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Sustained α-catenin Activation at E-cadherin Junctions in the Absence of Mechanical Force

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ABSTRACT Mechanotransduction at E-cadherin junctions has been postulated to be mediated in part by a force-dependent conformational activation of α-catenin. Activation of α-catenin allows it to interact with vinculin in addition to F-actin, resulting in a strengthening of junctions. Here, using E-cadherin adhesions reconstituted on synthetic, nanopatterned membranes, we show that activation of α-catenin is dependent on E-cadherin clustering, and is sustained in the absence of mechanical force or association with F-actin or vinculin. Adhesions were formed by filopodia-mediated nucleation and micron-scale assembly of E-cadherin clusters, which could be distinguished as either peripheral or central assemblies depending on their relative location at the cell-bilayer adhesion. Whereas F-actin, vinculin, and phosphorylated myosin light chain associated only with the peripheral assemblies, activated α-catenin was present in both peripheral and central assemblies, and persisted in the central assemblies in the absence of actomyosin tension. Impeding filopodia-mediated nucleation and micron-scale assembly of E-cadherin adhesion complexes by confining the movement of bilayer-bound E-cadherin on nanopatterned substrates reduced the levels of activated α-catenin. Taken together, these results indicate that although the initial activation of α-catenin requires micron-scale clustering that may allow the development of mechanical forces, sustained force is not required for maintaining α-catenin in the active state.

INTRODUCTION

Epithelial tissue integrity is maintained by the formation of adherens junctions between adjacent cells. These junctions are constituted by a calcium-dependent homophilic interaction between the transmembrane adhesion protein E-cadherin between apposing cells. A loss of E-cadherin function due to mutations in the protein results in a loss of cohesion between cells, which leads to epithelial tissue deformation and may contribute to cancer metastasis (1). E-cadherin is a multidomain protein consisting of five extracellular cadherin domains (ECD), a transmembrane domain, and a catenin-binding intracellular domain (ICD) (2–4). While the interaction between the E-cadherin ECDs from apposed cells physically holds the cells together, the E-cadherin-ICD interacts with the actin cytoskeleton by forming a tertiary complex with β-catenin and α-catenin (5–8). The integration of E-cadherin ECD (E-cad-ECD) and E-cad-ICD-mediated interactions allows the mechanical coupling of adhering cells in the epithelial tissue and provides the potential for intercellular communication (9,10).

Epithelial cells sense mechanical signals from the cellular microenvironment and remodel their adhesions as a consequence. Strengthening of adhesion has been postulated to be mediated by α-catenin, which undergoes a force-dependent activation from a closed to an open conformation, allowing increased binding to the F-actin cytoskeleton as well as to vinculin (11–15). Force-dependent conformational changes in α-catenin have been independently observed in a multitude of experiments. These include in vitro single-molecule force spectroscopy experiments using purified proteins (12,14), and binding of a conformation-specific antibody (α18) in cells (11) and in live cells using an intramolecular Förster resonance energy transfer (FRET)-based assay (13). Although the crystal structure of homodimeric α-catenin revealed structural details of the closed conformation (16), the structure of the open conformation is not known so far. In addition to α-catenin, vinculin, which is well known for its critical role in mechanical signal transduction at focal adhesions, is enriched at adherens junctions upon an increase in cellular actomyosin tension (17,18). Vinculin is structurally homologous to α-catenin and undergoes a similar closed-to-open conformational activation that allows it to interact

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with α-catenin via its head domain (15,19,20). However, in the absence of force, its tail domain outcompetes α-catenin for binding to its head domain (15,21).

Here, we used a nanopatterned supported lipid bilayer platform (22,23) to study the role of micron-scale assembly of E-cadherin clusters in mechanotransduction at adherens junctions. Supported lipid bilayers are suited for such studies because they allow membrane-associated proteins to be organized into micron-scale assemblies, a process that can be physically perturbed by the application of nanopatterned substrates to elucidate the role of assembly in signal transduction (24–28). Mobility-dependent assembly, and physical perturbation of such assemblies could not be achieved together in other cell adhesion assays such as monolayer cell cultures or those that involve display of protein on solid surfaces. Cellular extension and retraction of filopodia on E-cad-ECD-functionalized bilayers nucleates specific adhesion and clustering of cell-surface E-cadherin with bilayer-bound E-cad-ECD (22). These clusters then accumulate with previously formed clusters, resulting in the assembly of extended E-cadherin-mediated adhesions at the cell-bilayer interface.

