Larval billfish abundance in the Western Indian Ocean and future research endeavors

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ABSTRACT

Stock assessment of highly migratory species such as tunas and billfishes from fisheries data alone is challenging. Using fisheries-independent larval data may be useful as a supplement to those models. As of date, billfish spawning is only reported in the Pacific and eastern Indian Ocean and no scientific record of billfish spawning in the western Indian Ocean has been reported. Opportunistic larval fish surveys were conducted on January 2022 in Tromelin and Reunion EEZ and on April 2023 in the Mozambique Channel within the French EEZ. Larval billfishes were collected from surface and subsurface tows. Maximum density was 19.77 istiophorids / 1000 m^2 from the Mozambique Channel. Mean sea surface temperature was 28.79°C (± 0.69 SE). Istiophorids collected in 2022 were genetically identified to be blue marlin (*Makaira nigricans*, n = 25) and 2023 collection identified morphologically by pigment pattern and morphometrics relationships were blue marlin (n = 6) and Indo-Pacific sailfish (*Istiophorus platypterus*, n = 15). Monthly or bimonthly systematical sampling is recommended to reveal spatio-temporal spawning of the billfish species in the WIO and to better understand the role of the Mozambique Channel and adjacent water as spawning ground and nursery site for the billfish species.

KEYWORDS: larval distribution, Indo-Pacific sailfish, Blue marlin, Mozambique Channel, Reunion Island

Introduction

Billfish is one of the most important fish species group in the Western Indian Ocean (WIO) region, both socio-economically and culturally (Kadagi *et al*. 2022). Six billfish species are reported from the WIO: swordfish (*Xiphias gladius*), black marlin (*Makaira indica*), blue marlin (*Makaira nigricans*), striped marlin (*Tetrapturus audax*), shortbill spearfish (*Tetrapturus angustirostris*), and Indo-Pacific sailfish (*Istiophorus platypterus*). Of these, all but the shortbill spearfish are currently managed by the Indian Ocean Tuna Commission (IOTC 2022). Marlins are caught in artisanal fisheries for food and income, while swordfish hold commercial importance for large-scale international industrial fishing fleets. They are also highly sought after by recreational anglers throughout the world and have been attracting sport fisheries since the 1950's in Kenya (Howard & Starck II, 1975; Williams, 1970). The status of two of the six billfish species in the WIO is 'overfished' and 'subject to overfishing', where striped marlin have had such status for over a decade. Some level of uncertainty exists in their stock status for the rest of billfish species (IOTC 2022).

Despite their significant regional interest, comprehensive knowledge of biology of the billfish species and their fisheries still remains limited in most WIO countries for number of reasons. Catches are under-reported or not reported because most billfish species, other than swordfish, are considered as a bycatch in large-scale industrial fishing fleets targeting tuna and tuna-like fishes. Poor documentation and poor reporting of landings results in the uncertainty of fishing mortality from artisanal and recreational fisheries (Kadagi *et al*., 2020). The circum-global distribution and highly migratory behavior of billfishes add complexity to national-level assessments, requiring the consideration of fisheries impacts across different national jurisdictions. Without adequate fisheries-dependent data, stock assessment models have uncertainties in the stock evaluation.

Given the challenges to adequately assess the fisheries data on such highly migratory species, fisheries-independent data from larval surveys combined with modified close-kin mark-recapture would enhance stock assessment without using CPUE data—which are difficult to develop for bycatch species (Bravington *et al*., 2016; McDowell *et al*., 2022). Current knowledge about the spawning habitat of the Indo-Pacific billfish species in the IO is dated back to 1981 by Nishikawa *et al*. (1985) and they are mostly on the eastern side of the IO. Angler observations and gonad maturity, however, also suggests that the Mozambique Channel may also be a spawning ground for the black marlin (Kadagi *et al*., 2022).

