Mechanosensitive EPLIN-dependent remodeling of adherens junctions regulates epithelial reshaping

Katsutoshi Taguchi, Takashi Ishiuchi, and Masatoshi Takeichi

Introduction

Many classes of epithelial cells adhere to each other via zonula adherens (ZA), an epithelial form of the adherens junction (AJ). The ZA consists of E-cadherin–catenin complexes and associated actin filaments, called circumferential actin cables, and is located at a region near the apical end of lateral cell–cell contacts, showing a closed ring configuration (Fanquhar and Palade, 1963; Boller et al., 1985; Cavey and Lecuit, 2009; Meng and Takeichi, 2009). αE-catenin, which binds to E-cadherin via β-catenin, mediates the interactions between the E-cadherin–β-catenin complex and actin filaments (Kovacs and Yap, 2008; Nelson, 2008; Meng and Takeichi, 2009; Sawyer et al., 2009; Kwiatkowski et al., 2010). Although the E-cadherin–β-catenin–αE-catenin complex cannot directly bind to actin filaments (Drees et al., 2005; Yamada et al., 2005), other actin-binding proteins attached to αE-catenin appear to assist in their linkage. These proteins include EPLIN (epithelial protein lost in neoplasm; Abe and Takeichi, 2008) and vinculin (Geiger et al., 1980; Palade, 1963; Boller et al., 1985; Cavey and Lecuit, 2009; Meng and Takeichi, 2009). αE-catenin–bound EPLIN acts as a mechanosensitive regulator for this process.

The ZA, a type of adherens junction (AJ), plays a major role in epithelial cell–cell adhesions. It remains unknown how the ZA is remodeled during epithelial reorganization. Here we found that the ZA was converted to another type of AJ with punctate morphology (pAJ) at the margins of epithelial colonies. The F-actin–stabilizing protein EPLIN (epithelial protein lost in neoplasm), which functions to maintain the ZA via its association with αE-catenin, was lost in the pAJs. Consistently, a fusion of αE-catenin and EPLIN contributed to the formation of ZA but not pAJs. We show that junctional tension was important for retaining EPLIN at AJs, and another force derived from actin fibers laterally attached to the pAJs inhibited EPLIN–AJ association. Vinculin was required for general AJ formation, and it cooperated with EPLIN to maintain the ZA. These findings suggest that epithelial cells remodel their junctional architecture by responding to mechanical forces, and the αE-catenin–bound EPLIN acts as a mechanosensitive regulator for this process.
Two forms of AJs in epithelial colonies

Double immunostaining for E-cadherin and F-actin showed that colonies of DLD1 epithelial cells, a polarized epithelial cell line derived from human colon carcinoma, exhibit two forms of AJ. In the inner portions of the colonies, cells organized the typical, closed ZAs, where E-cadherin and F-actin are linearly aligned. At the margins of the colonies, in contrast, the junctions were opened toward cellular free edges (Fig. 1, A and B). Although these open junctions were structurally equivalent to the ZA at the interior portions, they lost the linearity near the free edges, where E-cadherin signals became discontinuous. In some of these peripheral cells, E-cadherin maintained a closed ring structure, even though these cells have no contact partners at the side of their leading edges (Fig. S1 A). From a morphological point of view, this ring is likely a remnant of the ZA. Cells with the closed and open E-cadherin signals were detectable within a single colony (Fig. S1 A), which suggests that these structures are convertible. Throughout this paper, we call the peripheral AJs punctate AJs (pAJs), and the inner AJs forming ZAs are called linear AJs (lAJs). E-cadherin and αE-catenin always colocalized at both types of AJ (Fig. S1 A).

To investigate the cytoskeletal backgrounds for these junctional structures, we looked at F-actin distributions. The peripheral cells organized actin bundles running in parallel with the cellular edge. These actin bundles terminated at the pAJs at right angles, where individual E-cadherin puncta were morphologically pulled toward the associated actin filaments (Fig. 1, A and B), as seen in mesenchymal or fibroblastic AJs (Miyake et al., 2006). This type of actin association pattern is shown in contrast with the parallel alignment of F-actin and E-cadherin at the lAJs. We also examined the distributions of myosin II isoforms IIA and IIB, which have recently been shown to work differently in AJ formation (Ivanov et al., 2007; Smutny et al., 2010). Myosin IIA was localized along the ZAs, as well as along the peripheral actin fibers targeting the pAJs (Fig. 1 C, top). Myosin IIB also densely delineated the ZAs, but it only locally decorated the peripheral actin fibers (Fig. 1 C, bottom). As found for E-cadherin or αE-catenin, myosin IIB often exhibited a closed ring configuration in the peripheral cells, which suggests that not only the authentic ZAs but also the peripheral ZA-like rings comprise actomyosin filaments and associated E-cadherin–αE-catenin complexes.

The pAJ-associated actin (or actomyosin) fibers were reminiscent of the purse string–like actin cables formed by cells undergoing wound closure (Yonemura et al., 1995; Adams et al., 1996; Vasioukhin et al., 2000). To confirm this, we purposely scratched DLD1 cell layers. We found that the cells whose neighbors were removed remodeled their linear junctions to form the punctate type with concomitant reorganization of actin fibers (Fig. S1 B). We therefore assumed that the linear-to-nonlinear conversion of AJs at colony margins was a common phenomenon in a wide variety of polarized epithelial cells, such as MDCK, EpH4, and Caco2 cells, which suggests that this process generally occurs in epithelial cells. We thus decided to study the mechanisms of this junctional remodeling and its role in epithelial closing behavior.
major E-cadherin–associated proteins and \( \beta \)-, \( \alpha \)-, and p120-catenins were localized in both forms of the junction in proportion to E-cadherin. Likewise, vinculin, ZO-1, and afadin, which are known to interact with αE-catenin directly or indirectly, were detected both along the ZAJs and at the pAJs (Fig. 2 A, left; and Fig. S2, A and B). Intriguingly, another αE-catenin–interacting protein, EPLIN, was detected only at the IAJs, not at the pAJs, although EPLIN decorated peripheral actin fibers in immediate proximity to E-cadherin puncta forming the pAJs (Fig. 2 A, right). We also noticed that some of the epithelial lines, including MCF10A cells, exhibit pAJ-type junctions throughout their colonies, and these junctions were devoid of EPLIN signals (Fig. S2 C). Thus, EPLIN seems to be localized to only IAJs across cell lines.

