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Reduced swimming and metabolic fitness of aquaculture-reared California Yellowtail (*Seriola dorsalis*) in comparison to wild-caught conspecifics

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ABSTRACT

Aspects of swimming and metabolic physiology were measured in aquaculture-reared California Yellowtail (*Seriola dorsalis*) in comparison to wild-caught individuals in order to examine potential differences in health and fitness associated with captive rearing, and to help identify areas for targeted improvement in *Seriola* aquaculture. Incremental swimming velocity trials using a swim tunnel respirometer on small yellowtail (mean body length = 18.9 cm, mass = 80.1 g) showed that aquaculture-reared fish had a significantly slower mean maximum sustainable swimming speed (U_{crit}) (4.16 ± 0.62 BL s^{-1}) in comparison to that of wild-caught fish (4.80 ± 0.52 BL s^{-1}). In addition, oxygen consumption ($\dot{M}_{O_2}$) measurements at varying swimming speeds allowed for estimation of standard metabolic rate, which was significantly higher in aquaculture-reared yellowtail (7.31 ± 2.32 vs. 3.94 ± 1.60 mg O_2 kg^{-1} min^{-1} at 18 °C). Aquaculture fish also had a lower aerobic scope (9.20 ± 3.44 mg O_2 kg^{-1} min^{-1}) in comparison to wild-caught yellowtail (15.80 ± 5.78 mg O_2 kg^{-1} min^{-1}), which likely contributed to their reduced capacity for fast sustainable swimming. Reduced physical fitness is commonplace in aquaculture-reared fishes, and the examination of wild-caught yellowtail in this study provides baseline metrics that can be used to gauge the health and fitness of future *S. dorsalis* production. In particular, the lower standard metabolic rate and higher aerobic scope of wild-caught fish represent desirable metabolic characteristics that if achievable in aquaculture through better-rearing practices could allow for increased feed conversion efficiencies and potentially faster growth. At a minimum, a 35–40% reduction in metabolic costs at low swimming speeds (to those observed for wild-caught yellowtail) should result in substantial cost savings for feed in aquaculture operations.

1. Introduction

Species of the carangid genus *Seriola*, or amberjacks, are becoming an increasingly popular target for aquaculture development due to their fast growth rates and high flesh quality. While species such as the Japanese Amberjack (*S. quinqueradiata*) and, to a lesser extent, the Greater Amberjack (*S. dumerili*) have been produced in aquaculture for several decades, these operations have been largely dependent on the regional harvest of wild-caught seed (from larvae to large juveniles) that are then reared to market size (Nakada, 2002; Ottolenghi et al., 2004). In most locations, however, the availability of wild seed is a major biological bottleneck that limits *Seriola* aquaculture outgrowth capacity. With recent advances in hatchery-production techniques, wild-captured or domestically-bred broodstock are now being used to produce eggs and larvae for growout operations. This has greatly increased prospects for further *Seriola* aquaculture development and expansion.

Much of this new work has focused on the yellowtail jack species complex (*S. lalandi*) (Abbink et al., 2012; Moran et al., 2007; Stuart and Drawbridge, 2013), which is allowing for *Seriola* expansion into more temperate waters in areas such as the United States, New Zealand, Australia, and Chile. While *S. lalandi* was previously recognized as a single circumglobal species, recent genetic research has revealed at least three closely-related but distinct species – *S. dorsalis* from the Northeast Pacific, *S. aureovittata* from the Northwest Pacific, and *S. lalandi* from the Southern Hemisphere (Martinez-Takeshita et al., 2015), the last of which should likely be further split between the waters of South Africa and the South Pacific (Purcell et al., 2015). The genetic distinction between these species is thought to represent local adaptation to regional environmental conditions and highlights the need for comparative studies of culturing methods as well as the need for caution when translocating seed between regions for aquaculture growout (Purcell et al., 2015).

Despite advances in hatchery production for several species, most of

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the world's current *Seriola* aquaculture production is still based on the capture of wild juveniles. This is largely because wild-caught seed is generally considered cheaper than hatchery production, especially in places such as Japan where the capture of wild seed has been the standard for more than 50 years (Nakada, 2002, 2008). In addition, hatchery-reared individuals are often thought to be of lower quality, showing higher incidences of deformity and having slower growth rates. Still, hatchery production is a much more sustainable practice that could help prevent overfishing naturally-occurring populations for aquaculture use and, in theory, circumvent the juvenile bottleneck on aquaculture expansion by allowing *Seriola* aquaculture in regions of the world where juveniles are not available for capture from the wild. These concepts, coupled with declines in the availability of wild seed (Nakada, 2008), highlight the need for continued research to optimize domestic production.

This study focuses on the health and fitness of the California Yellowtail, *S. dorsalis*, for which hatchery-rearing and growout protocols are currently being tested and are in high demand for US aquaculture development (Jirsa et al., 2011; Rotman et al., 2017; Stuart and Drawbridge, 2011; Stuart and Drawbridge, 2013; Stuart et al., 2010). To better understand the often-referenced inferiority of aquaculture-produced seed, this study establishes baseline physiological metrics of the health and fitness of aquaculture-reared *S. dorsalis* in comparison to wild-caught counterparts and in comparison to metrics available for other *Seriola* species. Specifically, we examine aspects of *S. dorsalis* swimming performance (e.g., critical swimming speed, cost of transport), which have been used as indicators of fish "physiological well-being" (Brauner et al., 1994; Claireaux et al., 2005; Farrell et al., 1997; Hammer, 1995; McKenzie et al., 2012; Plaut, 2001), and are likely representative of regular "health checks" made by large-scale *Seriola* aquaculture operations [fish health is regularly checked by observing the swimming speed and behavior of individuals during feeding, and the swimming activity of the school as a whole (Nakada, 2008)]. In addition, we compare the metabolic physiology of culture-reared and wild-caught individuals as differences in metabolic fitness affect fish growth, feed conversion efficiency, and likely provide insight into how exercise training and other manipulations may positively affect fish health and growth (Davison and Herbert, 2013; Palstra et al., 2015).

