Adhesion to a common ECM mediates interdependence in tissue morphogenesis in *Drosophila*

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Abstract

For organs to be functional, the cells and tissues that constitute them must effectively interact with each other and coordinate their behaviours. Halfway during Drosophila embryogenesis, two lateral epidermal sheets stretch to fuse at the dorsal midline; concomitant with this, the main tubes of the respiratory system also shift towards the dorsal side of the embryo. Here, we show that these two processes occur simultaneously and are coordinated by the adhesion of the epidermal sheets and a subset of cells of the tracheal trunks to a common extracellular matrix (ECM) that separates them. We show that during dorsal closure, tracheal trunk cells extend protrusions towards the ECM underneath the epidermis. These protrusions are under tension, suggesting they have a mechanical function. Additionally, perturbing adhesion between tracheal cells and the epidermis affects the development of both tissues. Altogether, our findings uncover a novel mechanism used for tissue coordination during development, one that is based on tissue adhesion towards a common ECM capable of transmitting forces across the embryo.

Introduction

Cells and tissues modify their behaviour according to extracellular signals. Many works have characterised the intracellular mediators and effectors of such signalling pathways and how they influence individual cell behaviour, leading to tissue-wide rearrangements. However, fewer studies have examined how seemingly independent tissues influence each other's behaviours. The development of the *Drosophila* tracheal system is a very useful model for looking at tissue interactions and how they influence cell and tissue remodelling since tracheal tubes branch and migrate over neighbouring tissues of different biochemical and mechanical properties to ventilate them. The variety of properties that tracheal cells are exposed to, suggests that the tracheal system has wide plasticity to adapt to specific environments, making it a useful model to study tissue interactions and how they influence development.

Tracheal development begins with the invagination of lateral tracheal pits. Once inside of the animal, these branch and fuse to form the main tubes of the tracheal system, among which are the dorsal trunks. Dorsal trunks are two multicellular tubes located on each side of the embryo. Smaller tubes sprout from these: dorsal branches grow towards the dorsal midline and ventral branches towards the nervous system and the gut (Hayashi and Kondo, 2018). Despite their name, dorsal trunks are initially formed on the lateral sides of the embryo. Only towards the end of development are they displaced to the dorsal side of the animal. However, how this remodelling takes place has not been studied.

It has been generally assumed that trunks reach the dorsal side of the embryo because of the pulling forces of dorsal branch migration. Dorsal branches migrate dorsally through the action of Branchless, a member of the fibroblast growth factor (FGF) family that is expressed in the dorsal region of the embryo where it acts as a chemoattractant in the tip cells of tracheal branches (Sohr et al., 2019; Sutherland et al., 1996). Trunk displacement temporally coincides with one of the major morphogenetic processes in *Drosophila* embryogenesis, dorsal closure of the epidermis. In this process, the two lateral epidermal sheets of the embryo surface stretch dorsally to meet and fuse at the dorsal midline of the embryo. A major player in this process is the amnioserosa, an epithelium in the dorsal surface bound to the epidermis that dies upon dorsal closure completion. The amnioserosa generates forces that drive its

own contraction and that act on the epidermal cells favouring their stretching (Hayes and Solon, 2017; Pasakarnis et al., 2016).

Here, we show that dorsal trunk displacement is mechanically coupled to the remodelling of the epidermis. Our experiments indicate that a subset of tracheal trunk cells lies near the epidermis, specifically, the ECM-rich tendon cells that mediate muscle attachment to the exoskeleton. Disrupting trachea-epidermis interactions affects the behaviour of both tissues and results in defective development. These results uncover a novel layer of regulation of morphogenesis in the *Drosophila* embryo, where adhesion of adjacent tissues mechanically couples whole-embryo remodelling.

Materials and methods

Fly lines

Fly stocks were kept under standard culture conditions. Experiments were carried out at 25°C or 29°C where stated. *btl-gal4, UAS*-mCD4::mIFP*; btl-gal4, UAS*-PH::mCherry*; btl-gal4, UAS*-Utrophin::GFP*,* and *Collagen-IV::GFP* were used before and come from Maria Leptin's Lab at EMBL, Heidelberg (Ríos-Barrera and Leptin, 2022; Ríos-Barrera et al., 2017). *en-gal4, UAS*-GFP is from Tobias Troost, Heinrich Heine University Düsseldorf, Germany; *sqh*-Gap43::mCherry is from Thomas Lecuit lab, IBDM, France (Rauzi et al., 2010); *kay*^{1} is a gift from Juan Riesgo-Escovar, UNAM, Mexico (Riesgo-Escovar and Hafen, 1997). Insertion of GFP at the *mys* locus (β-Integrin::GFP) was a kind gift from Nick Brown, MRC, UK (Klapholz et al., 2015). *UAS*-CD8::GFP is from Fanis Missirlis, CINVESTAV, Mexico. The following lines come from Bloomington Drosophila Stock Center: *sr-gal4* (#26663); *UAS-mys* TRiP (#27735); *en-gal4, UAS-nls::mCherry* (#38420); *UAS-NsImb-vhhGFP4* (#38421); *UAS-Mmp1.f2* (#58702); *UAS-Mmp2* (#58705); insertion of GFP at the *shg* locus (E-Cadherin::GFP, #60584); and *btl-moeRFP* (#64233). *UAS-Talin* RNAi comes from the Vienna Drosophila Resource Center (#40399).

Immunostainings

Immunostainings were carried out using standard methods. Embryos were dechorionated using 50% commercial bleach for 30 seconds, then washed with water and transferred to heptane. Afterward, embryos were fixed in 4% PFA for 25 minutes. For devitellinization, PFA was replaced with methanol, and vials were vigorously shaken for 1 minute. We rehydrated the embryos using PBS Triton X-100 0.3% and washed them three times. We then blocked using the washing solution and BSA 1%, and incubated overnight with the corresponding antibody at 4°C. The next day, we did three 10 min washes and incubated with secondary antibodies for 1 hour. These were then washed three times for 10 minutes each, and finally, we mounted them using Vectashield with DAPI. The antibodies used were: anti-GFP coupled to FITC (Abcam #6222, 1:500); anti-βIntegrin (Developmental Studies Hybridoma Bank [DSHB] #CF.6G11, 1:20); and Goat anti-mouse IgG coupled to Alexa647 (Thermofisher #A21235, 1:300). Samples were analysed in a Nikon A1R+ confocal microscope using a resonant scanner and a 60x/1.2 NA water objective and processed using Fiji (Schindelin et al., 2012).

Live microscopy

Embryos were dechorionated as above, washed with water and transferred to an agar plate to manually select the embryos of the desired stages. For confocal live imaging, embryos were glued to glass-bottom dishes using heptane glue and covered with halocarbon oil. Samples were analysed in

a Nikon A1R+ confocal microscope using a resonant scanner and a 60x/1.2 NA water objective and processed using Fiji (Schindelin et al., 2012). Z-step size was 0.213 μm.

For light-sheet imaging, dechorionated embryos were mounted in low-melting temperature agarose within a glass capillary. The gel cylinder containing the embryo was then pushed above the glass rim to have optical access to it. The position of the embryo was then fixed, and the glass capillary was placed inside the PBS-filled imaging chamber of a homebuilt MuVi-SPIM. 3D volumes were acquired as described previously (Caroti et al., 2018; Medeiros et al., 2015), using two 20x/1.0 NA water immersion detection objectives (Olympus XLUMPLFLN20XW) for fluorescence detection and two 10x/0.3 NA water immersion objectives (Nikon CFI Plan Fluor 10X W) for excitation. Images were processed in Fiji (Schindelin et al., 2012). The time resolution was 2 minutes per frame and the z-step was 1.5 μm.

Laser micro-dissection

Embryos were processed for live microscopy as above. Laser micro-dissection experiments were carried out as detailed previously (Sánchez-Cisneros et al., 2023). We used a Zeiss LSM 780 microscope, using a 63x/1.4NA oil objective. We used a femtosecond-pulsed two-photon laser at 950 nm, 75% (1540 mW) power. Micro-dissection was done using a single cycle on a single 1 μm thick z-stack.

Larval fixations

Third-instar larvae were heat-fixed by transferring them to halocarbon oil in a cover slide and a heating plate at 65°C for 15 seconds. Afterward, the larvae were oriented with the dorsal side down. Larvae were analysed in a Nikon A1R+ confocal microscope using a resonant scanner in mosaic mode and a 10x/0.25 NA air objective and processed using Fiji.

Electron microscopy

Embryos were processed according to the method of Tepass & Harstenstein (Tepass and Hartenstein, 1994). After dechorionation, the embryos were fixed in a glutaraldehyde 2% solution and heptane in a relation of 2:8 for 17 min in agitation. After fixed, we withdrew the vitelline membrane manually with insulin needles. Embryos were fixed in 2.5% glutaraldehyde in cacodylate solution (50mM, pH 7.2) and post-fixed in osmium tetroxide solution (2%). After fixations, the embryos were gradually dehydrated with alcohol solutions (70, 80, 90, 95, 100%) and acetone. The dehydrated embryos were incubated in acetone/Epon solution overnight and then were included in Epon solution for 2 days. Afterward, 1 μm semi-thin sections were stained with Methylene blue and visualized under a conventional light microscope to identify the regions of interest. Once these were identified, we did

ultrafine (50-100 nm) sections and analysed them using transmission electron microscopy. This was carried out using a JEOL JEM 1200 EXII electron microscope. Feature tracing was done in Inkscape.

