

Video Article

Capturing Tissue Repair in Zebrafish Larvae with Time-lapse Brightfield Stereomicroscopy

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Keywords: Developmental Biology, Issue 95, Fin regeneration, Time-lapse imaging, Stereomicroscope, Heated incubation chamber, Post-imaging analysis

Date Published: 1/31/2015

Citation: Lisse, T.S., Brochu, E.A., Rieger, S. Capturing Tissue Repair in Zebrafish Larvae with Time-lapse Brightfield Stereomicroscopy. *J. Vis. Exp.* (95), e52654, doi:10.3791/52654 (2015).

Abstract

The zebrafish larval tail fin is ideal for studying tissue regeneration due to the simple architecture of the larval fin-fold, which comprises of two layers of skin that enclose undifferentiated mesenchyme, and because the larval tail fin regenerates rapidly within 2-3 days. Using this system, we demonstrate a method for capturing the repair dynamics of the amputated tail fin with time-lapse video brightfield stereomicroscopy. We demonstrate that fin amputation triggers a contraction of the amputation wound and extrusion of cells around the wound margin, leading to their subsequent clearance. Fin regeneration proceeds from proximal to distal direction after a short delay. In addition, developmental growth of the larva can be observed during all stages. The presented method provides an opportunity for observing and analyzing whole tissue-scale behaviors such as fin development and growth in a simple microscope setting, which is easily adaptable to any stereomicroscope with time-lapse capabilities.

Video Link

The video component of this article can be found at <https://www.jove.com/video/52654/>

Introduction

The ability of an organism to orchestrate tissue repair processes after injury is crucial for its survival¹. While all animals have the capacity to heal their wounds, the extent to which tissues regenerate differs greatly among species. Vertebrate species such as zebrafish, salamanders and frog tadpoles have the remarkable ability to regenerate lost tissues, including their appendages, portions of their eyes, heart, and central nervous system²⁻⁴. Mammalian species, such as the African spiny mouse and rabbits, are capable of regenerating holes in their pinnae⁵⁻⁷, and humans and mice regenerate portions of their liver as well as their digit tips during fetal and juvenile stages⁸⁻¹². Although it is not well understood yet why and how certain species regenerate tissues more effectively than others, the presence of similar genetic pathways suggests that these mechanisms may lie dormant in species without great regeneration potential^{13,14}. Thus elucidating tissue repair and regeneration mechanisms in species with satisfactory regeneration outcomes will benefit regeneration in humans.

We have chosen the larval zebrafish tail fin as a paradigm to demonstrate its regeneration with time-lapse brightfield stereomicroscopy. The zebrafish larval tail fin is anatomically simple as compared to the more complex adult structures, consisting of a two-layered, infolded epithelium with somatosensory axons innervating the skin that surrounds medially located mesenchymal cells¹⁵. Despite the anatomical differences, larval tail fin regeneration is somewhat comparable to adult fin regeneration in terms of the molecular signatures and the outgrowth responses^{16,17}. As compared to the adult fin, imaging larval tail fin regeneration has however several advantages: 1) larval fin regeneration is completed within just 2-3 days¹⁶, 2) larvae can be mounted in low-melt agarose, and 3) larvae do not require feeding until ~ 5 days post fertilization (dpf) due to the presence of the yolk sac. This makes zebrafish larvae ideal for observing tissue repair dynamics *in vivo*.

The presented method enables the capture of detailed dynamics underlying the early processes of fin regeneration. Many studies have utilized fluorescence-based confocal microscopy to study cellular and subcellular biological processes in embryonic and larval zebrafish. Sophisticated confocal imaging setups are however often not accessible to everyone and highly expensive as compared to other imaging techniques. In contrast, the presented methodology utilizes a Discovery V12 stereomicroscope equipped with Axiovision software and a time-lapse module, thus providing a more affordable alternative to expensive imaging equipment to examine tissue behaviors. We demonstrate that this method can be utilized for imaging tissue regeneration with high temporal resolution at a minimal cost. The implications for this method could extend beyond basic biology to advance mammalian regeneration studies using organ cultures, for therapeutic development through pharmacological and genetic screens, and it can serve as a teaching tool in a classroom setting.

Protocol

Zebrafish (*nacre* strain) were bred and raised according to established protocols. All efforts were made to minimize suffering, using 0.4 mM Tricaine for anesthesia and 1 mM Tricaine for euthanasia. Zebrafish embryos and larvae were handled in strict accordance with good animal practice as approved by the appropriate committee (MDI Biological Laboratory animal core IACUC number 13-20). This study was approved by the National Human Genome Research Institute Animal Care and Use Committee, MDIBL Institutional Assurance # A-3562-01 under protocol # 14-09.

Note: The imaging procedure that captures fin regeneration in larval zebrafish is summarized in the following steps:

1. Raising of Zebrafish to Larval Stages

1. Collect the eggs and place approximately 50 eggs into a 100 x 25 mm Petri dish containing 0.03% Instant Ocean salt in deionized water supplemented with 0.00004% methylene blue. Incubate O/N in a 28.5 °C incubator.
2. The next morning remove the dead embryos with a glass pipette and rinse the eggs in a strainer with 0.03% Instant Ocean salt in deionized water (termed embryo medium).
Note: Medium such as Ringers¹⁸, Hanks¹⁸, E2¹⁹, E3²⁰, and Danieau²¹ may be preferred.
3. sup>Add fresh embryo medium to the dish. If using pigmented strains, optionally add 0.2 mM 1-phenyl-2-thiourea (PTU), as PTU will prevent melanogenesis and thus pigmentation of the larvae. Let the embryos further develop in the incubator until 2 days post fertilization or any other desired larval stage.