Oceanography

All year round, the South Equatorial Current from the eastern IO flows westward. The portion that passes through the northern edge of the Madagascar becomes the Northeast Madagascar Current (NEMC) and about a third breakoff north as the East African Coastal Current upon reaching the eastern coast of the African continent (Schott *et al*., 2009). The main flow of the NEMC, about 20 Sv (Donguy & Piton,1991), flows south along the coast of Mozambique. At the bottleneck of the Mozambique Channel, the flow splits northward and southward, where the northern current flows back along the western coast of Madagascar to form a large anti-

cyclonic eddy, rejoining NEMC. The southern flow passes through the Mozambique Channel as the Mozambique Current. Numerous anti-cyclonic gyres form as the current pass south along the eastern continental edge of Africa. The northern gyre is wind driven and has interannual and seasonal variability, whereas the southern system is thermohaline (Donguy & Piton, 1991). These gyres create complex system of mesoscale (50–200 km) and submesoscale (under 50 km) eddies in the region. Most of the Mozambique Channel is over 2000 m deep but there are also numerous shallow banks and seamounts derived from active underwater volcanic activities. Presence of these underwater seamounts and shallow basins further create complex currents in the region (Miramontes *et al*., 2019). These eddies would physically influence advection and retention of the larvae before settlement on reef and nursery sites to some degree. Current understanding of dispersal at early life history is that fish larvae are not passive plankton but they have well-developed behavioral and sensory abilities (hearing, smelling, vison) (Leis *et al*., 2011; Montgomery *et al*., 2006; Simpson *et al*., 2005) and larval behavior influence dispersal as much as physical processes; larval fish dispersal is therefore considered a biophysical process (Leis, 2015). Additionally, the cyclonic eddies' centers are known to create upwellings of nutrient-rich deepwater that creates temporary plankton blooms in the otherwise oligotrophic open ocean. Anti-cyclonic gyre sweeping along the shallow coast also delivers nutrient rich riverine waters off-shore (Quartly & Srokosz, 2004). Predator and prey accumulate around convergent edges of these eddies for an energy-efficient outcome for the voracious billfish larvae and zooplanktivorous predators of demersal and pelagic larval fish (Lindo-Atichati *et al*., 2012; Schumid *et al*., 2020; Shulzitski *et al*., 2016).

Methods and Materials

Zooplankton sampling took place onboard a French surveillance vessel OSIRIS II which is a 55.5 m long longliner that has been converted to fisheries control and scientific activities.
Two surveys were conducted: from January 17 to February 1st, 2022 in waters adjacent to Tromelin and Reunion Island, and April 4–20, 2023, within the French EEZ located in the Mozambique Channel. Zooplankton nets were cast opportunistically during the day, 2–3 times a day (Fig.1).

Ichthyoplankton collection

A 60 cm bongo frame, equipped with 500 μm mesh nets with codends and mechanical flowmeters (438 110, Hydro-Bios Kiel), was towed against the current at approximately 1.5 kts off a davit from the starboard side. Two different samples were taken at each station: a horizontal 10 minute neuston tow, and a 25 m oblique tow in 2022 and 100 m oblique tow in 2023. The latter was equipped with 40 kg weight (Murphy *et al*., 2014) and custom temperature and depth recorder. Net depth was estimated during the tow from line out and line angle. Time, GPS positions of nets' casts and retrievals, and filtered volumes were calculated from flowmeter counts and recorded. Temperature and depth profiles were recovered from the recorder after each tow.

