniformly produced at extremely shallow water depths (< 0.5 m) in the absence of oyster shell. A high backscatter above the substrata indicated the presence of SAV (Lefebvre et al. 2009). Additionally, bay shoreline was readily identifiable on
acoustic imagery and served as a basis for determination of marsh-edge habitat coverage.

To ground truth acoustically-derived data, field-based sampling was conducted at 522 locations distributed across a spatial grid at 100 m intervals within the survey area. Benthic habitat composition at the field locations was determined by either ponar grab (water depths ≤ 1.5 m; n = 236) or bottom poling (water depths > 1.5 m; n = 286). The 1.5 m depth threshold was based on the reported maximum depth of 1.3 m for the occurrence of SAV in the survey area (Cho and May 2008). Point-based data were collected in the field using ArcPad, version 6.0 (ESRI 2002), integrated with a Garmin GPS-17N dGPS. Habitat type was classified as either SAV, oyster shell, buried oyster shell or sand/mud based on the contents of the ponar sample (depths ≤ 1.5 m) or the firmness of substrate detected with the PVC pole (depths > 1.5 m). Point-based habitat data were overlaid onto the processed sidescan imagery to assess whether initial acoustic-based habitat designations assigned by the researcher concurred with substrate types identified from the ponar/poling survey. Areal coverage (m$^2$) was then determined for major habitat types based on each of the two mapping methods employed. Habitat-specific area was directly measured in ArcGIS for acoustically-derived data. Coverage for point-based sampling was estimated by extrapolating the proportional coverage for each habitat type estimated from its frequency of occurrence to the overall survey area. Although not a specific target habitat, areal coverage of marsh-edge in the survey area was determined using ArcGIS measurements of sonar-derived shoreline delineations, and a 50 m distance of adjacent water bottoms from the shoreline served as the basis for
areal coverage estimation (marsh-edge area = shoreline linear distance * 50 m of adjacent water bottom).

Results

A total of 92 km of transects was surveyed via sidescan sonar in Point aux Chenes Bay study area in 2011 (Figure 2). In 2012, sampling was limited to 14 transect km in the area of known SAV (Figure 3) in an effort to evaluate inter-annual variation in SAV coverage for that particular seagrass expanse and to finely delineate SAV and non-vegetated habitats for subsequent fish sampling. After post-processing but prior to data validation sampling, three particular areas were identified as distinct based on their acoustic reflections and visually classified as: “unknown return 1”, “unknown return 2”, and “suspected SAV” (Figure 4). Ground-truthing of the sidescan data via ponar grab and bottom poling illustrated the predominance of non-vegetated mud/sand in the survey area (Figure 5), with the exception of the northeastern-most portion of the bay. The area identified as “unknown return 1” in the western bay was a relatively shallow mud/sand flat area devoid of any SAV or shellfish resources.

Seven shell detections were encountered during validation sampling. Five of the detections were isolated relic shell or the margin of a shoreline oyster midden area. The two consecutive oyster shell detections occurred in the “unknown return 2” area and overlapped with the northern extent of an oyster reef created as part of a mitigation measure in 2004 (Figure 6). Supplemental bottom poling along five transects across the oyster reef revealed that, while exposed shells were present on the reef complex, the majority of the reef had become buried over time (Figure 7).
Figure 2. Sidescan sonar data acquisition in Point aux Chenes Bay, July 2011.
Figure 3. Sidescan sonar data acquisition in northeastern Point aux Chenes Bay, July 2012.
Figure 4. Initial habitat classifications based on visual interpretation of the sidescan data, July 2011.
Figure 5. Data validation (ponar grab and bottom poling) relative to sidescan data, July 2011.
Figure 6. Substrate type and sidescan data relative to 2004 oyster shell plant site location.
Figure 7. Supplemental data validation via bottom poling relative to sidescan data and 2004 oyster shell plant site location.
The northeastern portion of the survey region encompassed a relatively large area (~ 0.2 km$^2$) of designated SAV (“suspected SAV”) based on the initial classification of the acoustic data. Sidescan sonar detected in great detail the boundaries between SAV and non-vegetated habitats and facilitated visualization of individual clumps of SAV in many areas (Figure 8). Ponar grab sampling typically confirmed the presence of SAV at expected locations (Figure 9), and both *Ruppia maritima* and *Halodule wrightii* were present in grab samples, which coincides with previous reports of SAV species composition in the survey area (Cho and May 2008). However, six grabs (1.2% of total) identified as sand/mud actually fell within the SAV boundary, and one grab that yielded SAV fell outside of that boundary. Upon closer inspection of the acoustic data, the six sand/mud grabs occurred in barren, non-vegetated pockets within larger SAV patches, while the SAV grab outside of the delineated area likely fell within a small area of seagrass not evident in the imagery. Supplemental sampling again confirmed the expected habitat designations for SAV and non-vegetated bottoms (Figure 10).

A comparison of sidescan sonar data from 2011 and 2012 revealed the occurrence of SAV in the same general locations in both years (Figures 11 and 12), but SAV coverage was slightly greater in 2011 than in 2012 (Table 1). Among the three general zones (aggregations) of SAV, coverage was slightly greater in 2011 in the southern zone but slightly greater in the southeastern zone in 2012. For the northern zone, higher water levels during 2012 allowed the researchers to access an additional 11,500 m$^2$ of SAV habitat during mapping efforts, so total mapping extent was not directly comparable between years without excluding non-overlapping areas. Even so,
Figure 8. Overall survey area with SAV coverage delineated in green (top left), fine-scale inset of general SAV area with close-up area delineated in yellow (top right), and close-up of sidescan imagery depicting SAV beds and individual seagrass clumps (bottom).
Figure 9. Habitat validation data via ponar grab relative to sidescan data for initial SAV classifications.
Figure 10. Supplemental and baseline data validation via ponar grab relative to sidescan data for initial SAV classifications.
Figure 11. A comparison of image-derived SAV coverage between July 2011 and July 2012 surveys relative to 2011 survey data.
Figure 12. A comparison of image-derived SAV coverage between July 2011 and July 2012 surveys relative to 2012 survey data.
Table 1

Measurements of Annual SAV Spatial Coverage in Northeastern Point aux Chenes Bay, Mississippi, for July 2011 and July 2012 Sidescan Surveys

<table>
<thead>
<tr>
<th>Geographic Area</th>
<th>Coverage (m²) 2011 Survey</th>
<th>Coverage (m²) 2012 Survey</th>
<th>Coverage (m²) 2012 Survey*</th>
</tr>
</thead>
<tbody>
<tr>
<td>Northern</td>
<td>80,452</td>
<td>84,246</td>
<td>72,638</td>
</tr>
<tr>
<td>Southern</td>
<td>80,175</td>
<td>70,120</td>
<td>70,120</td>
</tr>
<tr>
<td>Southeastern</td>
<td>20,722</td>
<td>24,906</td>
<td>24,906</td>
</tr>
<tr>
<td>TOTAL</td>
<td>181,349</td>
<td>179,272</td>
<td>167,664</td>
</tr>
</tbody>
</table>

* excludes area surveyed in 2012 that does not overlap 2011 coverage

SAV coverage was somewhat greater in July 2011 due to an apparent general contraction of SAV toward the shoreline in July 2012.

Sonar-based mapping and point-based sampling produced relatively similar estimates of spatial coverages of non-vegetated and oyster shell habitat based on 2011 mapping (Table 2). There was little variation in oyster shell coverage between the two mapping methods when considering the larger oyster reef complex. Areal coverages of SAV aggregations, however, differed by 14% between the two survey methods, with SAV coverage being underestimated by 24,000 m² based on single point sampling.

Point-based sampling yielded six ponar collections of mud/sand substrate occurring in relatively small barren pockets within larger SAV aggregations, but those particular points were more accurately characterized as SAV in the context of ecological habitat when based on the broader-scale evaluation of comprehensive habitat coverage obtained from the sidescan sonar approach. Although not included as a target habitat, detailed
acoustic delineations of the bay shoreline allowed for the estimation of marsh-edge habitat coverage (0.196 km$^2$), wherein fish sampling was conducted as part of Chapter III.

**Discussion**

Acoustic detection and spatial delineation of the target habitats of SAV, shell material and non-vegetated soft bottoms in water depths < 2 m were successfully accomplished using a sidescan sonar sampling array. The bow mounted sonar unit operated most effectively in sea conditions of less than 0.5 m (wave height), the

**Table 2**

*Spatial Coverage of Major Habitat Types Calculated from Sonar-based Mapping and Estimated from Single Point Transect Sampling in Point aux Chenes Bay, Mississippi, for July 2011 Surveys*

<table>
<thead>
<tr>
<th>Sonar-Based Mapping Habitat Type</th>
<th>Measured Coverage (km$^2$)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Non-vegetated Sand/Mud</td>
<td>4.851</td>
</tr>
<tr>
<td>Oyster Shell</td>
<td>0.019</td>
</tr>
<tr>
<td>SAV</td>
<td>0.181</td>
</tr>
<tr>
<td>Marsh-Edge*</td>
<td>(0.196)</td>
</tr>
<tr>
<td>TOTAL</td>
<td>5.051</td>
</tr>
</tbody>
</table>

*marsh-edge included for descriptive purposes only; area not included in total coverage measurement*

<table>
<thead>
<tr>
<th>Point-Based Mapping Habitat Type</th>
<th>Frequency</th>
<th>% Frequency</th>
<th>Estimated Coverage (km$^2$)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Non-vegetated Sand/Mud</td>
<td>496</td>
<td>96.5%</td>
<td>4.874</td>
</tr>
<tr>
<td>Oyster Shell</td>
<td>2</td>
<td>0.4%</td>
<td>0.020</td>
</tr>
<tr>
<td>SAV</td>
<td>16</td>
<td>3.1%</td>
<td>0.157</td>
</tr>
<tr>
<td>TOTAL</td>
<td>514</td>
<td></td>
<td>5.051</td>
</tr>
</tbody>
</table>
threshold over which wave action negatively affected acoustic image quality. Integration of the sidescan unit with the SonarWiz.Map software facilitated data collection with the real-time positioning capability and track line production, and the program’s ability to display acoustic data in real-time allowed the surveyors to ensure sufficient data overlap between transects and to assess image quality relative to sea conditions.

As was found by Allen et al. (2005), the bow-mounted unit effectively detected the presence of exposed and buried oyster shells in the survey area. Aside from scattered relic shells primarily dispersed along shallow shoreline waters, the only oyster shells detected during mapping were associated with a reef created in 2004 as part of a mitigation action. Not surprisingly given the numerous storm events that have occurred within the region since that time, acoustic imagery and subsequent ground-truthing revealed that the majority of the created reef had become buried, with only a small fraction of oyster shell remaining exposed. Despite burial, the acoustic pulse of the sidescan unit was able to penetrate the sediment and detect thinly buried shells. Mapping projects specifically targeting exposed and buried shells may however require alternate sidescan frequency, range and/or gain settings, or added integration of a sub-bottom profiler, to differentiate between exposed and more deeply buried shell resources.

Acoustic detection and subsequent imaging of SAV was of high quality given the generally high relief of SAV relative to barren sand/mud substrates and corresponds with findings from recent sidescan-based mapping studies for various SAV species (Sagawa et al. 2008; Lefebvre et al. 2009; Parnum and Gavrilov 2009). Although not required for purposes of this research, acoustic image quality was sufficient to allow for fine-scale delineation of SAV complexity, including patch size and inter-patch
distances, which would support monitoring efforts to measure variability in SAV coverage over temporal scales (Moncreiff 2007; Carter et al. 2011). In addition, the bow-mounted unit operated effectively enough to provide quality data at the shallow water depths (< 1.3 m) at which SAV normally occurs within the survey area (Cho and May 2008).

Prior SAV mapping efforts in the survey area relied primarily on snorkeling to quantify relative SAV coverage along transects repeatedly sampled over time (Cho and May 2008). While such methodology is conducive to repeated seasonal sampling of SAV coverage along those particular transects, data acquisition via transect snorkeling is spatially limited to a small portion of the larger-scale SAV coverage. Thus transect method results may not be indicative of overall seasonal, annual or spatial trends in areal coverage. Comparison of the two mapping techniques used in this study support that assumption, as SAV coverage was underestimated based on single-point ponar grab sampling. Mapping efforts that require accurate and precise delineations of benthic habitats would benefit from the application of acoustic sampling. The use of the sidescan sonar integrated with SonarWiz.Map software described here offered a relatively cost-effective and time-efficient option for comprehensive mapping and areal quantification of SAV habitats using standard GIS software for precise calculations of habitat coverage.

Although not within the scope of this research, future efforts should be directed at developing classification techniques for acoustically-derived delineations of SAV occurring at ultra-shallow (< 1.5 m) depths, for discrimination between shell and buried shell resources, and for general habitat classifications in shallow bay systems (Rivera et
Allen et al. (2005) evaluated the utility of Imagine image analysis and an Isis training module as a means to classify acoustic data into two general categories. They found general application of those techniques for research and management purposes to be very satisfactory.

The use of sonar-derived acoustic data in ecological assessments is increasing. These data have been combined with measurements of environmental variables to comprehensively describe habitat and better predict abundances of fish and invertebrates (Yeung and McConnaughey 2008). Side-scan mapping has also been used in fishery stock enhancement to identify habitats that would promote survival of hatchery-reared fish released into the wild (Edsall et al. 1989). Acoustic imagery from the current study was used as a basis to determine habitat use patterns by late-juvenile spotted seatrout (*Cynoscion nebulosus*), a popular coastal fishery species.

In summary, sidescan sonar offered an excellent approach for identifying, characterizing and quantifying habitat coverage, as a prelude to targeted sampling of late-juvenile spotted seatrout within an important nursery area. The methods used in this study provided quality acoustic imagery of shallow estuarine habitats and allowed for accurate areal quantification of those benthic resources within the Grand Bay NERR. When compared to point-based data, sonar-derived imagery provided more accurate estimation of habitat coverage and provided the ability to precisely delineate habitat boundaries, particularly for SAV. Relative to other remote sensing applications, sidescan sonar provided quality imagery while operating in ultra-shallow (< 1.5 m) waters and in relatively turbid conditions. Overall, the bow-mounted sidescan is a viable alternative for shallow water mapping applications and should be implemented in future studies to
more accurately delineate estuarine habitats that provide various ecological services benefit. Of particular importance is locating, delineating and protecting spatially limited SAV in nearshore coastal waters, as those habitats are known to offer important feeding and refuge habitat for fishes but are not nearly as prevalent as other benthic habitats, such as non-vegetated sand/mud water bottoms. Coastal SAV habitat is too infrequently mapped at large spatial scales (Moncreiff 2007) but has shown considerable decadal-scale variations in spatial coverage (Moncrieff et al. 1998; Carter et al. 2011), so its relative importance as fish habitat likely varies on both temporal and limited spatial scales in Mississippi coastal waters.
LITERATURE CITED


CHAPTER III
HABITAT USE BY JUVENILE SPOTTED SEATROUT
IN A MISSISSIPPI BAY SYSTEM

Introduction

Optimal habitat selection affords organisms the opportunity to maximize net energy gain while avoiding both predators and competitors (Knieb 1987). As such, habitat is not defined merely by the physical structure in and around where a species occurs but also includes the physical-chemical properties of the surrounding water, as well as other co-occurring biota, including prey, predators and competitors. Together, all of these habitat factors determine whether conditions are suitable in terms of survival (e.g., through predator avoidance) and growth (e.g., through efficient and sufficient feeding) (Baltz et al. 2003). Habitat delineation and determination of associated ecological services are therefore necessary for assessing habitat requirements of fishes throughout their life history.

The spotted seatrout occurs almost exclusively within estuarine systems throughout its life cycle (Bortone 2003). Estuarine habitat use varies by life-history stage and based on the availability of alternative habitat types within a given estuary (Helser et al. 1993). Numerous studies have evaluated habitat use by various life-history stages of spotted seatrout, but relatively little data are available for the late juvenile stage. Spotted seatrout appear to show an affinity for structured habitats. In Florida where extensive seagrass meadows occur, spotted seatrout reportedly spawn over areas of submerged aquatic vegetation (SAV) (McMichael and Peters 1989). Habitat for early juvenile spotted seatrout was characterized by Tabb (1966) as brackish waters...
containing extensive areas of seagrass adjacent to deep-water areas (for refuge from extreme temperatures). McMichael and Peters (1989) collected early juveniles less than 75 mm standard length (SL) primarily in shallow *Spartina* marsh-edge shoreline or SAV habitats. Seagrass is also preferred larval habitat for spotted seatrout in Texas coastal waters (Tolon et al. 1997).

