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Natural spawning and scaling-up of yellowtail snapper (*Ocyurus chrysurus*): Larval rearing for the mass production of juveniles

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ABSTRACT

The natural spawning and scaling-up the larval rearing of yellowtail snapper (*Ocyurus chrysurus*) are described. We obtained fertilized eggs via the volitional spawning of broodstock acclimated for two years in two 12-m^3 tanks. Of the eggs collected 10 to 14 h after spawning, 87.3% were floating, and 90% of these were transparent and presented a live embryo (egg diameter of 824 ± 77 μm and oil droplet diameter of 137 ± 14 μm). The incubation period was 18 h at 27 °C and 38 g l^{-1} salinity. The average normal larvae hatching rate was 88.3 ± 3.3%, and the notochord length of the newly hatched larvae was 2.5 ± 0.5 mm. Larval survival at 48-h post-hatching was 48.1 ± 3.7%. The initial mean stocking density in the 2500 l rearing tanks was 206.3 ± 24.5 floating eggs l^{-1}, which resulted in 99 ± 43 larvae l^{-1} per tank. A total of 261,736 juveniles were harvested at a mean of 50 ± 3 days post-hatching (dph) from ten 4.5-m^3 cylindrical fibreglass tanks, which presented a mean of 26,174 ± 3211 juveniles per tank (5.8 ± 0.7 juveniles l^{-1}). The mean survival in the ten tanks was 7.0 ± 1.4% from the time of hatching and 15.0 ± 2.9% as of the first feeding (48-h post-hatching). Two critical periods with high larval mortality occurred during rearing: between 4 and 7 dph and during the third and fourth weeks (20–28 dph). At harvest, the mean total length (TL) was 3.8 ± 0.2 cm (range: 2.9–5.4 cm) and the mean total weight was 0.9 ± 0.2 g (range: 0.3–2.1 g). Larval growth increased from a mean of 0.24 mm day^{-1} during the first 14 dph (specific growth rate (SGR) of 6.7% day^{-1}) to a mean of 1.73 mm day^{-1} (SGR of 9.6% day^{-1}) in the subsequent 21 rearing days. The mean final biomass was 4.9 ± 1.3 kg m^{-3}. Finally, the results and current and future larval rearing practices for this species are discussed.

1. Introduction

Yellowtail snapper (*Ocyurus chrysurus*) is a marine species distributed in the tropical and subtropical western Atlantic Ocean from southern Massachusetts, USA (45°N) to southeastern Brazil (16°S) and throughout the Gulf of Mexico and the Caribbean (Riley et al., 1995; Froese and Pauly, 2016), including the Bahamas and Bermuda. Yellowtail snapper is common in commercial catches and for sport fishing around the Gulf of Mexico and the Caribbean (Munro et al., 1973; Turano et al., 2000), and it is a highly valued commercial fish (Cummings, 2004; Gutierrez Benitez, 2012) and has been recommended for cultivation (Watanabe et al., 2005). Yellow snapper is a gonochorist fish, with females and males becoming sexually mature at 250 to 310 mm (McClellan and Cummings, 1998).

Soletchnik et al. (1989) and Alvarez-Lajonchère et al. (1992) described yellowtail snapper as a promising candidate for aquaculture and stock enhancement. The positive aquacultural features of this fish include (a) its high food quality, consumer acceptance and high price and demand; (b) its high tolerance to crowding; (c) its wide environmental tolerance; and (d) its adaptability to practical formulated dry diets. However, mass production of juveniles remains a problem, which is mainly because of the absence of reliable and stable juvenile mass production technology. Several institutions are working on the artificial reproduction and larval rearing of yellowtail snapper (Soletchnik et al., 1989; Alvarez-Lajonchère et al., 1992; Riley et al., 1995; Turano et al., 2000; Allen et al., 2003; Faulk et al., 2005; Ibarra-Castro et al., 2010); however, technology for mass producing juveniles of this species has not been reported.

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Over the past two years in Mexico, the main objective of juvenile production research has been rearing yellow snapper to marketable sizes. In 2010, a joint research programme on larval rearing at the laboratory scale was initiated by the Faculty of Science of the National Autonomous University of Mexico, the Mexican government, and the private sector (Sanchez-Zamora, unpublished data). The present study reports the results from the natural spawning of broodstock matured in captivity at the Sisal Laboratory and the scaling-up of the best results previously obtained for the species. This work followed the adjusted mass production protocols that presented successful results for other marine fish, such as striped patao (Alvarez-Lajonchère et al., 1996), fat snook (Alvarez-Lajonchère et al., 2002), common snook (Ibarra-Castro et al., 2011), and spotted rose snapper (Alvarez-Lajonchère et al., 2012).

2. Materials and methods

2.1. Egg production

Fertilized eggs were obtained from the natural spawning of captive broodstock. The mean body weights (BW) were 1.2 kg for females and 1 kg for males, with a total female biomass of 13 kg. The fish were held in two cylindrical fibreglass 12-m^3 spawning tanks (4-m wide and 1.0-m deep) with a flow-through system (three tank volumes day^{-1}) and strong aeration. White sailcloth was suspended 3.5 m above the tanks, which were located in a 12 × 30 m greenhouse at the Sisal Yucatan Unit of the National Autonomous University of Mexico (21° 09″N; 90° 02″ W, 45 km from Merida City, Yucatán State). Broodstock were fed a laboratory-prepared mixture of 60% commercial dry diet (Fish Breed-M®, INVE Aquaculture, Inc.) and 40% oily fish flesh (skipjack), which was supplied daily at a rate of 2% BW.