**EPLIN localizes only at IAJs and is responsible for their formation**

To study the mechanisms of the junctional conversion, we investigated the molecular differences between these two forms of AJs. Major E-cadherin–associated proteins and \( \beta \)-, \( \alpha \)-, and p120-catenins were localized in both forms of the junction in proportion to E-cadherin. Likewise, vinculin, ZO-1, and afadin, which are known to interact with αE-catenin directly or indirectly, were detected both along the IAJs and at the pAJs (Fig. 2 A, left; and Fig. S2, A and B). Intriguingly, another αE-catenin–interacting protein, EPLIN, was detected only at the IAJs, not at the pAJs, although EPLIN decorated peripheral actin fibers in immediate proximity to E-cadherin puncta forming the pAJs (Fig. 2 A, right).

Live imaging of actin-EGFP introduced into DLD1 cells showed that the peripheral actin fibers were highly dynamic, as observed for pAJ-forming E-cadherin, whereas ZA-associated F-actin was much more stable (Video 3). Arrays of actin clusters, which are assumed to correspond to those associated with pAJs, were pulled in directions parallel to the cellular edges, which suggests that strong tension is operating on them. Some of these actin clusters moved inward, which suggests that the observed E-cadherin flow might have been driven by these actins via their linkages.

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junctions. Because the pAJs are unique in their association with peripheral actin fibers, we attempted to mechanically ablate these fibers. We prepared DLD1 cells transfected with EPLIN-EGFP and E-cadherin–mKOR, in which the exogenous EPLIN-EGFP widely decorated actin fibers. Then, we laser-irradiated a peripheral cell, focusing on a tiny point on the pAJ-targeting actin fibers (Fig. 3 A). The treated actin fibers immediately retracted toward the cell–cell boundary, where they anchored (Fig. 3 B and Video 5). Simultaneously, the cell connected with the laser-treated cell via the pAJ also contracted, likely because of the loss of a tension balance between the two cells. At the cell–cell boundary between these laser-treated and untreated cells, EPLIN signals were quickly up-regulated in a form of lAJ at the original pAJ sites, which suggests that removal of the peripheral actin fiber–derived force is sufficient to restore lAJs at these sites. This up-regulation of EPLIN accompanied a simultaneous increase of E-cadherin at the same cell boundaries (Fig. S4 [A and B] and Video 6), which indicates that EPLIN and E-cadherin behave together during these changes.

We previously reported that EPLIN depletion caused disorganization of the ZA (Abe and Takeichi, 2008). We confirmed this in the present study by immunostaining EPLIN-depleted cells for myosin IIA. The resultant junctions were similar to pAJs (Fig. 2 B); that is, both junctions were associated with myosin IIA fibers at right angles. Time-lapse recording of E-cadherin–EGFP showed that the junctions of EPLIN-depleted cells were highly dynamic, constantly changing their shapes (Video 4), just as seen in pAJs in control cells. These observations suggest that the pAJs were generated via a mechanism that also removes EPLIN from AJs.

EPLIN localization at AJs is mechanosensitive

We then asked how EPLIN was lost from pAJs. We found that, even when EPLIN was overexpressed, this protein was never recruited to the pAJs (Fig. S3 A), which indicates that there must be an active mechanism to suppress its association with the peripheral junctions. Because the pAJs are unique in their association with peripheral actin fibers, we attempted to mechanically ablate these fibers. We prepared DLD1 cells transfected with EPLIN-EGFP and E-cadherin–mKOR, in which the exogenous EPLIN-EGFP widely decorated actin fibers. Then, we laser-irradiated a peripheral cell, focusing on a tiny point on the pAJ-targeting actin fibers (Fig. 3 A). The treated actin fibers immediately retracted toward the cell–cell boundary, where they anchored (Fig. 3 B and Video 5). Simultaneously, the cell connected with the laser-treated cell via the pAJ also contracted, likely because of the loss of a tension balance between the two cells. At the cell–cell boundary between these laser-treated and untreated cells, EPLIN signals were quickly up-regulated in a form of lAJ at the original pAJ sites, which suggests that removal of the peripheral actin fiber–derived force is sufficient to restore lAJs at these sites. This up-regulation of EPLIN accompanied a simultaneous increase of E-cadherin at the same cell boundaries (Fig. S4 [A and B] and Video 6), which indicates that EPLIN and E-cadherin behave together during these changes.
such as tension support the proper accumulation of EPLIN and E-cadherin at IAJs. In these experiments, we also noticed that EPLIN signals fluctuate in nontreated junctions contiguous to the laser-treated junction via a vertex (Fig. 3 D, arrowheads in the center panel; and Video 7), where we suppose that the tension acting on the former was altered because of the laser ablation of the latter.

To confirm the role of tension, we used a stretching machine to test whether external forces can also affect the EPLIN and E-cadherin association with IAJs (Iwaki et al., 2009). We cultured DLD1 colonies consisting of several cells on an elastic chamber, and stretched them unidirectionally to ~150% of their original length. Live images of transfected EPLIN-EGFP and E-cadherin–mKOR showed that their signals were sharpened or intensified at the majority of the junctions (Fig. 3 E; see the figure legend for quantification). These observations suggest that the accumulation of EPLIN and E-cadherin such as tension support the proper accumulation of EPLIN and E-cadherin at IAJs. In these experiments, we also noticed that EPLIN signals fluctuate in nontreated junctions contiguous to the laser-treated junction via a vertex (Fig. 3 D, arrowheads in the center panel; and Video 7), where we suppose that the tension acting on the former was altered because of the laser ablation of the latter.