Image processing and analysis

Except for larval measurements, image processing and analyses were performed using Fiji. We registered all time-lapse micrographs using the 'Correct 3D drift' function. Cartographical projections of the embryo's surface were done using a custom-made Fiji macro that extracts the curvature of the embryo and reslices over 16 z planes throughout the time-lapse. To measure tissue coordination, we used particle image velocimetry (PIV) using the Fiji plugin described in (Tseng et al., 2012), with an interrogation window size of 128x128 px (6.25% of the image) with an overlap of 64 px, and a correlation threshold of 0.60. Cross-correlation analyses were done using a custom-made R code, using the average of magnitudes in the y-axis generated by the PIV plugin.

Embryonic dorsal trunk length measurements were done manually in Fiji, and these were normalised to the minimum distance between the initial and final measurement points. Larval dorsal trunk measurements were normalized in a similar way, but these were done in 3D using Imaris under a trial license. Rates of dorsal closure progression were measured using the Manual Tracking plugin from Fiji, following the signal corresponding to the leading edge for the epidermis, and a dorsal edge of the tracheal trunk. In both cases, we followed the same segment corresponding to the center of the amnioserosa (Abdominal segments 3-4). We plotted displacement in the y axis, normalizing the data with the initial and final time points set to 0 and 100, respectively. To measure signal intensity in the deGradFP experiments, we used the 'plot profile' function in Fiji. Then, we calculated the correlation coefficient between the signal from the driver (*btl*>CD4::IFP or *en*>nls::mCherry) with respect to the signal of the target protein, β-integrin::GFP. All data were plotted using ggplot2 in R Studio or Prism. For statistical analyses, data normal distribution was tested in Prism.

Results

The epidermis and tracheal trunks displace dorsally at the same time during embryogenesis.

Halfway in development, tracheal trunks, which originally form at the sides of the embryo, are displaced dorsally acquiring their final position. Simultaneously with trunk displacement, dorsal branches elongate in response to FGF (Figure 1A-A'', Video S1). Only the tip cells of the tracheal system respond to FGF. It is generally assumed that trunk displacement occurs due to tip cells pulling on the trunks so that they follow their path dorsally. However, in addition to dorsal branches, the trunk is also subject to pulling forces in the opposite direction, since tip cells in ganglionic and visceral branches migrate at the same time to reach the gut and the nervous system (Figure 1B-B'', Video S1). With two opposing forces pulling the trunks other factors likely participate in their dorsal displacement, but so far these have remained unstudied.

Tracheal trunk displacement occurs at similar stages as epidermal dorsal closure, so we wondered if these two processes could be actively coordinated, and if the epidermis could participate in trunk displacement. To study this, we first carried out double-tissue labelling and live imaging using a line that expresses the Moesin actin-binding domain fused to a red fluorescent protein (Moe::RFP) under direct control of the tracheal-specific promoter of *btl* (*btl*-Moe::RFP). We used the Gal4/*UAS* system to label spaced epidermal stripes using GFP (*en*>GFP). In these recordings, we confirmed that trunk displacement coincided with epidermal dorsal closure (Figure 1D-D''). We quantified this observation by doing PIV analyses over consecutive timepoints for each of the tissues, followed by cross-correlation analysis of both tissue displacements. We found a very strong positive correlation (Figure 1E), suggesting that epidermal dorsal closure and trunk displacement are actively coordinated.

The dorsal displacements of the trunks and the epidermis could be controlled by extracellular signals that act on both tissues. However, while signals from the epidermis participate in tracheal development, these act mainly in fate determination or tip cell migration (Sánchez-Cisneros et al., 2025). Similarly, the signals that regulate epidermal dorsal closure do not participate in tracheal development, or vice versa (Letizia et al., 2023; Reichman-Fried et al., 1994). Therefore, we hypothesized that coordinated displacement could be mediated by

adhesion, as the tracheal system lies beneath the epidermis. Tissue adhesion could be mediated by an intermediate extracellular matrix (ECM) layer. To explore this, we did time-lapse microscopy of the tracheal system together with an ECM marker, an endogenous insertion of GFP to the *viking* locus, which codes for Collagen IV α2 subunit (from now on referred to as Col IV::GFP, Figure 1F-F''). Col IV::GFP was enriched surrounding cardioblasts, haemocytes, and at muscle attachment sites as reported previously (Borchiellini et al., 1996). Similar to what we found with the epidermal marker, the Col IV::GFP signal displacements and the tracheal reporter had a strong positive correlation (Figure 1G).

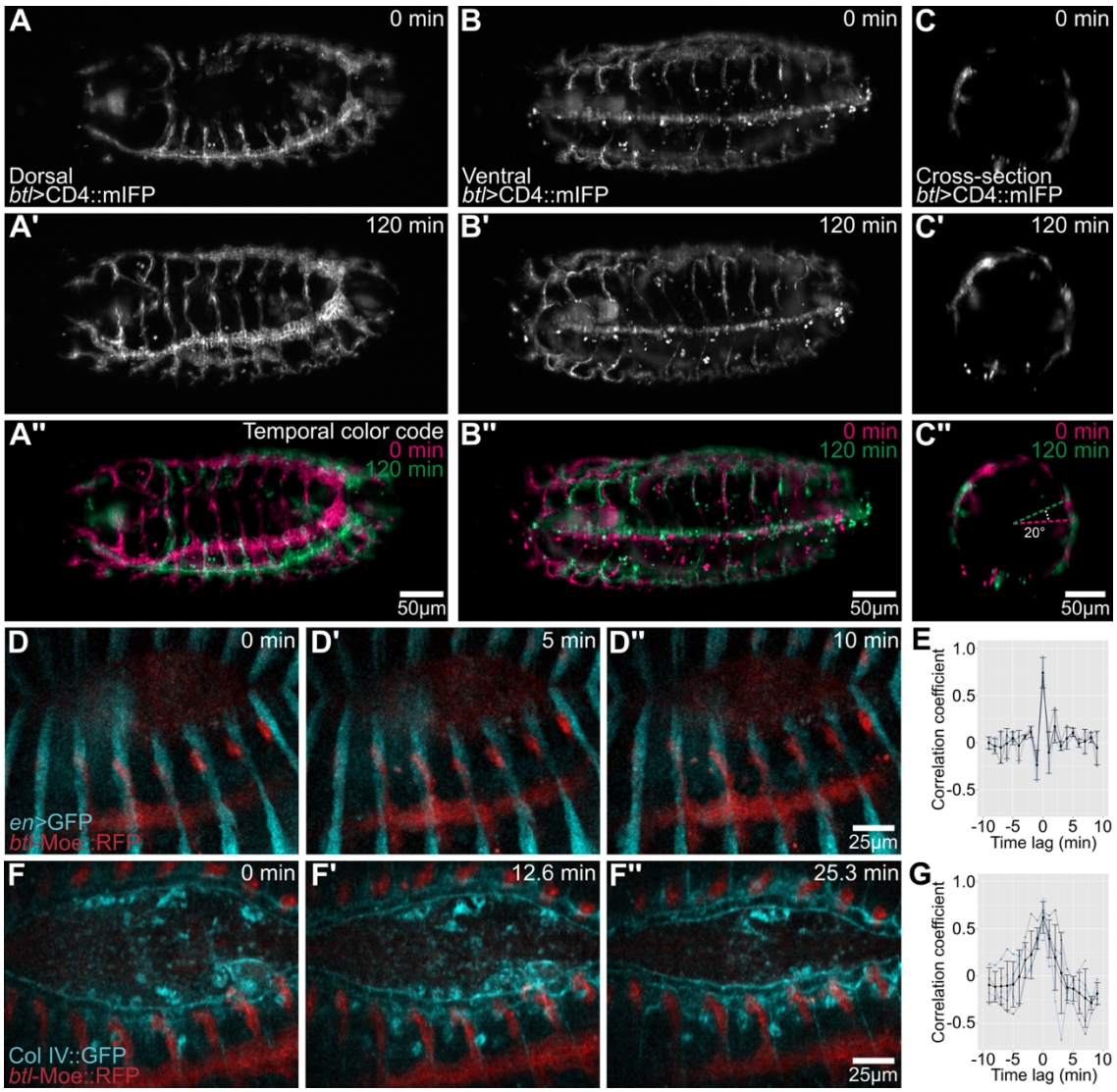

Figure 1. Coordination analyses of tracheal, epidermal and ECM movements during dorsal closure stages. (A – C') Time-lapse imaging of tracheal development using MuVi-SPIM. The tracheal system is marked with *btl*>CD4::mIFP. (A – A') Dorsal view; (B – B') Ventral view; (C – C') Transversal view. (A'' – C'') Overposition of the initial (pink) and final (green) imaging time-point of the movie. (D – D'') Cartographical maximum intensity projection of confocal stacks showing stripes of epidermis using *en*>GFP and the tracheal system using *btl*-Moe::RFP. (E) Cross-correlation analysis of the signal displacement of both markers throughout time. Plot shows means +/- SD. n=3. (F – F'') Cartographical maximum intensity projection of confocal stacks showing Collagen IV::GFP and the tracheal system using *btl*-Moe::RFP. (G) Cross-correlation analysis of the signal displacement of both markers throughout time. Plot shows means +/- SD, n=5.