These adhesion assemblies contain E-cadherin in complex with key adaptor proteins, including β- and α-catenin, and can be distinguished as peripheral or central assemblies based on their spatial localization. Although the peripheral assemblies associate with F-actin, vinculin, and phosphorylated myosin light chain, the central assemblies do not. Both types of E-cadherin assemblies, however, contain α-catenin in the active, open conformation, as determined by binding of the isolated vinculin-head domain or the conformation-specific α18 antibody. Furthermore, the release of cellular applied tension by pharmacological inhibition of actomyosin contractility also fails to switch α-catenin to the inactive, closed state. The use of nanopatterned supported bilayers, in which grids of metallic structures fabricated onto the underlying substrate create barriers to the lateral mobility of supported-bilayer components, leads to a systematic reduction in the amount of active α-catenin. On these patterned substrates, E-cad-ECD in the supported bilayer is free to assemble locally into small-scale clusters, but micron-scale movements, including the filopodia-mediated nucleation process, are blocked (22,25,29,30). This suggests that α-catenin becomes activated during the nucleation process that initially activates E-cadherin assembly and adhesion. Once activated, however, α-catenin does not revert to the inactive state, even in the absence of mechanical force. The consequences of this observation on our understanding of the mechanism by which cadherin-mediated adherens junctions sense force will be discussed.

MATERIALS AND METHODS

Details regarding the materials and methods used in this work are available in Supporting Materials and Methods in the Supporting Material. Briefly, vesicles prepared using 1,2-dioleoyl-*sn*-glycero-3-phosphocholine (DOPC) and 1,2-dioleoyl-*sn*-glycero-3-[(N-(5-amino-1-carboxypentyl)iminodiacetic acid)succinyl] nickel salt (Ni-NTA-DOGS) were used to prepare bilayers by the vesicle fusion method (31). E-cad-ECD containing a C-terminal His_6 was attached to bilayers via Ni-NTA-poly-His interaction (32). FRAP was performed by illuminating a small field of view (15–30 μm diameter) at high intensity, followed by continued imaging to observe recovery of fluorescence. Hybrid E-cadherin adhesions were reconstituted by seeding MKN28-E-cad-GFP cells (22), and adhesion was observed using a 100× objective in an Eclipse Ti inverted microscope in either total internal reflection fluorescence (TIRF) or confocal mode. Images were collected in Metamorph (Molecular Devices, Sunnyvale, CA) and analyzed with either ImageJ (NIH, Bethesda, MD) or Fiji (33). For ratiometric analysis, images were registered using the TurboReg plugin (34). Micron-scale patterned substrates were prepared by deep UV-etching PLL-PEG coating (35), and nanoscale chromium-patterned substrates were prepared by nano-imprinting (24). Cells were stained with phalloidin to visualize F-actin or with protein-specific antibodies to visualize their localization. Activated α-catenin was visualized either by expressing vinculin-head-mCherry (chicken; amino acid residues 1–258) (12) or by staining with the conformation-specific α18 antibody (11). Cells were treated with 50 μm of either Y-27632 or blebbistatin for 30 min to reduce actomyosin tension.

RESULTS

In vitro reconstitution of E-cadherin adhesion in a live cell/supported lipid bilayer system

Here, we studied the interaction between E-cadherin and the actin cytoskeleton in a hybrid setting wherein one of the cells was replaced with a nanopatterned supported lipid bilayer displaying E-cad-ECD (Fig. 1 *A*). We employed a previously described strategy to display purified, recombinant human E-cad-ECD (Fig. S1 *A*) on supported lipid bilayers (22,32). Epithelial MKN28 cells expressing GFP-tagged E-cadherin (MKN28-E-cad-GFP) (8,22) were used to enable visualization of junctions that formed between cells and the bilayer. Functionalizing bilayers with poly-His containing E-cad-ECD at high density resulted in a reduction in mobility, as seen in a fluorescence recovery after photobleaching (FRAP) experiment (Fig. S1, *B* and *C*), thereby creating a proper physical microenvironment for junction formation (22).

A significantly higher number of cells remained adhered to bilayers containing E-cad-ECD compared with control bilayers that did not contain E-cad-ECD (Fig. 1 *B*), indicating that the protein that was displayed on the bilayer surface was adhesive. We utilized TIRF microscopy to observe enrichment and clustering of E-cadherin-GFP at the cell-bilayer interface (36). Cells seeded on E-cad-ECD-functionalized bilayers showed an enrichment of cellular E-cadherin-GFP at the cell-bilayer interface (Fig. 1 *C*), which was not observed on bilayers lacking E-cad-ECD (but containing Ca^{2+} ions) or in the absence of Ca^{2+} ions (Fig. 1 *C*). Additionally, cells seeded on E-cad-ECD bilayers showed significantly larger contact areas compared with cells on bilayers without E-cad-ECD (Fig. S1 *D*). Mature adhesions were formed by the