Eggs from the volitional spawning were collected in 100-l collector tanks with a 0.3-mm mesh screen under the spawning tank outflow. All eggs were removed from the collectors 10–14 h after spawning at the embryo somitogenesis stage. Floating egg numbers from the larval rearing trials were estimated volumetrically with a 500-ml graduated cylinder, which generated an estimated mean of 1435 ± 52 eggs ml^{-1}, with 50 eggs from 10 selected spawnings. The viability (% floating eggs with live embryos) and egg and oil droplet diameters (to the nearest 10 μm) were determined from a sample of over 120 floating eggs from the 10 seed spawnings.

2.2. Larval and juvenile rearing environment management

Incubation and larval rearing were conducted in five 5-m^3 cylindrical fibreglass tanks with black walls and white bottoms, with a culture tank volume of 4.5 m^3. Mean stocking density was 206.3 ± 24.5 floating eggs l^{-1}, initially with 2.5 m^3 of water. Hatching rates were determined by incubating 50 eggs with live embryos in six 1-l beakers and recording the number of newly hatched larvae the next morning as well as their 48-h post-hatching survival. Rearing tanks were in the same room as the spawning tanks.

Seawater was treated by pressurized sand filters followed by multicartridge (20 μm–10 μm–5 μm relative retention) filtration and a continuous-flow UV lamp (30,000 μW scm^{-2}). The water quality parameters during egg incubation and larval rearing were as follows: temperature, 28 ± 1 °C; salinity, 38 g l^{-1}; dissolved oxygen, 6 ± 0.5 mg l^{-1} (saturation: 94.2 ± 7%); pH, 8.2 ± 0.2; and NH_3, less than 0.6 mg l^{-1}. The photoperiod was 16 h light:08 h dark.

Environmental management and feeding protocols were established for egg incubation, larval rearing, and weaning (Figs. 1 and 2, respectively), and a daily work plan was written for the environmental control practices, cleaning, feeding, and behavioural observations, which were performed according to Alvarez-Lajonchère and Hernández Molejón (2001).

2.3. Live feed production and larval and juvenile feeding regimen

Microalgae (*Nannochloropsisoculata* and *Isochrysis* sp.) culturing was performed indoors in 18 semi-transparent 250-l cylindrical fibreglass tanks (0.30-m diameter) with Guillard F/2 medium. Rotifers (*Brachionus rotundiformis*) were cultured in continuous culture systems indoors with an initial density of 50 rotifers ml^{-1} in 1000-l tanks, Culture Selco 3000® (INVE Aquaculture Inc., Mazatlan, Mexico) at 0.3 g per $million^{-1}$ rotifers was provided three times daily, and pure oxygen was supplied. The tank was initially harvested when it reached a mean density of 700 rotifers ml^{-1}.

The harvested rotifers were enriched with Protein Selco Plus® (INVE Aquaculture Inc.) at 0.3–0.36 g l^{-1} × 10^6 rotifers for 18 h. Newly hatched premium-quality *Artemia* spp. (Great Salt Lake, INVE Aquaculture, Inc., Mazatlan, Mexico) and 36-h metanauplii enriched with DC DHA Selco® (INVE Aquaculture Inc.) were provided per the manufacturer's instructions.

The microalgae additions and the feeding schedule are shown in Fig. 2. Live food organisms were added to the tanks five times daily at 0800 h, 1100 h, 1400 h; 1700 h and 2000 h after counting the organisms remaining in the larval rearing tank.

Enriched *Brachionus rotundiformis* rotifers were fed from 2 dph to 18 dph. *Artemia* was supplied from 10 dph to 30 dph in the form of newly hatched nauplii for the first 5 days and enriched metanauplii for the last 15 days. *Artemia* were supplied twice daily, with 50% of the day's ration provided after tank cleaning and the other 50% stored at low temperature (4–8 °C) and supplied at 1600 h. Weaning to formulated food (OTOHIME®, ProAqua-Mexico) began at 10 dph, with complete weaning occurring by 35 dph (~15-mm TL). Artificial feed was supplied every 2 h beginning at 0600 h, with a final feeding at 2200 h.

2.4. Larval sampling

Larval growth was monitored every 7 days. In each sampling, the TL was measured on 25–110 randomly collected individuals from each tank. Following collection, fish larvae were anesthetized in a petri dish with a drop of clove oil. Digital images were taken using a Nikon SMZ-800® stereomicroscope fitted with a Nikon DS-L camera, and the TL was obtained with ImageJ 1.30v software (http://rsb.info.nih.gov/ij/). Samples taken at 35–45 dph were measured with a digital calliper (precision: 0.01 mm).

Prior to harvest, 5 samples of 50 juveniles were collected. The juveniles were weighed and counted to obtain the average weight per juvenile. To facilitate harvest, the juveniles were lightly sedated using clove oil at a concentration of 1 ppm. The specific growth rates (SGRs) were calculated by the following equation: SGR (% day^{-1}) = 100 (ln L_1 − ln L_o) t_{-1}, where L_o and L_1 are the TL at the beginning of a set period and at the end of that period, respectively, and t is the number of days in the period. Different groups were weighed to obtain the total juvenile harvest weight. The total number of harvested juveniles was obtained by dividing the total biomass by the average weight per juvenile.

3. Results

3.1. Egg production and quality

From May to October 2012, eggs were collected from different natural spawns from the captive breeding stock. A total of 26.7 × 10^6 eggs were collected (2.05 × 10^6 eggs per kg of female spawned biomass) 12–14 h after spawning, and all egg stock in each larval rearing tank were from a single spawning. The mean floating eggs was 87.3%, of which 90% had live embryos. A mean of 515,686 ± 61,142 floating eggs was initially stocked in each rearing tank with a volume of 2500 l. The average diameter of 120 eggs from each spawning used for the