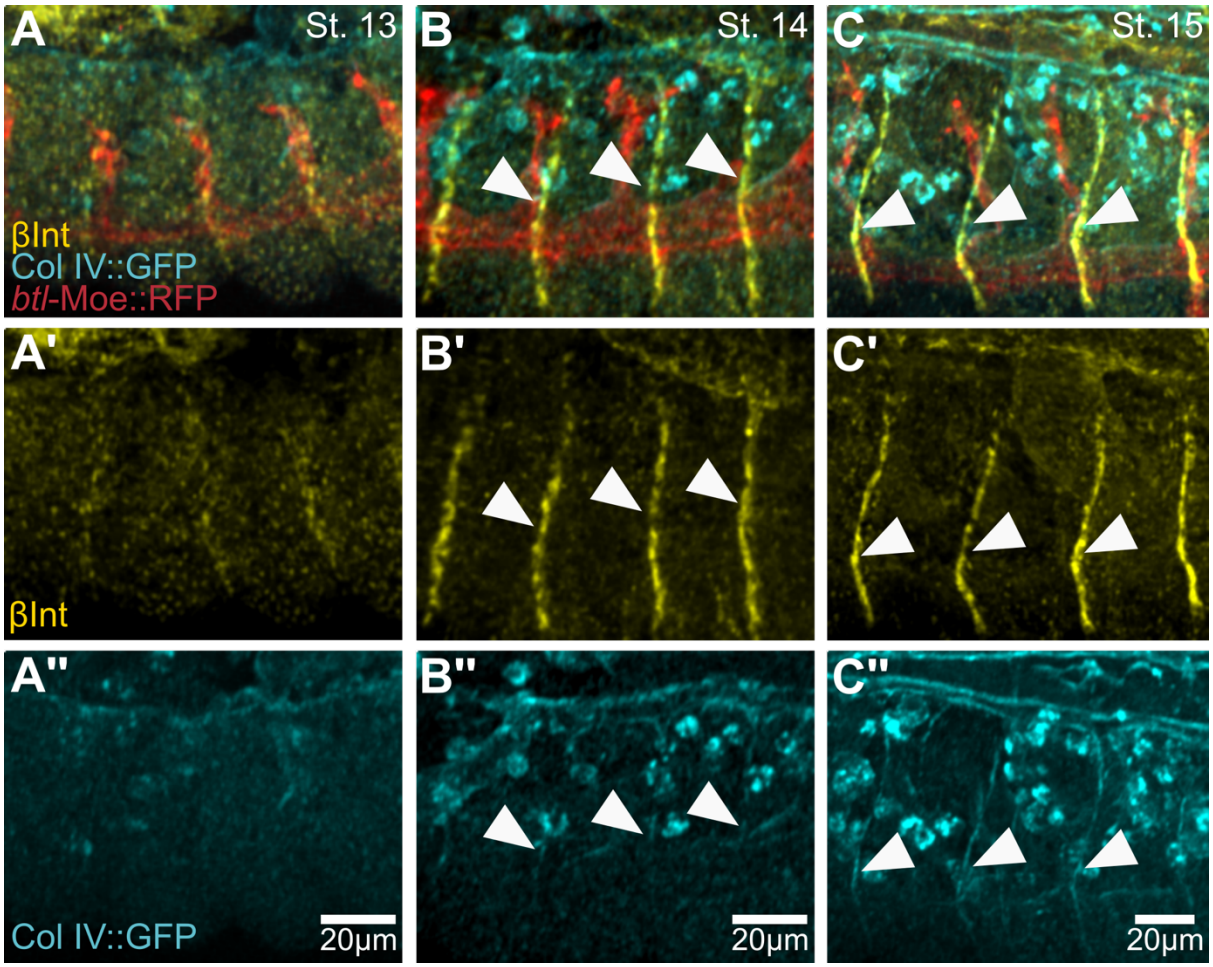

Figure S1. Distribution of Collagen IV::GFP and β-Integrin in stage 13-15 embryos. β-integrin (yellow) and Collagen IV::GFP stained for GFP (cyan) in relation to the tracheal system labelled with *btl*-MoeRFP (red). (A-A') Stage 13; (B-B') Stage 14; (C-C') Stage 15. Arrowheads point to muscle attachment sites at their intersection with tracheal trunks.

The tracheal trunk cells contact tendon cells of the epidermis during dorsal closure.

Since we could not detect a particular enrichment of Col IV::GFP around the trunks using live imaging, we stained embryos against β-integrin and against GFP to detect Col IV::GFP. We could not detect any enrichment for these components at the tracheal trunks during the stages of dorsal closure but only after this process is completed, as had been reported already [Figure S1; (Klußmann-Fricke et al., 2022)]. Instead, at the onset of dorsal closure, Col IV::GFP and β-integrin were only detectable at muscle attachment sites (Figure S1B-B'').

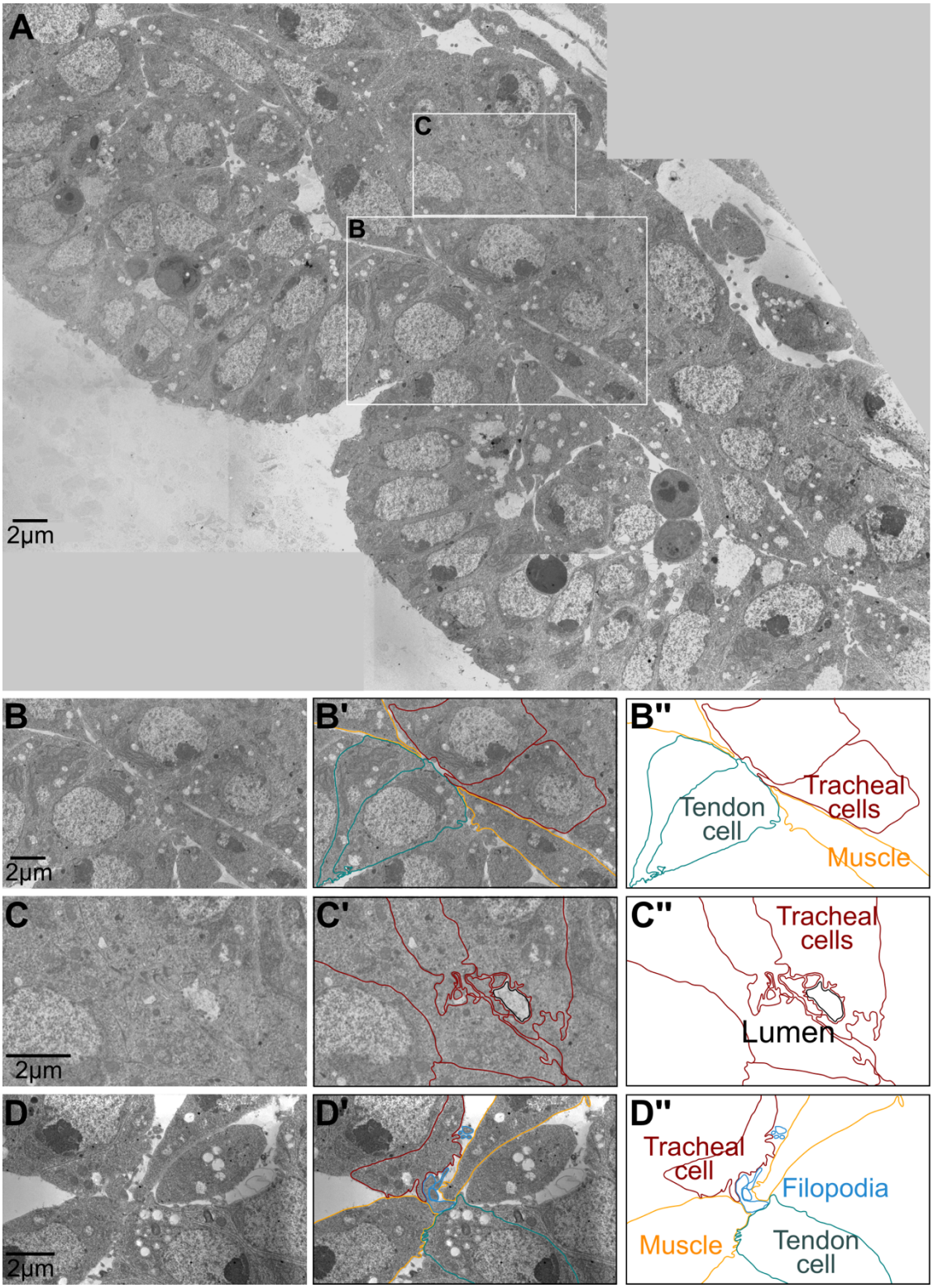

Figure 2. Contact points between the epidermis and the tracheal trunks. (A) Electron micrograph of a longitudinal section of the dorsal trunk, stage 15 embryo. The regions indicated by the white squares are magnified in (B-C). (B) Magnification of a contact point between tracheal trunks and a tendon cell. (B', B'') Manual tracings of the elements present in (B). (C) Magnification of a region of the tracheal trunk where the lumen is visible. (C', C'') Manual tracings of the elements present in (C). (D) Electron micrograph of tracheal cells in proximity to tendon cells and muscles. (D', D'') Manual tracings of the elements present in (D).