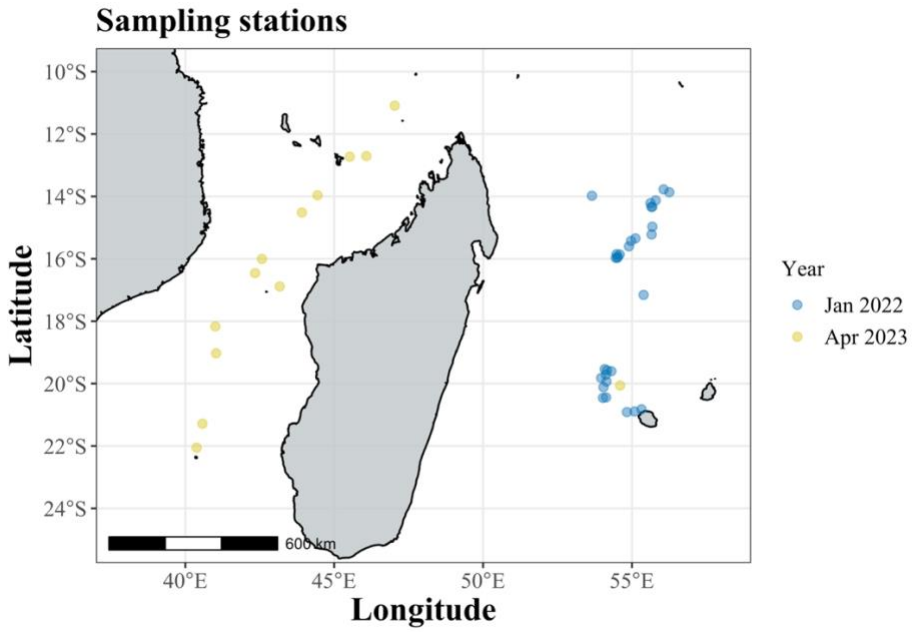

Figure 1. Sampling stations and survey years. All stations in the Mozambique Channel were conducted within the French EEZ.

Visual identification, sample sorting, and preservation

Samples were immediately preserved in 95% EtOH and stored in an air conditioned room. The initial preservative was changed 24 hours post-collection to compensate for dilution due to sample dehydration. Zooplankton displacement volumes were recorded and all samples were sorted to separate fish larvae. All fish larvae where identified to the family level using various references (Leis & Carson-Ewart, 2004; Okiyama, 2014; Richards, 2005). Due to difficulties of identifying larval marlins to species level, genetic identification was used for the marlin larvae collected in 2022. Prior to dissection, dorsal, ventral, and lateral view photographs were taken. Dorsal and lateral view photographs were used for standard length (SL) and morphometric measurements using Image J software and ventral views were used for lower jaw pigments identification. For the genetic identification, the eyeball was primarily used as a tissue sample (right eyeball if both were intact), or the tail was severed with a sterilized scalpel if both eyes were missing (Richardson *et al*., 2007). All results were plotted using R (R Core Team, 2023).

Genetic Identification

The DNeasy Tissue Kit (Qiagen, Germany) was used to make the DNA extraction as per the manufacturer's instructions, except for the elution step where the same 100 µL of elution buffer (EB) was applied twice on the column to concentrate the DNA, whereas they recommend to put 200 µL of EB. The extracted DNA concentration was measured using an Invitrogen™ Qubit™ 4 Fluorometer (Waltham, USA).

PCR primers used during the experiments were based on a literature review. Six species-specific pairs were found to amplify different genes from mitochondrial DNA sequences. Hyde *et al*. (2005) designed six species-specific pairs of primers for sailfish *(Istiophorus platypterus*),

blue marlin *(Makaira nigricans*), black marlin (*Makaira indica*), shortbill spearfish *(Tetrapturus angustirostris*), striped marlin (*Tetrapturus audax*), and swordfish (*Xiphias gladius)* (Table 1). For each species the same forward primer was used (UniversalF) whereas the reverse one was species-specific.

Table 1. The seven original primer pair sets for multiplex PCR, their sequences, the lengths of PCR amplicons (Hyde *et al*., 2005).

Primers	Direction	Sequence	T°m	Amplicon size
UniversalF	Forward	ART GAA TYT GAG GHG GYT TCT C	54,6	/
I .platypterusR	Reverse	GTT AGG CCT CGC TGT TTA GAG	55,3	463bp
M. indicaR	Reverse	ACA CCC CCT AGT TTR TTA GGA ATC	55,1	388bp
M. nigricansR	Reverse	GGA GGT HAG ACC AAT TAG RAG A	53,1	239bp
T. angustirostrisR	Reverse	GTA AAG TTG TCA GGA TCA CCA	52,3	287bp
T. audaxR	Reverse	ATT TTA TCT GCG TCT GAG TTT AGC	53,4	169bp
X. gladiusR	Reverse	GTG AAT AAT GGT TGC GGC TAT G	54,4	104bp

For multiplex PCR, a mix of reverse primers were prepared by adding 2 µL of each primer for a total volume of 20 µL. The final volume of PCR reaction was 20 µL, containing 4 µL of extracted DNA, 1 µM of each reverse primer (from the mix), 0.5µM of UniversalF, 800 µM of dNTP, 2.5 U of Taq DNA Polymerase (Qiagen, Germany), 1X AllTaq PCR buffer with $MgCl_2$ and complete with Milli-Q Water. Multiplex PCR was carried out in an Applied Biosystems 2720 Thermal Cycler (Waltham, USA) using the following program: 94°C for 2 minutes, followed by 35 to 50 cycles (depending on the DNA extracted concentration) of 30 seconds at 94 °C, 30 seconds at 55 °C, 30 seconds at 72 °C. The final extension step at 72 °C was performed for 3 minutes or longer.