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AJs (Fig. S3 C), although vinculin still localized at these Y-27632–treated junctions (Fig. S3 D). Similar results were obtained when myosin IIA or IIB was knocked down (Fig. S5 A): as previously described (Ivanov et al., 2004; Smutny et al., 2010), myosin IIA depletion caused disorganization of E-cadherin and F-actin distributions, and EPLIN no longer colocalized with E-cadherin in these cells (Fig. S5 B). In myosin IIB–depleted cells, their junctional morphology became similar to that of pAJs, and EPLIN disappeared from these junctions.

Rho kinases (ROCKs) play a major role in the contraction of actomyosin filaments localized in ZAs (Hildebrand, 2005; Nishimura and Takeichi, 2008), and this contractility is likely a major cause of junctional tension. We found that ROCK1 is localized along the IAJs but not at pAJs (Fig. S3 B). When DLD1 cells were treated with a ROCK inhibitor, Y-27632, the junctional morphology was converted to the pAJ type, and EPLIN was completely abolished from these AJs (Fig. S3 C), although vinculin still localized at these Y-27632–treated junctions (Fig. S3 D). Similar results were obtained when myosin IIA or IIB was knocked down (Fig. S5 A): as previously described (Ivanov et al., 2004; Smutny et al., 2010), myosin IIA depletion caused disorganization of E-cadherin and F-actin distributions, and EPLIN no longer colocalized with E-cadherin in these cells (Fig. S5 B). In myosin IIB–depleted cells, their junctional morphology became similar to that of pAJs, and EPLIN disappeared from these junctions.

Figure 4. Exogenous expression of an αE-catenin–EPLIN fusion in R2/7 cells. (A) αE-catenin constructs tagged with EGFP. αE(1–906) corresponds to the full-length αE-catenin. The full-length EPLIN was fused to αE(1–508). (B) Junctional organization in R2/7 cells transfected with αE(1–906), αE(1–508), or αE(1–508)EPLIN. Contrasted with the balanced organization of IAJs and pAJs in αE(1–906)-transfected colonies, αE(1–508) transfectants show pAJs even in the interior. The inset shows an enlarged view of the boxed region. αE(1–508)EPLIN organize only IAJ-like junctions. Dotted signals probably represent overexpressed molecules deposited in the cytoplasm. (C and D) Junctional organization in R2/7 cells transfected with αE(1–508) or αE(1–508)EPLIN. Cells were double-stained for EGFP and E-cadherin (C) or F-actin (D). αE(1–508) organizes IAJs and pAJs at the central and peripheral regions, respectively, whereas αE(1–508)EPLIN organizes only IAJs. Arrowheads indicate pAJs. Bars, 10 µm.
These observations suggest that the ROCK-mediated contraction of AJ-associated actomyosin is required for the localization of EPLIN to AJs. Because EPLIN is recruited to AJs via its association with αE-catenin (Abe and Takeichi, 2008), we examined whether the αE-catenin–EPLIN interaction is maintained or not in Y-27632–treated cells by immunoprecipitation experiments. We first noticed that the detergent solubility of EPLIN was increased after Y-27632 treatments (Fig. S3 E, left), which suggests that the binding of EPLIN to actin fibers is ROCK dependent at least in part. Then, we found that a similar amount of EPLIN coprecipitated with αE-catenin in the control and Y-27632–treated cells, which indicates that their interaction was not affected by ROCK inhibition. These results indicated that the pAJs exclude the entire αE-catenin–EPLIN complex from themselves.

Altogether, these findings suggest that the IAJs maintain the αE-catenin–EPLIN complex (presumably the E-cadherin–αE-catenin–EPLIN complex) in a tension-sensitive way, and this process is blocked by peripheral actin fibers attached to the AJs from the lateral sides. At the pAJs, only the EPLIN-free E-cadherin–αE-catenin complexes are allowed to function.

αE-catenin–EPLIN fusion proteins exclusively contribute to IAJ formation

To further study the specific roles of the αE-catenin–EPLIN complex, we constructed a fusion protein between αE-catenin and EPLIN using the αE(1–508) fragment of αE-catenin, in which the EPLIN-binding VHI3 domain and adjacent portions were removed (Fig. 4 A). To characterize this fusion protein (αE(1–508)EPLIN), we prepared R2/7 cells transfected with this construct (αE(1–508)EPLIN-R2/7 cells), and also those transfected with αE(1–508) as a control (αE(1–508)-R2/7 cells). R2/7 transfectants with the full-length αE-catenin (αE(1–906)-R2/7 cells) were also used for comparison. We confirmed that both αE(1–508) and αE(1–508)EPLIN coprecipitate with E-cadherin upon immunoprecipitation (Fig. 5 A).

As described previously (Watabe-Uchida et al., 1998), αE(1–508) can organize AJ-like junctions. However, αE(1–508)-R2/7 cells were different from the αE(1–906)-R2/7 cells or DLD1 cells, in that they form nonlinear junctions even in the interior of the colony (Fig. 4 B, left and middle). This suggests that the deleted portions are required for a balanced IAJ-pAJ organization. However, αE(1–508)EPLIN-R2/7 cells exhibited only linear-type junctions, and curiously αE(1–508)EPLIN signals were not detectable at peripheral regions (Fig. 4 B, right). For more precise analysis of these junctional structures, we examined a single cell–cell border formed between a pair of cells. αE(1–508) or associated E-cadherin exhibited both the linear and nonlinear configurations within the border (Fig. 4, C and D), confirming that αE(1–508) can organize into both forms of AJ. In the case of αE(1–508)EPLIN-R2/7 cells, this construct sharply delineated the cell–cell borders together with E-cadherin or F-actin (Fig. 4, C and D), but it never showed any punctate-type distribution (Fig. 4, C and D). In addition, the αE(1–508)EPLIN-mediated contacts appeared unstable, as these junctions slid by each other, and even broke up during cultures (Video 8). Intriguingly, when αE(1–508) and αE(1–508)EPLIN were coexpressed in R2/7 cells, they transiently organized a typical combination of pAJs and IAJs (Video 9): in these double transfectants, αE(1–508)EPLIN showed only ZA-like distributions, whereas αE(1–508) was localized to both pAJs and IAJs. These observations suggest that αE(1–508)EPLIN contributes to the formation of IAJ-type junctions, whereas αE(1–508) participates in both forms of AJ, and the presence of both constructs promotes the organization of AJs resembling those in normal cells.