2. Preparation of the Imaging Chamber

1. Method 1: Imaging chambers made from PVC or Teflon tubing (**Figure 1**)
Note: This method is similar to Concha and Adams (1998)²².
 1. Acquire drinking-water grade plastic or Teflon tubing from a hardware store with a 25 mm outer and a 20 mm inner diameter. Cut the tubing to make rings of approximately 10 mm thickness with an even surface on each side. Use > 200 grit sandpaper to smoothen the edges.
 2. Clean the rings with warm water and 70% ethanol and let them air dry.
 3. With a pipette tip, apply silicon grease to one half of a ring and attach the ring to a 75 mm x 25 mm glass cover slip. Alternatively, use a 3 ml syringe filled with silicon grease instead of a pipette tip.
Note: Because it is difficult to insert the silicon grease into the 3 ml syringe, add the silicon grease to a 30 ml syringe first and use this for filling the 3 ml syringe.
2. Method 2: Preparation of a Petri dish as an imaging chamber.
 1. Acquire 35 or 60 mm diameter Petri dishes with a glass coverslip attached to the lid (**Figure 2A**). Alternatively, as shown in **Figure 3** of Distel & Koester (2007)²³, drill an opening small enough to hold a coverslip into the lid of a small Petri dish and apply silicon grease to the outside with a 3 ml syringe. Using a clean pipette tip, carefully attach a round or square coverslip of desired thickness to the outside (**Figure 2B**).
 3. To ensure that the agarose being used for mounting stays firmly attached during the imaging procedure, attach a fine plastic mesh inside the ring. First, cut the mesh made of window screen obtained from a hardware store, into the size of the inner ring diameter using fine scissors with an angle. Then cut a small rectangle > twice the size of the larva into the middle of the mesh (**Figure 1, 2**).
 4. Apply four small dots of non-toxic silicon grease to the interface between the cover slip and the chamber ring (**Figure 1A**).
 5. Use forceps to attach the mesh firmly to the bottom of the glass coverslip.

3. Mounting and Imaging of the Pre-injured Larva (this Step is Optional)

Note: This step is suitable for comparisons between the amputated and regenerated fin length, as the amputation plane after fin regeneration is not recognizable in zebrafish larvae.

1. Prepare a 0.5-1.2% low-melt agarose solution in embryo medium for immobilization of the specimen.
2. Heat the agarose in a microwave and transfer the liquid agarose into 1.5 ml tubes that are pre-warmed to 42 °C in a heating block.
3. Let the hot agarose cool down to 42 °C, which can be maintained over several weeks at this temperature. Try to avoid pipetting the larvae into agarose above 42 °C, as this will be detrimental to the animals.
4. Anesthetize several larvae using 10 ml of 0.4 mM Tricaine (pH 7) in embryo medium in a 60 mm diameter Petri dish. Prepare the Tricaine according to the Zebrafish Book Recipes¹⁸.
 1. Alternatively, use 10 ml of a 1:1000 dilution of 99% 2-Phenoxyethanol in a 60 mm diameter Petri dish. Poke the larvae with a capped microloader pipette tip to assess their touch response before proceeding. Use only non-responsive larvae.
5. Transfer one larva into the 42 °C agarose solution using a glass Pasteur pipet. Do not transfer excessive liquid into the agarose, otherwise the agarose will be too diluted and not solidify.
6. Discard the remaining liquid from the pipet and transfer the larva with one drop of agarose into a small Petri dish (35 or 60 mm diameter). Position the larva on its side for imaging of the tail fin.
7. Allow the agarose to solidify. Assess the agarose with a plastic pipette tip; the tip will immerse into the agarose if it is too liquid. Once the agarose has solidified, a small indentation will be visible upon touching with a pipette tip. Following solidification, add Tricaine solution and proceed to the stereomicroscope that will be used later for time-lapse imaging.
Note: This step may also be performed by placing the anesthetized larva on a Petri dish coated with 1.5% agarose in embryo medium.

8. Use a stereomicroscope with time-lapse software. Select an appropriate objective and magnification, which will be used later for time-lapse imaging. Here, use a 3.5 x, 16 mm working distance objective lens on a stereomicroscope. As desired, utilize alternative microscopes and objective lenses but choose a proper magnification to account for potential xy-drift and growth of the fin during the imaging procedure.
9. Select the camera detection mode on the microscope.
10. In the software, select the 'live' mode to view the larva on the screen.
11. Open the 'Properties' window to automatically detect the brightness.
12. Manually adjust the contrast at the microscope trans-illumination base.
13. Move the larva out of the field of view and select the Shading Correction feature to minimize background noise.
14. Position the larva back into the field of view and take a snapshot. Save the image.
15. Remove the agarose from the larva by first scraping the agarose off the head. This way the larva can be slipped out of the agarose by gently pulling the head away from the remaining agarose with a capped microloader pipet tip or an insect pin.
16. With a glass Pasteur pipet transfer the larva into fresh Tricaine solution.

4. Amputation Assay

1. Prepare a 1.5% agarose solution using embryo medium and pour a thin layer into a Petri dish. Let the agarose solidify.
2. Under a stereomicroscope, place the larva sideward onto the solidified agarose and amputate the tail fin with a 23 G syringe needle with slight pressure (**Figure 3A**).