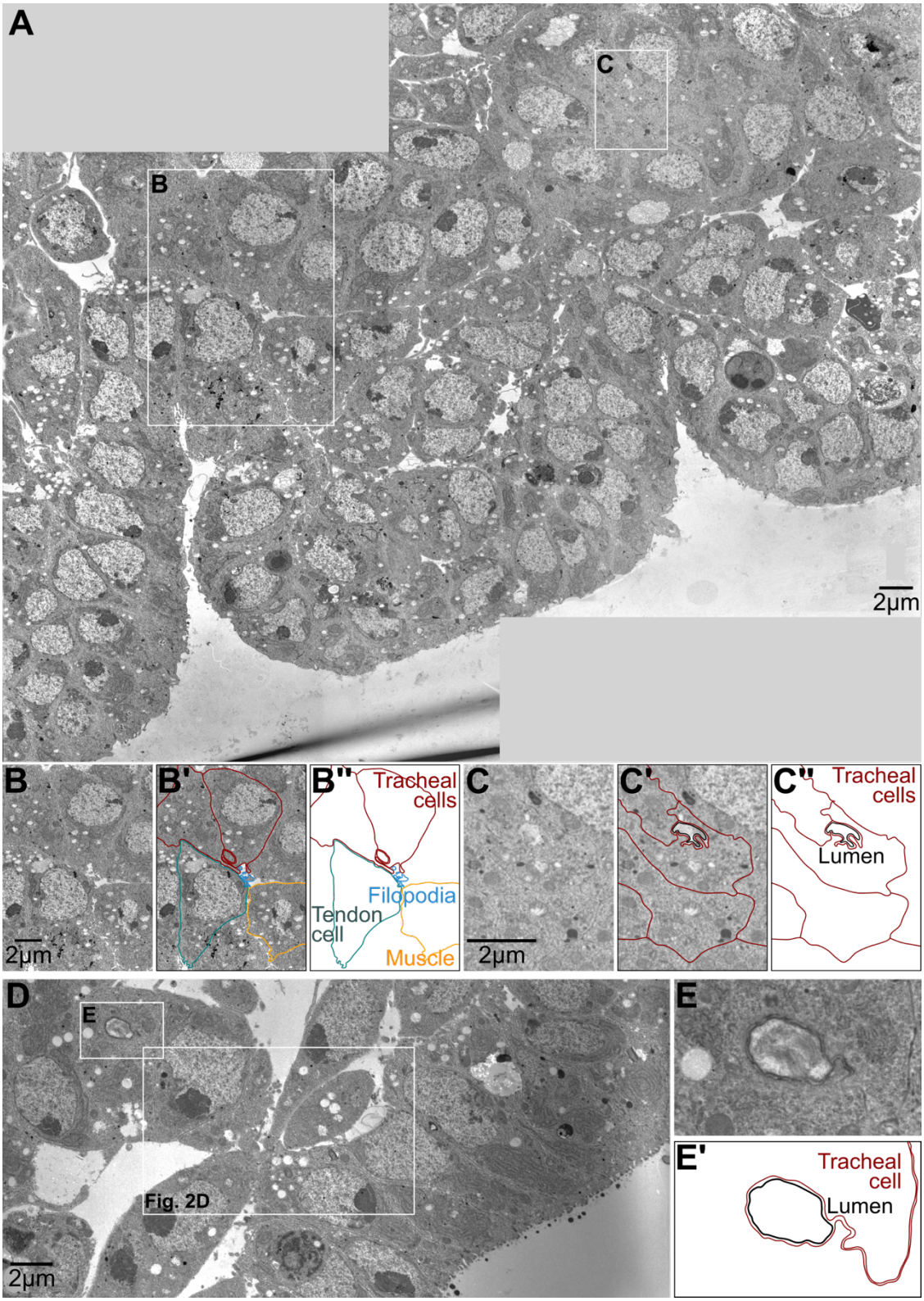

Figure S2. Contact points between the epidermis and the tracheal trunks (other examples). (A) Electron micrograph of a longitudinal section of the dorsal trunk, stage 15 embryo. The regions indicated by the white squares are magnified in (B-C). (B) Magnification of a contact point between tracheal trunks and a tendon cell. (B', B'') Manual tracings of the elements present in (B). (C) Magnification of a region of the tracheal trunk where the lumen is visible. (C', C'') Manual tracings of the elements present in (C). (D) Overview image of the micrograph magnified in Figure 2D. (E) Magnified view of the tracheal lumen illustrated in (D). (E') Manual tracings of the elements present in (E).

Muscles attach to epidermal tendon cells through a dense meshwork of ECM; we figured that tracheal cells could also interact with the epidermis at these points. Classical anatomical descriptions of the embryo report that muscles separate the tracheal trunks from the epidermis (Hartenstein, 1993). Nevertheless, we decided to obtain higher resolution images of these regions by using transmission electron microscopy. With this approach, we observed that as reported, the dorsal trunks and the epidermis are separated by muscles and other mesenchymal cells in most parts of the embryo (Figure 2A, S2A). However, in the regions where tendon cells are found, trunks are no longer separated from the epidermis by other tissues but are instead found adjacent to the epidermis (Figure 2B-B'', S2B-B''). Tendon cells are easily recognizable in these images as they localise at the segmental borders and associate to muscles. We identified tracheal cells based on their epithelial morphology and the presence of a lumen within the epithelium (Figure. 2C-C'', S2C-E'). These experiments revealed a novel anatomical contact between the epidermis and tracheal trunks that had not been previously reported and whose relevance had not been studied up to now.

A subpopulation of dorsal trunk cells forms tensile protrusions towards the epidermis. Electron micrographs also suggested high filopodial activity at the interphase between tendon cells and tracheal trunks (Figure 2D, S2B). Muscles extend filopodia as they attach to tendon cells (Maartens and Brown, 2015), leading us to examine whether tracheal cells could also form filopodia to make contact with tendon cells. Time lapse imaging using MuVI-SPIM already suggested protrusive activity at tracheal trunks during dorsal closure stages (Figure 3A, Video S2). We performed high resolution live imaging of dorsal trunk cells to better visualize this protrusive behaviour. For this, we used *btl*>CD4::IFP to label the tracheal system and a ubiquitously expressed membrane marker, *sqh*-Gap43::mCherry to label the epidermis. In time-lapse movies, we observed that trunk cells form protrusions in the direction of the epidermis (Figure 3B, Video S3). These protrusions persist throughout epidermal dorsal closure and are consistent with the attachment points observed through electron microscopy. Notably, the cells that form protrusions have a very regular distribution, as they are evenly distributed at the trunks, near the base of each dorsal branch (Figure 3A, B, Video S2). These same positions are where tracheal trunks intersect with muscle attachment sites (Figure 1F, S1B). We propose that this subset of tracheal cells mediate the interaction between the trunks and tendon cells, and because of their morphology, we will refer to them as protruding cells.

If protruding cells adhere to the ECM of muscle-attachment sites, we hypothesised that they should be under tension. We tested this by doing laser micro-dissection directly over these structures and measuring the resulting recoil speed using PIV analyses. We found that seconds after doing the laser cut there was a significant recoil in the direction of the trunk (Figure 3C, E; Video S4), which was comparable to the recoil seen upon laser micro-dissection of tracheal tip cells [Figure 3D, E, Video S4; (Ríos-Barrera and Leptin, 2022)]. These results show that protruding cells are under tension, likely due to their role as mediators of trachea-epidermis interactions.