Next, we examined the effects of the expression of αE(1–508) and αE(1–508)EPLIN in DLD1 cells on their junctional organization, designating them as αE(1–508)-DLD1 and αE(1–508)EPLIN-DLD1 cells, respectively. E-cadherin–mKOR was also cotransfected in these cells. When E-cadherin was immunoprecipitated from these transfectants, we detected both endogenous and mutant αE-catenins in the precipitates (Fig. 5 A), which suggests that E-cadherin binds either form of αE-catenin in the cells. Expression of αE(1–508) did not show any particular effects on the junctional organization in these cells. αE(1–508) EPLIN-DLD1 cells organized epithelial colonies, and the fusion protein was localized along their ZA (or IAJs), colocalizing with E-cadherin. However, in these colonies, most of the peripheral cells did not form lateral contacts, assuming a flower petal–like arrangement (Fig. 5 B). In time-lapse movies of these cells, we could detect E-cadherin signals forming pAJ-type junctions between peripheral cells. Notably, these E-cadherin signals did not colocalize with αE(1–508)EPLIN, although this fusion protein was detectable adjacent to the E-cadherin signals in a diffuse fashion. These αE(1–508)EPLIN-free E-cadherin–mediated pAJs were a transient structure, as they subsequently moved into the ZA ring formation (Fig. 5, C and D; and Video 10), as observed in wild-type cells (Video 1). In contrast to the control cells, however, once the pAJs were absorbed into the rings, the cell peripheries were never replenished with new pAJs, which explains why pAJs were not observable in fixed samples. The fusion proteins that were detectable in proximity to the initial pAJs might have competed with the natural E-cadherin–αE-catenin complex, resulting in the inhibition of further pAJ formation. In addition, we noticed that even the IAJs were unstable in αE(1–508)EPLIN-DLD1 cells; these junctions were often disrupted (Video 10).

Vinculin and EPLIN in AJ formation

The above results indicate that EPLIN is involved in the formation of IAJs but not pAJs, which suggests that other molecules cooperate with αE-catenin to organize the AJs. Because αE(1–508) is active in general AJ formation, molecules that bind this αE-catenin construct are candidates. We and others previously found that αE-catenin mutants, equivalent to αE(1–508), can bind vinculin in vitro pull-down assays (Watabe-Uchida et al., 1998; Yonemura et al., 2010), and a fusion between these αE-catenin mutants and vinculin could organize AJs (Watabe-Uchida et al., 1998), which suggests that vinculin works together with αE-catenin. However, the role of vinculin in AJ formation has not been thoroughly investigated through loss-of-function approaches. We therefore reexamined the function of vinculin in the formation of cell–cell contacts.
We first looked at the effects of vinculin depletion on the junctions of αE(1–508)-R2/7 and αE(1–508)EPLIN-R2/7 cells. In both transfectants, vinculin colocalized with these mutant molecules at cell junctions, whereas this did not occur at the contact sites between parent R2/7 cells (Fig. 6 A). This supports the idea that the vinculin-binding site is preserved in both constructs. In vinculin-specific siRNA-treated cells, vinculin levels were considerably down-regulated in an siRNA probe-specific way (Fig. 6, B and C). In these cells, vinculin remained at focal contacts, but became undetectable at many portions of the cell junctions. We found that vinculin-depleted αE(1–508)-R2/7 cells were vigorously dispersed in a vinculin level–dependent fashion, which indicates that vinculin is essential for AJ-dependent cell–cell contacts in these cells. In the case of αE(1–508)EPLIN-R2/7 cells, vinculin depletion did not particularly disrupt the junctions (Fig. 6 D), which suggests that EPLIN dominates over vinculin in maintaining the transient αE(1–508)EPLIN-mediated cell–cell contacts.

Next, we examined the effects of vinculin depletion in DLD1 cells, comparing this with EPLIN depletion. In vinculin-null junctions, E-cadherin signals became fragmented, and each fragment showed filopodia-like or dotted shapes, in contrast with the more condensed puncta representing the EPLIN-depleted junctions (Fig. 7 A). These vinculin-free E-cadherin signals colocalized only with faint amorphous actin signals, whereas EPLIN-depleted junctions were linked with tensile actin fibers (Fig. 7 B). These observations suggest that vinculin plays a role in linking the E-cadherin–catenin complex to the tensile actin fibers. Consistent with this idea, vinculin was still localized at the punctate junctions in EPLIN-depleted cells (Fig. 7 A). When vinculin and EPLIN were both depleted, filopodia-like E-cadherin signals appeared to have increased, but their phenotypes were essentially identical to those observed in vinculin-depleted cells. We noted that EPLIN was not detectable on the vinculin-free E-cadherin signals, even in vinculin-depleted cultures (Fig. 7 C). This explains why the effects of vinculin depletion and vinculin/EPLIN double depletion were not so different. The loss of EPLIN from vinculin-depleted cells might be a phenomenon equivalent to its unstable association with AJ's that lost tension, which was observed in laser
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more dynamic form. Our analysis of this junctional conversion demonstrated that this process is regulated through the interactions of α-E-catenin with EPLIN and vinculin.