5. Mounting the Larva for Time-lapse Imaging

1. Proceed as described in steps 3.1 – 3.5 (**Figure 3B**).
2. Transfer a drop of 0.5-1.2% liquid agarose at 42 °C containing the larva into the imaging chamber ring (step 2.1), orient the larva and let the agarose solidify. Fill the ring with Tricaine solution. Alternatively, if the Petri dish chamber (step 2.2) is utilized, mount the larva onto the lid coverslip and fill the lid with Tricaine solution.
3. To allow proper wound healing or tissue regeneration to occur, carefully scrape off the agarose surrounding the distal tail fin using a capped microloader pipette tip or an insect pin. Try not to injure the fin repeatedly (**Figure 3C**).
4. Decant the Tricaine solution containing the removed agarose and fill the chamber ring with fresh Tricaine solution.
5. Apply silicon grease to the top of the chamber ring and attach a 75 mm x 25 mm glass slide. Try to avoid air pockets in the chamber, as they will interfere with brightfield imaging and desiccate the larva over time.
6. If using a Petri dish as an imaging chamber, apply silicon grease to the top rim of the bottom chamber and fill the bottom chamber with Tricaine solution. Carefully decant the Tricaine solution in the lid and turn the lid over to immerse the larva into the Tricaine solution of the bottom chamber at a slight angle to avoid air pockets. The chamber will be sealed due to the silicon grease.

6. Time-lapse Imaging

1. Assemble a heated incubation chamber as described in ^{23,24} (**Figure 4**). Place the incubation chamber around the microscope and turn on the heat. Adjust the temperature to 28 °C for about 10 - 20 min or until the temperature has stabilized.
2. Open the front of the heated incubation chamber and place the imaging chamber onto the microscope stand with the coverslip facing upward toward the objective.
3. Position the larval fin in a way that 2/3 of the field of view remains unoccupied. This ensures the capture of the growth and regeneration of the fin over the course of the imaging procedure without having to reposition the larva.
 1. Adjust the mounted larva to 28 °C for ~ 30 min prior to starting the time-lapse recording to avoid changes in brightfield intensity or potential shifts of the agarose. Alternatively, utilize pre-warmed buffer to start imaging after shorter adjustment time.
4. To set up the time-lapse recording, open the 6D Multidimensional Acquisition window in the Axiovision software and select the z-stack and time-lapse option.
5. Optional) In the z-stack tab and Slice Mode, select the slice thickness and then select the Start/Stop mode.
6. Define the upper and lower position of the stack.
7. In the time-lapse tab, select the interval and the duration of the movie and then start the movie by pressing the start button. We found that 30 min intervals are sufficient and this interval does not generate excessive data; however shorter intervals may be used.
8. Check the position and z-stack dimensions during the first hour if the larva was not pre-adjusted. If necessary, reposition the larva again after a day, as the larva may have shifted.
9. Save the file at the end of the time-lapse recording and proceed with the post-processing and quantifications using available image analysis software, such as Imaris ²⁵ or the open source software packages Image J ²⁶ and Fiji ²⁷.

7. Data Analysis

1. Determining the fin length.
 1. Open the time-lapse movie in the imaging software and save the files in the proprietary file format to enhance software performance.
 2. Select the orthogonal view to display individual sections as projected stacks. For drift correction, under the Fiji menu, select Plugins, then Registration, and 'Correct 3D drift'. This will open a Fiji window and perform the drift corrections. Correct rotational drift in the Imaris Spots function. Alternatively, install the StackReg and TurboReg plugins in Image J and import to Fiji. Choose the desired transformation algorithm in the StackReg plugin.
 3. To measure distances (e.g., wound diameter or fin length), select the 'Add New Measurement Points' option in the upper left tool bar.
 1. Under 'Configure list of visible Statistics Values' in the bottom left menu, select the statistics values to be displayed.
 2. In the 'Line Mode' under the settings tab select 'Pairs (AB, CD ...)'.

3. In 'Labels Properties' select 'Name' and 'Distance', to display the distance between points A and B next to the measurement line.
 4. Switch to the 'Edit' mode and hold down the shift button to select the first point at the end of the notochord. Then use the same configuration to select the second point at the distal fin margin.
 5. Under the 'Statistics' tab, select the disk button (Export All) at the lower right to display and export the distance in the image.
 6. Repeat the measurements at selected times by moving the slider below the image to the right. Instead of creating new Measurement Points, the previous ones can be repositioned by first selecting the point with the left mouse button, then simultaneously pressing the shift and the left mouse button at the new position.
4. Alternatively to the Measurement Points option, utilize the slice viewer to measure distances. In the slice view mode, scroll to a desired position and click on the first and second position with the left mouse button. The distance will be displayed. This option however does not allow for data export.
2. Determining fin length and area in ImageJ.
 1. Open the time-lapse movie in ImageJ using a plugin that recognizes the .zvi file format. Alternatively, load in a QuickTime file or tiff sequence.
Note: If using an uncompressed tiff file format, the image dimensions do not need to be specified.
 2. If opening a different file format without the file information, select 'Set Scale' under the 'Analyze' menu to first define the image distance and unit. In the 'Set Scale' menu, type in the 'Distance in pixels', below type in the 'Known distance' for the pixel value (this can be obtained by measuring the number of pixels on a scale bar that was added to the image, then click Measure to obtain the result), and the 'Unit of length' (typically ' μm '). Then click OK.
 3. For fin area measurements select the 'Freehand Selection' tool in the tool bar and outline the fin area by holding the left mouse button down while drawing along the outline of the fin. For fin length measurements, select the 'Straight' line tool and draw a line between the desired points to be measured.
 4. Click the 'Measure' option under 'Analyze' to display the area and length of the fin. Repeat this step as often as necessary for multiple time points of the movie.
 3. Using statistics software the data can be graphically displayed.