2. Materials and methods

2.1. Fish collection and husbandry

Aquaculture-reared *S. dorsalis* were derived from eggs produced by wild-captured broodstock housed at the Hubbs-SeaWorld Research Institute and reared according to current protocols (Stuart and Drawbridge, 2013). Young juveniles were transferred to the Southwest Fisheries Science Center (SWFSC) at approximately 5–6 cm body length (= total length, BL) where they were housed in 300 × 150 × 90 cm ($l \times w \times h$) oval tanks (approximately 3200 l) and held at ambient ocean temperature. Wild-caught yellowtail were captured at 12–15 cm BL by hook and line from local waters (< 20 km from shore) associated with drifting kelp. Upon transfer to the SWFSC these fish were housed in identical tanks and temperatures as the aquaculture-reared individuals. Both aquaculture-reared and wild-caught yellowtail were fed commercial pellet fish feed (BioTrout, Bio-Oregon, Longview, WA) to satiation 1–2 times daily six days a week. Wild-caught fish were held 2–4 months in captivity before swim tunnel experimentation. All yellowtail husbandry and experimentation was conducted in accordance with protocol SW1401 of the SWFSC Animal Care and Use Committee.

2.2. Swim tunnel experimentation

Exercise endurance testing and respirometry measurements were made on 10 aquaculture-reared (18.63 ± 1.12 cm BL, 90.19 ± 19.88 g; condition factor = 1.38 ± 0.14; means ± standard deviation) and seven wild-caught (19.31 ± 1.35 cm BL, 65.80 ± 13.35 g, condition factor = 0.91 ± 0.07) *S. dorsalis* following methods established for active pelagic fishes [including the closely related *S. lalandi* (Clark and Seymour, 2006; Brown et al., 2011)] using a Brett-style swim tunnel respirometer. The acrylic, variable-speed 5.4 l swim tunnel respirometer (Loligo Systems, Tjele Denmark) had a 30 × 7.5 × 7.5 cm working section and was contained within a surrounding buffer tank for thermal insulation and to provide properly aerated seawater for flushing the system between respirometry measurements. The buffer tank was fed by a continuous influx of ambient filtered seawater that averaged 17.64 ± 1.61 °C across experiments. Fish were starved for approximately 24 h before being placed in the swim tunnel, where they were allowed to swim steadily at a preferred, low, sustainable speed (typically about 20–25 cm s^{-1}) with a continuous inflow of aerated seawater for a 1 h acclimation period prior to experimentation.

The maximum sustainable swimming speed (critical swimming speed, U_{crit}) was determined by increasing the velocity of the swim tunnel water by 5 or 10 cm s^{-1} steps every 30 min until the fish could no longer maintain steady swimming (defined as no longer being able to stay off the screen at the back of the working section) or erratic and non-directional burst activity occurred. U_{crit} was determined by the equation:

$$U_{crit} = U_i + U_{ii} (T_i / T_{ii}) \quad (1)$$

where U_i is the fastest swimming speed maintained for the full 30 min, U_{ii} is the velocity step (5 or 10 cm s^{-1}), T_i is the time swimming at the highest velocity (at which fatigue occurred), and T_{ii} is the length of time for each step in swimming speed (30 min). Water velocity within the swim tunnel was calibrated using a vane wheel flow probe connected to a handheld flowtherm NT (Höntzsch GmbH, Waiblingen, Germany). Following experimentation, swim tunnel water velocity was corrected for both the cross-sectional area of the vane probe and for the solid blocking effect of the fish in the tunnel according to Bell and Terhune (1970) resulting in true velocity step increases of 3.8 to 4.1 and 7.6 to 10.2 cm s^{-1} depending on fish size.

The oxygen consumption rate ($\dot{M}_{O_2}$) was determined for each fish at each swimming speed using a fiber optic oxygen sensor within the swim tunnel respirometer connected to a Fibox 3 fiber optic oxygen transmitter (PreSens Precision Sensing GmbH, Regensburg, Germany). Shortly (1–2 min) after the step increase in swimming speed, the swim tunnel respirometer was temporarily sealed from the inflow and outflow of fresh seawater from the surrounding buffer tank using manual valves, and the oxygen concentration within the now closed system was recorded once every 5 s. The oxygen level was allowed to drop until the swim tunnel water reached approximately 80% saturation, at which point the system was flushed (to return the oxygen saturation level to near 100%) and then resealed to continue $\dot{M}_{O_2}$ measurements if time allowed within that velocity step.

2.3. Calculation of oxygen consumption, standard metabolic rate, aerobic scope, and cost of transport

Typically, 2–3 sequential $\dot{M}_{O_2}$ traces (from ~100–80% oxygen saturation, $R^2 > 0.990$) were recorded during each 30 min velocity step, and these were combined to determine a single mean oxygen consumption rate for each individual at each speed. Following the completion of the experiment, the fish was removed from the swim tunnel for mass and length measurements, and the respirometer was resealed to measure background respiration, which was subtracted from fish $\dot{M}_{O_2}$.

To compensate for differences in fish mass and water temperature between trials, $\dot{M}_{O_2}$ measurements were adjusted to a water/body temperature of 18 °C (using $Q_{10} = 2$) and to a body mass (M) of 80 g (using $M^{0.80}$) (Pirozzi and Booth, 2009; Schmidt-Nielsen, 1984). Oxygen consumption data were then used to create an exponential