In the highly turbid estuaries of Mississippi and Louisiana, SAV is limited and spotted seatrout occupy other habitat types (Lorio and Perret 1980). Peterson (1986) found habitat for early juveniles (< 125 mm TL) in Caminada Bay to be shallow *Spartina* marsh-edge and unvegetated shoreline with soft to medium-soft mud/detritus substrate. He further noted that marsh edge may be preferable habitat for fish up to 50 mm TL in areas devoid of SAV. In the Barataria Basin, microhabitat use by three size classes of early juvenile spotted seatrout (all ≤ 100 mm SL) was consistent in that fish occurred more frequently than expected in samples from emergent (marsh-edge) vegetation (Rakocinski et al. 1992). In a later study in Barataria Bay, MacRae (2006) found no significant difference in catch-per-unit-effort (CPUE) of large juvenile (overall mean SL > 220 mm) spotted seatrout sampled from marsh-edge, oyster shell and soft mud habitats, but fish were more abundant in oyster shell and mud habitats than in marsh-edge. Contrary to Lorio and Perret (1980) who largely dismissed the importance of SAV to seatrout along mainland Louisiana, Thompson and Verret (1980) found that young-of-the-year (YOY) spotted seatrout utilized SAV within Lake Pontchartrain in summer and fall, and SAV and adjacent shallow pools were shown to be preferred habitats for this species at the Chandeleur Islands (Laska 1973).
Habitat use by spotted seatrout is not likely focused exclusively on SAV throughout its life history or in areas where SAV is not abundant, which may explain the ambiguous information regarding its importance. Habitat use assessments should consider relative abundance of structural habitat types and also hydrological (physical-chemical) conditions where a species occurs, particularly salinity and temperature, as those factors have metabolic consequences for growth and survival. For example, MacRae (2006) noted that CPUE of large juvenile and sub-adult spotted seatrout correlated significantly with temperature and salinity. Furthermore, Herke et al. (1984) observed that salt marsh may not provide suitable habitat for YOY spotted seatrout in Louisiana unless accompanied by suitable salinity, as juveniles from Caminada Bay were most frequently found at salinities between 8 and 25. Conversely, state-wide seine data revealed high relative juvenile abundances at salinities lower than 10 (Thomas 1999). Chester and Thayer (1990) contended salinity was not a major factor affecting the distribution of juvenile spotted seatrout in Florida Bay, where mean salinities were higher (> 35.4) and more homogeneous relative to north-central GoM bay systems.

It is evident from the wide range of habitats and physical conditions in which spotted seatrout occur that ecological conditions of this species can vary spatially, ontogenetically and temporally. Use of multiple habitat types by adults is a consequence of their limited movements within estuarine systems (Hendon et al. 2002). Also, they occur in discrete subpopulations that are spatially divided among different bay systems in the northern GoM (Gold et al. 1999). Although SAV and emergent vegetation provide habitat for early juvenile spotted seatrout (Peterson 1986; McMichael and Peters 1989; Rakocinski et al. 1992), MacRae (2006) found large juveniles (mean SL between 220
and 254 mm) more common at deeper oyster shell and soft mud sites than at shallow marsh-edge sites. This suggests an ontogenetic shift to deeper waters by large juveniles and sub-adults. However, SAV was not present at her study sites (MacRae 2006). Peterson (1986) also noted that large juveniles may shift into deeper waters away from the marsh edge and form schools, particularly in winter when cold shallow-water temperatures are not conducive to survival. Gunter (1938) also reported high numbers of large YOY spotted seatrout from deeper bay waters in winter months.

Although considerable data exist for larval, early juvenile and adult stages of spotted seatrout in coastal waters of Mississippi and nearby states, sufficient information on the occurrence and ecology of late-juvenile spotted seatrout in the 125 to 275 mm TL range is still lacking. In a study on the distribution of larval and small juvenile spotted seatrout in Mississippi coastal waters, Warren et al. (2001) found fish 2.7 to 131.0 mm SL to be more abundant at mainland stations with vegetated (marsh) mud than at those with vegetated and non-vegetated sand. Differences in abundance were not significant, however, and no mainland SAV stations were sampled in this study. Notably, early juvenile spotted seatrout were significantly more abundant in SAV than in non-vegetated sand at barrier island stations.

Because of the spatial variability in spotted seatrout distribution, studies on regional or even bay-specific scales are necessary to define habitat use and hydrological associations relevant to the management of local populations. In Mississippi, such data are needed for late juveniles to address gaps in life-history information of the species and for more effective management through habitat protection. This would be especially important for bay systems, such as Point aux Chenes Bay, that potentially
serve as important nursery source areas within Mississippi coastal waters (Comyns et al. 2008). Furthermore, the Seatrout Population Enhancement Cooperative (SPEC), a joint hatchery rearing venture between Mississippi agencies, was established in 2004. Data on habitat use by juvenile spotted seatrout are needed in order to understand the fate of hatchery-reared fish that are released into the wild. The objective of this study was to compare relative abundances of late juvenile spotted seatrout within SAV, marsh-edge, and non-vegetated sand/mud habitats delineated from sidescan mapping as a means to infer their preferred habitat use within the Point aux Chenes Bay system of Mississippi.

Materials & Methods

All collections were made in Point aux Chenes Bay, Mississippi, within of the Grand Bay National Estuarine Research Reserve (Figure 13). Based on otolith microchemical analyses, Comyns et al. (2008) found that the Grand Bay system represented an extensive nursery area for spotted seatrout in Mississippi and may be a major source area for the state's population of the species. Of the adults (mostly ages 1 and 2) collected in 2002 from Grand Bay, 85% had the Grand Bay otolith chemical signature. Forty-four percent of adults collected from the Pascagoula drainage, 39% collected from Biloxi Bay and 22% from St. Louis Bay also had the Grand Bay signature. This suggests Grand Bay is a vital spawning and nursery area providing a source of recruitment to Mississippi's spotted seatrout population, presumably through dispersal at the YOY stages based on the limited movements observed for adults.

Traditional fish sampling gear has been largely ineffective for collecting late-juvenile spotted seatrout, defined as fish between 125 and 275 mm TL. Previous studies have collected juvenile spotted seatrout with seines (Peterson 1986; McMichael and
Peters 1989; Rutherford et al. 1989; Warren et al. 2001; Powell et al. 2004), trawls
(Chester and Thayer 1990; Powell et al. 2004; Dorval et al. 2005) and drop sampler
(Rakocinski et al. 1992; Baltz et al. 1998); yet those gear types collected fish primarily
in the 5 to 125 mm range, 20 to 100 mm range, and 5 to 100 mm range, respectively.
Additionally, Powell et al. (2004) noted that juveniles greater than approximately 90
days old (70 mm SL) appeared to avoid their gear (otter trawl and seine). Considering
those studies and the target sizes sought in this study, sampling was conducted with an
experimental gill net measuring 122.0 m long by 1.8 m deep and consisting of four 30.5
m panels of 1.3, 1.6, 1.9 and 2.5 cm (square) mesh. An identical net measuring 1.2 m
deep was used in depths shallower than 1.2 m. Samples were collected weekly from
August to November 2011 and July to November in 2012. Initiation of sampling in late summer allowed for the collection of early-spawned age-0 fish and anticipated increases in juvenile recruitment through the fall. Sampling ceased once low water levels associated with seasonal northerly wind patterns prevented effective sampling in each of the habitat types.

As described in Chapter II, sidescan sonar mapping was conducted in early July of both years to identify and delineate target habitats within the study area, especially SAV. A weighted stratified random sampling design was initially established in which strata were represented by habitat types (n = 3), and the initial weighting factor was proportional to prior knowledge of the coverage by habitats within the study area, as determined from sidescan mapping. However, (1) given the disproportionately high coverage of non-vegetated (NV) habitats within the study area relative to SAV and marsh-edge (ME), (2) considering the difficulty in safely and effectively collecting all samples in a given day, and (3) given the need for a minimum of three replicates per habitat per sampling event, the final allocation of gill net sampling stations representing the target habitats was six stations per event for NV, three per event for SAV and three per event for ME. Based on habitat mapping validation samples, SAV habitats comprised a combination of *Ruppia maritima* and *Halodule wrightii*, which agrees with previous reports of SAV species composition in the Grand Bay Reserve (Cho and May 2008). Marsh-edge within the study area was almost exclusively erosional in nature; depositional marsh habitats typically occurred within the more wave-protected bayou systems north of Point aux Chenes Bay.
A grid layer of stations spaced at 100 m intervals was overlaid onto the final habitat classification map, and gill net stations were then assigned to a stratum based on their relative positions to target habitats (Figure 14). To alleviate potential edge effects, a 150 m minimum buffer strip was placed between/among adjacent habitats. Additionally, one oyster reef area identified from sidescan mapping was excluded from the gill net station selection process and the 150-m buffer was also applied to that area.

During each weekly field sampling event, surface salinity (PSU), temperature (°C) and dissolved oxygen (mg/L) were measured with a YSI® 85 meter, and surface turbidity (NTU) was measured with a LaMotte® 2020i turbidity meter. Water depth (cm) was measured with a depth-calibrated pole, and tide height (cm) for each station was

![Map depicting final gill net sampling station allocation among habitats in Point aux Chenes Bay, with 150-m minimum buffer among habitat types and excluded oyster reef area depicted.](image)

**Figure 14.** Final gill net sampling station allocation among habitats in Point aux Chenes Bay, with 150-m minimum buffer among habitat types and excluded oyster reef area depicted.
determined based on predicted tide height for the Point aux Pines, Mississippi location, obtained from WWW Tide and Current Predictor (http://tbone.biol.sc.edu/tide/).

Fish were collected at randomly selected stations each week by utilizing the gill net as a *strike* net, following a prescribed set of guidelines. The net was deployed from the bow of the vessel at each of the sampling sites, starting at the shoreline for the marsh-edge sites. Because of depth variation among habitat types (i.e., ME shoreline vs. deeper NV habitat), the configuration of the net could differ somewhat. The net was set in a crescent shape for SAV and NV habitats and set parallel to the shoreline for ME sites. After deployment, the vessel was circled around the perimeter and inside of the net two complete times, gradually decreasing the distance to the net at each pass. After the two rotations, the net was retrieved. All organisms were removed from the net and identified to species; for each species, total number, total weight (0.01 kg), and minimum and maximum lengths (mm) were recorded for each panel. All spotted seatrout were anesthetized using MS-222, bagged, labeled and returned to the laboratory for processing. Specimens of all other species were released immediately after processing.

*Statistical Analyses*

The null hypotheses were defined as no difference in catch-per-unit-effort (CPUE) in number, CPUE in biomass, and size (TL) of spotted seatrout among habitat types or between years. CPUE values were standardized relative to effort in terms of the number of minutes the net was fished. A two-way analysis of variance (ANOVA) tested whether the dependent variables of fish size or CPUE in abundance or biomass differed among habitats (3 levels) and years (2 levels) as fixed factors, with the type-I error rate
set to α = 0.05. Weekly samples of replicate sites within each habitat type and within each year were compared as the basis for among-habitat and between-year variation. If no interaction occurred between the habitat factors, but significant differences existed among levels of either factor, \textit{a posteriori} multiple comparisons were made with a Tukey’s HSD (equal variance assumed) or Games-Howell (equal variances not assumed) test. Habitat variables (salinity, temperature, dissolved oxygen, turbidity, water depth, and tide height) were similarly tested among habitat and year factors. To estimate potential density-dependent effects, total fish density, defined as the CPUE in weight of all co-occurring species inclusive of spotted seatrout, was included as a habitat variable in the analyses. Catch-per-unit-effort data were $\log(x+0.01)$-transformed where appropriate to alleviate violations of the test assumption of normality.

Because relationships involving correlations of habitat variables and seatrout occurrence may be meaningful, Principal Components Analysis was run with all habitat-related data to reduce collinear variables into fewer latent variables for ease of interpretation. Data reduction was considered informative at a Kaiser-Meyer-Olkin Measure of Sampling Adequacy (KMO) value ≥ 0.700 (Leech et al. 2011) and a statistically significant Bartlett’s Test of Sphericity (p < 0.05) suggesting positive variable correlation. Factors with eigenvalues ≥ 1.00 were extracted, varimax (orthogonal) rotation was applied to maximize differences among factors, and resulting rotated factor scores were saved to the associated database. Variables that loaded at or above | 0.5 | were considered informative for that particular factor. The relationships between factors and seatrout presence/absence were analyzed by binary logistic regression to identify any significant associations that may have constrained the
distribution of fish. Bray-Curtis community dissimilarity calculated among sample
events based on log-transformed CPUE-biomass data for the juvenile fish community
was compared using analysis of similarity (ANOSIM) to test for differences in fish
community structure between samples in which spotted seatrout were present versus
samples in which they were absent. Non-metric multi-dimensional scaling (MDS)
graphically displayed sample dissimilarities in reduced 3-D space. Furthermore, a
similarity percentage test (SIMPER) was conducted following a significant ANOSIM
test to identify which species were primarily responsible for between-group differences
in assemblage structure. MDS plots were considered informative at stress values less
than 0.20 (Clarke and Warwick 2001). For further insights, this procedure was repeated
to assess general fish community dissimilarity among the three habitat treatments.

All univariate, regression and PCA analyses were conducted with SPSS
Statistics, version 20.0 (IBM 2011). Multivariate analyses (ANOSIM, MDS and
SIMPER) were conducted in PRIMER-E, version 6 (Clarke and Gorley 2006). All tests
were considered significant at p < 0.05.

Results

Habitat Conditions

Data were collected at 278 stations in 2011 (n=119) and 2012 (n=159) (Table 3).Mean monthly values for salinity followed a parabolic pattern from summer through fall
for both years (Figure 15); temperature, turbidity, tide height and total fish biomass
decreased over the period (Figures 16 - 19). Mean monthly dissolved oxygen varied
between years, with an increase observed from August to November in 2011 but a
decreasing trend was found after September in 2012 (Figure 20). Water depth was the
Table 3

Descriptive Statistics for Habitat Variables for All Samples Pooled, Among Habitats, and Between Years

<table>
<thead>
<tr>
<th></th>
<th>ALL SAMPLES POOLED</th>
<th></th>
<th>AMONG HABITATS</th>
<th></th>
<th></th>
<th></th>
<th></th>
<th></th>
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</thead>
<tbody>
<tr>
<td></td>
<td>n</td>
<td>Mean</td>
<td>S.E.</td>
<td>Minimum</td>
<td>Maximum</td>
<td>n</td>
<td>Mean</td>
<td>S.E.</td>
</tr>
<tr>
<td>Surface Salinity</td>
<td>278</td>
<td>24.3</td>
<td>0.20</td>
<td>11.1</td>
<td>31.0</td>
<td>70</td>
<td>24.4</td>
<td>0.35</td>
</tr>
<tr>
<td>Surface Temperature (°C)</td>
<td>270</td>
<td>26.1</td>
<td>0.28</td>
<td>13.4</td>
<td>33.7</td>
<td>70</td>
<td>25.6</td>
<td>0.58</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen (mg/L)</td>
<td>270</td>
<td>6.0</td>
<td>0.07</td>
<td>3.4</td>
<td>9.4</td>
<td>67</td>
<td>5.5</td>
<td>0.16</td>
</tr>
<tr>
<td>Surface Turbidity (NTU)</td>
<td>278</td>
<td>14.5</td>
<td>0.45</td>
<td>3.0</td>
<td>48.2</td>
<td>70</td>
<td>15.9</td>
<td>0.83</td>
</tr>
<tr>
<td>Water Depth (cm)</td>
<td>278</td>
<td>123.7</td>
<td>2.55</td>
<td>57.9</td>
<td>222.5</td>
<td>70</td>
<td>92.4</td>
<td>2.15</td>
</tr>
<tr>
<td>Tide Height (cm)</td>
<td>278</td>
<td>33.3</td>
<td>0.91</td>
<td>6.1</td>
<td>64.0</td>
<td>70</td>
<td>38.7</td>
<td>1.73</td>
</tr>
<tr>
<td>CPUE (biomass, kg)</td>
<td>278</td>
<td>0.29</td>
<td>0.02</td>
<td>0.0</td>
<td>2.4</td>
<td>70</td>
<td>0.32</td>
<td>0.04</td>
</tr>
</tbody>
</table>

|                          | Marsh-Edge         |                          |                |                          |                      |                      |                      |                      |
|                          | n                  | Mean                     | S.E.           | Minimum                  | Maximum              | n                  | Mean                     | S.E.           | Minimum                  | Maximum              |
| Surface Salinity         | 68                 | 24.3                     | 0.43           | 13.7                     | 30.1                 | 66                 | 26.4                     | 0.58           | 14.3                     | 33.3                 |
| Surface Temperature (°C) | 66                 | 26.4                     | 0.58           | 14.3                     | 33.3                 | 66                 | 6.2                      | 0.15           | 3.5                      | 9.4                  |
| Surface Dissolved Oxygen (mg/L) | 66 | 6.2 | 0.15 | 3.5 | 9.4 | 68 | 16.9 | 1.07 | 3.5 | 48.2 |
| Surface Turbidity (NTU)  | 68                 | 16.9                     | 1.07           | 3.5                      | 48.2                 | 68                 | 89.6                     | 2.45           | 57.9                     | 128.0                |
| Water Depth (cm)         | 68                 | 89.6                     | 2.45           | 57.9                     | 128.0                | 68                 | 31.8                     | 1.82           | 6.1                      | 64.0                 |
| Tide Height (cm)         | 68                 | 31.8                     | 1.82           | 6.1                      | 64.0                 | 68                 | 0.33                     | 0.04           | 0.0                      | 2.3                  |
Table 3 (continued).