Representative Results

The presented technique is suitable to elucidate tissue repair dynamics in response to amputation. The movie demonstrates that amputation of the fin initially triggers a purse-string effect, characterized by contractions via actin-myosin cables that are present in the fin-fold²⁸ (**Figure 5 A,B**). Concomitantly, cells are extruded from the wound (see movie). The contraction may thus be a means to expel cells that are likely destined to undergo cell death. Our results further show that the developmental growth of the larva occurs independently of regeneration (movie), whereas fin regeneration does not initiate until about 14 hr post amputation as measured by fin length and area over the time course of 36 hr following amputation (**Figure 5C, D**). The total regenerative fin growth after 1.5 days was about 60% of the original fin length (**Figure 5E**). Taken together, these results demonstrate that amputation triggers fin contraction, extrusion of cells from the wound, and a temporally delayed regenerative response. While extruded cells are likely destined to undergo cell death, the nature of these cells needs to be further clarified.

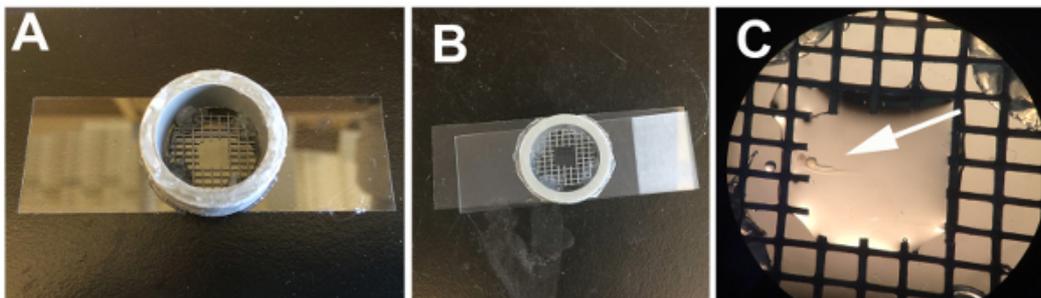


Figure 1. Imaging chamber ring assembly

(A) Shown is a plastic ring that is attached to a coverslip with silicon grease. A plastic mesh is attached to the inside of the chamber with four small dots of silicon grease. (B) The chamber containing the mounted larva is filled with Tricaine solution and a glass slide is attached to the top. (C) A mounted 2 day-old larva (arrow) is shown at higher magnification to depict its size in relation to the mesh. [Please click here to view a larger version of this figure.](#)

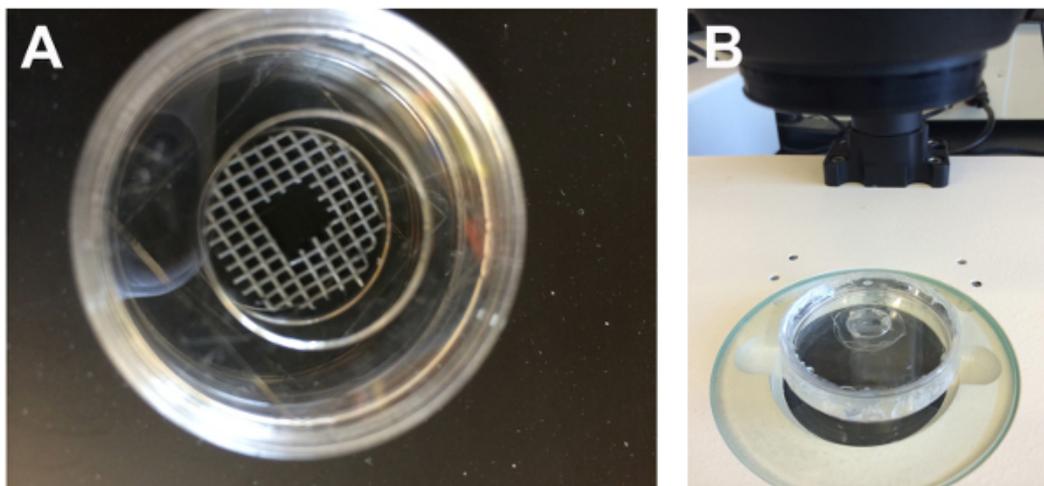


Figure 2. Imaging chamber assembly made from Petri dishes. (A) Shown is a commercial glass top glass bottom petri dish with a plastic mesh attached to the glass coverslip. (B) Shown is a self-constructed Petri dish chamber with a hole drilled into the lid and a coverslip attached from the outside with silicon grease. The mesh and larva are mounted inside the chamber containing Tricaine solution. To seal the chamber, silicon grease is applied to the upper, outer rim of the bottom chamber and the top lid attached. [Please click here to view a larger version of this figure.](#)