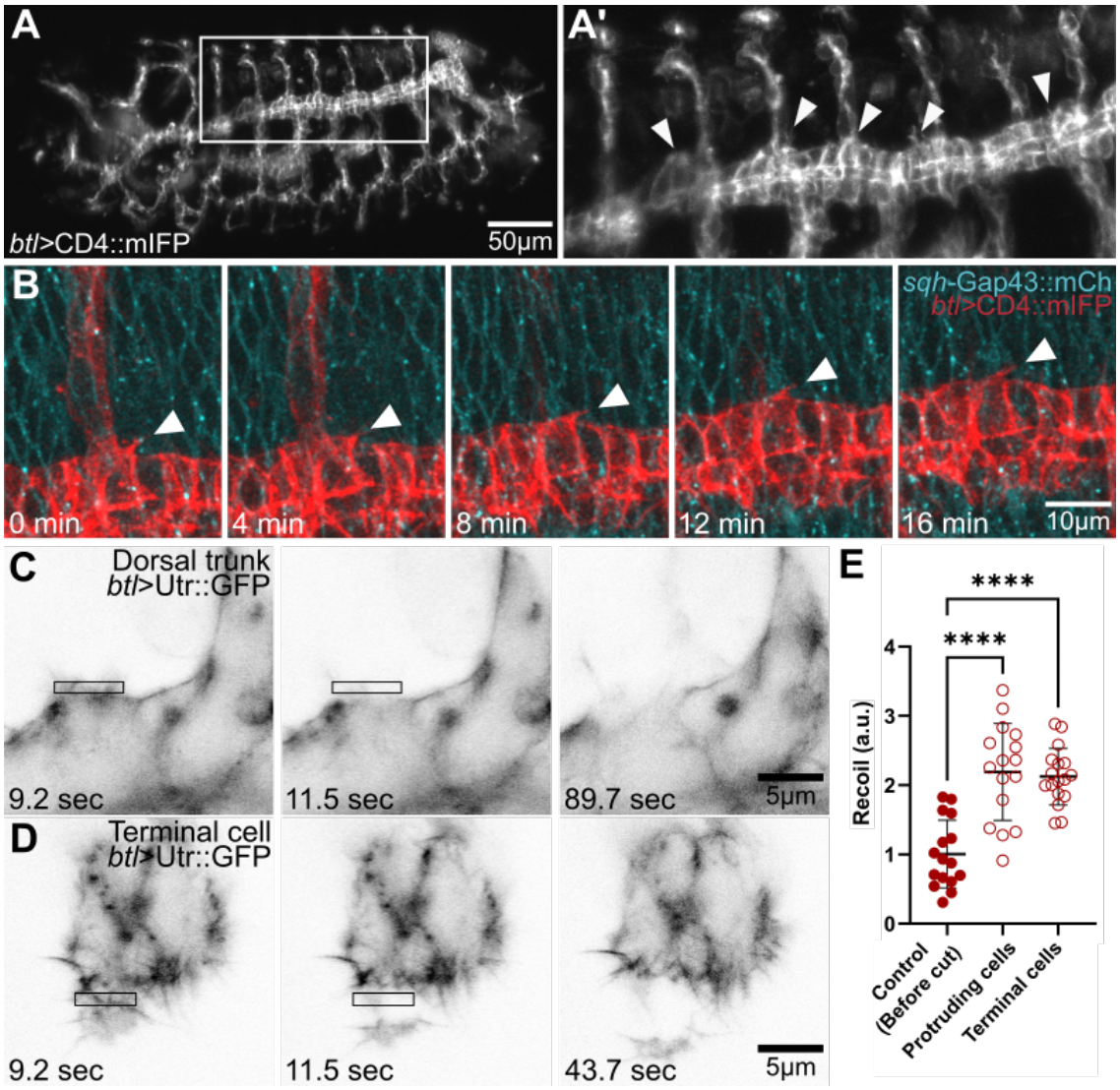

Figure 3. Live imaging of protruding cells and laser micro-dissection experiments. (A-A') Time-lapse imaging of an embryo expressing CD4::mIFP under *btl-gal4* and imaged using MuVi-SPIM. (B) Time-lapse imaging of an embryo expressing CD4::mIFP under *btl-gal4* and (red) and Gap43::mCherry under direct control the *sqh* promoter (cyan) and imaged using confocal microscopy. Arrowheads in (A-B) point to protruding cells of the dorsal trunks. (C) Laser cut in the protruding cells of the dorsal trunk expressing *btl*>Utr::GFP. (D) Laser cuts in the terminal cells of the dorsal branch expressing *btl*>Utr::GFP. Laser cuts were done in the regions marked with black squares in (C – D). (E) Quantification of the recoil using PIV analysis. Plot shows means +/- SD. Control, n=16; Protruding cells, n=16; Terminal cells, n=17. Significance was determined using ANOVA and Dunnett correction for multiple comparisons. ****p<0.0001.

Integrin-ECM adhesion complexes participate in the interaction between dorsal trunks and the epidermis.

To test the relevance of the adhesion between protruding cells and muscle-attachment sites, we combined *btl*-Moe::RFP to follow the tracheal system, together with a tendon cell driver (*stripe; sr-gal4*) that allowed us to visualise these cells and carry out ECM perturbation experiments. We over-expressed matrix metalloproteinases (Mmp1 and Mmp2), enzymes that degrade ECM components (Jia et al., 2014; Llano et al., 2002), and observed their effect on tracheal development. In control embryos we observed evenly distributed stripes of tendon cells across the anterior-posterior axis of the embryo that traversed perpendicularly the tracheal trunks (Figure 4A – A'). In embryos where we over-expressed Mmp1 or Mmp2 in tendon cells, the general organization of tendon cells remained the same and tracheal trunks were still displaced dorsally; however, they developed tortuosities over time, a morphology that is not seen in control embryos of similar developmental stages (Figure 4B-C). Consequently, experimental embryos showed significantly longer dorsal trunks compared to controls (Figure 4D). As Mmp2 is a membrane-anchored protein (Llano et al., 2002) its protease activity is limited to the ECM adjacent to the tendon cells, reducing the possibility that Mmp2 expressed by the tendon cells could have a direct effect on tracheal trunks by diffusing through the extracellular space.

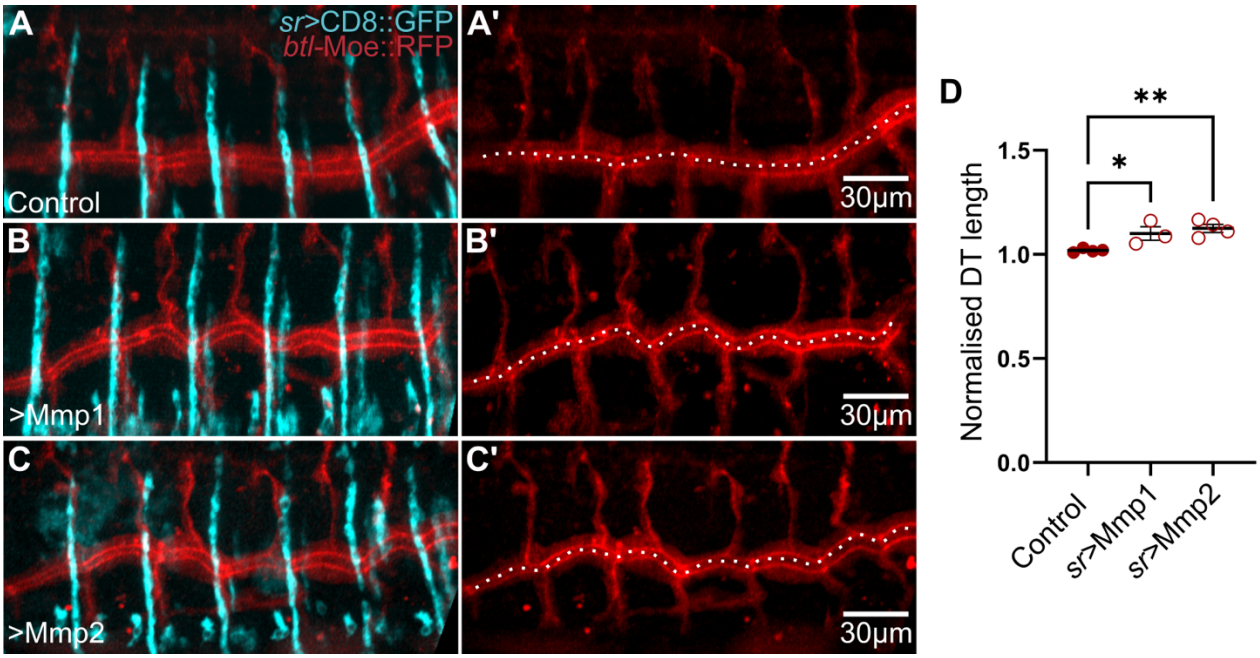

Figure 4. Effect of over-expressing MMPs in tendon cells on tracheal trunk morphology. (A-C) Live-imaging of embryos expressing CD8::GFP under the tendon cell driver *sr-gal4* (cyan) and *btl*-Moe::RFP (tracheal system, red). (A) Control; (B) Embryo additionally over-expressing *UAS*-Mmp1; (C) Embryo additionally over-expressing *UAS*-Mmp2. (D) Normalised dorsal trunk (DT) length. Plot shows means +/- SD. Control, n=4; *sr*>Mmp1, n=3; *sr*>Mmp2, n=4. Significance was determined using ANOVA and Dunnett correction for multiple comparisons. *p<0.05, **p<0.01.

We wondered if the tracheal defects caused by Mmp2 expression in tendon cells would persist in later stages of development, so we looked at tracheal trunks in third instar larvae. We found a similar phenotype to the one we observed during embryogenesis, with dorsal trunks acquiring a wavy phenotype resulting in significantly longer trunks compared to controls (Figure S3A, C). These results show that perturbing the ECM that lies below the tendon cells influences tracheal trunk morphology.