Discussion

Epithelial cells are linked together via the ZAs to form stable tissues. In embryonic or pathogenetic processes, however, epithelial sheets undergo dynamic remodeling including the closure of open spaces. We showed that the ZAs are converted to another form of AJ, pAJ, at the peripheries of cell colonies. The pAJs were involved in nascent cell–cell contacts, which suggests that the IAJ-to-pAJ conversion must be a process for epithelial cells to reorganize their junctional architecture into a

ablation experiments. Thus, EPLIN cannot maintain the stable ZAs in the absence of vinculin, although it can transiently organize IAJ-like contacts in the absence of vinculin, as shown in Fig. 6 D.

Mechanosensitive processes for the maintenance and remodeling of AJs

Previous studies suggested that the cadherin adhesion system or AJ components have mechanosensitive properties (Schwartz and DeSimone, 2008; Fernandez-Gonzalez et al., 2009; Ladoux et al., 2010; le Duc et al., 2010). Our observations indicate that at least two kinds of forces are involved in the maintenance and remodeling of epithelial AJs. One is the force derived from peripheral actin fibers targeting the pAJs. Laser ablation of the peripheral actin fibers was sufficient to convert pAJs to IAJs, which suggests that the IAJs may be a default form of AJ that
is altered when pulled by actin fibers, allowing its binding to vinculin (Yonemura et al., 2010). From a morphological point of view, one would expect $\alpha$-E-catenin to take a stretched form at pAJs, where vinculin is abundant and actin fibers are terminating in a pulling fashion. We could speculate that EPLIN is unable to bind such stretched $\alpha$-E-catenin molecules, and that the $\alpha$-E-catenin–EPLIN complex cannot interact with the pulling actin fibers; therefore, this complex is excluded from the pAJs (Fig. 8). Actually, in our previous experiments, we detected $\alpha$-E-catenin–EPLIN binding in vitro, where tensile forces are not expected to work on these molecular complexes (Abe and Takeichi, 2008). However, vinculin is present not only at pAJs but also at lAJs, where EPLIN localizes, which suggests that $\alpha$-E-catenin may also be stretched at the lAJs. This seems to contradict the model in which that EPLIN may bind only the nonstretched form of $\alpha$-E-catenin. However, if the ZA contains gets reorganized by these actin fibers. The other force is the tension of IAJs, produced by ROCK-dependent contraction of themselves or by external forces. Inhibition of ROCKs or myosin II depletion abolished the IAJs, and external tension upregulated the accumulation of EPLIN and E-cadherin on IAJs. We also showed that the solubility of EPLIN increased when cells were treated with a ROCK inhibitor, which suggests that EPLIN prefers the contracting actomyosin, when it binds to actin fibers. These suggest that EPLIN maintains the IAJs sensing their tension, and that this system is impaired by the peripheral actin fibers (Fig. 8). Similar tension-dependent recruitment of AJ components was also reported for myosin II (Fernandez-Gonzalez et al., 2009).

How do the peripheral actin fibers block the association of the $\alpha$E-catenin–EPLIN complex with AJ? It is noted that $\alpha$E-catenin itself is a mechanosensitive protein; its conformation is altered when pulled by actin fibers, allowing its binding to vinculin (Yonemura et al., 2010). From a morphological point of view, one would expect $\alpha$E-catenin to take a stretched form at pAJs, where vinculin is abundant and actin fibers are terminating in a pulling fashion. We could speculate that EPLIN is unable to bind such stretched $\alpha$E-catenin molecules, and that the $\alpha$E-catenin–EPLIN complex cannot interact with the pulling actin fibers; therefore, this complex is excluded from the pAJs (Fig. 8). Actually, in our previous experiments, we detected $\alpha$E-catenin–EPLIN binding in vitro, where tensile forces are not expected to work on these molecular complexes (Abe and Takeichi, 2008). However, vinculin is present not only at pAJs but also at IAJs, where EPLIN localizes, which suggests that $\alpha$E-catenin may also be stretched at the IAJs. This seems to contradict the model in which that EPLIN may bind only the nonstretched form of $\alpha$E-catenin. However, if the ZA contains

Figure 7. The effects of vinculin depletion on DLD1 cells. (A) Vinculin and EPLIN depletion causes fragmentation of E-cadherin signals. Vinculin remains to colocalize with E-cadherin in EPLIN-depleted cells, as indicated by arrows. (B) In vinculin-depleted and vinculin/EPLIN double-depleted cells, the fragmented E-cadherin signals no longer associate with tense actin fibers. (C) After vinculin depletion, E-cadherin no longer colocalizes with EPLIN. Bars, 10 µm.
by competing with the endogenous αE-catenin. Although it is unclear how much the artificial fusion proteins mimic the natural αE-catenin–EPLIN complex, some of the properties expressed by the fusion may reflect those of the natural complex. For example, the natural ZA may also be static, and not dynamic enough to engage in new contact formation; this could be a reason why cells need to convert the ZAs to pAJs at colony peripheries, so as to interact with other cells.

EPLIN and vinculin cooperate in AJ formation

The results obtained with the αE-catenin–EPLIN fusion proteins imply that, although the αE-catenin–EPLIN linkage is important for stabilizing the circumferential actin cables, its adhesion-sustaining ability may not be strong. We therefore thought that there must be other proteins to support αE-catenin functions, and we reexamined the role of vinculin. αE-catenin mutants lacking the C-terminal EPLIN-binding region, such as αE(1–508), are able to organize AJs through the binding to vinculin (Watabe-Uchida et al., 1998; Yonemura et al., 2010). We showed that vinculin knockdown in αE(1–508)-R2/7 cells severely disrupted their junctions, which indicates that vinculin is essential for the adhesion-supporting ability of this construct. Normal DLD1 cells also responded to vinculin depletion, although these cells were not dissociated. In vinculin-depleted DLD1 cells, E-cadherin signals were no longer associated with pulling actin fibers, which suggested that vinculin plays a role in the E-cadherin–F-actin linkage, as observed in the integrin-mediated focal contacts (Geiger et al., 2009; Parsons et al., 2010). The reason why the junctions of DLD1 cells were more resistant to vinculin depletion than those of αE(1–508)-R2/7

Characteristics of EPLIN-bearing AJs

EPLIN is known to bundle and stabilize actin fibers. This ability of EPLIN is likely a key process in maintaining ZAs. However, it inhibits Arp2/3-dependent branching of actin filaments (Maul et al., 2003), which suggests that EPLIN-bundled actin fibers are a rather static structure. This probably explains why the EPLIN-associated AJs look more static than EPLIN-free AJs.