<table>
<thead>
<tr>
<th>Non-Vegetated</th>
<th>n</th>
<th>Mean</th>
<th>S.E.</th>
<th>Minimum</th>
<th>Maximum</th>
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<tbody>
<tr>
<td>Surface Salinity</td>
<td>140</td>
<td>24.4</td>
<td>0.29</td>
<td>11.1</td>
<td>31.0</td>
</tr>
<tr>
<td>Surface Temperature (°C)</td>
<td>134</td>
<td>26.2</td>
<td>0.38</td>
<td>14.9</td>
<td>32.5</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen (mg/L)</td>
<td>134</td>
<td>6.2</td>
<td>0.10</td>
<td>3.9</td>
<td>8.7</td>
</tr>
<tr>
<td>Surface Turbidity (NTU)</td>
<td>140</td>
<td>12.7</td>
<td>0.56</td>
<td>3.0</td>
<td>34.7</td>
</tr>
<tr>
<td>Water Depth (cm)</td>
<td>140</td>
<td>155.8</td>
<td>2.86</td>
<td>61.0</td>
<td>222.5</td>
</tr>
<tr>
<td>Tide Height (cm)</td>
<td>140</td>
<td>31.3</td>
<td>1.28</td>
<td>6.1</td>
<td>61.0</td>
</tr>
<tr>
<td>CPUE (biomass, kg)</td>
<td>140</td>
<td>0.25</td>
<td>0.03</td>
<td>0.0</td>
<td>2.4</td>
</tr>
</tbody>
</table>

### BETWEEN YEARS

#### 2011

<table>
<thead>
<tr>
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<th>Minimum</th>
<th>Maximum</th>
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<tbody>
<tr>
<td>Surface Salinity</td>
<td>119</td>
<td>25.6</td>
<td>0.22</td>
<td>20.3</td>
<td>30.2</td>
</tr>
<tr>
<td>Surface Temperature (°C)</td>
<td>119</td>
<td>26.8</td>
<td>0.39</td>
<td>18.9</td>
<td>33.7</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen (mg/L)</td>
<td>119</td>
<td>6.2</td>
<td>0.10</td>
<td>3.4</td>
<td>9.4</td>
</tr>
<tr>
<td>Surface Turbidity (NTU)</td>
<td>119</td>
<td>15.1</td>
<td>0.66</td>
<td>3.5</td>
<td>48.2</td>
</tr>
<tr>
<td>Water Depth (cm)</td>
<td>119</td>
<td>120.6</td>
<td>3.56</td>
<td>57.9</td>
<td>213.4</td>
</tr>
<tr>
<td>Tide Height (cm)</td>
<td>119</td>
<td>33.6</td>
<td>1.29</td>
<td>15.2</td>
<td>64.0</td>
</tr>
<tr>
<td>CPUE (biomass, kg)</td>
<td>119</td>
<td>0.38</td>
<td>0.04</td>
<td>0.0</td>
<td>2.4</td>
</tr>
</tbody>
</table>

#### 2012

<table>
<thead>
<tr>
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<th>Mean</th>
<th>S.E.</th>
<th>Minimum</th>
<th>Maximum</th>
</tr>
</thead>
<tbody>
<tr>
<td>Surface Salinity</td>
<td>159</td>
<td>23.3</td>
<td>0.28</td>
<td>11.1</td>
<td>31.0</td>
</tr>
<tr>
<td>Surface Temperature (°C)</td>
<td>151</td>
<td>25.6</td>
<td>0.39</td>
<td>13.4</td>
<td>33.3</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen (mg/L)</td>
<td>148</td>
<td>5.9</td>
<td>0.10</td>
<td>3.4</td>
<td>9.0</td>
</tr>
<tr>
<td>Surface Turbidity (NTU)</td>
<td>159</td>
<td>14.1</td>
<td>0.62</td>
<td>3.0</td>
<td>41.1</td>
</tr>
<tr>
<td>Water Depth (cm)</td>
<td>159</td>
<td>126.0</td>
<td>3.57</td>
<td>57.9</td>
<td>222.5</td>
</tr>
<tr>
<td>Tide Height (cm)</td>
<td>159</td>
<td>33.0</td>
<td>1.27</td>
<td>6.1</td>
<td>57.9</td>
</tr>
<tr>
<td>CPUE (biomass, kg)</td>
<td>159</td>
<td>0.22</td>
<td>0.02</td>
<td>0.0</td>
<td>2.3</td>
</tr>
</tbody>
</table>

Note that technical problems with the YSI meter prevented measurements of temperature and/or dissolved oxygen for a small number of stations.
Figure 15. Mean monthly (± SE) surface salinity (PSU) for the survey periods.

Figure 16. Mean monthly (± SE) surface temperature (°C) for the survey periods.
Figure 17. Mean monthly (± SE) surface turbidity (NTU) for the survey periods.

Figure 18. Mean monthly (± SE) tide height (cm) for the survey periods.
**Figure 19.** Mean monthly (± SE) CPUE in biomass (kg, raw data) of all species for the survey periods.

**Figure 20.** Mean monthly (± SE) surface dissolved oxygen (mg/L) for the survey periods.
most variable of all the environmental components evaluated, with monthly means fluctuating between 115 and 135 cm (Figure 21).

Two-way ANOVA’s of habitat variables detected significant habitat*year interactions for both dissolved oxygen and CPUE-biomass (Table 4). The interaction effect for dissolved oxygen was driven by lower measurements in SAV for 2012 samples relative to 2011 (Figure 22). Log CPUE-biomass differed among habitats (p = 0.002) and between years (p < 0.001), with the interaction effect largely reflecting habitat-specific differences for 2012 samples (Figure 23).

Among-habitat differences were detected for turbidity, water depth and tide height (Table 4). Turbidity was significantly lower and water depth significantly higher at NV sites than at SAV (p = 0.008 and p < 0.001, respectively) and ME sites (p < 0.001, for both), while SAV and ME means did not differ for either variable (p = 0.707 and

![Figure 21](image-url). Mean monthly (± SE) water depth (cm) for the survey periods.
Table 4

*Results of Two-way ANOVA Comparisons of Habitat Variables Among Habitats and Between Years*

<table>
<thead>
<tr>
<th>Dependent Variable</th>
<th>Habitat*Year Effects</th>
<th>Habits Effects</th>
<th>Years Effects</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>df</td>
<td>F</td>
<td>p</td>
</tr>
<tr>
<td>Surface Salinity (PSU)</td>
<td>2</td>
<td>0.20</td>
<td>0.821</td>
</tr>
<tr>
<td>Surface Temperature (°C)</td>
<td>2</td>
<td>0.65</td>
<td>0.522</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen (mg/L)</td>
<td>2</td>
<td>11.61</td>
<td>&lt; 0.001</td>
</tr>
<tr>
<td>Surface Turbidity (NTU)</td>
<td>2</td>
<td>0.41</td>
<td>0.662</td>
</tr>
<tr>
<td>Water Depth (cm)</td>
<td>2</td>
<td>1.51</td>
<td>0.222</td>
</tr>
<tr>
<td>Tide Height (cm)</td>
<td>2</td>
<td>0.30</td>
<td>0.738</td>
</tr>
<tr>
<td>CPUE (biomass, g) (log)</td>
<td>2</td>
<td>6.24</td>
<td>0.002</td>
</tr>
</tbody>
</table>

*p = 0.685, respectively* (Figures 24 and 25). For tide height, means differed between SAV and ME (*p = 0.019*) and SAV and NV (*p = 0.002*), with tide height being higher at SAV for both comparisons; tide height was not different between ME and NV (*p = 0.975*) (Figure 26). Excluding dissolved oxygen and CPUE-biomass for which interactive effects were detected, between-year effects were significant only for salinity and temperature (Table 4), with both means being higher in 2011 than in 2012 (*p < 0.001*; Table 3).
Figure 22. Mean annual dissolved oxygen among habitat types. Matching number annotations indicate a significant difference between habitat types. Note that year and habitat*year effects were also significant.

Figure 23. Mean annual CPUE in biomass (log-transformed) among habitat types. Matching number annotations indicate a significant difference between habitat types. Significant year and habitat*year effects were also detected.
Figure 24. Mean annual turbidity among habitat types. Matching number annotations indicate a significant difference between habitat types.

Figure 25. Mean annual water depth among habitat types. Matching number annotations indicate a significant difference between habitat types.
Principal components analysis (PCA) reduced the seven habitat variables into three meaningful factors, as supported by significant Bartlett’s Test (p < 0.001); however, the calculated KMO level of 0.569 was marginal and below the 0.700 threshold considered to be informative. Overall, 65.5% of the variance in habitat variables was explained by Factors I, II and III. Positive loadings on Factor I accounted for seasonal trends in temperature, turbidity and log(CPUE biomass), and loadings on Factor II included primarily daily fluctuations in water depth and tide height; whereas Factor III comprised positive (salinity) and negative (DO) loadings (Table 5).

**Spotted Seatrout**

Gill net sets were made from August to November 2011 (n=119) and July to November 2012 (n=159), with 5,776 fish weighing 344.1 kg collected (Table 6). Of the total, 55 individuals were spotted seatrout. The majority (65.4%) of spotted seatrout...
Table 5

PCA Rotated Factor Loadings for Habitat Variables with Initial Eigenvalues Listed in Parentheses Under Each Factor

<table>
<thead>
<tr>
<th>Habitat Variable</th>
<th>Factor I (2.154)</th>
<th>Factor II (1.280)</th>
<th>Factor III (1.148)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Surface Temperature</td>
<td>0.856</td>
<td>0.238</td>
<td>0.002</td>
</tr>
<tr>
<td>Surface Turbidity</td>
<td>0.706</td>
<td>-0.468</td>
<td>0.027</td>
</tr>
<tr>
<td>Total Fish Biomass (log)</td>
<td>0.804</td>
<td>-0.094</td>
<td>0.034</td>
</tr>
<tr>
<td>Water Depth</td>
<td>-0.083</td>
<td>0.837</td>
<td>-0.162</td>
</tr>
<tr>
<td>Tide Height</td>
<td>0.111</td>
<td>0.548</td>
<td>0.455</td>
</tr>
<tr>
<td>Surface Salinity</td>
<td>-0.330</td>
<td>-0.030</td>
<td>0.658</td>
</tr>
<tr>
<td>Surface Dissolved Oxygen</td>
<td>-0.280</td>
<td>0.056</td>
<td>-0.731</td>
</tr>
<tr>
<td>% Variance Explained</td>
<td>30.8%</td>
<td>18.3%</td>
<td>16.4%</td>
</tr>
<tr>
<td>Cumulative % Variance Explained</td>
<td>—</td>
<td>49.1%</td>
<td>65.5%</td>
</tr>
</tbody>
</table>

Note. Variables with loadings ≥ | 0.5 | were considered informative for that particular factor.

collected were target-sized late juveniles (125 – 275 mm TL) (Figure 27), and all but one individual (485 mm TL) was in the 138 to 354 mm TL size range. With respect to mesh size, only two spotted seatrout were collected in the 1.3-cm mesh panel (138 and 354 mm TL), while the remaining three panels were more effective at capturing target-size juveniles (Figures 28 and 29). Sizes for late juveniles ranged from 146 to 232 mm TL for the 1.6 cm mesh, 183 to 268 mm for 1.9 cm mesh, and 240 to 275 mm for 2.5 cm mesh (Table 7).

Spotted seatrout were collected from each of the three habitat types (Figure 30 - 32), however, total abundance and total biomass were greatest in SAV (Figure 33).

When including only target-size juveniles, total abundance was still higher at SAV
Table 6

Total and Percent Number, Total and Percent Weight (kg), Length Measurement Type, and Minimum and Maximum Size (mm) for All Fish Species Collected

<table>
<thead>
<tr>
<th>Species</th>
<th>Number</th>
<th>% Number</th>
<th>Weight (kg)</th>
<th>% Weight</th>
<th>Type</th>
<th>Min.</th>
<th>Max.</th>
</tr>
</thead>
<tbody>
<tr>
<td>Brevoortia patronus</td>
<td>1258</td>
<td>21.8%</td>
<td>73.8</td>
<td>21.4%</td>
<td>Fork</td>
<td>67</td>
<td>273</td>
</tr>
<tr>
<td>Micropogonias undulatus</td>
<td>837</td>
<td>14.5%</td>
<td>40.4</td>
<td>11.7%</td>
<td>Total</td>
<td>97</td>
<td>297</td>
</tr>
<tr>
<td>Ariopsis felis</td>
<td>816</td>
<td>14.1%</td>
<td>32.6</td>
<td>9.5%</td>
<td>Fork</td>
<td>65</td>
<td>330</td>
</tr>
<tr>
<td>Leiostomus xanthurus</td>
<td>492</td>
<td>8.5%</td>
<td>31.4</td>
<td>9.1%</td>
<td>Total</td>
<td>76</td>
<td>211</td>
</tr>
<tr>
<td>Bairdiella chrysoura</td>
<td>295</td>
<td>5.1%</td>
<td>9.0</td>
<td>2.6%</td>
<td>Total</td>
<td>75</td>
<td>254</td>
</tr>
<tr>
<td>Hyporthampus macki</td>
<td>272</td>
<td>4.7%</td>
<td>11.4</td>
<td>3.3%</td>
<td>Fork</td>
<td>115</td>
<td>288</td>
</tr>
<tr>
<td>Harengula jaguana</td>
<td>253</td>
<td>4.4%</td>
<td>7.3</td>
<td>2.1%</td>
<td>Fork</td>
<td>69</td>
<td>165</td>
</tr>
<tr>
<td>Lagodon rhomboides</td>
<td>248</td>
<td>4.3%</td>
<td>19.7</td>
<td>5.7%</td>
<td>Fork</td>
<td>106</td>
<td>234</td>
</tr>
<tr>
<td>Elops saurus</td>
<td>221</td>
<td>3.8%</td>
<td>34.5</td>
<td>10.0%</td>
<td>Fork</td>
<td>186</td>
<td>606</td>
</tr>
<tr>
<td>Dorosoma petenense</td>
<td>154</td>
<td>2.7%</td>
<td>2.1</td>
<td>0.6%</td>
<td>Fork</td>
<td>72</td>
<td>260</td>
</tr>
<tr>
<td>Bagre marinus</td>
<td>129</td>
<td>2.2%</td>
<td>4.6</td>
<td>1.3%</td>
<td>Fork</td>
<td>69</td>
<td>345</td>
</tr>
<tr>
<td>Caranx hippos</td>
<td>127</td>
<td>2.2%</td>
<td>7.8</td>
<td>2.3%</td>
<td>Fork</td>
<td>94</td>
<td>236</td>
</tr>
<tr>
<td>Opisthonema oglinum</td>
<td>109</td>
<td>1.9%</td>
<td>0.9</td>
<td>0.3%</td>
<td>Fork</td>
<td>69</td>
<td>191</td>
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<tr>
<td>Strongylura marina</td>
<td>81</td>
<td>1.4%</td>
<td>5.5</td>
<td>1.6%</td>
<td>Fork</td>
<td>253</td>
<td>605</td>
</tr>
<tr>
<td>Mugil curema</td>
<td>81</td>
<td>1.4%</td>
<td>12.0</td>
<td>3.5%</td>
<td>Fork</td>
<td>101</td>
<td>256</td>
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<tr>
<td>Mugil cephalus</td>
<td>75</td>
<td>1.3%</td>
<td>12.3</td>
<td>3.6%</td>
<td>Fork</td>
<td>98</td>
<td>323</td>
</tr>
<tr>
<td>Scomberomorus maculatus</td>
<td>75</td>
<td>1.3%</td>
<td>13.0</td>
<td>3.8%</td>
<td>Fork</td>
<td>149</td>
<td>480</td>
</tr>
<tr>
<td>Cynoscion nebulosus</td>
<td>55</td>
<td>1.0%</td>
<td>9.0</td>
<td>2.6%</td>
<td>Total</td>
<td>140</td>
<td>485</td>
</tr>
<tr>
<td>Menticirrhus americanus</td>
<td>53</td>
<td>0.9%</td>
<td>3.9</td>
<td>1.1%</td>
<td>Total</td>
<td>105</td>
<td>269</td>
</tr>
<tr>
<td>Chloroscombus chrysurus</td>
<td>43</td>
<td>0.7%</td>
<td>0.7</td>
<td>0.2%</td>
<td>Fork</td>
<td>63</td>
<td>158</td>
</tr>
<tr>
<td>Cynoscion arenarius</td>
<td>32</td>
<td>0.6%</td>
<td>3.1</td>
<td>0.9%</td>
<td>Total</td>
<td>106</td>
<td>296</td>
</tr>
<tr>
<td>Sciaenops ocellatus</td>
<td>10</td>
<td>0.2%</td>
<td>2.4</td>
<td>0.7%</td>
<td>Total</td>
<td>235</td>
<td>340</td>
</tr>
<tr>
<td>Oligoplites saurus</td>
<td>7</td>
<td>0.1%</td>
<td>0.1</td>
<td>0.0%</td>
<td>Fork</td>
<td>94</td>
<td>181</td>
</tr>
<tr>
<td>Caranx cryos</td>
<td>6</td>
<td>0.1%</td>
<td>0.3</td>
<td>0.1%</td>
<td>Fork</td>
<td>116</td>
<td>147</td>
</tr>
<tr>
<td>Menticirrhus saxatilis</td>
<td>6</td>
<td>0.1%</td>
<td>0.7</td>
<td>0.2%</td>
<td>Total</td>
<td>182</td>
<td>271</td>
</tr>
<tr>
<td>Orthopristis chrysoptera</td>
<td>6</td>
<td>0.1%</td>
<td>0.5</td>
<td>0.1%</td>
<td>Fork</td>
<td>137</td>
<td>199</td>
</tr>
<tr>
<td>Pogonias cromis</td>
<td>5</td>
<td>0.1%</td>
<td>0.7</td>
<td>0.2%</td>
<td>Fork</td>
<td>147</td>
<td>273</td>
</tr>
<tr>
<td>Rhizoprionodon terraenovae</td>
<td>5</td>
<td>0.1%</td>
<td>1.6</td>
<td>0.5%</td>
<td>Fork</td>
<td>320</td>
<td>360</td>
</tr>
<tr>
<td>Sardinella aurita</td>
<td>4</td>
<td>0.1%</td>
<td>0.1</td>
<td>0.0%</td>
<td>Fork</td>
<td>118</td>
<td>139</td>
</tr>
<tr>
<td>Anchoides hepsetus</td>
<td>3</td>
<td>0.1%</td>
<td>0.0</td>
<td>0.0%</td>
<td>Fork</td>
<td>107</td>
<td>—</td>
</tr>
<tr>
<td>Lutjanus griseus</td>
<td>3</td>
<td>0.1%</td>
<td>0.1</td>
<td>0.0%</td>
<td>Total</td>
<td>82</td>
<td>189</td>
</tr>
<tr>
<td>Rachycentron canadum</td>
<td>3</td>
<td>0.1%</td>
<td>0.3</td>
<td>0.1%</td>
<td>Fork</td>
<td>219</td>
<td>300</td>
</tr>
<tr>
<td>Trichiurus lepturus</td>
<td>3</td>
<td>0.1%</td>
<td>0.4</td>
<td>0.1%</td>
<td>Total</td>
<td>195</td>
<td>673</td>
</tr>
<tr>
<td>Archosargus probatocephalus</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>1.0</td>
<td>0.3%</td>
<td>Fork</td>
<td>370</td>
<td>—</td>
</tr>
<tr>
<td>Chaetodipterus faber</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.2</td>
<td>0.0%</td>
<td>Total</td>
<td>163</td>
<td>—</td>
</tr>
<tr>
<td>Citharichthys spiloterus</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.0</td>
<td>0.0%</td>
<td>Total</td>
<td>117</td>
<td>—</td>
</tr>
<tr>
<td>Echeneis naucrates</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.1</td>
<td>0.0%</td>
<td>Total</td>
<td>256</td>
<td>—</td>
</tr>
<tr>
<td>Eucinostomus gula</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.0</td>
<td>0.0%</td>
<td>Fork</td>
<td>97</td>
<td>—</td>
</tr>
<tr>
<td>Mycteroperca microlepis</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.1</td>
<td>0.0%</td>
<td>Total</td>
<td>197</td>
<td>—</td>
</tr>
<tr>
<td>Paralichthys lethostigma</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.5</td>
<td>0.1%</td>
<td>Total</td>
<td>337</td>
<td>—</td>
</tr>
<tr>
<td>Peprilus alepidotus</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>0.2</td>
<td>0.1%</td>
<td>Fork</td>
<td>174</td>
<td>—</td>
</tr>
<tr>
<td>Prionotus tribulus</td>
<td>1</td>
<td>&lt; 0.1%</td>
<td>&lt; 0.1</td>
<td>&lt;0.1%</td>
<td>Total</td>
<td>151</td>
<td>—</td>
</tr>
</tbody>
</table>