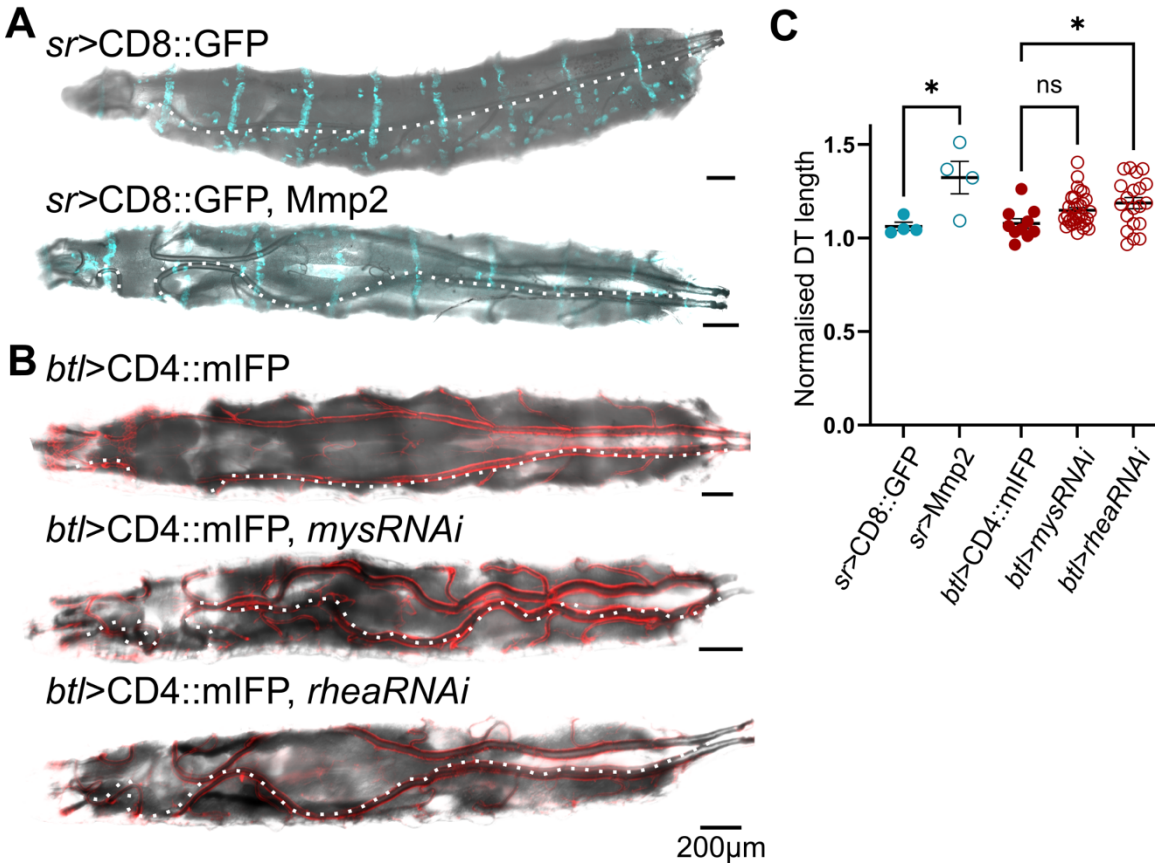

Figure S3. The role of adhesion complexes and ECM in larval dorsal trunk morphology. (A-B) Overlay of maximum-intensity projections of fluorescent reporters and minimum-intensity projections of reflected light images (for bright field) of heat-fixed third instar larvae. (A) Larvae expressing GFP under *sr-gal4*. Top panel, control; bottom panel, over-expression of Mmp2. (B) Larvae expressing CD4::mIFP under *btl-gal4*. Top panel, control; middle panel, *mys* (which codes for β-integrin) RNAi; bottom panel, *rhea* (which codes for Talin) RNAi. (C) Normalised dorsal trunk (DT) length. Plot shows means +/- SD. *sr*>CD8::GFP, n=4; *sr*>Mmp2, n=4; *btl>CD4::mIFP*, n=10 ; *btl>mysRNAi*, n=33 ; *btl>rheaRNAi*, n=20 . Significance was determined using ANOVA and Kruskal-Wallis test for multiple comparisons. *p<0.05.

To determine the role of the interaction between the tracheal system and the ECM in dorsal trunk development, we removed β-Integrin specifically in the tracheal system. Studies on integrins in the embryonic tracheal system have been hampered by abundant maternal contribution that may obscure early phenotypes, and/or by the early lethality of germ-line clones for mutations on components of the integrin complex (Brown, 1994; Wieschaus and

Noell, 1986). Therefore, we used the deGradFP system that targets GFP-labelled proteins to proteasomal degradation. This is achieved by expressing UAS-NsImb::Vhh4, a modified ubiquitin ligase (NsImb) fused to a nanobody against GFP (Vhh4) that can be expressed under any particular driver (Caussinus et al., 2011). For these experiments, we targeted an endogenous insertion of GFP in the gene *mys*, that codes β-Integrin (Klapholz et al., 2015). β-Integrin::GFP also allowed us to visualise the epidermis and other tissues in time-lapse experiments. Since β-Integrin::GFP is a transmembrane protein, it was possible that it could be unsensitive to proteasomal degradation using deGradFP. We tested this with *en-gal4* in stage 12-13 embryos and found that the β-Integrin::GFP signal significantly decreased in the regions were *en-gal4* was expressed (Figure S4A-B, E). Testing this using *btl-gal4* was technically more challenging, since by the time *btl-gal4* is active, the β-Integrin::GFP signal is more abundant in other tissues. Nevertheless, we also found a significant decrease in the GFP intensity (Figure S4CD, F).

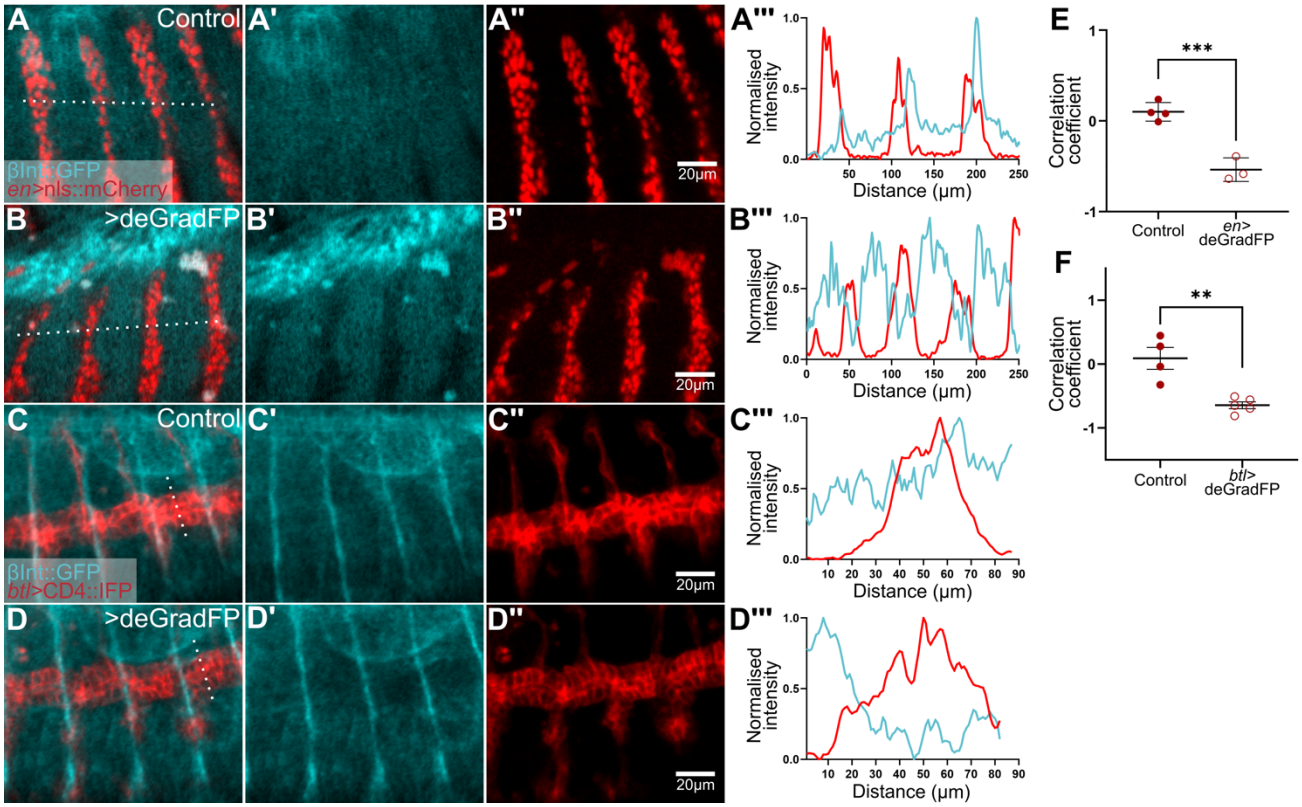

Figure S4. Effect of deGradFP expression on β-Integrin::GFP fluorescence intensity. (A-D) Maximum intensity projections of embryos expressing β-Integrin::GFP (cyan). (A-B) Embryos additionally expressing nls::mCherry (red) under *en-gal4*. (C-D) Embryos additionally expressing CD4::mIFP under *btl-gal4*. (A, C) Controls; (B, D) deGradFP over-expression. (A'''-D''') Fluorescence intensity profiles across the dotted white lines drawn in (A-D). (E-F) Correlation analyses of fluorescence intensity profiles comparing signals from the *en*>deGradFP (E; Control, n=4; *en*>deGradFP, n=3) and the *btl*>deGradFP (F; Control, n=4; *btl*>deGradFP, n=5) experiments. Plots show means +/- SD. Significance was determined using t-tests. ***p>0.001, **p>0.01.

When doing time-lapse microscopy, in control embryos we observed that tracheal trunks move to the dorsal side of the embryo in coordination with the epidermis, as determined by cross-correlation analyses (Figure 5A-C, G, Video S5, left). When we degraded β-Integrin::GFP in the tracheal system, as in our Mmp experiments, we observed coordinated tissue displacement (Figure 5D-G, Video S5, right) and tracheal trunks also showed a wavy lumen phenotype, resulting in significantly longer trunks (Figure 5H). We corroborated these results in third instar larvae using RNAi against *mys* and *rhea* (which code for β-Integrin and its intracellular mediator Talin, respectively). As with Mmp2 over-expression using *sr-gal4*, silencing these genes under *btl-gal4* resulted in wavy dorsal trunks (Figure S3B-C). These results show that cell-ECM interactions are important for proper trunk morphology, but not for its displacement.