We found that the αE-catenin–EPLIN fusion can form only IAJ-like contacts, which is consistent with the observation that native EPLIN participates in IAJ, but not pAJ, formation. The αE-catenin–EPLIN fusion-mediated junctions appeared unstable. Moreover, this artificial protein even disrupted the preformed junctions when introduced in DLD1 cells, probably

Figure 8. Summary and model. E-cadherin (red) and AJ-associated F-actin (green) distributions, observed in the present study, are schematically summarized at the left. At the right, a working model to explain the present observations is shown. The shapes of molecules are arbitrarily drawn. It is not determined whether EPLIN and vinculin bind to the same or different αE-catenin molecules. Black arrows indicate tension.
cells can be explained by the previous observation that the C-terminal domain of αE-catenin, which is deleted in αE(1–508), has some ability to support αE-catenin–mediated cell adhesion, as assayed in fibroblastic cells (Imamura et al., 1999). Thus, αE-catenin seems to have multiple pathways for supporting cell–cell adhesion.

We previously proposed that EPLIN works as a linker between αE-catenin and F-actin. The present observations suggest that the αE-catenin–vinculin and αE-catenin–EPLIN complexes have distinct roles. It seems that the former is important for general AJ formation, and the latter is specified for ZA formation. To organize the complete set of AJs, they probably complement each other. It should be noted that αE(1–508) can organize both IAJs and pAJs, despite the absence of an EPLIN-binding site, which suggested that EPLIN need not be bound to αE-catenin for maintaining IAJs; that is, EPLIN could maintain ZA-associated actin filaments independently of the cadherin–catenin complex. However, the positioning of IAJs and pAJs in αE(1–508) transfectants is not well organized. This indicates that EPLIN assists the ordered ZA organization most efficiently through its binding to αE-catenin. For example, EPLIN senses tensed actomyosin fibers and recruits more cadherins to specific junctional sites via their linkages, strengthening the cell–cell adherence there.

We have not determined whether vinculin and EPLIN bind together to a single αE-catenin or bind separately to different αE-catenin molecules when functioning at the ZAs. Notably, in αE(1–508)EPLIN-R2/7 cells, vinculin was concentrated at cell–cell junctions, probably through its binding to αE(1–508)EPLIN. This vinculin does not appear to be functional, however, as vinculin depletion did not affect these junctions. Thus, EPLIN and vinculin might compete with one another, even when they are linked to an αE-catenin molecule. In addition, we showed that αE(1–508) and αE(1–508)EPLIN cotransfected into R2/7 cells organized a set of pAJ- and IAJ-like junctions, which suggests that EPLIN and vinculin can work through separate αE-catenin molecules. In conclusion, our findings suggest that EPLIN and vinculin cooperate in the proper organization of two forms of epithelial AJs, and that the mechanosensitive removal of the αE-catenin–EPLIN complex from AJs is involved in their morphological conversion, which is likely important for the reshaping of epithelial architecture at colony peripheries.

Materials and methods

Antibodies and reagents

For cadherin detection, rat monoclonal anti-E-cadherin (ECCD2; Shirayoshi et al., 1986) and mouse monoclonal anti-E-cadherin (HECD-1; Shimoyama et al., 1989) antibodies were used. Immunoglobulin fractions containing ECCD2 were purified from rat ascites fluids by 50% sodium ammonium sulfate fractionation and DEAE column chromatography. Antibodies against ROCK1 have been described previously (Nishimura and Takeichi, 2008). The following antibodies were purchased: rabbit polyclonal anti-YAP (Cell Signaling Technology), rabbit polyclonal anti-myosin IIA and IIB, and rabbit polyclonal anti-l-α-fadin antibodies (Sigma-Aldrich); rabbit polyclonal anti-myosin IIB antibody (Covance); rabbit polyclonal anti-EGF antibody (MBL); mouse monoclonal anti-EPLIN antibody (BD); rabbit polyclonal anti-ZO-1 antibody (Invitrogen); and mouse monoclonal anti-glyceroldehyde 3-phosphate dehydrogenase (GAPDH; Millipore). F-actin was detected by Alexa Fluor 488– or Alexa Fluor 594–conjugated phalloidin (Invitrogen).

Plasmid construction

pCA-IRES-neomycin-mKOR and pCA-IRES-hygromycin-EGFP were used as the backbone plasmids for stable expression in eukaryotic cells, in which a neomycin-or hygromycin-resistant gene was driven by the IRES element. For construction of the full-length and mutant αE-catenins, 1–906 and 1–508 fragments were amplified from mouse αE-catenin cDNA (Kametani and Takeichi, 2007) by PCR, respectively. These DNA fragments were inserted into the EcoRV site of the backbone plasmids. Construction of EPLIN-EGFP expression vectors was described previously (Abe and Takeichi, 2008). Briefly, the full-length cDNA of mouse EPLIN, obtained from the Institute of Physical and Chemical Research FAN TOM cDNA database, was amplified by PCR, and inserted into the NotI site of pcDNA-EGFP. The expression of E-cadherin–EPLIN–E(1–508)-EGFP, an enzyme site located in the 3′ side of the EcoRV site. Before this ligation, the NotI site was blunted with Blunting high (TOYOBO). These constructs were sequenced for conformation. Mouse E-cadherin was also cloned into the backbone vectors (constructed by Y. Kametani in our laboratory, RIKEN Center for Developmental Biology, Minatojima-Minamimachi, Chuo-ku, Kobe, Japan).