TOTAL 5776 344.1
Figure 27. Total length frequency (25-mm increments) for all spotted seatrout collected in 2011 and 2012.

Figure 28. Total length (mm) frequency by mesh size for all spotted seatrout collected in 2011 and 2012. Note that 1.3-cm mesh is not displayed for the one 126-mm interval and one 351-mm interval fish collected therein; also, one 476-mm interval fish not displayed for the 1.6-cm panel.
Figure 29. Mean (± SE) total length (mm) by mesh size for all spotted seatrout (top) and all spotted seatrout ≤ 275mm TL (bottom). Note that y-axis scale differs between the two graphs.
Table 7

*Descriptive Statistics for Spotted Seatrout TL (mm) Relative to Gill Net Mesh Size for All Spotted Seatrout (top) and Spotted Seatrout ≤ 275 mm TL (bottom)*

<table>
<thead>
<tr>
<th>Panel Size (cm)</th>
<th>n</th>
<th>Mean</th>
<th>S.E.</th>
<th>Min.</th>
<th>Max.</th>
</tr>
</thead>
<tbody>
<tr>
<td>1.3</td>
<td>2</td>
<td>246.0</td>
<td>108.0</td>
<td>138</td>
<td>354</td>
</tr>
<tr>
<td>1.6</td>
<td>16</td>
<td>222.6</td>
<td>21.5</td>
<td>146</td>
<td>485</td>
</tr>
<tr>
<td>1.9</td>
<td>10</td>
<td>233.6</td>
<td>15.1</td>
<td>183</td>
<td>322</td>
</tr>
<tr>
<td>2.5</td>
<td>27</td>
<td>273.0</td>
<td>3.7</td>
<td>240</td>
<td>337</td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>Panel Size (cm)</th>
<th>n</th>
<th>Mean</th>
<th>S.E.</th>
<th>Min.</th>
<th>Max.</th>
</tr>
</thead>
<tbody>
<tr>
<td>1.3</td>
<td>1</td>
<td>138</td>
<td>—</td>
<td>—</td>
<td>—</td>
</tr>
<tr>
<td>1.6</td>
<td>12</td>
<td>184.0</td>
<td>8.6</td>
<td>146</td>
<td>232</td>
</tr>
<tr>
<td>1.9</td>
<td>8</td>
<td>215.3</td>
<td>10.9</td>
<td>183</td>
<td>268</td>
</tr>
<tr>
<td>2.5</td>
<td>15</td>
<td>260.5</td>
<td>2.8</td>
<td>240</td>
<td>275</td>
</tr>
</tbody>
</table>

Habitat. However, NV habitat yielded the highest raw numbers of target-size fish in 2011 (i.e., not standardized by sampling effort) (Figure 34).

Considering all sizes of spotted seatrout collected, two-way ANOVA’s of log(CPUE) yielded significant habitat effects (Table 8), but CPUE responses were not statistically different between years (Figure 35). Overall, mean CPUE-number was significantly higher for SAV habitat than means for both ME (p = 0.043) and NV habitats (p = 0.003) (Figure 36), while no difference was found between ME and NV habitats (p = 0.609) for mean CPUE-number. For CPUE-biomass, the only significant
Figure 30. Annual spatial distribution of collection locations for all spotted seatrout (top) and spotted seatrout \( \leq 275 \) mm TL (bottom) collected in the survey area in 2011 (yellow) and 2012 (orange).
Figure 31. Spatial distribution (yellow) and total number by station for all spotted seatrout collected in the survey area in 2011 (top) and 2012 (bottom).
Figure 32. Spatial distribution (yellow) and total number by station for spotted seatrout ≤ 275 mm TL collected in the survey area in 2011 (top) and 2012 (bottom).
Figure 33. Total number (top) and total biomass (g, bottom) of spotted seatrout collected by habitat and year. Numbers represent total number of net sets made.
Figure 34. Total number of spotted seatrout ≤ 275 mm TL collected by habitat and year. Numbers represent total number of net sets made.

Table 8

Results of Two-way ANOVA Comparisons of Spotted Seatrout CPUE and Size Among Habitats and Between Years for All Spotted Seatrout Collected

<table>
<thead>
<tr>
<th>Dependent Variable</th>
<th>Habitat*Year df F p</th>
<th>Habitats Effects df F p</th>
<th>Years Effects df F p</th>
</tr>
</thead>
<tbody>
<tr>
<td>Log CPUE (number) All Fish</td>
<td>2 1.70 0.185</td>
<td>2 7.72 0.001</td>
<td>1 0.79 0.374</td>
</tr>
<tr>
<td>Log CPUE (biomass) All Fish</td>
<td>2 1.59 0.206</td>
<td>2 7.75 0.001</td>
<td>1 1.07 0.301</td>
</tr>
<tr>
<td>Log Total Length (mm) All Fish</td>
<td>2 3.86 0.028</td>
<td>2 1.03 0.365</td>
<td>1 0.09 0.766</td>
</tr>
</tbody>
</table>
Figure 35. Mean (± SE) CPUE (top) for number and biomass (g, bottom) between years for all spotted seatrout.
Figure 36. Mean (± SE) CPUE for number (top) and biomass (g, bottom) among habitats for all spotted seatrout collected in 2011 and 2012. Matching number annotations indicate a significant difference among/between habitat types based on comparisons of log-transformed data.
habitat treatment effect was evident in the pairwise comparison between SAV and NV habitats ($p = 0.003$) (SAV vs. ME, $p = 0.072$; ME vs. NV; $p = 0.414$). A significant habitat*year interaction was detected for log-transformed total length of all spotted seatrout collected (Table 8), the interaction of which can be attributed to a larger mean size in NV habitats in 2012 (Figure 37). Overall, neither independent main factor effect was significant for total length ($p > 0.379$).

Considering only late-juvenile spotted seatrout within the target size range, two-way ANOVA of log(CPUE) for spotted seatrout $\leq 275$ mm TL yielded a significant habitat effect (Table 9) but the lack of a significant difference between years (Figure 38). Overall, mean CPUE-number was again significantly higher for SAV habitats compared to both ME ($p = 0.020$) and NV habitats ($p = 0.031$) (Figure 39), whereas no difference was found between ME and NV habitats ($p = 0.917$). Similar results were

![Figure 37](image)

*Figure 37.* Mean annual total length among habitat types for all spotted seatrout collected. A significant habitat*year interaction was detected for log-transformed data, but no differences existed among/between the habitat and year treatments.
Table 9

*Results of Two-way ANOVA Comparisons of Spotted Seatrout CPUE and Size Among Habitats and Between Years for Spotted Seatrout ≤ 275 mm TL*

<table>
<thead>
<tr>
<th>Dependent Variable</th>
<th>Habitat*Year Effects</th>
<th>Habitats Effects</th>
<th>Years Effects</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>df  F  p</td>
<td>df  F  p</td>
<td>df  F  p</td>
</tr>
<tr>
<td>Log CPUE (number) Fish ≤ 275mm TL</td>
<td>2 1.80 0.167</td>
<td>2 6.10 <strong>0.003</strong></td>
<td>1 0.17 0.167</td>
</tr>
<tr>
<td>Log CPUE (biomass) Fish ≤ 275mm TL</td>
<td>2 2.08 0.127</td>
<td>2 5.68 <strong>0.004</strong></td>
<td>1 0.00 0.983</td>
</tr>
<tr>
<td>Log Total Length (mm) Fish ≤ 275mm TL</td>
<td>2 0.07 0.798</td>
<td>2 3.18 0.055</td>
<td>1 0.08 0.783</td>
</tr>
</tbody>
</table>

obtained for comparisons of CPUE-biomass among treatments, with juvenile seatrout biomass in SAV habitats exceeding both ME (p = 0.040) and NV habitats (p = 0.029), but with no difference in biomass between ME and NV habitats (p = 0.999). Total length of spotted seatrout ≤ 275 mm TL was marginally significant among habitats (p = 0.055), and fish from NV habitat were generally smaller in 2011 and were absent from 2012 collections (Figure 40).

Although the KMO value for the PCA of the habitat variables was less than ideal, a logistic regression of presence/absence of for all spotted seatrout relative to the PCA factors proved to be significant relative to all three components (p ≤ 0.020). Although generally informative, predictive power of the components was not surprisingly low (Nagelkerke $R^2 = 0.163$). For spotted seatrout ≤ 275 mm TL, the logistic regression between presence/absence and the PCA factors was significant only
Figure 38. Mean (± SE) CPUE (top) for number and biomass (g, bottom) between years for spotted seatrout ≤ 275 mm TL.
Figure 39. Mean (± SE) CPUE for number (top) and biomass (g, bottom) among habitats for spotted seatrout ≤ 275 mm TL collected in 2011 and 2012. Matching number annotations indicate a significant difference among/between habitat types based on comparisons of log-transformed data.
Figure 40. Mean annual total length among habitat types for spotted seatrout ≤ 275 mm TL.

for Factor I (p = 0.041) and even less predictive (Nagelkerke R^2 = 0.110) than when considering all sizes.

Comparisons of Bray-Curtis dissimilarity using ANOSIM revealed the lack of any difference in community structure between samples with and without spotted seatrout (R = 0.059; p = 0.892), and subsequent evaluation of MDS plots (3D stress = 0.16) confirmed the lack of any discernible visual pattern attributable to spotted seatrout presence/absence (Figure 41). The null relationship did not change when considering only the presence or absence of late juveniles (≤ 275 mm TL); again the ANOSIM test was non-significant (R = -0.021; p = 0.606) and MDS plots illustrated a null relationship between the two treatments in terms of community associations (Figure 42).
Figure 41. 3D MDS plot of fish community dissimilarity relative to spotted seatrout presence/absence (3D stress = 0.16). To aid in comprehensive interpretation, the bottom plot is a rotated view of the top plot with an approximate 180° revolution of the data points.
Figure 42. 3D MDS plot of fish community dissimilarity relative to the presence/absence of spotted seatrout ≤ 275 mm TL (3D stress = 0.16). To aid in comprehensive interpretation, the bottom plot is a rotated view of the top plot with an approximate 180° revolution of the data points.
For the overall comparison of fish community dissimilarity among the three habitat types, ANOSIM detected a significant treatment effect (R = 0.185; p = 0.001), and separation of samples based on habitat type was evident in 3-D MDS plots (Figure 43). All pairwise comparisons between habitats were also significant (R > 0.129; p = 0.001). SIMPER found NV vs. SAV habitat to be most dissimilar in community structure (\( \bar{X} \) dissimilarity = 76.96), followed by ME vs. NV (\( \bar{X} \) dissimilarity = 73.58) and ME vs. SAV habitat comparisons (\( \bar{X} \) dissimilarity = 70.73). For the SAV vs. NV comparison, community differences primarily reflected higher mean log(CPUE-biomass) of *Ariopsis felis*, *Lagodon rhomboides*, and *Hyporhampus meeki* in SAV, and higher means for *Micropogonias undulatus* and *Brevoortia patronus* in NV (Table 10). Those same species and trends also accounted for key differences in species dissimilarity for SAV vs. ME habitats, with the exception of *Elops saurus* which replaced *B. patronus* (NV) as the second highest mean log(CPUE-biomass) in ME. Differences between ME and NV fish communities were again driven predominantly by the more abundant species, with higher means in ME for *E. saurus* and *A. felis*, compared to higher means for *B. patronus* and *M. undulatus* in NV habitats.