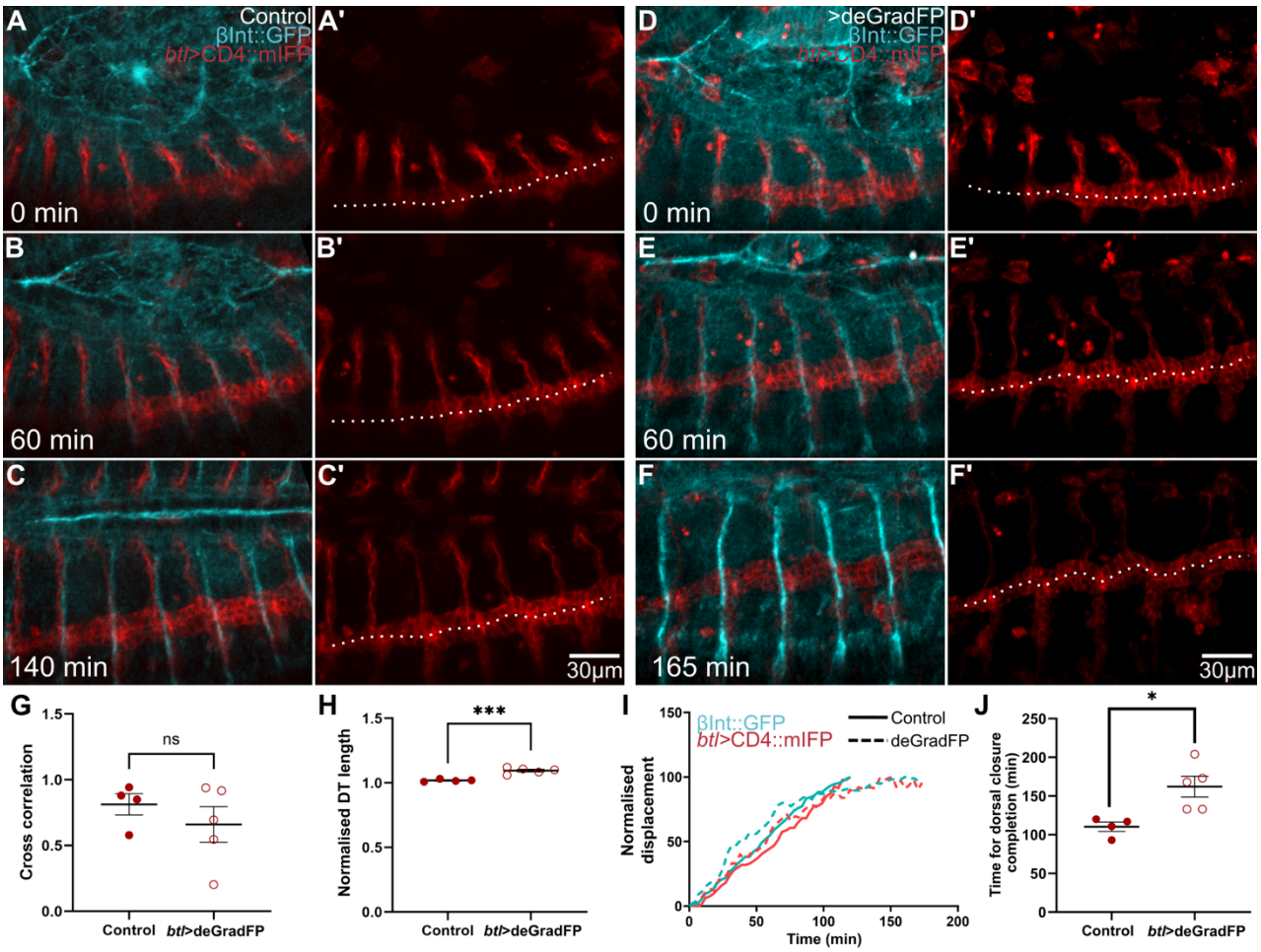

Figure 5. Effect of deGradFP expression in tracheal morphology of β-Integrin::GFP embryos. (A-F) Maximum intensity projections and time-lapse imaging of embryos expressing β-integrin::GFP (cyan) and CD4::mIFP under *btl-gal4* (red). (A-C) Control; (D-F) deGradFP over-expression. Dotted lines in (A'-F') highlight the tracheal trunk lumen. (G) Quantification of epidermis-dorsal trunk coordination using PIV and cross-correlation analyses. (H) Normalized dorsal trunk (DT) length, ***p<0.001. (I) Normalised displacement of epidermis (cyan) and dorsal trunk (red). (J) Time for dorsal closure completion, *p<0.05. Control, n=4; *btl*>deGradFP, n=5. Significance was assessed using t-tests. Plots in (G, H, J) show means +/- SD.

Affecting dorsal closure results in aberrant tracheal morphogenesis and vice versa. In our deGradFP experiments, we noticed that in addition to the effects on tracheal trunks, epidermal dorsal closure also appeared to be affected upon depletion of β-Integrin::GFP from the tracheal system (Figure 5F, Video S5, right). We quantified the time required for dorsal closure completion in these embryos, and we found a significant delay compared to controls (Figure 5I-J). Albeit unexpected, these results suggested an interdependence between epidermal and tracheal development. We decided to test an inverse scenario, where we affected dorsal closure and observed an effect on tracheal development. To do this, we used mutant embryos where epidermal dorsal closure was affected. Mutations in genes that code for components of the JNK signalling pathway either lead to a complete failure or to a delay in dorsal closure. We used *kay*^{1} (which codes for Fos) mutant embryos and focused on embryos with a delayed dorsal closure phenotype to see if this would have an effect in tracheal morphogenesis. We employed *btl*>CD4::mIFP to visualize the tracheal system, together with E-Cad fused to GFP to follow the progression of epidermal dorsal closure.

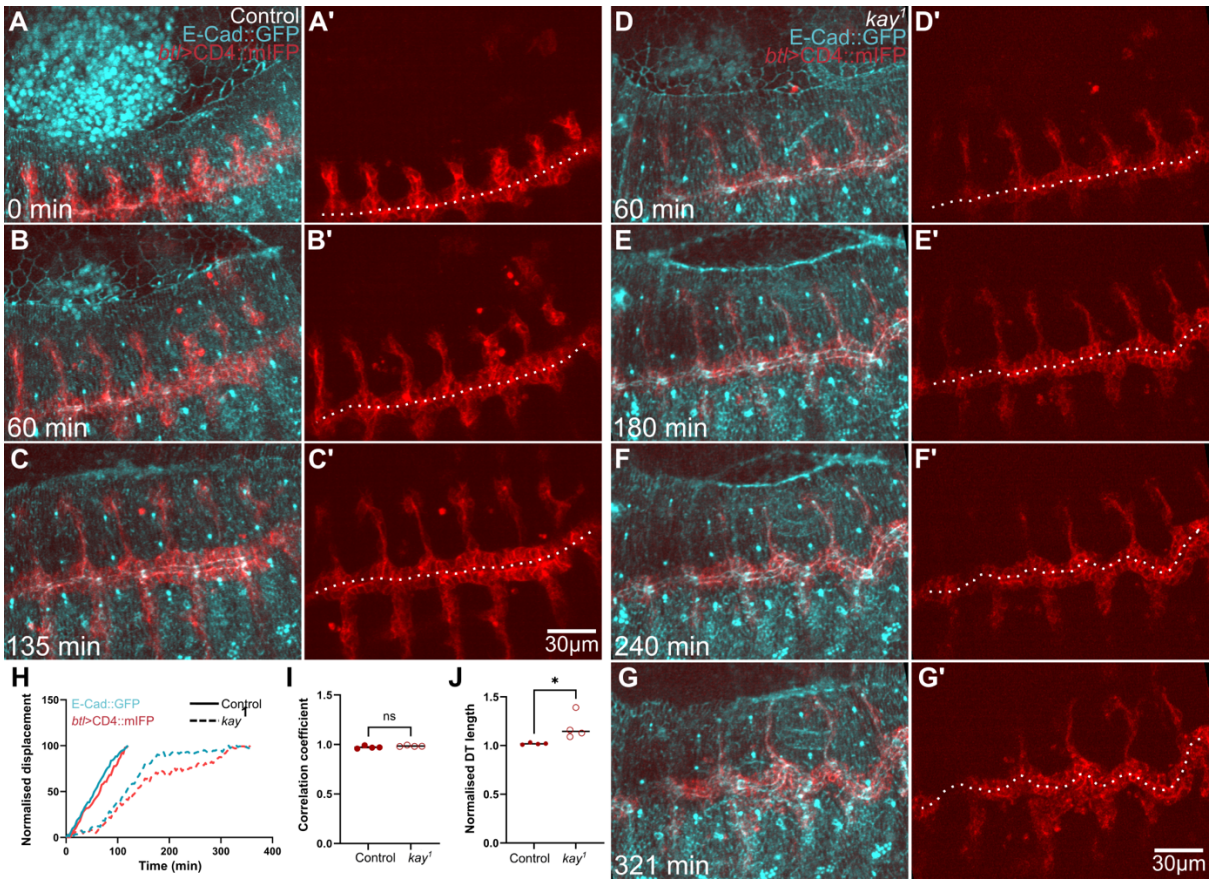

Figure 6. Tracheal trunk morphology in *kay*^{1} mutant embryos with a delayed dorsal closure phenotype. (A-G) Maximum intensity projections and time-lapse imaging of embryos expressing E-Cad::GFP (cyan) and CD4::mIFP under *btl-gal4* (red). (A-C) Control embryo; (D-G) *kay*^{1} mutant embryo. (H) Normalised displacement of the epidermis (cyan) and dorsal trunk (red). (I) Quantification of epidermis-dorsal trunk coordination using PIV and cross-correlation analyses. (J) Normalised dorsal trunk (DT) length, *p<0.05. Plots show means +/- SD. Control, n=4; *kay*^{1}, n=4. Significance was assessed using t-tests.

Like in control embryos, where we observed that dorsal closure progressed in parallel to trunk displacement (Figure 6A-C, H-I, Video S6, left), in *kay*[1] mutant embryos with a delayed closure phenotype, tracheal trunk displacement still occurred at the same rate as epidermal dorsal closure (Figure 6D-I, Video S6, right). However, in *kay*[1] mutant embryos, tracheal trunks developed malformations that were not observed in controls, which consisted in tortuosities that changed in shape over time and mimicked movements also observed in the epidermis. Again, in this mutant condition, tracheal trunks were longer than in controls (Figure 6J). These experiments show that the trachea-epidermis association is present even in conditions of delayed dorsal closure. Also, they support that proper trunk development depends on timely epidermal dorsal closure.