Ten microliters of PCR product and 2 µL of DNA-dye Non-Tox were mixed and loaded onto a 2% agarose gel containing 8 µL of Midori Green. The electrophoresis was running in the TE buffer at 100 V for 90 minutes. The DNA bands were observed under ultraviolet light and photographed using InfinityCapt (Vilber Lourmat Sté, France, Fig. 2).

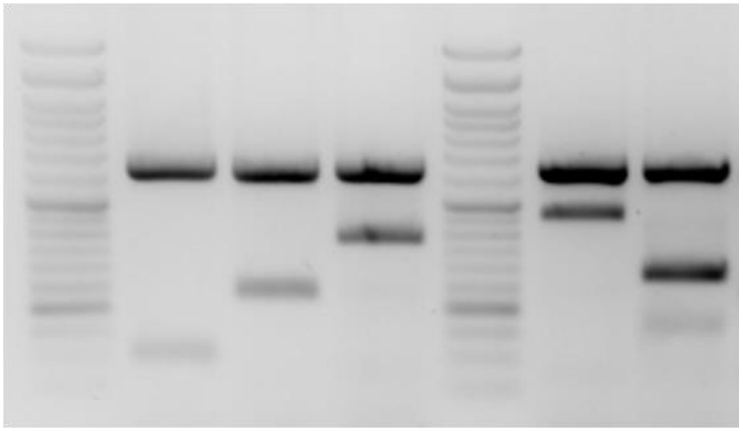

Figure 2. Specificity of multiplex PCR under UV light (from left to right): *X. gladius* (SWO, 104bp), *M. nigricans* (BUM, 239bp), *M. indica* (BLM, 388bp), *I. platypterus* (SFA, 463bp), and *T. angustirostris* (SSP, 287bp). No samples of striped marlin were available at the time of testing.

Results

During the January 2022 survey, 419 and 1632 ichthyoplankton from 21 and 31 families were collected from 30 surface and 33 subsurface plankton tows , respectively. Larval istiophorids were collected from 14 of the 33 stations (n = 17 and 9 for surface and subsurface tows, respectively), whereas a single xiphid larva was collected from one station (n = 1 from subsurface). Mean ichthyoplankton density was twice as much higher subsurface, but mean istiophorid densitiy was 3.71 times higher at the surface. The size range of istiophorid larvae were 2.30–11.79 (mean = 4.03 ± 2.44 SE) mm SL (Figure 3). Of the 28 larvae collected in January 2022, 25 were identified as blue marlin and 3 larvae <3 mm SL were unidentifiable.

During the April 2023 survey, 13 surface and 12 subsurface tows were conducted and 2459 and 2683 ichthyoplankton from 35 and 75 families were collected, respectively. Istiophorid larvae were collected from four of the 13 stations (n = 20 and 2 for surface and subsurface tows, respectively), and xiphid larvae were collected from the surface tows conducted at two stations (n = 3). Mean ichthyoplankton density was three times higher at the surface than subsurface and mean istiophorid density was 35 times higher at the surface. The size range of istiophorid larvae were 2.61–12.91 (mean = 8.11 ± 2.59 SE) mm SL (Figure 3). Genetic identification is still pending on these larvae.

Sea surface temperature (SST) during both surveys ranged from 27.0–29.9°C (mean = 28.79 ± 0.69 SE). There was no significant difference in SST between istiophorid positive and negative stations (df = 41, p> 0.05, two sample t-test).