Cell culture and transfection

DLD1 and R2/7 (Watabe-Uchida et al., 1998) cells were cultured in a 1:1 mixture of DME and Ham's F12 medium [(i)wak]] supplemented with 10% FCS, and maintained in 5% CO2 at 37°C. Cells were transfected using Lipofectamine 2000 (Invitrogen) according to the manufacturer's instructions. For isolating stable transfectants, cells were selected by exposure to 200 µg/ml of HydroGold (InvivoGen) and/or 400 µg/ml of G418. Mixtures of heterogeneous antibiotic-resistant clones were used for the analysis of transfectants to avoid clonal variations in the phenotypes.

Y-27632 treatment was performed with the culture medium containing 20 µM of the reagent for 3 h in 5% CO2 at 37°C. MCF 10A cells were cultured with the MEGM Bullet kit [Takara Bio Inc.] containing 100 ng/ml cholera toxin (Sigma-Aldrich).

RNAi

Human EPLIN depletion was performed by using Stealth siRNAs (Invitrogen) whose sequence has been described previously (Abe and Takeichi, 2008). Transfection of EPLIN-specific siRNAs was performed with RNAiMAX (Invitrogen) according to the manufacturer’s instructions: cells were treated with siRNA–RNAiMAX complexes for 4–5 h. Human vinculin depletion was performed using Mission siRNAs (Sigma-Aldrich). Sequences for the knockdown of vinculin were 5′-CAAGAUGAUUGACGAGAGATT-3′ and 5′-GAU- UUAACUGCCGCUGGUUT3′, designated as Nos. 3 and 7, respectively (Fig. 6B). For the effective depletion of vinculin, the standard protocol was modified: cells were first treated with siRNA–RNAiMAX complexes for 3–5 h, and further cultured with fresh medium for 12–15 h. These cells were then trypsinized and replated on collagen-coated dishes (i)wak]] or coverslips (i)wak]] in the medium containing siRNA–RNAiMAX complexes.

After 3–5 h, this medium was changed with fresh medium. After incubating for another 12–15 h, cells were subjected to analyses. When EPLIN and vinculin were co-depleted, the mixture of these siRNAs was used. Universal negative controls for each siRNA were obtained from Invitrogen and Sigma-Aldrich. For knockdown of myosin IIA and IIB, we used Mission siRNAs (Sigma-Aldrich) with the same protocol as used for vinculin depletion. Sequences for the knockdown of myosin IIA were 5′-GACAGAA-AUGCGUGAUUGUGAAUUGUAC-3′, designated as Nos. 1 and 2, respectively (Fig. S5A). For depletion of myosin IIB, sequences were 5′-GUCGUGAAUGCUUUGUAGA-3′ and 5′-GAGAAGUGCAUGCUUUG-3′, designated as Nos. 2 and 3, respectively (Fig. S5A). These siRNAs could deplete the expression of each isoform specifically.

Immunoprecipitation

Cells were washed with TMCN buffer (20 mM Tris-HCl, pH 7.4, 1 mM MgCl2, 1 mM CaCl2, and 0.15 M NaCl) containing 1 mM PMFS. These cells were harvested and dissolved in TMCN buffer containing 1% Nonidet P-40 and 1 mM PMFS on ice. This crude mixture was centrifuged at 200,000 × g for 10 min at 4°C. The supernatant was recovered. This supernatant was precleared with 200 µg/ml of HydroGold (InvivoGen) according to the manufacturer's instructions: cells were treated with siRNA–RNAiMAX complexes for 4–5 h, and further cultured with fresh medium for 12–15 h. These cells were then trypsinized and replated on collagen-coated dishes (i)wak]] or coverslips (i)wak]] in the medium containing siRNA–RNAiMAX complexes. After 3–5 h, this medium was changed with fresh medium. After incubating for another 12–15 h, cells were subjected to analyses. When EPLIN and vinculin were co-depleted, the mixture of these siRNAs was used. Universal negative controls for each siRNA were obtained from Invitrogen and Sigma-Aldrich. For knockdown of myosin IIA and IIB, we used Mission siRNAs (Sigma-Aldrich) with the same protocol as used for vinculin depletion. Sequences for the knockdown of myosin IIA were 5′-GACAGAA-AUGCGUGAUUGUGAAUUGUAC-3′, designated as Nos. 1 and 2, respectively (Fig. S5A). For depletion of myosin IIB, sequences were 5′-GUCGUGAAUGCUUUGUAGA-3′ and 5′-GAGAAGUGCAUGCUUUG-3′, designated as Nos. 2 and 3, respectively (Fig. S5A). These siRNAs could deplete the expression of each isoform specifically.
IgG (Beckman Coulter) was used. The complexes were precipitated with protein G–conjugated Sepharose beads, and eluted by SDS sample buffer (62.5 mM Tris-HCl, pH 6.8, 2% SDS, 0.003% Pyronin Y, and 10% glycerol) at 56°C for 15 min. Precipitates were separated by SDS-PAGE and analyzed by Western blotting. For detection of E-cadherin on the blots, HEC-D1 antibody was used.

SDS-PAGE and Western blotting

Samples were denatured by heating with SDS sample buffer containing 1% 2-mercaptoethanol at 95°C for 3 min. Proteins were separated by SuperSep polyacrylamide gel (Wako Chemicals USA), and transferred to polyvinylidine difluoride membranes (Bio-Rad Laboratories). Membranes were blocked with 5% skim milk in TBS for 1 h at room temperature. Horseradish-conjugated secondary antibodies were purchased from GE Healthcare. Protein bands were detected by Western lightning plus ECL (PerkinElmer).