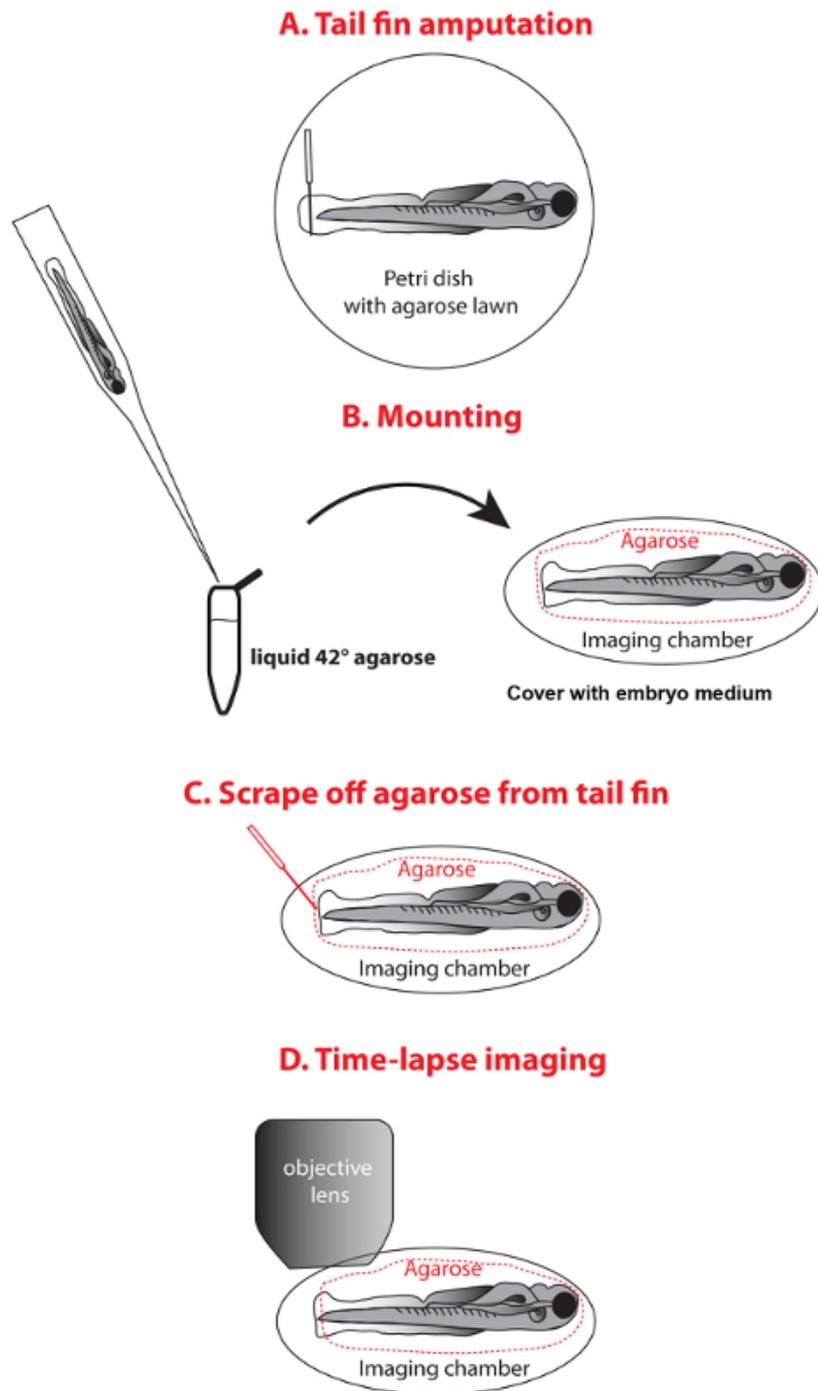


Figure 3. Scheme of amputation and mounting of a larva for imaging. (A) For amputation, place an anesthetized larva onto an agarose-coated Petri dish and amputate the tail fin with a syringe needle. (B) For mounting, transfer the larva with a transfer pipette into a 1.5 ml tube filled with 42 °C liquid agarose and pipette a drop containing the larva into the imaging chamber, orient the fish and cover the solidified agarose with embryo medium. (C) Scrape off the agarose from the tail fin using a capped microloader pipette tip or similar tool and replace embryo medium with fresh medium. (D) Image the tail fin under a stereomicroscope. [Please click here to view a larger version of this figure.](#)

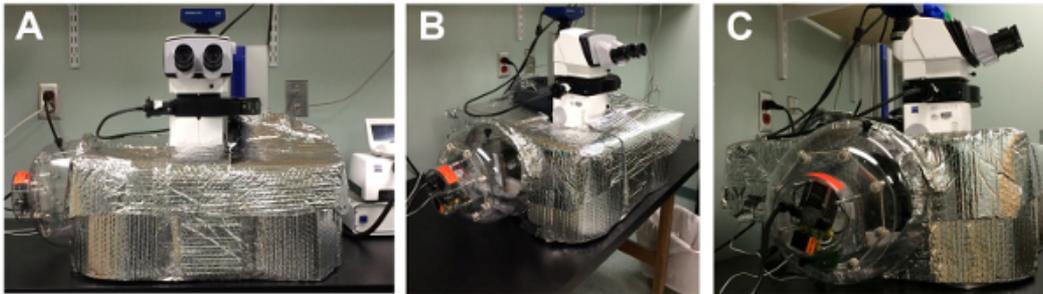


Figure 4. Self-constructed heated incubation chamber. (A-C) Shown is a heated incubation chamber made of cardboard, bubble wrap and Velcro. A wired dome heater (originally designed for chicken egg incubation) is attached to the chamber using aluminum tape. [Please click here to view a larger version of this figure.](#)