**Discussion**

Numerous studies have identified SAV as being important for spawning, larval settlement and early juvenile habitat for spotted seatrout throughout the northern GoM and eastern U.S. coast. The significantly higher CPUE for late juveniles in SAV habitats of this study area suggests that the preference for SAV also extends beyond the early juvenile stage. Overall, mean CPUE in both number and biomass was statistically higher in SAV than NV habitats for all spotted seatrout collected and for all target-sized late-
Figure 43. 3D MDS plot of fish community dissimilarity among habitat types (3D stress = 0.16). To aid in comprehensive interpretation, the bottom plot is an inverted and rotated view of the top plot with an approximate 180° revolution of the data points.
Table 10

Pairwise Comparisons of Average Dissimilarities in Overall Fish Community Structure
Among Habitat Types Based on SIMPER Analysis

<table>
<thead>
<tr>
<th>Species</th>
<th>NV - ME</th>
<th>ME - NV</th>
<th>SAV - NV</th>
<th>X - SAV</th>
<th>Diss/SD</th>
<th>% Contr.</th>
<th>Cum. %</th>
</tr>
</thead>
<tbody>
<tr>
<td>Ariopsis felis</td>
<td>1.19</td>
<td>2.90</td>
<td>8.22</td>
<td>1.02</td>
<td>10.68</td>
<td>10.68</td>
<td></td>
</tr>
<tr>
<td>Micropogonias undulatus</td>
<td>3.02</td>
<td>1.93</td>
<td>7.85</td>
<td>0.99</td>
<td>10.20</td>
<td>20.88</td>
<td></td>
</tr>
<tr>
<td>Brevoortia patronus</td>
<td>2.40</td>
<td>1.50</td>
<td>7.22</td>
<td>1.03</td>
<td>9.38</td>
<td>30.36</td>
<td></td>
</tr>
<tr>
<td>Lagodon rhomboides</td>
<td>0.17</td>
<td>2.45</td>
<td>7.17</td>
<td>0.99</td>
<td>9.32</td>
<td>39.58</td>
<td></td>
</tr>
<tr>
<td>Hyporthampus meeki</td>
<td>0.42</td>
<td>2.32</td>
<td>6.83</td>
<td>1.07</td>
<td>8.88</td>
<td>48.46</td>
<td></td>
</tr>
<tr>
<td>Leiostomus xanthurus</td>
<td>1.70</td>
<td>1.19</td>
<td>5.52</td>
<td>0.93</td>
<td>7.51</td>
<td>55.64</td>
<td></td>
</tr>
<tr>
<td>Strongylura marina</td>
<td>0.15</td>
<td>1.15</td>
<td>4.92</td>
<td>0.58</td>
<td>6.26</td>
<td>61.89</td>
<td></td>
</tr>
<tr>
<td>Bairdiella chrysoura</td>
<td>0.68</td>
<td>1.26</td>
<td>4.19</td>
<td>0.88</td>
<td>5.44</td>
<td>67.34</td>
<td></td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>Species</th>
<th>NV - ME</th>
<th>ME - SAV</th>
<th>SAV - ME</th>
<th>X - ME</th>
<th>Diss/SD</th>
<th>% Contr.</th>
<th>Cum. %</th>
</tr>
</thead>
<tbody>
<tr>
<td>Ariopsis felis</td>
<td>1.82</td>
<td>2.90</td>
<td>6.91</td>
<td>1.09</td>
<td>9.77</td>
<td>9.77</td>
<td></td>
</tr>
<tr>
<td>Micropogonias undulatus</td>
<td>2.65</td>
<td>1.93</td>
<td>6.52</td>
<td>0.99</td>
<td>9.22</td>
<td>18.99</td>
<td></td>
</tr>
<tr>
<td>Elops saurus</td>
<td>2.44</td>
<td>1.07</td>
<td>6.48</td>
<td>0.96</td>
<td>9.16</td>
<td>28.15</td>
<td></td>
</tr>
<tr>
<td>Lagodon rhomboides</td>
<td>1.34</td>
<td>2.45</td>
<td>6.43</td>
<td>1.01</td>
<td>9.10</td>
<td>37.25</td>
<td></td>
</tr>
<tr>
<td>Hyporthampus meeki</td>
<td>0.83</td>
<td>2.32</td>
<td>5.81</td>
<td>1.10</td>
<td>8.21</td>
<td>45.45</td>
<td></td>
</tr>
<tr>
<td>Brevoortia patronus</td>
<td>1.55</td>
<td>1.50</td>
<td>5.32</td>
<td>0.96</td>
<td>7.52</td>
<td>52.98</td>
<td></td>
</tr>
<tr>
<td>Strongylura marina</td>
<td>1.13</td>
<td>1.15</td>
<td>4.77</td>
<td>0.81</td>
<td>6.74</td>
<td>59.72</td>
<td></td>
</tr>
<tr>
<td>Leiostomus xanthurus</td>
<td>1.41</td>
<td>1.19</td>
<td>4.62</td>
<td>0.92</td>
<td>6.35</td>
<td>66.24</td>
<td></td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>Species</th>
<th>ME - NV</th>
<th>NV - ME</th>
<th>SAV - ME</th>
<th>X - SAV</th>
<th>Diss/SD</th>
<th>% Contr.</th>
<th>Cum. %</th>
</tr>
</thead>
<tbody>
<tr>
<td>Elops saurus</td>
<td>2.44</td>
<td>0.80</td>
<td>7.59</td>
<td>0.92</td>
<td>10.32</td>
<td>10.32</td>
<td></td>
</tr>
<tr>
<td>Brevoortia patronus</td>
<td>1.55</td>
<td>2.40</td>
<td>7.43</td>
<td>1.01</td>
<td>10.09</td>
<td>20.41</td>
<td></td>
</tr>
<tr>
<td>Micropogonias undulatus</td>
<td>2.65</td>
<td>3.02</td>
<td>7.42</td>
<td>0.91</td>
<td>10.09</td>
<td>30.50</td>
<td></td>
</tr>
<tr>
<td>Ariopsis felis</td>
<td>1.82</td>
<td>1.19</td>
<td>6.05</td>
<td>0.97</td>
<td>8.23</td>
<td>38.73</td>
<td></td>
</tr>
<tr>
<td>Leiostomus xanthurus</td>
<td>1.41</td>
<td>1.70</td>
<td>5.80</td>
<td>0.98</td>
<td>7.88</td>
<td>46.60</td>
<td></td>
</tr>
<tr>
<td>Lagodon rhomboides</td>
<td>1.34</td>
<td>0.17</td>
<td>4.50</td>
<td>0.65</td>
<td>6.12</td>
<td>52.72</td>
<td></td>
</tr>
<tr>
<td>Strongylura marina</td>
<td>1.13</td>
<td>0.15</td>
<td>4.45</td>
<td>0.62</td>
<td>6.05</td>
<td>58.77</td>
<td></td>
</tr>
<tr>
<td>Mugil cephalus</td>
<td>1.21</td>
<td>0.29</td>
<td>4.32</td>
<td>0.56</td>
<td>5.87</td>
<td>64.63</td>
<td></td>
</tr>
</tbody>
</table>

Column headings: “X - ??” = habitat group means for log(CPUE-biomass); “X Diss” = mean dissimilarity across all pairs of samples; “Diss/SD” = mean dissimilarity / standard deviation; “% Contr” = percent contribution of each species to total dissimilarity; “Cum. %” = cumulative percentage dissimilarity
juveniles (≤ 275 mm TL) collected, while CPUE did not differ between years for any of the metrics assessed. The importance of SAV to early life history of spotted seatrout has been documented regionally in Gulf waters for post-settlement (Rooker et al. 1998) and early juveniles (< ~ 50 mm SL, Neahr et al. 2010) in Texas, YOY in Louisiana (Laska 1973; Thompson and Verret 1980), juveniles < 131 mm SL in Mississippi (Warren et al. 2001), and larvae and juveniles in Florida (McMichael and Peters 1989; Chester and Thayer 1990). Similar findings for the U.S. east coast by Powers (2012) noted juveniles 30-160 mm TL were positively associated with SAV in North Carolina and by Smith et al. (2008), who used trawls to collect spotted seatrout 20 - 140 mm SL from SAV habitats in Chesapeake Bay. Despite the relative wealth of knowledge on early stage juveniles, the present study is one of few, if not the only quantitative one, to assess habitat use by and relative abundance of late juveniles of this species. Results of this study support and expand upon prior findings of the preference for SAV by YOY spotted seatrout.

Marsh-edge is another important habitat for larvae and juveniles of numerous resident and transient estuarine fishes in north-central GoM waters, including spotted seatrout (Peterson et al. 2000). Peterson (1986) found shallow *Spartina* marsh-edge in Caminada Bay, Louisiana, to be a preferred habitat of juveniles < 125 mm TL, and Rakocinski et al. (1992) reported similar findings for spotted seatrout ≤ 100 mm SL in Barataria Bay. However, in the present study, late-juvenile CPUE was statistically higher in SAV than in ME habitats for three of the four related metrics (CPUE-number for all seatrout, CPUE-number and CPUE-biomass for seatrout ≤ 275 mm TL). No significant difference was detected between SAV and ME for CPUE-biomass for all
spotted seatrout ($p = 0.072$). The lack of a significant effect for that metric can be attributed to a higher variance for those particular biomass data. Aside from the general lack of SAV in Louisiana coastal waters, the apparent disparity between the two referenced Louisiana studies and the present study may be a function of differences in shoreline morphology, ontogenetic habitat shifts, or a combination of both. With regard to shoreline morphology, virtually all marsh habitats in the Point aux Chenes study area consisted of erosional marsh-edge, whereas the marsh habitats sampled in Louisiana almost certainly included depositional marsh-edge shorelines. McIvor and Odum (1988) found more nekton associated with depositional as opposed to erosional marsh-edge, as access to the vegetated marsh structure is physically and temporally more readily available for utilization in depositional areas. As such, the SAV-ME difference found in this study may not be directly comparable with prior findings which encompass the depositional marsh-edge. Regarding ontogenetic effects, in a study similar to the present one, MacRae (2006) examined habitat use by spotted seatrout among non-vegetated, marsh-edge and oyster shell habitats in Louisiana. She found larger juveniles (220-254 mm SL) to be more common, although not significantly different, at deeper oyster shell and non-vegetated mud sites than at marsh-edge locations. That finding suggests an ontogenetic shift by larger juveniles to deeper, non-shoreline habitats; a finding which is not supported by this study. Submerged aquatic vegetation, however, was not included as a habitat treatment in MacRae’s (2006) study. In the Point aux Chenes Bay area, relatively shallow SAV is preferred habitat for late juvenile spotted seatrout, particularly relative to the erosional marsh-edge found therein. Where SAV covers relatively less area or is lacking, the suitability of marsh-edge and other alternative habitats may be
higher albeit ultimately not as ecologically valuable as SAV, particularly with respect to erosional marsh-edge.

When scaled to habitat availability as determined from sidescan sonar within the study area, SAV appeared to be non-essential for late-juvenile spotted seatrout. Although the CPUE of late juveniles was highest in SAV ($\bar{X} = 0.071$), SAV was limited in spatial coverage within the study area. Based on calculations of areal cover derived from the sonar-based imagery, NV habitats accounted for 92% of the survey area (4.66 km$^2$) excluding NV substrata adjacent to marsh-edge, whereas SAV (0.18 km$^2$) and ME (0.20 km$^2$) each comprised less than 5% of the total area from which juveniles were sampled. Thus within this particular bay, habitat utilization scaled to habitat availability would actually be highest for the more widespread NV habitat, because its low mean CPUE ($\bar{X} = 0.019$) would be extrapolated according to the disproportionately high amount of available NV habitat. At the wider population level, habitat use relative to habitat availability would likely be even more skewed toward NV habitat (and emergent marsh vegetation) given the general lack of SAV in other turbid bay systems along the Mississippi coastline. Although SAV may be preferred where available, as shown in this study, its large-scale absence in nearshore Mississippi coastal waters indicates that SAV is probably not essential to the regional spotted seatrout population, and its true value will not be known until survival and growth benefits are completely evaluated for this spatially limited habitat.

Certain habitat variables are often identified as positive correlates with fish abundance, and MacRae (2006) found significant relationships between spotted seatrout CPUE and both temperature and salinity in her study. Similarly, Herke et al. (1984)
identified high-salinity regime marsh as important habitat for juveniles of this species. The findings of the present study, however, generally concur with those of Chester and Thayer (1990), who found salinity was not a contributing factor to juvenile seatrout distribution in Florida Bay. A more limited range of relatively homogeneous salinities in that study was likely consistent with salinities in Point aux Chenes Bay. By contrast, Louisiana estuaries are subject to greater freshwater discharge and thus more heterogeneous salinity trends. In the present study, the seven habitat variables were reduced to three PCA factors explaining 65.5% of the variability in all habitat data; however, the logistic relationships between PCA-reduced habitat factors and presence/absence of late juvenile seatrout were not predictive (Nagelkerke $R^2 = 0.163$ and 0.110) but were significant. Relative to typical bay systems of Louisiana and Mississippi, the salinity of Point aux Chenes Bay would be considered more saline, homogeneous and stable, primarily because there is less riverine input into this system than in bay systems to the west. As such, the lack of a predictive model of seatrout abundance based on the PCA habitat factors is not surprising for this particular bay system. Additionally, spotted seatrout presence/absence was not associated with overall fish community structure, wherein habitat-specific differences were driven primarily by $A. felis$, $M. undulatus$, $L. rhomboides$, $E. saurus$, $B. patronus$ and $H. meeki$. Taken as a whole, these findings suggest structural habitat properties are more important ecological correlates for juvenile seatrout in this system.

Among-habitat physical-chemical differences were detected only for water depth, turbidity and tide height, the first two of which were positively related and the latter of which was largely a function of sampling logistics. In the survey area, SAV
occurred at depths < 1.3 m (Cho and May 2008) and marsh-edge was also shallow; non-vegetated bottom was predominant throughout all depths in the survey area. Turbidity is typically higher at shallow depths due to greater disturbance of bottom sediments in higher wave/wind conditions, so significant differences of inverse magnitudes of those two variables among habitats in not surprising. With regard to tide height, which was based on time of day, the occurrence of SAV in shallow waters necessitated the sampling of SAV stations during rising, high or falling tidal conditions, in order to deploy the net properly.

The relatively small-mesh experimental gill net applied in this study was effective at collecting target sizes of late juvenile spotted seatrout that have been largely lacking from other collections of smaller juveniles (Warren et al. 2001) and from collections of larger adults of the species (Hendon et al. 2012). Relatively high numbers of larger fish were also collected, but large fish were generally not gilled; instead, they became entangled by the upper jaw canine teeth or one side of the maxilla. Exclusion of such large entangled fish from selectivity analyses would be warranted as they were not technically caught and secondary analyses were used to exclude those individuals from habitat and year comparisons. While epibenthic sleds (e.g., Neahr et al. 2010), seines (e.g., McMichael and Peters 1989) and trawls (e.g., Powell et al. 2004) effectively sample early juvenile spotted seatrout, and larger mesh gill nets are commonly used for monitoring and assessment of adults of the species (GSMFC 2001; Hendon et al. 2012), efficient quantitative sampling of the historically elusive late juvenile spotted seatrout has proven problematic. Application of the experimental gill net used in this study is recommended for future research efforts to better quantify ecological characteristics of
late-juvenile spotted seatrout and other important species and to assess post-release survival, movements and abundance of hatchery-reared juveniles released into the wild for stock enhancement purposes.

Overall, results from this study support prior findings that identify SAV as a preferred habitat, where available, for spotted seatrout throughout its larval and juvenile stages. However, the contribution of SAV in areas like Grand Bay where seagrass coverage is limited requires a more detailed study of growth and survival to better define population-level habitat value. The abundance of late-juveniles in significantly higher numbers in SAV than in erosional marsh-edge and non-vegetated habitats expands on the knowledge base of the relative importance of SAV to the life history of this species, as fish in the 125 to 275 mm TL size range are largely absent from prior studies evaluating habitat use. Extending the relevance of SAV to the late-juvenile stage of spotted seatrout further underscores the long-standing need for more comprehensive mapping efforts to identify, delineate, and protect ecologically beneficial habitats, particularly those like SAV whose spatial coverage can fluctuate at varying temporal scales (Moncrieff et al. 1998; Carter et al. 2011). Toward that end, studies of habitat use which are predicated on quantitative surveys of available habitat make habitat assessments even more insightful, as was done using sidescan sonar prior to sampling of spotted seatrout in this study. With regard to stock enhancement applications, future enhancement practices for Mississippi’s spotted seatrout population should further investigate the use of SAV as a release habitat for hatchery-reared fish, as the higher relative abundances of wild juveniles in SAV may be indicative of favorable ecological conditions that could promote post-release survival of hatchery-reared individuals.
LITERATURE CITED


Powers JP. 2012. Distribution patterns of juvenile spotted seatrout (Cynoscion nebulosus) and red drum (Sciaenops ocellatus) along shallow beach habitats in Pamlico River, North Carolina [thesis]. [Greenville, (NC)]: East Carolina University.


CHAPTER IV
HABITAT-SPECIFIC GROWTH AND FEEDING OF HATCHERY-REARED
JUVENILE SPOTTED SEATROUT IN A MISSISSIPPI BAY SYSTEM

Introduction

The spotted seatrout \( (Cynoscion nebulosus) \) is one of the most important recreational and commercial fisheries species in the northern Gulf of Mexico (GoM) and is the most targeted recreational finfish in Mississippi coastal waters (Deegan 1990). Fishery landing estimates for 2010 through 2012 indicate that spotted seatrout was the most heavily exploited recreational saltwater species in Mississippi waters, averaging 447,307 kg of landed weight annually during that period (NMFS 2013). Given the importance of the fishery, long-term monitoring of adults was implemented in Mississippi coastal waters to provide annual population demographic data (most recently summarized in Hendon et al. 2012). The latest assessment of those data suggested more conservative management actions were needed to ensure sufficient recruitment in the face of the heightened directed fishing effort on the species (Fulford and Hendon 2010). Recently, stock enhancement has also been explored as a means to supplement overly-exploited fish populations in conjunction with traditional management efforts and habitat restoration (Lorenzen 2008). Accordingly, a state-collaborative stock enhancement program, the Seatrout Population Enhancement Cooperative (SPEC), was created at the University of Southern Mississippi’s Gulf Coast Research Laboratory (USM-GCRL) to develop procedures for enhancing Mississippi’s heavily exploited spotted seatrout population through the release of cultured fish into the wild.
The ultimate goal of stock enhancement is to increase population sizes of overly exploited species. In order to successfully achieve that goal, two primary underlying conditions must be achieved: (1) survival of hatchery-reared (HR) fish in the wild, and (2) non-displacement of wild fish by stocked individuals (Leber et al. 1995; Huntingford 2004). Three separate and necessary elements are associated with survival of HR fish: (1) acclimation to natural environmental conditions, (2) transition to feeding on novel wild prey, and (3) learning to avoid predators. If differences exist between HR and wild seatrout in any of those outcomes, it could undermine the success of the enhancement program and potentially result in failure if not rectified through alternative culture and/or release strategies. Responses of newly released HR fish to natural conditions should be evaluated through post-release monitoring/modeling and directed field-based experiments to assess post-release survival behaviors (Blankenship and Leber 1995; Leber et al. 1995; Walters and Martell 2004; Lorenzen 2006; Hervas et al. 2010). Additionally, information gained through such efforts can be used to optimize post-release survival of HR fish, as well as to determine and implement appropriate release strategies (Munroe and Bell 1997; Mahnken et al. 2004) relative to optimal release size (Leber 1995), timing of release (Leber et al. 1997), pre-release conditioning of fish (Brennan et al. 2006; Brown et al. 2013), and suitable release habitat (Stunz and Minello 2001; Andersen et al. 2005). Determination of advantageous release habitat is the focus of the present study. Selection of habitat types for this field experiment was informed by habitat mapping and fish survey results described in Chapters II and III.