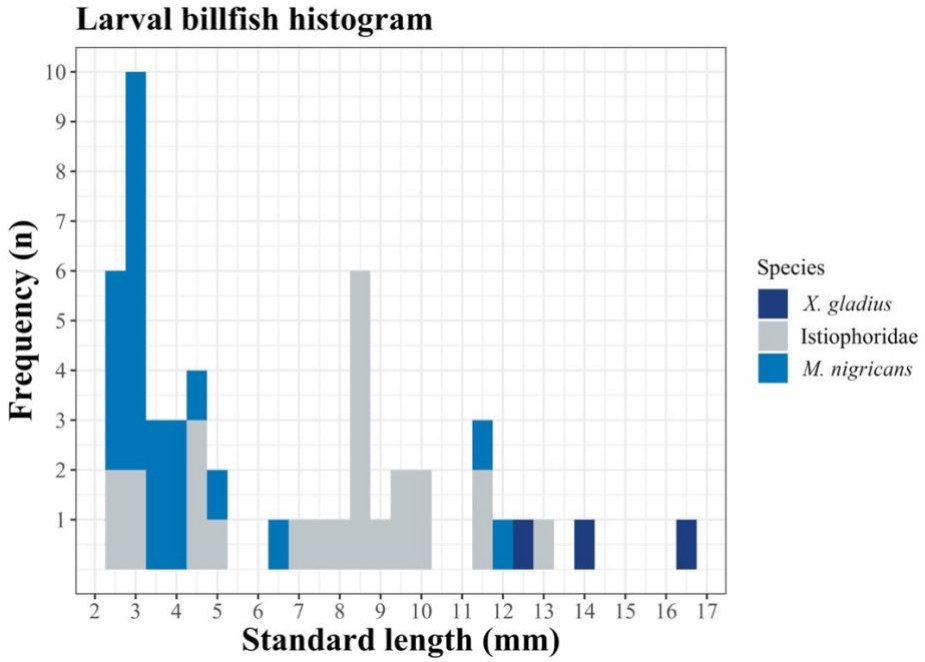

Figure 3. Histogram of all billfish collected from the two preliminary surveys. All unidentified istiophorid from April 2023 survey, except for three individuals <3 mm SL. All *X. gladius* were from the April 2023 survey.

Discussion

Density and mean length was higher for larvae collected from the Mozambique Channel. Standard length range for the majority of those larvae was 7–10 mm SL, which is probably about 10–15 days post hatch (DPH), based on age length relationships for the Atlantic istiophorids (Simms *et al.*, 2010; Sponaugle *et al.*, 2005; Tidwell *et al.*, 2008). The origin of those larger larvae are unknown but we also captured a larva that is 2.61 mm SL (2–3 DPH, 18.16°S 41°E), which most likely have spawned within the Mozambique Channel. The cause for the higher density in the Mozambique Channel in April compared to adjacent water east of Madagascar in January is unknown. Zooplankton displacement volume measurements, however, showed higher zooplankton biomass in the Mozambique Channel, which would be favorable for the voracious billfish larvae (Fig. 4).