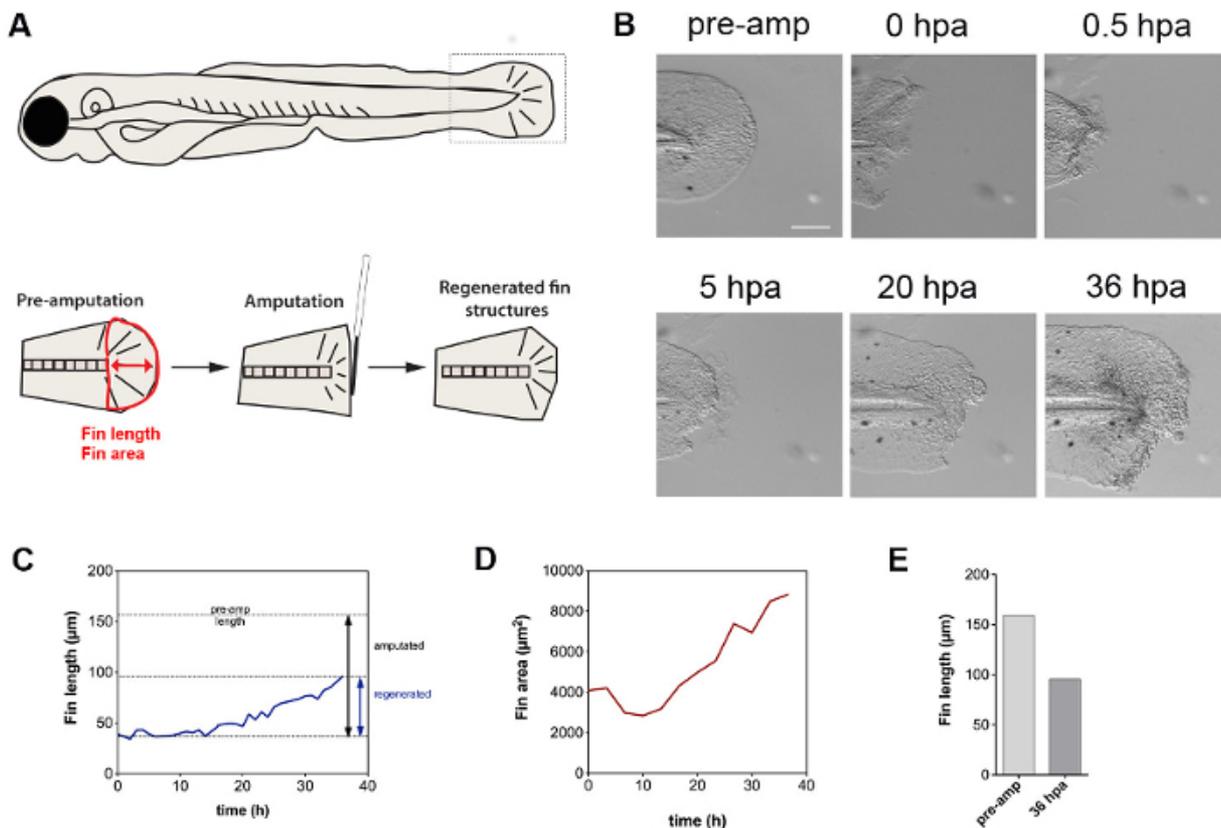


Figure 5. Fin regeneration dynamics. (A) Scheme of the utilized tail fin amputation assay and quantification method to determine the fin length (red arrow) and area (red outline of the fin). (B) Tail fin amputation initially triggers contraction of the fin, followed by regenerative tissue outgrowth. The fin also undergoes developmental growth, as evidenced by the lateral size increase. (C) Shown is the fin length as a function of time, revealing a linear regenerative growth starting at ~14 hpa. (D) Quantification of the fin area reveals an initially decrease in size, which can be attributed to the contraction of the fin. After ~14 hr, the fin size increases at a linear rate. (E) Comparison of the fin length before amputation and after 36 hr shows ~60% regrowth. Scale bar: 100 μm Abbreviations: pre-amp, pre-amputation; hours post amputation, hpa; regen, regeneration; amp, amputation [Please click here to view a larger version of this figure.](#)

Movie. Fin regeneration over the time course of 36 hr. Shown is a tail fin of a 2.5-day old larva during the course of regeneration. Starting 30 min post amputation regenerative growth is imaged in 30 min intervals on a stereomicroscope using a 3.5x objective lens.

Discussion

The presented method allows for observing wound healing and tissue regeneration in living zebrafish larvae with *in vivo* time-lapse imaging on a brightfield stereomicroscope, using a comparatively simple set-up. This procedure requires certain important aspects that we have tested, which will optimize the outcome: 1) Low agarose concentrations (~0.5%) will minimize growth impediments of the continually growing larval zebrafish, 2) Removal of the agarose around the fin is important not to obscure the healing process, 3) Trapping the agarose in a plastic mesh retains the agarose and animal in a stable position throughout the procedure, and 4) A proper temperature-controlled environment, which is essential for larval viability. We have adapted a heated incubation chamber^{23,24}, which utilizes bubble wrap that is taped onto cardboard, and a wired dome heater to control the temperature and proper air circulation with minimal fluctuations during the imaging procedure. This simple and cost-

efficient chamber can be prepared to fit any microscope. A similar heated incubation chamber has been also utilized for imaging mice and chick development^{24,29}.

We suggest that pre-amputated larvae are mounted for a pre-amputation image, demounted for amputation, and remounted for time-lapse imaging. Although it is feasible to perform these steps in a single step in the final imaging chamber, in our experience we found that amputating the tail fin on a glass coverslip is not optimal, as it tears the tissue and does not result in a clean cut. The agarose-based amputation method using a syringe needle was originally described by Kawakami and colleagues (2004)¹⁶ and is also, in our experience, ideal to perform the amputations. Thus, the rather complicated series of steps that we presented is well justified and ensures an optimal regeneration outcome.