Growth during the early life stages of fishes is a critical factor in mediating survival. Rapid growth generally equates to enhanced predator avoidance capabilities,
better tolerance of environmental stress and a competitive advantage in feeding success (Houde 1987; Sogard 1992; Piet et al. 1998), each of which can mitigate mortality rate (Sogard, 1997; Rooker et al. 1999). Higher quality estuarine habitats are presumed to promote faster growth in fishes through some combination of abundant food resources, available refuge from predators, and favorable physical conditions (Meng et al. 2000). Consequently, growth rate may be a good proxy indicator of habitat quality (Sogard, 1992; Meng et al. 2000; Necaise et al. 2005; Shervette and Gelwick 2007). Structural habitat appears to be ecologically important for the successful recruitment of spotted seatrout. In Florida Bay, Florida, Hettler (1989) reported seagrass to be the most valuable feeding habitat for juvenile spotted seatrout. Likewise, Peebles and Tolley (1988) suggested that low larval mortality in Fakahatchee Bay, Florida, could be attributed to the greater extent of seagrass cover relative to Naples Bay. In Louisiana, where SAV habitat is limited, the most rapid growth of small juvenile spotted seatrout was observed from habitats with organic detrital sediments (Baltz et al. 1998).

Abundance of late-juvenile spotted seatrout in Mississippi was significantly higher in SAV when compared to marsh-edge and non-vegetated habitats in the Grand Bay National Estuarine Research Reserve, suggesting that SAV likely provides valuable habitat in that area. Shervette and Gelwick (2007) found juvenile pinfish (*Lagodon rhomboides*) in the same bay system grew significantly faster in vegetated (emergent marsh) habitat than in oyster shell and non-vegetated habitats, but they did not evaluate the importance of SAV to juvenile growth.

Given the need to evaluate the effectiveness of current stock enhancement endeavors and considering the large gap in general knowledge on juvenile spotted
seatrout ecology in Mississippi coastal waters and elsewhere, the purpose of this study was to assess overall and habitat-specific growth and diet of HR spotted seatrout within a shallow Mississippi bay system that likely serves as a source of recruitment (Comyns et al. 2008). The null hypothesis that growth of HR fish does not differ among habitat types was tested for SAV and non-vegetated habitats delineated from side-scan mapping. Diets of HR fish enclosed within selected habitat types were also compared with diets of wild-caught juveniles from the same area to evaluate if prey use coincided between HR and wild fish.

Materials and Methods

Cage Design & Study Area

Enclosure experiments were conducted within Grand Bay and Point aux Chenes Bay (Figure 44). The latter area contains the only extensive expanse of SAV remaining along the Mississippi coastal mainland (Moncreiff 2007). Both bay systems were characterized by relatively high salinity regimes and extensive seagrass beds of *Ruppia maritima* and *Halodule wrightii* (Cho and May 2008). The Grand Bay area, however, was subject to higher wave energy because of its proximity to the shoreline and prevailing winds (Cho 2007).

For the enclosure studies, 1.8 m (diameter) by 1.2 m (height) circular cages were constructed of 1.9 cm square plastic mesh netting supported by 2.5 cm diameter PVC piping and plastic tubing. A pilot growth experiment in 2011 used cages with no bottom panels to allow fish to interact freely with the benthic substrata. Plastic sleeves attached to the bottom circumference of the cages were pushed into the sediment to prevent fish from escaping and preclude predators from the cages. Twenty cages were constructed
Figure 44. Map of cage study areas (hashed polygons) in Point aux Chenes Bay, Mississippi, and Grand Bay, Alabama, USA. Inset shows general survey location (star) relative to the northeastern Gulf of Mexico.

For the pilot trial, half of which were allocated to each bay system.

For the pilot experiment, five cages per bay were randomly sited within SAV habitat, and the other five per bay were placed in adjacent non-vegetated bottom habitat. Cages were secured by attaching them to 10 cm diameter PVC poles driven approximately 1.3 m into the substrate. Five fish were introduced into each cage and held for 14 d, a period estimated to be long enough for fish to adjust to feeding in the wild. The 14 d experimental period provided a balance between allowing sufficient time for the commencement of feeding and successful retrieval of live fish. Survival could not be accurately checked during the experiment as the cage lacked a bottom panel. One hundred fish were introduced into the cages at the start of the initial experiment, but
only 21 were retrieved after the 14 d period. In Point aux Chenes Bay, 11 of the original 50 fish were collected, and large predatory blue crabs (*Callinectes sapidus*) were discovered in all but one of the ten cages. In Grand Bay, only 10 of the 50 fish were retrieved as most fish apparently escaped from the cages after the enclosures became partially dislodged from the sediment, presumably as a result of the higher wave action in that area. Two of the ten cages could not be located. Apparent predation by blue crabs and escapement from dislodged cages yielded too few fish to allow for meaningful comparisons from the 2011 pilot experiment.

In light of retention and predation issues, bottom panels of the same mesh material as the tops and sides were installed in all cages for the main experiment in 2012. This cage modification allowed for intermittent monitoring of mortality and replacement of missing individuals with placeholder HR fish over the duration of the experiment. It was assumed that the relatively large mesh size of the cage material (1.9 cm² square opening) would allow *R. maritima* and *H. wrightii* stems to enter the cage through the bottom mesh. However, *in situ* preliminary trials with flat mesh panels revealed that virtually all of the SAV was depressed by the mesh material, due to the flaccid nature of the relatively long SAV stems. To compensate for the effect of the bottom mesh panel on the physical structure of the natural SAV habitat and to standardize the habitat structure effect, artificial seagrass consisting of green polypropylene ribbon was fixed to the interior bottom panels of each SAV cage. The stem height and surface area of artificial seagrass used in each cage were based on measurements of stem height and total stem width obtained from random samples of SAV collected from the study area. Because artificial seagrass did not have the same
width dimensions as that of naturally-occurring SAV, the amount of artificial seagrass applied to each cage was based on stem surface area (rather than stem density) in order to provide amounts of artificial stem surface area consistent with that of natural SAV on a per unit area of water bottom. Final metrics for the amount of artificial seagrass allocated to each cage equated to a mean stem height of 275.8 mm (range of natural SAV samples = 238-293 mm) and a mean total stem width of 75.3 mm (range of natural SAV samples = 59-93 mm) per 6.4 cm$^2$ of bottom area.

To allow sufficient time for artificial seagrass to become conditioned and colonized by macrofauna, SAV cages were randomly deployed within each of the two SAV areas in Grand Bay and Point aux Chenes Bay on 19 July 2012, 56 d prior to the addition of experimental fish. Cages were placed fully within the boundaries of an SAV patch. All five SAV cages in the higher-energy Grand Bay area and two non-vegetated cages in Point aux Chenes Bay were dislodged and lost, some as a result of Tropical Storm Isaac. Consequently, it was decided to focus the experiment solely within the lower wave energy area of northeastern Point aux Chenes Bay, despite pseudoreplication concerns based on deploying within a single study area. Cages with artificial SAV in Point aux Chenes Bay had remained undisturbed since their initial deployment, and the availability of extra cages allowed the extension of the experiment to an additional habitat type. The remaining eight cages were allocated between non-vegetated shoreline (n=4, “NVS”) and non-vegetated open water (n=4, “NVO”) treatments within the study area (Figure 45). Due to low tidal level concerns, depositional marsh-edge was not a viable treatment option for this growth study. The NVS habitat was adjacent to erosional marsh-edge in the study area with cage locations
Figure 45. Experimental cage locations by habitat type in northeastern Point aux Chenes Bay relative to 2012 SAV spatial coverage. (Image: Google Earth Pro, version 7.0)

comparable to the marsh-edge (“ME”) treatment sites sampled in Chapter III.

Experimental Fish

Late juvenile spotted seatrout reared as part of the SPEC program were used to assess survival and growth within the selected habitats. Fish ranged in size from 144 to 188 mm TL and thus allowed for the use of relatively uniform sizes of fish for the growth trial. On 14 August 2012, 190 juveniles were anesthetized in an aerated tank treated with MS-222, and a uniquely encoded Biomark© Passive Integrated Transponder (PIT) tag (12mm long, 134.2-kHz frequency) was injected into the abdominal cavity of
each fish, just beneath the epidermis. The use of PIT tags enabled tracking of individual growth rates rather than having to rely on overall growth per enclosure. Tagged fish were placed in a separate holding tank to assess tag retention. Prior to preparing fish for release into field enclosures, tank conditions at the SPEC facility were matched to field conditions by gradually adjusting water temperature and salinity over a three-day period in order to acclimate fish to the appropriate physio-chemical conditions.

One month after tagging HR fish (on 13 September 2012), juveniles were removed from the holding tank, anesthetized and scanned with a Biomark 601 hand-held reader to ensure tag retention and to determine the unique PIT tag codes for experimental fish. Each fish was weighed (wet weight) to the nearest 0.1g, measured [standard length (SL)] to the nearest mm, and randomly allocated to a numbered enclosure. Fish were placed in buckets lined with plastic fish-transport bags that were filled with tank seawater and labeled with the appropriate cage number. Oxygen was injected into the bags prior to sealing them, and the fish were transported to the study sites in the bag-lined buckets. Remaining PIT-tagged fish were left in the designated holding tank at the SPEC facility to serve as replacements for cage mortalities (fish that died in cages during the experimental period). Estimates of natural fish densities have not been calculated for juvenile spotted seatrout in Mississippi waters. Five fish per cage (cage area = 2.5 m²) were used to ensure sufficient sample sizes for subsequent analyses, in anticipation of some expected mortality during the experiment. Cage density was, however, likely higher than that for fish under free-ranging natural conditions based on gill net collections of late juveniles (Chapter III) that generally yielded one or two fish per set of the 122 m long net.
Cages were checked weekly for mortalities by gently lifting each cage up to a point where the bottom was just below the water surface. Any dead fish found in the cage were removed, and counts were made of the number of live fish remaining. All dead or missing fish were replaced with placeholder HR fish to maintain equitable densities of five fish per cage. After a four-week period (12 October 2012), all fish were recovered from the cages, anesthetized and immediately taken to shore, where they were scanned, weighed and measured. All fish were then placed in labeled bags on ice and subsequently frozen for diet analysis.

**Stomach Content Analyses**

Experimental fish were later thawed and scanned to identify the unique tag number relating each fish to the appropriate post-experiment (pre-freezing) weight and length data. Stomachs from all HR fish were removed and the contents examined to determine abundance and diversity of prey items to the lowest possible taxonomic level. Additionally, archived stomachs of 29 wild-caught late-juvenile spotted seatrout collected from the same bay system and associated habitats (SAV, n = 20; NVS, n = 1; NVO, n = 8) were examined. Where possible, prey types were enumerated, and relative volumetric contributions based on water displacement were measured as percent volume for each prey type (%V, in mL); total weights of stomach contents [wet weight (WW), g] were also taken for determination of stomach fullness. For many fish, only unidentified remains and miscellaneous prey parts were present in stomachs, thus prey-type enumeration was based on the presence of identifiable pieces, such as telsons, carapaces, heads and/or eyes. For stomachs that contained only unidentifiable remains due to digestion, a total number of one prey item for the diet of that individual was
assigned for subsequent analytical purposes even though accurate counts were not possible. As a result, prey counts may be underestimations of actual quantities.

Statistical Analyses

Possible differential environmental effects on fish growth among habitats were considered by comparing weekly means for salinity, temperature (°C), dissolved oxygen (mg/L) and turbidity (NTU) among treatments with a one-way ANOVA. Additionally, to obviate any potential effect of differential natural exposure time on growth, only fish that survived for the entire duration of the experiment were used for growth and feeding comparisons. For purposes of evaluating survival, it was assumed that all fish missing from cages had died and decomposed rather than escaped.

Change in wet weight was used as the dependent variable for habitat comparisons of growth since it is considered to be a more sensitive indicator than length for short-term growth studies (Weatherly 1972; Able et al. 1999; Necaise et al. 2005; Lanier and Scharf 2007). Individual fish growth was calculated as the specific (instantaneous) growth rate (G):

$$ G = \frac{\log_{10} W_f - \log_{10} W_i}{t} \times 100 $$

where $W_f$ was final fish wet weight, $W_i$ was initial fish wet weight, and $t$ was the experiment duration in days (Ricker 1975). For making comparisons to other studies for which *C. nebulosus* growth rates in SL have been inferred from otolith increments, absolute daily growth rate (ADG) [mm (SL) d\(^{-1}\)] was also computed as

$$ ADG = \frac{(SL_f - SL_i)}{t} $$
where $SL_f$ was final standard length (mm), $SL_i$ was initial standard length, and $t$ was the experiment duration in days.

In order to test (and control) for potential cage effects and for differential initial fish size, a Nested ANCOVA was used to compare $G$ among habitat types, including initial weight as the covariate. Standard assumptions (normality; homogeneity of variance) were tested prior to the comparison, and the ANCOVA assumption of homogeneity of slopes was also tested by checking for a significant interaction between the covariate (initial weight) and the habitat factor within the hierarchical model. If the interaction was not significant (i.e., $p > 0.05$), growth was compared across habitat treatments using a linear mixed model procedure in SPSS to fit the model with $G$ as the dependent variable. The cage effect was the nested term of cage nested within habitat (fixed factor), habitat was the main factor (fixed), and initial fish weight was the covariate. In the event of an overall difference ($p < 0.05$) among treatments, *a posteriori* pairwise comparisons of estimated marginal means were made using the SIDAK adjustment to account for multiple comparisons. This procedure was repeated to compare $G$ based on SL among the three habitat treatments, using initial SL as the covariate.

To determine if fish sample sizes were sufficient to adequately describe their diets, cumulative prey curves were constructed for the fish groups (Ferry et al. 1997). The order in which diet samples were evaluated was first randomized and the number of total unique prey categories plotted as a function of sample size. The presence of an asymptote in a cumulative prey curve would indicate that the sample size was sufficient for characterizing the diet richness.
For all wild-caught late juveniles and experimental HR fish examined, standard diet metrics of percent frequency of occurrence (%F), percent number (%N) and percent volume (%V) were calculated for all prey types in stomachs that contained prey (Hyslop 1980), and the Index of Relative Abundance (IRI) (Pinkas et al. 1971) was calculated for major taxonomic categories as

\[
IRI = (%N + %V) \times %F
\]

in order to obtain a well-rounded composite diet index incorporating the three different prey metrics (Hyslop 1980). A stomach fullness index (FI) was also calculated as

\[
FI = \frac{S_i}{W_i} \times 100
\]

where \(S_i\) was the wet weight of the stomach contents (g) and \(W_i\) was the wet somatic weight of the fish (g). Because data for both groups were still not normal after data transformation, FI was compared among habitats using non-parametric Kruskal-Wallis tests. Stomach contents of HR fish ranging from 144 to 188 mm TL were compared to all wild late juveniles and to those of wild fish < 200 mm TL. For descriptive purposes, diets of wild fish were also considered over the entire late juvenile size range (125-275 mm TL), and frequency of occurrence of prey was also compared among habitat types.

Analyses were conducted with IBM SPSS Statistics, version 20.0, (IBM 2011) and all tests were considered significant at \(p < 0.05\). Where presented, means are accompanied by \(\pm\) one standard error.

Results

Of the original 65 HR fish released into cages on 13 September 2012, 45 survived the duration of the experiment which concluded on 12 October 2012. Retention of the injected PIT tags was high in experimental fish: 119 of the 124 tank-maintained
fish (96.0%) retained their tags over the one-month holding period. Tag retention was 100% for fish that survived the field experiment. Total percent survival within the confines of the cages was lowest for the first week of the experiment (73.8%), after which survival leveled off and remained relatively stable (week-4 mean total mortality = 69.2%). Cumulative survival ranged from 20% to 100% among individual cage replicates at the end of the experiment (Table 11).

Physical conditions did not vary significantly among habitat types over the experimental period, suggesting similar conditions prevailed among treatments throughout the study period. Differences in weekly grand means were not significant for surface water temperature ($F_{df=2} = 0.374; p = 0.698$) (Figure 46), salinity ($F_{df=2} = 0.008; p = 0.992$) (Figure 47), dissolved oxygen ($F_{df=2} = 0.942; p = 0.425$) (Figure 48), or turbidity ($F_{df=2} = 0.367; p = 0.702$) (Figure 49).

Table 11

<table>
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Figure 46. Mean (± SE) weekly surface temperature (°C) among habitats for the 29 d experimental period.

Figure 47. Mean (± SE) weekly surface salinity (ppt) among habitats for the 29 d experimental period.
Figure 48. Mean (± SE) weekly dissolved oxygen (mg/L) among habitats for the 29 d experimental period.