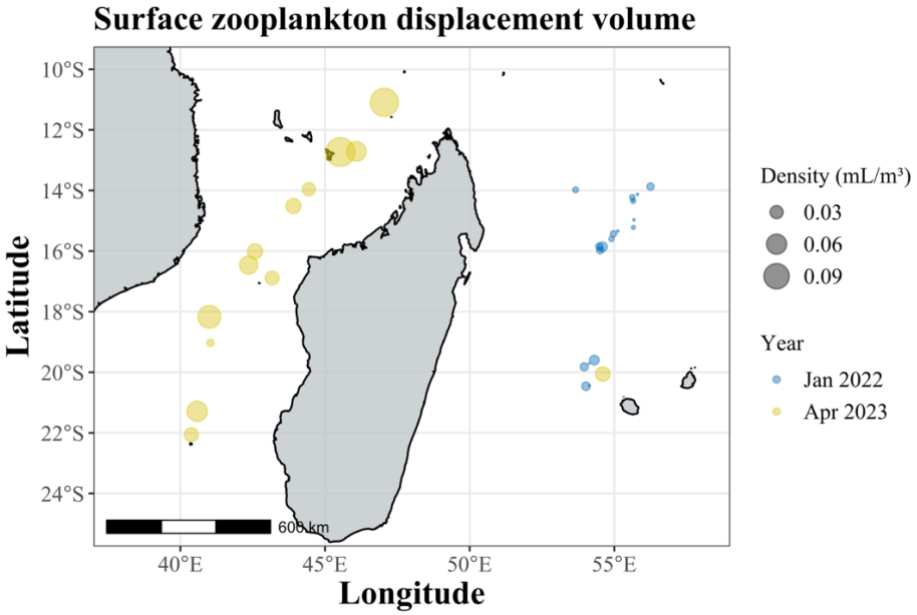

Figure 4. Zooplankton displacement volume (mL m^{-3}) from surface tows. Zooplankton biomass was generally higher in the Mozambique Channel than at adjacent water east of Madagascar. SST showed no significant differences between the years.

From age length analysis of the blue marlin from the Exuma Sound and Straits of Florida in the Atlantic, Sponaugle *et al*., (2005) found that larval growth varied by site for larvae that were 5–10 days old, most likely due to ontogenetic change to more nutritious prey from copepods to fish larvae (Govoni *et al*., 2003; Sponaugle *et al*., 2005). Since the ambient temperature between sites did not show significant difference, variance in turbidity may have affected the prey availability and accessibility, resulting in different growth rate. Hence, it is important to use location specific growth curve or directly age the larvae for spatio-temporal analysis of spawning (Sponaugle *et al*., 2005).

Morphological identification was attempted for the istiophorid larvae collected in 2023, following identification keys available. Based on the pigment pattern, ten larvae were identified as sailfish, nine were identified as blue marlin, and two were unidentifiable. Following Luthy *et al.* (2005), metrics relationship of snout length over orbit diameter to SL were plotted (Fig. 5). Three larvae that were morphologically identified as blue marlin and two that were morphologically unidentifiable followed the trend for sailfish. These metrics relationship may be useful but further analysis is required with larger sample size to find the metrics relationship for WIO as the growth rate may affect the outcome of the correlation line.

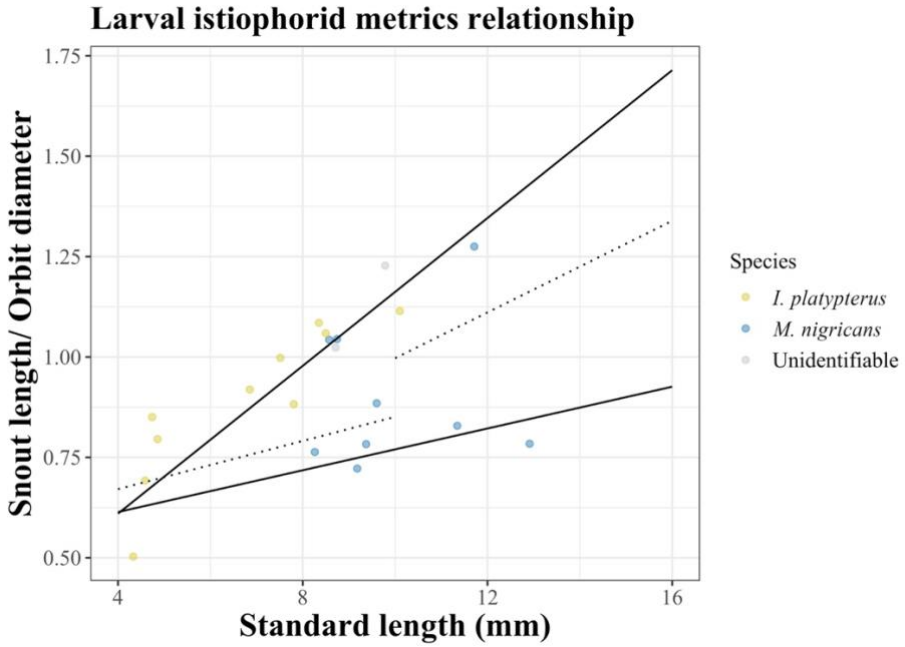

Figure 5. Morphologically identified larvae were plotted on morphometrics relationship by Luthy *et al*. (2005). Solid line indicates correlation line for sailfish (top) and blue marlin (bottom) from the Atlantic species. Dotted line indicates the separation used in identification key.