We showed that larval zebrafish at 2 dpf can be imaged up to 1.5 days in agarose and Tricaine solution. We used pH-optimized Tricaine (pH7) solution prepared with Instant Ocean salt, which does not interfere with the specimen's health for the presented imaging period. We previously however also demonstrated that using Tricaine in Danieau medium permits time-lapse imaging of 2.5 dpf larval zebrafish on a confocal microscope for at least 2 days³⁰. Thus, optimal buffer conditions can extend larval health and the length of imaging. Alternatively, lower Tricaine concentrations may be used for anesthesia, or 2-phenoxyethanol, which we found is well tolerated during larval and adult stages at 28 °C for at least 60 hr.

To avoid defects in fin regeneration, we removed the agarose from the tail fin prior to imaging. Our data shows that within 1.5 days the fin has regenerated to about 60%. This regeneration rate is consistent with a previous study defining 3 days as an average time for tail fin regeneration in zebrafish larvae up to 6 dpf¹⁶. Alternative methods to agarose could however be utilized to mount the fish for imaging. For example, thin plasma clots³¹ or fluorinated ethylene propylene (FEP) tubes coated with methylcellulose and filled with very low agarose concentrations (0.1%) have been recommended for light sheet microscopy³² and may be suitable for our presented method. However, we do not recommend methylcellulose and 0.1% agarose, as they require that the specimen are mounted at the bottom of the chamber due to the lack of solidification of these media. Very high concentrations of methylcellulose will moreover generate air pockets based on our experience, and these may interfere with the imaging procedure. If these media are preferred with using the bottom chamber, it is important that an appropriate working distance between the objective lens and the specimen is present. It should be noted that methylcellulose as a mounting medium is recommended only for up to 1 day, as it may interfere with larval health³².

Mounting the specimen in the lid may result in a slow gravitational downward drift. It is therefore recommended to image multiple sections at each time point, which can either be projected into a single plane or only images that are in the focal plane may be extracted for assembling the final movie. Imaging the specimen at the bottom chamber could be an alternative methodology to avoid potential downward drift. Plasma clots could be useful to avoid drift, as the plasma will stick to the outer enveloping layer (EVL, periderm)³¹ and therefore may stabilize the specimen. This however needs to be tested, as well as how long larval zebrafish can be maintained in plasma clots without interfering with larval health or fin regeneration.

Our movie was assembled utilizing individual sections (26 µm) of a recorded z-stack, which covered the full thickness of the fin (~10 µm) and which accounted for potential z-drift of the fin during the imaging procedure. In order to retain 3-D information, it is also possible to project z-stacks into single images. Because this may result in blurriness of the image, brightfield deconvolution may be desired. Software, such as Deconvolve or Autoquant X3 could be utilized for this purpose. Alternatively, mathematical algorithms (described in Tadrous³³) can be applied for obtaining a point-spread function of high signal-to-noise ratio (SNR). Obtaining a high SNR represents one of the major hurdles in brightfield deconvolution. Although this method requires high contrast and thin sample thickness, it would be appropriate for imaging of the tail fin due to its reduced width.

A clear advantage of the presented imaging method is that it is rapidly adaptable to any stereomicroscope equipped with a CCD camera and time-lapse software and offers a low-cost alternative to more expensive confocal imaging systems. While this method does not utilize fluorescence for cell detection, it can be extended for such applications by utilizing an automated system for shutter control and post-imaging deconvolution software³⁴. This would enable users to further observe wound repair and regeneration processes with single cell or subcellular resolution over longer time periods.

The optical clarity and ease with which embryonic and larval zebrafish can be handled, and the adaptability of this method to any stereomicroscope makes it suitable for teaching basic vertebrate biology in a classroom setting. This method can provide students with a better understanding of the basic biological processes underlying tissue repair and regeneration. Other biological processes that have been captured with a similar method are zebrafish embryonic development^{23,34} and cardiac function (unpublished). This method also offers the possibility for monitoring wound repair and regeneration in larvae that have been genetically and pharmacologically manipulated.

Disclosures

The authors have nothing to disclose.

Acknowledgements

We thank the MDI Biological Laboratory animal core service facility for zebrafish maintenance. Research reported in this publication was supported by Institutional Development Awards (IDeA) from the National Institute of General Medical Sciences of the National Institutes of Health under grant numbers P20GM104318 (for COBRE) and P20GM103423 (INBRE) and Department of Defense – USAMRAA (W81XWH-BAA-1) grant.