Figure 49. Mean (± SE) weekly turbidity (NTU) among habitats for the 29 d experimental period.
**Growth Analyses**

Overall, all but five HR fish lost weight in absolute terms (gross change in weight), and one of those five showed no change in weight. Weight change values ranged from -15.4 g to 4.4 g. Fish from SAV habitat lost the least weight (-2.55 g ± 0.769), followed by NVS (-3.13 g ± 0.696) and NVO habitats (-6.34 g ± 1.095) (Table 12a). Fish from SAV generally lost half as much weight as fish from NVO during the experimental period. Mean $G$ (specific growth) followed the same relative trend as absolute growth among the habitat types. Overall, $G$-values ranged from -0.457 to +0.183 (Table 12b).

For comparison of $G$ among habitats, the covariate (initial weight) had a significant effect on $G$ ($F_{df=1} = 6.664; p = 0.015$) as did the cage nested within habitat effect ($F_{df=10} = 2.548; p = 0.022$). Moreover, a significant overall effect was also found.

Table 12

**Descriptive Statistics for Total and Habitat-Specific Absolute Weight-based Growth (a) and Weight-based Specific Growth Rates (b)**

<table>
<thead>
<tr>
<th>(a) Absolute Growth</th>
<th>N</th>
<th>Minimum</th>
<th>Maximum</th>
<th>Mean</th>
<th>S.E.</th>
</tr>
</thead>
<tbody>
<tr>
<td>Total</td>
<td>45</td>
<td>-15.4</td>
<td>4.4</td>
<td>-4.06</td>
<td>0.571</td>
</tr>
<tr>
<td>SAV</td>
<td>17</td>
<td>-10.2</td>
<td>4.4</td>
<td>-2.55</td>
<td>0.769</td>
</tr>
<tr>
<td>NV-Shoreline</td>
<td>12</td>
<td>-7.5</td>
<td>1.0</td>
<td>-3.13</td>
<td>0.696</td>
</tr>
<tr>
<td>NV-Open Water</td>
<td>16</td>
<td>-15.4</td>
<td>0.4</td>
<td>-6.34</td>
<td>1.095</td>
</tr>
</tbody>
</table>

<table>
<thead>
<tr>
<th>(b) Specific Growth Rate</th>
<th>N</th>
<th>Minimum</th>
<th>Maximum</th>
<th>Mean</th>
<th>S.E.</th>
</tr>
</thead>
<tbody>
<tr>
<td>Total</td>
<td>45</td>
<td>-0.457</td>
<td>0.183</td>
<td>-0.1621</td>
<td>0.0197</td>
</tr>
<tr>
<td>SAV</td>
<td>17</td>
<td>-0.321</td>
<td>0.183</td>
<td>-0.1037</td>
<td>0.0283</td>
</tr>
<tr>
<td>NV-Shoreline</td>
<td>12</td>
<td>-0.301</td>
<td>0.054</td>
<td>-0.1309</td>
<td>0.0274</td>
</tr>
<tr>
<td>NV-Open Water</td>
<td>16</td>
<td>-0.457</td>
<td>0.023</td>
<td>-0.2475</td>
<td>0.0332</td>
</tr>
</tbody>
</table>
among habitat treatments after controlling for initial size and cage effects ($F_{df=2} = 10.326; p < 0.001$). Tests revealed no assumption violations, as data were normally distributed (Shapiro-Wilk test, $p = 0.988$) and variances were not statistically different across groups (Levene’s test, $p = 0.653$). In addition, the treatment by covariate interaction (habitat * initial weight) was not significant ($p = 0.198$) within the context of the full model, indicating homogeneity of regression slopes among habitat types. Pairwise post hoc comparisons of estimated marginal means revealed $G$ to be significantly greater in SAV compared to NVO habitat ($p < 0.001$) and in NVS versus NVO ($p = 0.013$), whereas the difference in $G$ between SAV and NVS was not significant ($p = 0.627$) (Figure 50). Comparison of mean residuals of $G$ by cage from the $G$ by initial weight relationship illustrates the relative cage-specific growth differences for each habitat (Figure 51), wherein $G$ was more frequently greater relative to mean initial weight for SAV and NVS habitats.

Regarding changes in absolute length, overall growth in length for all fishes pooled averaged $+4.00 \pm 0.392$ mm for the 29-day period, and changes in length among individual fish ranged from a low of -1 mm to a high of +12 mm in total SL. Habitat-specific means were $+4.53 \pm 0.743$ mm for SAV, $+3.50 \pm 0.452$ mm for NVS, and $+3.81 \pm 0.703$ mm for NVO. When standardized by time, overall mean ADG was $+0.14 \pm 0.014$ mm d$^{-1}$, and ADG ranged from -0.03 to +0.31. Mean ADG was highest in SAV ($+0.16 \pm 0.026$), followed by NVO ($+0.13 \pm 0.024$) and NVS ($+0.12 \pm 0.016$) habitats. No significant effects were detected within the linear mixed model for length-based comparisons of $G$ among habitats for the habitat treatment ($p = 0.116$), cage nested within habitat ($p = 0.451$), or the covariate, initial SL ($p = 0.823$). All appropriate test
Figure 50. Estimated marginal mean (± SE) specific growth rates (G) among habitats for hatchery-reared fish surviving the duration of the experiment. Matching number annotations indicate a significant difference in pairwise comparisons.

assumptions were satisfied prior to analysis.

Diet Analyses

Of the 45 HR fish that survived the entire 29d experiment period, 22 (49%) contained prey material, whereas 22 of the 29 (76%) wild fish in the 125 to 275 mm TL size range contained prey. Inspection of the cumulative prey curves for wild and HR fish indicated sample sizes were insufficient for adequately representing entire ranges of prey types within diets (asymptotic relationship not achieved for any species-area curve) (Figure 52). As such, diet results should be regarded as descriptive of presumed major diet constituents.

Teleost fish, crabs and shrimp (or the remains thereof) occurred in the diets of both experimental and wild fish groups, but the state of digestion of prey in HR fish
precluded definitive identifications of shrimp items and all but one fish (Table 13). Ten taxa were identified to the family level in the diets of wild-caught late juveniles (125-275 mm TL). Items assumed to be incidental to the feeding process were found in the stomachs of both groups, including SAV, detritus and mud in wild fish, and mud in HR fish. No SAV was found in the stomachs of fish collected from non-SAV habitat.

Overall, IRI values were relatively similar between the wild and experimental groups for the generic teleost and crab prey types, but shrimp were more prevalent in late-juvenile wild fish than HR individuals (Figure 53). Mean fullness (FI) ranks were highly variable and marginally different among habitats for wild fish ($\chi^2 = 5.752; p = 0.056$) (Figure 54), which partly reflects the low power associated with the small sample sizes, but
Figure 52. Cumulative species area (prey) curves for wild juvenile spotted seatrout < 276 mm TL, wild juveniles < 201 mm TL and HR spotted seatrout; sample order was randomized prior to accumulating prey types across the sample.

Mean fullness was relatively consistent across habitats for HR fish ($\chi^2 = 1.134; p = 0.567$).

In order to restrict the diet comparison to similar size classes, only wild fish < 200 mm TL (n = 11) were evaluated, of which eight (73%) had items in their stomachs (Table 14). Exclusion of the large wild juveniles eliminated crabs (or associated remains) from the diets of wild fish and also virtually excluded shrimp as a food item. Excluding large juveniles also heightened the importance of teleosts in the diet of comparable sized wild juvenile seatrout (Figure 55). Consequently, IRI values were highest for teleost fish for both groups when comparing similar sizes, notwithstanding limited sample sizes.
Table 13

*Diet Metrics (Non-empty Stomachs) for HR Seatrout that Survived the Entire 29 d Experiment Period and Wild Fish in the Late Juvenile Target Size Range (125-275 mm TL) Collected in Point aux Chenes Bay, Mississippi*

<table>
<thead>
<tr>
<th>Taxon</th>
<th>Hatchery-Reared (n = 22)</th>
<th>Wild (n = 22)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Freq.</td>
<td>% F</td>
</tr>
<tr>
<td>Carangidae</td>
<td>1</td>
<td>5%</td>
</tr>
<tr>
<td>Gobiidae</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Sciaenidae</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Unidentified</td>
<td>14</td>
<td>64%</td>
</tr>
<tr>
<td>Portunidae</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Callinectes sp.</td>
<td>2</td>
<td>9%</td>
</tr>
<tr>
<td>C. similis</td>
<td>2</td>
<td>9%</td>
</tr>
<tr>
<td>Penaeidae</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Farfantepenaeus sp.</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Palaemonetes sp.</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Alpheus sp.</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Hippolyte sp.</td>
<td></td>
<td></td>
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<tr>
<td>Mysidae</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Unidentified</td>
<td>2</td>
<td>9%</td>
</tr>
<tr>
<td>SAV</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Detritus</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Mud</td>
<td>1</td>
<td>5%</td>
</tr>
</tbody>
</table>
Figure 53. IRI values for major prey groups in the diets of HR (n = 22) and wild spotted seatrout between 125 and 275 mm TL (n = 22) from Point aux Chenes Bay, Mississippi.

Unidentified fish was the most frequently observed prey category in diets of both HR and wild fish. This unidentified status is fairly typical in feeding studies of piscivorous predators, given the relatively high digestion rate of soft (fish) versus hard (crab/shrimp carapace) prey. Only three fish prey could be identified to family level, two of which occurred in wild fish (Gobiidae and Sciaenidae) and one in HR fish (Carangidae). *Callinectes* prey occurred in 18% of HR diets, but were completely lacking in the diets of wild fish < 200 mm TL. In addition, two unidentified shrimp occurred in the diet of HR fish, and one mysid occurred in a wild fish stomach. Incidental material indicative of the feeding habitat (SAV, detritus and mud) was more common in wild fish than in HR fish (mud only).

Although interpretation of diets for HR fish was confined only to individuals surviving the duration of the cage experiment, it is noteworthy that all of the replacement HR fish (introduced weekly to maintain consistent densities among cages)
had empty stomachs. The complete lack of prey within the stomachs of all 15 replacement fish (25% of all fish retrieved) suggests a protracted period (>21 days) may be required for late juvenile fish to transition from pelletized feed in captivity to natural prey in the wild.

Figure 54. Mean ranks for stomach fullness index (FI) among habitats for wild (top) and HR fish (bottom).
Table 14

*Diet Metrics (Non-empty Stomachs) for HR Seatrout that Survived the Entire 29 d Experiment Period and Comparable-sized Wild Fish (< 200 mm TL) Collected in Point aux Chenes Bay, Mississippi*

<table>
<thead>
<tr>
<th>Taxon</th>
<th>HR (n = 22)</th>
<th></th>
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</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Freq.</td>
<td>% F</td>
<td>N</td>
<td>%N</td>
<td>V</td>
<td>%V</td>
<td>TW-g</td>
<td>Freq.</td>
<td>% F</td>
<td>N</td>
<td>%N</td>
<td>V</td>
<td>%V</td>
<td>TW-g</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Carangidae</td>
<td>1</td>
<td>5%</td>
<td>1</td>
<td>5%</td>
<td>0.4</td>
<td>11%</td>
<td>0.40</td>
<td></td>
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<td></td>
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<tr>
<td>Gobiidae</td>
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<tr>
<td>Sciaenidae</td>
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</tr>
<tr>
<td>Unidentified Teleost</td>
<td>14</td>
<td>64%</td>
<td>14</td>
<td>64%</td>
<td>1.0</td>
<td>26%</td>
<td>0.87</td>
<td>8</td>
<td>100%</td>
<td>13</td>
<td>81%</td>
<td>1.2</td>
<td>38%</td>
<td>0.85</td>
<td></td>
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</tr>
<tr>
<td>Callinectes sp.</td>
<td>2</td>
<td>9%</td>
<td>2</td>
<td>9%</td>
<td>0.7</td>
<td>19%</td>
<td>0.48</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
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<td></td>
<td></td>
</tr>
<tr>
<td>C. similis</td>
<td>2</td>
<td>9%</td>
<td>2</td>
<td>9%</td>
<td>1.3</td>
<td>36%</td>
<td>0.94</td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td></td>
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<tr>
<td>Mysidae</td>
<td></td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
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<td></td>
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<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Unidentified Shrimp</td>
<td>2</td>
<td>9%</td>
<td>3</td>
<td>14%</td>
<td>0.3</td>
<td>7%</td>
<td>0.23</td>
<td></td>
<td></td>
<td></td>
<td></td>
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<td></td>
</tr>
<tr>
<td>SAV</td>
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<tr>
<td>Detritus</td>
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<td></td>
<td></td>
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<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Mud</td>
<td>1</td>
<td>5%</td>
<td></td>
<td></td>
<td>0.1</td>
<td>1%</td>
<td>0.01</td>
<td>2</td>
<td>25%</td>
<td></td>
<td></td>
<td>0.1</td>
<td>3%</td>
<td>0.08</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>
Discussion

In this study, the use of HR juvenile spotted seatrout provided the opportunity to evaluate two of the three core requirements necessary for stock enhancement to succeed. Within the confines of the experimental enclosures, HR fish from the USM-GCRL SPEC program were able to (1) acclimate to and survive in fluctuating natural conditions, and (2) transition to feeding on wild prey. The third ecological requirement, predator avoidance, was not examined in this study, as predators were excluded. Prior field-based research on post-release condition of HR fish has focused almost exclusively on salmonid species, where studies have evaluated feeding for HR brown trout (*Salmo trutta*) (Bachman 1984) and Chinook salmon (*Oncorhynchus tshawytscha*) (Johnson 2008) released into the wild. More extensive research has been conducted for HR Atlantic salmon (*S. salar*) relative to feeding (Sosiak et al. 1979; Larsson et al. 2011).
and growth and survival (Jonsson et al. 2003) in natural conditions. However, little focus has been directed toward post-release conditions of HR fish in GoM waters, as was evaluated in this study.

**Survival**

Cumulative survival over the four-week cage study was relatively high, particularly considering the experimental fish had not been subjected to pre-conditioning or to natural conditions. Overall, 73.8% of the original HR fish survived the initial transition to natural conditions during the first week, and survival remained fairly stable thereafter as 69.2% of fish initially introduced into enclosures survived the duration of the experiment. Detrimental effects of handling/transport and acclimation to field conditions probably explain the initial mortality during the first week.

**Growth**

Almost all HR fish lost weight (i.e., negative weight-specific growth) during the experimental period, yet those fish were likely in better condition than wild fish when released, giving them a potential buffer energy reserve for acclimation. Despite the presumed reserve, HR fish still lost weight in every habitat, indicating either that they were generally underperforming at gaining energy or that cage artifacts precluded them from performing adequately. Mean specific growth rate was, however, significantly higher (less negative) in the both SAV and NVS habitats than in the NVO treatment, a trend consistent with prior research that found higher relative growth in vegetated habitats for caged juvenile red drum (Stunz et al. 2002) and pinfish (Shervette and Gelwick 2007). Those results support the view that SAV, marsh-edge, and shoreline habitats are beneficial habitat for juvenile spotted seatrout in GoM and U.S. Atlantic
waters (Laska 1973; Thompson and Verret 1980; Peterson 1986; McMichael and Peters 1989; Chester and Thayer 1990; Rakocinski et al. 1992; Rooker et al. 1998; Smith et al. 2008; Neahr et al. 2010; Powers 2012). Moreover, the finding extends the view about the importance of SAV to the later juvenile stage, about which little is known in terms of ecology.

Temperature, salinity, dissolved oxygen and turbidity did not differ among habitats during the experimental period, suggesting that habitat structure may have played a role in the growth differential among habitats. The lack of a difference in relative growth rates between SAV and NVS treatments, however, is not consistent with the habitat structure interpretation. Of all the habitat characteristics, depth (and by inference, distance from shore) was the only habitat-related metric that differed noticeably among habitat types in a way that was consistent with relative growth; depth was greater at NVO than at both SAV and NVS cage locations, whereas weight loss was significantly lower in SAV and NVS. In the absence of SAV or other structural habitat in deep water, shallow water may serve as alternate refuge for small prey species (Ruiz et al. 1993). Accordingly, the absence of SAV at the relatively greater depths in which NVO cages were sited suggests that prey may have been less abundant in association with those particular enclosures relative to NVS and SAV cages located at shallower depths.

In this study, HR fish were not pre-conditioned before release, but prior research has shown that the feeding success of solitary juvenile HR fish improves with repeated exposure to novel prey (grass shrimp), in terms of prey recognition, strikes per foraging event and prey capture success (Jackson et al. 2013). Jackson et al. (in review) also
showed that large juvenile spotted seatrout (comparable to the sizes used in this study) rapidly learned to feed on grass shrimp within a mesocosm setting, and further, that prior exposure to novel wild prey enhanced their feeding success within mesocosms with novel emergent vegetation. Greater water depths at NVO cages may have led to potentially lower prey abundance and to lower prey encounter frequency, considering the greater total search volume at NVO habitats. The combination of potentially reduced prey resources and the increased energetic cost with detecting and capturing those prey could have contributed to the lower relative growth.

Although not preferred for studies of short-term growth (Weatherly 1972; Able et al. 1999; Necaise et al. 2005; Lanier and Scharf 2007), standard length (absolute daily rates) was also measured for experimental fish for comparison with previous studies based on this metric for *C. nebulosus*. Overall mean daily growth (ADG) for HR juvenile fish measured in this study (119-155 mm SL) was low (0.14 ± 0.014 mm d\(^{-1}\)) compared to otolith-derived growth in length of wild spotted seatrout in other systems (Table 15). Perhaps this is not surprising, given that wild fish had been fully acclimated to natural conditions throughout their life, so their growth rates would be expected to be higher compared to HR fish having been exposed to natural environmental conditions within confined conditions for only one month. Nevertheless, positive growth in length did occur for HR fish in this study, and although non-significant, growth in length was higher in SAV than in the other two habitats.