Billfish larvae are relatively rare in zooplankton samples, even if the bio-physical characteristics appear favorable to sustain the larvae (Richardson et al., 2009). Younger larvae may be collected with few others but they become sparser as they grow due to mortality from starvation, cannibalism by older and larger cohorts, or by predation from other piscivorous predators. Because they are abundant in the surface layer during the day (Llopiz & Cowen, 2008), conventionally, a 2 m x 1 m horizontally rectangular neuston frame is used for larval billfish collection, which has about 3.42 times larger mouth size than our 60 cm bongo frame used during our surveys. It is worthy to note that even with the gear used, we found larval istiophorid density comparable to some of the spawning sites in the Atlantic. Using 2 m x 1 m neuston frame equipped with 500 and 1200 μm mesh, Tidwell *et al*. (2008) and Simms *et al*. (2010) reported maximum density of 52.83 billfish larvae 1000 m^{-2} and 51.4 sailfish larvae 1000 m^{-2} from their survey in the Gulf of Mexico, respectively. In comparison, our maximum density using a gear one third in size was 19.77 istiophorids 1000 m^{-2} from the Mozambique Channel (Fig. 6). In order to evaluate the importance of Mozambique Channel as spawning habitat of the billfish larvae, further sampling using gear equal in size is necessary.

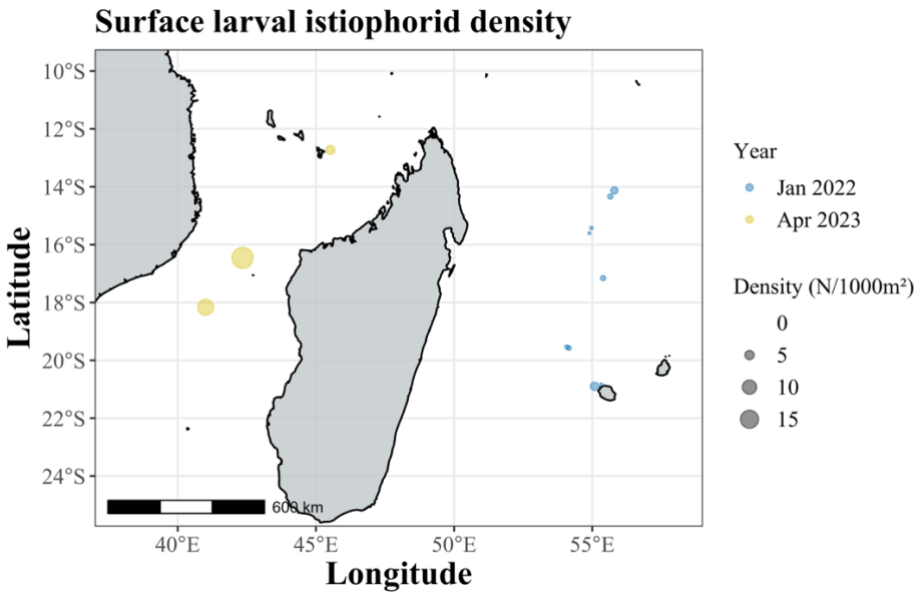

Figure 6. Larval istiophorid density (ind. 1000 m^{-2}) using 60 cm bongo nets. Maximum density of 19.77 ind. 1000 m^{-2} was recorded from the Mozambique Channel.

Conclusion

Opportunistic zooplankton sampling in the WIO collected billfish larvae, especially istiophorids in abundance. Since there were no historical record of billfish spawning in the region, and previous reports from the adjacent waters in the 1980's does not mention occurrence of these larvae, whether these catches are due to chance, climate change induced habitat change, or due to poor larval survey effort in the region, is unknown. Spawning sites within the habitat range and the proportion of the population that uses those spawning locations needs to be determined through combined effort in adult tagging, otolith microchemistry analysis, and genetics, to improve the use of these larval data for stock assessment. Further monthly or bimonthly sampling in the region is needed to determine the timing and location of billfish spawning and to better understand the role of the Mozambique Channel and adjacent water as spawning ground and nursery site for billfish species.

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