References

- San Miguel-Ruiz, J. E., & García-Arrarás, J. E. Common cellular events occur during wound healing and organ regeneration in the sea cucumber *Holothuria glaberrima*. *BMC Dev Biol.* **7**, 115, doi: 10.1186/1471-213X-7-115 (2007).
- Poss, K. D., Keating, M. T., & Nechiporuk, A. Tales of regeneration in zebrafish. *Dev Dyn.* **226**, 202-210, doi: 10.1002/dvdy.10220 (2003).
- Akimenko, M. A., Mari-Beffa, M., Becerra, J., & Géraudie, J. Old questions, new tools, and some answers to the mystery of fin regeneration. *Dev Dyn.* **226**, 190-201, doi: 10.1002/dvdy.10248 (2003).
- Slack, J. M. Regeneration research today. *Dev Dyn.* **226**, 162-166, doi: 10.1002/dvdy.10232 (2003).
- Seifert, A. W. *et al.* Skin shedding and tissue regeneration in African spiny mice (*Acomys*). *Nature.* **489**, 561-565, doi: 10.1038/nature11499 (2012).
- Goss, R. J., & Grimes, L. N. Epidermal downgrowths in regenerating rabbit ear holes. *J Morphol.* **146**, 533-542, doi: 10.1002/jmor.1051460408 (1975).
- Williams-Boyce, P. K., & Daniel, J. C. Comparison of ear tissue regeneration in mammals. *J Anat.* **149**, 55-63 (1986).
- Allan, C. H. *et al.* Tissue response and *Msx1* expression after human fetal digit tip amputation *in vitro*. *Wound Repair Regen.* **14**, 398-404, doi: 10.1111/j.1743-6109.2006.00139.x (2006).
- Borgens, R. B. Mice regrow the tips of their foretoes. *Science.* **217**, 747-750 (1982).
- Han, M., Yang, X., Lee, J., Allan, C. H., & Muneoka, K. Development and regeneration of the neonatal digit tip in mice. *Dev Biol.* **315**, 125-135, doi: 10.1016/j.ydbio.2007.12.025 (2008).
- Muneoka, K., Allan, C. H., Yang, X., Lee, J., & Han, M. Mammalian regeneration and regenerative medicine. *Birth Defects Res C Embryo Today.* **84**, 265-280, doi: 10.1002/bdrc.20137 (2008).
- Takeo, M. *et al.* Wnt activation in nail epithelium couples nail growth to digit regeneration. *Nature.* **499**, 228-232, doi: 10.1038/nature12214 (2013).
- Akimenko, M. A., Johnson, S. L., Westerfield, M., & Ekker, M. Differential induction of four *msx* homeobox genes during fin development and regeneration in zebrafish. *Development.* **121**, 347-357 (1995).
- Reginelli, A. D., Wang, Y. Q., Sassoon, D., & Muneoka, K. Digit tip regeneration correlates with regions of *Msx1* (*Hox 7*) expression in fetal and newborn mice. *Development.* **121**, 1065-1076 (1995).
- Brien, G. S. *et al.* Coordinate development of skin cells and cutaneous sensory axons in zebrafish. *Journal of Comparative Neurology.* **520**, 816-831, doi: 10.1002/cne.22791 (2012).
- Kawakami, A., Fukazawa, T., & Takeda, H. Early fin primordia of zebrafish larvae regenerate by a similar growth control mechanism with adult regeneration. *Dev Dyn.* **231**, 693-699 (2004).
- Yoshinari, N., Ishida, T., Kudo, A., & Kawakami, A. Gene expression and functional analysis of zebrafish larval fin fold regeneration. *Dev Biol.* **325**, 71-81 (2009).
- Westerfield, M. *The zebrafish book. A guide for the laboratory use of zebrafish (Danio rerio)*. Fourth edn, Univ. of Oregon Press, Eugene (2000).
- Detrich, H. W., Westerfield, M., & Zon, L. I. The zebrafish: cellular and developmental biology, part A. Preface. *Methods Cell Biol.* **100**, xiii, doi: 10.1016/B978-0-12-384892-5.00018-9 (2010).
- Dahm, N.-V. a. *Zebrafish: A Practical Approach, Issue 975*. illustrated, reprint edn, 303 Oxford University Press (2002).
- Detrich, H., Westerfield, M., and Zon, L., Eds. *The Zebrafish: 2nd Edition Genetics, Genomics and Informatics*. 2 edn, (2005).
- Concha, M. L., & Adams, R. J. Oriented cell divisions and cellular morphogenesis in the zebrafish gastrula and neurula: a time-lapse analysis. *Development.* **125**, 983-994 (1998).
- Distel, M., & Köster, R. W. *In vivo* time-lapse imaging of zebrafish embryonic development. *CSH Protoc.* **2007**, pdb.prot4816 (2007).
- Kulesa, P. M., & Kasemeier-Kulesa, J. C. Construction of a Heated Incubation Chamber around a Microscope Stage for Time-Lapse Imaging. *CSH Protoc.* **2007**, pdb.prot4792 (2007).
- Bitplane. *Imaris V 6.1.0 Reference Manual*. (2008).
- Abramoff, M.D., Magalhaes, P.J., & Ram, S.J. *Image Processing with ImageJ*. 36-42 Biophotonics International (2004).
- Schindelin, J. *et al.* Fiji: an open-source platform for biological-image analysis. *Nat Methods.* **9**, 676-682, doi: 10.1038/nmeth.2019 (2012).
- Mateus, R. *et al.* *In vivo* cell and tissue dynamics underlying zebrafish fin fold regeneration. *PLoS One.* **7**, e51766, doi: 10.1371/journal.pone.0051766 (2012).
- Jones, E. A. *et al.* Dynamic *in vivo* imaging of postimplantation mammalian embryos using whole embryo culture. *Genesis.* **34**, 228-235, doi: 10.1002/gene.10162 (2002).
- Rieger, S., Senghaas, N., Walch, A., & Köster, R. W. Cadherin-2 controls directional chain migration of cerebellar granule neurons. *PLoS Biol.* **7**, e1000240, doi: 10.1371/journal.pbio.1000240 (2009).
- Langenberg, T., Brand, M., & Cooper, M. S. Imaging brain development and organogenesis in zebrafish using immobilized embryonic explants. *Dev Dyn.* **228**, 464-474, doi: 10.1002/dvdy.10395 (2003).
- Kaufmann, A., Mickoleit, M., Weber, M., & Huisken, J. Multilayer mounting enables long-term imaging of zebrafish development in a light sheet microscope. *Development.* **139**, 3242-3247, doi: 10.1242/dev.082586 (2012).
- Tadrous, P. J. A method of PSF generation for 3D brightfield deconvolution. *J Microsc.* **237**, 192-199, doi: 10.1111/j.1365-2818.2009.03323.x (2010).
- Distel, M., Babaryka, A., & Köster, R. W. Multicolor *in vivo* time-lapse imaging at cellular resolution by stereomicroscopy. *Dev Dyn.* **235**, 1100-1106 (2006).