The use of enclosures to confine fish to specific habitats potentially introduced some artifacts (Peterson and Black 1994). The protection from predators and competitors that was provided to experimental fish may have promoted artificially high
Table 15

*Comparison of Daily Growth Rate Measurements/Estimates for Larval and Juvenile Spotted Seatrout from GoM and U.S. Atlantic Waters*

<table>
<thead>
<tr>
<th>Study (Fish Source)</th>
<th>Area</th>
<th>Fish Size (mm SL)</th>
<th>Basis</th>
<th>Growth Rate (mm d⁻¹)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Present Study (HR)</td>
<td>Point aux Chenes Bay, Mississippi</td>
<td>119 - 155</td>
<td>direct SL difference</td>
<td>0.14</td>
</tr>
<tr>
<td>Baltz et al. 1998 (Wild)</td>
<td>Barataria Basin, Louisiana</td>
<td>3 - 29</td>
<td>otolith</td>
<td>1.07</td>
</tr>
<tr>
<td>McMichael and Peters 1989 (Wild)</td>
<td>Tampa Bay, Florida</td>
<td>&lt; 50</td>
<td>otolith, length frequency</td>
<td>0.51</td>
</tr>
<tr>
<td>Powell et al. 2004 (Wild)</td>
<td>Florida Bay, Florida</td>
<td>12 - 88</td>
<td>otolith</td>
<td>0.84</td>
</tr>
<tr>
<td>Smith et al. 2008 (Wild)</td>
<td>Chesapeake Bay</td>
<td>35 - 120</td>
<td>otolith</td>
<td>1.44</td>
</tr>
</tbody>
</table>

Note. For fish source, “HR” indicates hatchery-reared fish, and “Wild” indicates wild-caught samples.

growth (less rapid weight loss) through (1) more time for foraging without the threat of predation (via predator exclusion), (2) decreased competition for food resources (via exclusion of animals normally exploiting similar prey items), and/or (3) increased availability of prey via attraction by enclosure structures (Lanier and Scharf 2007). Conversely, enclosures may have negatively influenced growth by restricting widespread movement of fish to other profitable foraging areas or to areas of more suitable water quality (Phelan et al. 2000). Negative effects on growth (or weight loss)
would also result from depletion of prey inside experimental units (prey not replaced at natural rates by colonization) (Sogard 1992). For this study, each of the aforementioned artifacts are admittedly possible confounding factors in interpreting growth results, and observed survival was undoubtedly inflated by the exclusion of predators. Additionally, habitat-dependent growth can vary interactively among regions and over time within the same enclosure study (Sogard 1992; Able et al. 1999; Phelan et al. 2000), thus illustrating the need for spatial and/or temporal replication of experimental conditions. Although spatial replication of the entire experiment within two independent areas was attempted, the loss of most cages in the higher energy area of Grand Bay precluded inclusion of experimental units from that area. While insightful, the growth results from this study should be validated with subsequent trials.

Another artifact in this study may have been introduced by the installation of bottom mesh panels on enclosures, which potentially reduced access to benthic substrates and associated prey. An earlier deployment of cages without bottom panels resulted in the loss of too many fish. Several factors apparently contributed to the loss, including effects of *C. sapidus* burrowing under bottom sleeves, loss of experimental fish to predation, and effects of cage dislodging and subsequent escapement in Grand Bay. These factors as well as the inability to routinely check for fish mortality (and thus increase exposure time) necessitated the use of bottom panels. Some former studies have also used fully enclosed cages to assess habitat specific growth (Able et al. 1999; Necaise et al. 2005; Lanier and Scharf 2007). In one such study, Able et al. (1999) documented the penetration of mesh bottoms into soft sediments using underwater video, although the use of mesh bottom might be more problematic in complex SAV
habitat. Gallagher (2001) found negligible cage effects on prey density and community structure in soft sediments like those in non-vegetated treatments of this study, although the presence of cages did affect water flow and fine-scale sediment composition. The mesh size (1.9 cm square) of cages was relatively large (the maximum size possible while preventing fish escapement) in the present study, and all non-vegetated cages were pushed into bottom sediments before firmly securing them to PVC poles. Also, because mud was present on bottom panels every time cages were checked each week, fish apparently were not excluded from bottom substrata by the mesh panels; the presence of mud in the stomach of one HR seatrout further supported that perception.

An additional potential artifact was the use of artificial seagrass as a proxy for natural SAV that was displaced by cage bottoms within SAV habitat. Initial test deployments revealed that virtually all of the SAV was depressed and compacted by the panels instead of protruding through the bottom mesh. To address the displacement of SAV and to standardize the SAV habitat experimental units, artificial seagrass was fixed to the bottom mesh panels of SAV treatment cages. Preliminary measurements of natural SAV within the study area ensured that the SAV units provided comparable SAV surface area to that of naturally-occurring SAV, after which cages were then sited and left for a two-month period to allow for colonization. Previous research using the same artificial seagrass material applied in the present study showed that artificial seagrass units (not associated with cages) were rapidly colonized by macrofauna in areas of SAV in Indian River Lagoon, Florida, wherein peak abundance and species richness was reached within four to eight days (Virnstein and Curran 1986). Likewise, profuse macrofauna was associated with artificial SAV in this study. After a four-week
colonization period, Bologna and Heck (1999) found no difference in macrofaunal richness or abundance between artificial and natural SAV treatments (*Thalassia testudinum*) once they were colonized by epiphytes in St. Joseph Bay, Florida. Taken together, these findings substantiate the use of artificial seagrass as a viable surrogate for natural SAV in the current study.

**Feeding**

Diet analysis indicated that HR fish were feeding somewhat successfully within the experimental period and that diets of late juvenile HR and wild fish were comparable. Species area curves showed that sample sizes were insufficient to fully characterize the diets of HR and wild groups, thus more research is needed to better define the feeding ecology of this important life stage. Most prey items of HR fish were digested beyond the point of specific identification, and unidentified fish was the major diet component of both HR and wild fish. Both HR and wild spotted seatrout < 200 mm TL preyed primarily on teleost fishes, but this focus on fish prey was greater for wild (IRI = 1.82) than for HR fish (IRI = 0.72). Juvenile *C. similis* and unidentified *Callinectes* crabs were consumed by HR fish in NVO habitats but not by wild fish from any habitat. Only one other specific prey identification could be made for HR prey (family Carangidae).

For purposes of evaluating stock enhancement potential for the SPEC program, the fact that 49% of HR fish that survived the entire experimental period contained food in their stomachs is promising. The ability of HR *C. nebulosus* to transition to live novel prey was also supported in laboratory and field studies by Jackson et al. (2013; in review), who demonstrated that HR spotted seatrout possess the inherent plasticity to
switch from a pellet feed to novel live prey. Although they documented initial
deficiencies in feeding performance skills at an earlier juvenile stage in a solitary
setting, later staged juvenile HR spotted seatrout readily learned to feed on live novel
prey in a group setting. Prior exposure to live novel prey also carried over to better
feeding success when fish were subjected to emergent vegetated structure in a group
setting.

Ten prey taxa were identified to the family, genus or species level (Table 13)
from wild *C. nebulosus* over the entire late juvenile size range in this study, and all of
the prey types represented nekton common to the survey area (Peterson et al. 2007).
Teleosts and shrimp were equally important diet items for wild fish in terms of
frequency of occurrence, but shrimp displayed a greater importance (IRI = 0.80) than
fish (IRI = 0.51) when number and volume contributions were taken into account.
Considering the small sample size, these findings are not inconsistent with prior research
on feeding habits of this species in Mississippi coastal waters. Overstreet and Heard
(1982) reported a relatively even occurrence of fish (65%) and crustaceans (47%,
primarily penaeids) in the diets spotted seatrout ranging from 73 to 249 mm (SL). Diet
data in this study also compare well with studies in other GoM estuaries that
documented the primary importance of fish and penaeid shrimp in the diet of late
juvenile spotted seatrout in Florida (Moody 1950; Hettler 1989; Llanso et al. 1998) and
Louisiana (Darnell 1958). The relative dietary importance of shrimp found in this study
is further supported by Minello and Zimmerman (1984), who found that juvenile spotted
seatrout 160-210 mm TL actively chose brown shrimp (*Farfantepenaeusaztecus*) over
spot (*Leiostomus xanthurus*) in a laboratory setting, even in the presence of substrate in which shrimp could burrow.

Overall results from this study show that HR juvenile spotted seatrout have the capacity to both survive fluctuating environmental conditions and transition to feeding on wild prey in a natural system, indicating that two of the three survival elements necessary for successful stock enhancement can effectively be attained by this species. Those adaptations were achieved, however, in the absence of both predation risk and conspecific competition, both of which would elevate post-release mortality. Further research is needed to assess predator-prey interactions (the final survival element) and competitive feeding success for HR fish. Despite small sample sizes, major prey taxa were similar between late juvenile HR and wild fish within the same general size range and habitats, suggesting that reared fish can effectively transition to feeding habits generally comparable to those of wild juvenile spotted seatrout. With regard to habitat, the lower rate of weight loss shown by fish from SAV versus NVO habitat suggests that habitat complexity may be an important factor in post-release feeding. This finding corresponds to habitat use trends found for cultured *C. nebulosus* juveniles in Texas (Neahr et al. 2010) and for the occurrence of wild fish observed in an earlier part of this study (Chapter III). Considering that predators were excluded by enclosures in this study, the role of SAV for HR seatrout would likely extend beyond feeding benefits, as SAV also provides critical refuge from predators (Rozas and Odum 1988; Jordan et al. 1996). Directed studies on relative habitat benefits for HR spotted seatrout are warranted to better understand how to maximize post-release condition of HR fish. From a
management perspective, emphasis should continue to be placed on comprehensive estuarine habitat delineation to identify and protect ecologically beneficial resources.
LITERATURE CITED


Jackson LA, Rakocinski CF, Blaylock RB. In review. Effects of prior exposure to novel prey on the feeding success of hatchery-reared spotted seatrout, Cynoscion nebulosus (Cuvier, 1830), within a group setting. Submitted to Fisheries Management and Ecology.


CHAPTER V
GENERAL CONCLUSIONS

Conclusion

The objective of this study was two-fold: (1) to address significant deficiencies in the knowledge of habitat use and general feeding ecology of late-juvenile spotted seatrout in Mississippi coastal waters, and (2) to evaluate post-release growth and feeding of late juvenile hatchery-reared (HR) seatrout subjected to natural environmental conditions. Sidescan sonar mapping described in Chapter II provided acoustic imagery of benthic habitats available to late-juvenile spotted seatrout in shallow depths (< 2 m) and relatively turbid conditions in Point aux Chenes Bay, Mississippi. Field-based benthic sampling confirmed image-derived habitat classifications, which revealed that relative coverage in the survey area was greatest for non-vegetated water bottoms but did not differ considerably between SAV and marsh-edge habitats. Comprehensive sidescan mapping allowed for more accurate estimation of SAV coverage than did point-based ponar/poling sampling, which underestimated SAV area. Additionally, inter-annual differences in SAV spatial coverage were evident based on analysis of 2011 and 2012 acoustic imagery. Future directed habitat research in shallow coastal waters would benefit from the techniques employed here, particularly with respect to delineating coastal SAV that provides important habitat for many estuarine species but is seldom mapped at large spatial scales (Moncreiff 2007). Such comprehensive habitat assessments provide an effective first step for detailed ecological studies.
Directed gill net sampling (Chapter III) collected late-juvenile spotted seatrout (125-275 mm) that have been missing from most studies, particularly in Mississippi. Late-juveniles were five times more abundant in SAV than along erosional marsh-edge, even though those habitats had similar areal coverages in the survey area based on sidescan mapping. Juvenile abundance was lowest in non-vegetated habitats, where CPUE was eight times less than that for SAV. Those findings further underscore the importance of SAV, where available, for YOY of the species in GoM and U.S. Atlantic waters (Laska 1973; Thompson and Verret 1980; McMichael and Peters 1989; Chester and Thayer 1990; Rooker et al. 1998; Smith et al. 2008; Powers 2012). Results from this study also suggest that the role of SAV as preferred habitat for spotted seatrout extends beyond the early juvenile stage to late juveniles of the species. Submerged aquatic vegetation in Mississippi coastal waters, however, is likely not essential at the spotted seatrout population level due to its large-scale absence in nearshore waters. Accordingly, in turbid waters where SAV does not typically occur, such as those along the western Mississippi coast and coastal Louisiana, physio-chemical conditions may be more important factors than structural habitat in governing distribution of the late-juvenile stage of this species (MacRae 2006), although structural habitat is clearly preferred habitat for post-larval and juvenile spotted seatrout.

Through the implementation of a field enclosure study (Chapter IV) conducted in habitats similar to those sampled in previous chapters, HR juvenile spotted seatrout exhibited the ability to survive introduction into natural environmental conditions and transition to wild food sources, in the absence of predators and competitors. Hatchery-reared fish generally lost weight during the experimental period, but specific growth rate
was highest for fish in SAV. Growth was, however, not statistically different between SAV and non-vegetated shoreline habitats, which corresponded to marsh-edge sampled for Chapter III. Although sample sizes were too low for comprehensive evaluations of feeding habits, 49% of HR fish that survived the trial period contained prey, and major prey taxa were similar to those found in the diets of wild fish. Feeding habits by wild spotted seatrout were also consistent with those reported by other GoM studies which found fish and penaeid shrimp to be primary dietary components of late juveniles (Moody 1950; Darnell 1958; Overstreet and Heard 1982; Hettler 1989; Llanso et al. 1998). Overall, the results illustrate that HR spotted seatrout have the innate ability to transition from hatchery to wild conditions for the key ecological functions of survival and feeding. The latter of these abilities was also shown by Jackson et al. (in review) for HR fish feeding on wild prey in a laboratory setting. Future research focusing on predator-prey interactions would help in the selection of release habitats that would maximize survival potential of juvenile HR spotted seatrout, particularly relative to SAV and emergent vegetation that provide refuge from predators (Rozas and Odum 1988) and may also promote faster growth, as suggested in this study.

Study results suggest that management of spotted seatrout would benefit from expanded research to further identify, delineate and ecologically characterize coastal habitats and their relative importance. Results of such studies in conjunction with routine population monitoring would assist in determining if, and to what degree, regional spotted seatrout stocks are habitat- or recruitment-limited, knowledge of which is key to the effective management of heavily exploited populations. Habitat-based ecological surveys would also allow for more thorough assessments of habitat value at
the population level. In a broader sense, comprehensive mapping efforts will help support best coastal zone management practices that are crucial to the conservation of ecologically beneficial habitats that may foster survival, growth and recruitment of important fishery species.

Considering stock enhancement as a management tool, findings from this study can be applied in developing release strategies that maximize post-release survival of HR fish. For instance, SAV yielded higher CPUE of wild fish compared to marsh-edge and non-vegetated habitats, and specific growth of HR individuals was higher in SAV and shoreline stations relative to non-vegetated open water. Those results suggest SAV and areas in close proximity to emergent vegetation provide ecological characteristics that may promote survival and growth of fish introduced into the wild. Accordingly, stock enhancement goals to supplement exploited wild populations would be better achieved by identifying and utilizing beneficial habitats that likely contain abundant prey resources and provide refuge from predators.
LITERATURE CITED


Jackson LA, Rakocinski CF, Blaylock RB. In review. Effects of prior exposure to novel prey on the feeding success of hatchery-reared spotted seatrout, *Cynoscion nebulosus* (Cuvier, 1830), within a group setting. Submitted to Fisheries Management and Ecology.


APPENDIX A

INSTITUTIONAL ANIMAL CARE AND USE COMMITTEE APPROVAL FORM

The proposal noted below was reviewed and approved by The University of Southern Mississippi Institutional Animal Care and Use Committee (IACUC) in accordance with regulations by the United States Department of Agriculture and the Public Health Service Office of Laboratory Animal Welfare. The project expiration date is noted below. If for some reason the project is not completed by the end of the three year approval period, your protocol must be reactivated (a new protocol must be submitted and approved) before further work involving the use of animals can be done.

Any significant changes (see attached) should be brought to the attention of the committee at the earliest possible time. If you should have any questions, please contact me.

PROTOCOL NUMBER: 11051201
PROJECT TITLE: Ecology of Juvenile Spotted Seatrout
PROPOSED PROJECT DATES: 05/12/2011 to 03/31/2012
PROJECT TYPE: New Project
PRINCIPAL INVESTIGATOR(S): Joseph Read Hendon, Ph.D.
COLLEGE/DIVISION: College of Science & Technology
DEPARTMENT: Center for Fisheries Research & Development
FUNDING AGENCY/SPONSOR: Mississippi Department of Marine Resources/Tidelands Trust Fund
IACUC COMMITTEE ACTION: Full Committee Review Approval
PROTOCOL EXPIRATION DATE: 09/30/2013

Jodie M. Jawor, Ph.D. IACUC Chair

[Signature]

Date