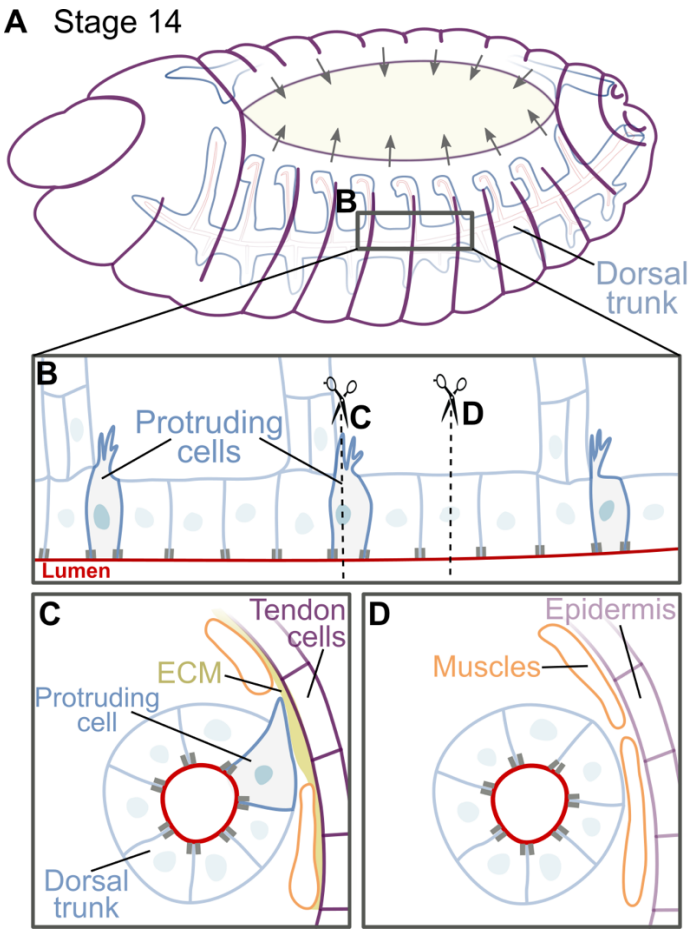

Figure 7. Model of trachea-epidermis interactions. (A) Overview of a stage 14 embryo at the onset of dorsal closure. The black square at the dorsal trunk is magnified in (B). (B) Protruding cells distribution at the dorsal trunk. The dotted lines are presented as cross-sections in (C) and (D). (C) Just below tendon cells, protruding cells extend filopodia towards the basement membrane of tendon cells. (D) Regions of the trunks not near to tendon cells are separated from the epidermis by muscles and other mesenchymal cells.

Altogether, we conclude that adhesion of the dorsal trunks to the tendon cells through the ECM is not required for dorsal displacement, as this movement still takes place when the interaction of the two tissues is altered. Instead, it plays a stabilising role in maintaining the straightness of dorsal trunks during their repositioning to the dorsal side of the embryo. This interaction is important for the tracheal system morphogenesis, but it also has an effect in epidermal dorsal closure.

Discussion.

Here, we demonstrated that epidermal closure and tracheal trunk displacement are coordinated events, with evidence suggesting that ECM integrity plays a key role in their regulation. In this model, the ECM would act at the same time as a milieu where extracellular signals can diffuse and act in different tissues, and as a scaffold that transmits forces, linking tissues as they undergo complex rearrangements. In the case of trachea-epidermis interactions, a number of classical studies have shown that signals produced by the epidermis act in the epidermis itself but also regulate tracheal patterning [reviewed in (Sánchez-Cisneros et al., 2025)]. Our experiments show that besides these well-characterised signals, mechanical interactions between the two tissues also fine-tune tracheal behaviour.

Emergence of a 'wavy trunk' phenotype upon disruption of trachea-epidermis interactions.

Our results demonstrate that perturbing trachea-epidermis interactions by different mechanisms (weakening the ECM through Mmp over-expression, targeting tracheal β-integrin to degradation, or delaying epidermal dorsal closure), consistently result in similar phenotypes: trunks are still displaced dorsally but they acquire a wavy appearance. This suggests that tracheal trunk displacement may be in fact achieved by the pulling forces of dorsal branches as they migrate in response to FGF. Instead to what we initially predicted, which was that adhesion to the epidermis would allow trunks to displace dorsally, our results suggest that this interaction may provide stabilising points that counteract the pulling forces generated by dorsal branches.

The wavy trunk phenotype has been already reported as a result of loss of laminins (Klußmann-Fricke et al., 2022). Despite the phenotype that manifests in laminin (*LanB1*) mutant embryos is stronger than the ones that we report here, we interpret it at least partly as a consequence of the loss of the interaction with the epidermis. The stronger phenotype described in laminin mutant embryos could be explained by an additive effect of cell-autonomous defects in cell polarity, as previously suggested (Klußmann-Fricke et al., 2022), and by the presence of other adhesion complexes that rely on laminin besides the integrins complex. The mechanism behind the wavy trunk phenotype of *kay*^{1} mutant embryos is not immediately obvious. It is possible that *kay* has a role in tracheal development, but earlier studies show that *kay* mRNA is not detectable in tracheal trunks (Souid and Yanicostas, 2003). We propose that upon loss of *kay,* zipping of the epidermis is delayed but trunk

maturation (thickening of the lumen and elongation of the trunks) continues. If the trunks are attached to tendon cells, their growth could be restricted by these interactions. The role of *bsk*, which codes for JNK, has been tested in tracheal development. While in these embryos there is no affection on dorsal branch intercalation or tip cell extension, trunks do seem to acquire a wavy morphology (Caussinus et al., 2008). Nevertheless, over-expression of a JNK dominant negative construct under *btl-gal4* does not affect tracheal development (Letizia et al., 2023), supporting that the effects of loss of this signalling pathway are non-cell autonomous.

Interactions between protruding cells and tendon cells.

According to our model, trachea-epidermis interactions occur at specific contact points distributed along the antero-posterior axis of the embryo, where tracheal trunks intersect with tendon cells of the epidermis. The protruding cells we describe here are frequently found directly in front of tracheal dorsal branches, which we believe has prevented their characterisation as they are difficult to visualise with general tracheal reporters.

Tendon cells are well-known force transmitters as they interact with the exoskeleton and muscle cells through apical and basal ECM layers, respectively, enabling locomotion as muscles contract (Chu and Hayashi, 2021; Martin-Bermudo, 2000; Weitkunat et al., 2014). Our analyses suggest that these points also mediate the adhesion of tracheal trunk cells, the protruding cells, to transmit forces relevant for tracheal remodelling. Previous studies have shown that tip cells of dorsal branches generate tension as they migrate, and it has been proposed that this tension is responsible for the cell intercalation that allows dorsal branch elongation (Caussinus et al., 2008; Letizia et al., 2019; Ochoa-Espinosa et al., 2017). It is possible that adhesion towards tendon cells also counteracts the pulling forces that regulate stalk cell intercalation. Our experiments do not reflect defects in cell intercalation, but these could be compensated by the looping of the trunks upon pulling interactions from tracheal branches.

Our results do not address if protruding cells possess different gene-expression profiles compared to their neighbours or if they acquire a distinctive morphology due to their proximity to the specialised ECM found near tendon cells. Previous works have shown that tendon cells secrete FGF which in turn regulates tracheal development (Butí et al., 2014; Dorfman et al., 2002). However, it is unlikely that tendon cell-derived FGF is responsible for the formation of

these protrusions, as tracheal trunk cells do not activate landmarks of FGF signalling like SRF expression or ERK phosphorylation (Letizia et al., 2023). Previous studies have induced tendon cell differentiation by over-expression of Stripe in other epidermal cells (Dorfman et al., 2002). Because of the effect on FGF expression, it is difficult to assess the effect on trunk cell behaviour from these experiments.

Trachea-epidermis interactions and their role in epidermal dorsal closure.

Besides the effects on the tracheal system, we found that targeting β-integrin in the tracheal system also resulted in a delay in epidermal dorsal closure. Previously, it was shown that dorsal tip cell migration is independent from epidermis displacement (Caussinus et al., 2008). Our results do not contradict these experiments; we see adhesion only at the level of tracheal trunks, and from our experiments there are no effects on tip cell morphology or migration. It has been shown that the forces produced by the amnioserosa are the main components that allow epidermal dorsal closure; even when the epidermis forms a supracellular actomyosin ring at dorsal-most row of epidermal cells, this only controls the seamless sealing of the epidermal sheets at the dorsal midline (Pasakarnis et al., 2016). It was suggested recently that the epidermis itself acts as a counter-force in response to the pulling forces of the amnioserosa (Lv et al., 2022). Our results support this view and suggest that adhesion of the trunks to the epidermis may contribute to balance the interplay of forces that participate in epidermal dorsal closure.

The ECM as a coordinator of morphogenesis.

Mechanical interactions between adjacent tissues and how they shape morphogenesis have been described in a wide range of processes. These include the above-mentioned epidermis-amnioserosa interactions in *Drosophila* development (Hayes and Solon, 2017; Kiehart et al., 2017) to epidermis-blood vessels in the rhinarium of dogs and other species (Dagenais et al., 2024). In many of these processes, the ECM, its mechanical properties and its remodelling, play important roles in force transmission and/or coordinating tissue behaviour (Barrera-Velázquez and Ríos-Barrera, 2021; Goodwin et al., 2016; Goodwin et al., 2017; Inoue and Hayashi, 2007; Münster et al., 2019). We suggest that ECM-mediated tissue adhesion may have a role in general embryo development, integrating morphogenetic processes of seemingly unrelated tissues.

Author contributions:

Conceptualization, LDRB; Methodology and investigation, LESC, MBV, DK, HML, LDRB; Formal Analysis, LESC, MBV, PB, LDRB; Writing-first draft, LESC & LDRB; Writing—Review and Editing, LESC, MBV, DK, PB, HML, LDRB; Visualization, LESC & LDRB; Supervision, LDRB; Funding Acquisition, LDRB.

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