Immuno-staining and confocal laser-scanning microscopy

Cells were fixed with 1% PFA in cultured medium for 10 min at room temperature. The fixed cells were permeabilized with 0.1% Triton X-100 in TBS, pH 7.4, for 10 min, and blocked with 5% skim milk in TBS for 10 min. Next, cells were incubated with primary antibodies in the blocking solution for 1–2 h. These cells were then washed with TBS and further treated with secondary antibodies. For double-staining, Alexa Fluor 488– or Alexa Fluor 594–conjugated antibodies (Invitrogen) were used as secondary antibodies. To detect ECD2, CF Dye488A– or CF Dye594–conjugated antibodies (Biotium) were used. In the case of triple staining, ECD2 was detected by Cy5-conjugated antibody (Millipore). These cells were washed with TBS and then with milliQ water (Millipore), and finally mounted with FluorSave (EMD). Images were acquired as a z stack (20–25 z sections, 0.3–0.5 µm apart, 1,024 × 1,024 pixels) through a Plan-Apochromat 63×/1.40 NA oil differential interference contrast objective lens (Carl Zeiss) with an inverted laser-scanning confocal microscope (LSM510; Carl Zeiss).

Live-cell imaging

Live-cell imaging was performed by using a DeltaVision microscope (Applied Precision), unless otherwise specified. Before the operation, cells were cultured on collagen-coated glass-based dishes (Iwaki) with L-15 medium (Invitrogen) supplemented with 10% FCS for 1 h at 37°C. The dish was placed on the stage of an inverted microscope (IX70 or IX71; Olympus) equipped with a cooled charged-coupled device camera (Series 300 CH330 or CoolSNAP HQ2; Photometrics). The temperature of the microscope room was maintained by a thermal controller (Sanyo). Time-lapse images (acquired as 6–10 z sections, 0.8–1.0 µm apart, 512 × 512 pixels, and binning 2 × 2) were obtained through Plan-Apochromat 60×/1.40 NA oil objective lenses (Carl Zeiss) with an inverted laser-scanning confocal microscope (LSM510; Carl Zeiss).

Wound healing

Cells were cultured at confluent densities on collagen-coated cover slips (Iwaki) or glass-based dishes (Iwaki). Wound edges were generated by scraping cells with a plastic tip. After scraping, cells were cultured for another 3 h, fixed, and subjected to analyses. For live-cell imaging, cells were cultured with L-15 medium supplemented with 10% FCS for 30 min, scraped to generate wounds, and further cultured for 1 h. Time-lapse images were then collected for 4 h.

Laser ablation

Laser ablation was performed by using Fluoview FV1000 laser-scanning confocal microscope (Olympus) equipped with a laser ablation system (provided by T. Kondo and G. Hayashi). A 355-nm laser beam was irradiated at a specific spot. Time-lapse images (acquired as 4–8 z sections, 0.3–0.5 µm apart, 512 × 512 pixels, and binning 2 × 2) were obtained through a LUCPlanFLN 60×/0.70 NA objective lens (Carl Zeiss) with an inverted laser-scanning confocal microscope (LSM510; Carl Zeiss).

Immunostaining

For E-cadherin–mKOR and EPLIN–EGFP during cell-cell junction remodeling

Fig. S1 shows the distribution of AJ proteins in natural or scratched DLD1 colonies. Fig. S2 shows the distribution of various junctional proteins in DLD1 or MCF10A cells. Fig. S3 shows the distribution of exogenous EPLIN, and the ROCK dependence of EPLIN–AJ association. Fig. S4 shows the effect of laser ablation of peripheral actin fibers on E-cadherin distribution. Fig. S5 shows the effect of myosin II depletion on EPLIN localization in AJs. Video 1 shows time-lapse movies of E-cadherin–mKOR in DLD1 colonies. Video 2 shows a time-lapse movie of ε-catenin–EGFP during wound closure. Video 3 shows a time-lapse movie of actin–EGFP in DLD1 cells. Video 4 shows time-lapse movies of E-cadherin–mKOR in a confluent culture of control or EPLIN-depleted DLD1 cells. Video 5 shows a time-lapse movie of EPLIN–EGFP after ablation of peripheral actin fibers. Video 6 shows a time-lapse movie of EPLIN–EGFP and E-cadherin–mKOR after laser ablation of peripheral actin fibers. Video 7 shows a time-lapse movie of EPLIN–EGFP after ablation of a part of the ZA. Video 8 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP introduced into R2/7 cells. Video 9 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP and ε(E1–508)EPLIN–EGFP doubly introduced into R2/7 cells. Video 10 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP and E-cadherin–mKOR doubly introduced into DLD1 cells.

Online supplemental material

Fig. S1 shows the distribution of AJ proteins in natural or scratched DLD1 colonies. Fig. S2 shows the distribution of various junctional proteins in DLD1 or MCF10A cells. Fig. S3 shows the distribution of exogenous EPLIN, and the ROCK dependence of EPLIN–AJ association. Fig. S4 shows the effect of laser ablation of peripheral actin fibers on E-cadherin distribution. Fig. S5 shows the effect of myosin II depletion on EPLIN localization in AJs. Video 1 shows time-lapse movies of E-cadherin–mKOR in DLD1 colonies. Video 2 shows a time-lapse movie of ε-catenin–EGFP during wound closure. Video 3 shows a time-lapse movie of actin–EGFP in DLD1 cells. Video 4 shows time-lapse movies of E-cadherin–mKOR in a confluent culture of control or EPLIN-depleted DLD1 cells. Video 5 shows a time-lapse movie of EPLIN–EGFP after ablation of peripheral actin fibers. Video 6 shows a time-lapse movie of EPLIN–EGFP and E-cadherin–mKOR after laser ablation of peripheral actin fibers. Video 7 shows a time-lapse movie of EPLIN–EGFP after ablation of a part of the ZA. Video 8 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP introduced into R2/7 cells. Video 9 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP and ε(E1–508)EPLIN–EGFP doubly introduced into R2/7 cells. Video 10 shows a time-lapse movie of ε(E1–508)EPLIN–EGFP and E-cadherin–mKOR doubly introduced into DLD1 cells.

Online supplemental material is available at http://www.jcb.org/cgi/content/full/jcb.201104124/DC1.

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