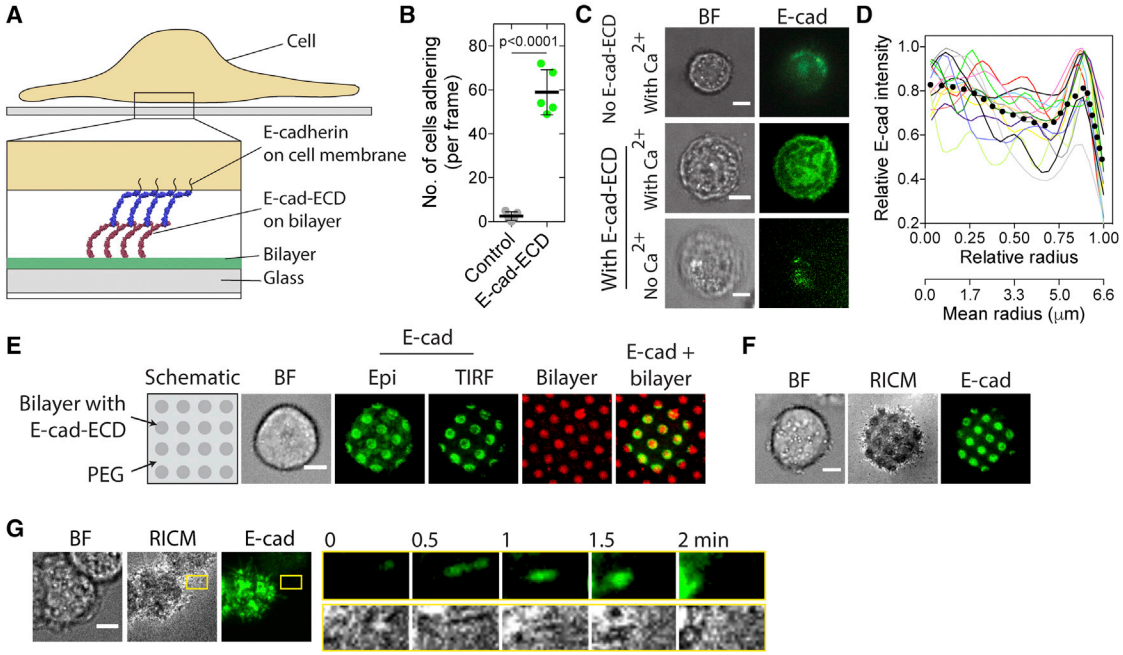

FIGURE 1 E-cadherin adhesion on a supported lipid bilayer. (*A*) Schematic representation of a cell forming an adhesion on an E-cad-ECD-functionalized bilayer. (*B*) Graph showing a comparison of the number of cells adhering to control (without E-cad-ECD) versus E-cad-ECD-functionalized bilayers. The data shown are from multiple independent experiments and the *p*-value was obtained from an unpaired *t*-test. (*C*) Bright-field (BF) and TIRF microscopy images of E-cadherin in cells seeded on control (without E-cad-ECD) or on E-cad-ECD-functionalized bilayers in the absence and presence of 2 mM $CaCl_2$, respectively. Note the extensive zone of cellular E-cadherin enrichment on E-cadherin bilayers in the presence of $CaCl_2$. (*D*) Plot showing multiple individual radial E-cadherin intensity profiles obtained from confocal images of adhesions formed by cells on E-cad-ECD-functionalized bilayers, revealing a peak at the cell periphery and random distribution of E-cadherin clusters within the adhesions. Original individual intensity profiles were smoothed with the average of three data points. The lower *x* axis panel shows the average cell contact radius, revealing micron-scale features of E-cadherin in individual radial profiles. The dotted black curve shows the average profile of all the 15 cells shown individually. (*E*) Schematic of a micropatterned substrate containing 2 μm discs of supported lipid bilayers functionalized with E-cad-ECD on a PEG surface. BF, epifluorescence (Epi), and TIRF images of an adhering cell on the micropatterned substrate. Bilayer discs are shown in red. Note that the enrichment of cellular E-cadherin coincides with regions of the substrate containing E-cad-ECD-functionalized bilayer discs. (*F*) BF, RICM, and TIRF images of an adhering cell on micropatterned substrate containing 2 μm discs of bilayers functionalized with E-cad-ECD, showing loss of RICM intensity due to interference at regions containing E-cadherin clusters. (*G*) TIRF and RICM images of a cell forming an adhesion on an E-cad-ECD bilayer, showing the addition of E-cadherin clusters formed by retracting filopodia, leading to the formation of large assemblies of E-cadherin at the adhesion. Scale bar, 5 μm.

dynamic movement of clusters at the cell-bilayer interface (Fig. S1 *E*), and the fluorescence intensity measured from the time-lapse TIRF images plateaued in ~30 min, with a mean $t_{1/2}$ for junction formation of 7.5 min (Fig. S1 *F*). The kinetics of adhesion formation observed in the hybrid format here is consistent with time frames reported for junction formation between two live cells (37). Additionally, mature adhesions showed very little recovery of E-cadherin fluorescence intensity in a FRAP experiment (Fig. S1 *G*), indicating that a large fraction of E-cadherin is immobilized upon adhesion formation, which is a characteristic feature of stable E-cadherin junctions (37).

Cells forming adhesion on the E-cadherin bilayers typically showed a ring of E-cadherin clusters at the periphery and some clusters at the central part of the cell-bilayer contact. Indeed, an analysis of E-cadherin intensity at a large number of adhesions by radial intensity profiling revealed a consistent peak at the periphery in all cells and a random distribution of micron-scale features of E-cadherin within the central part of adhesions (Fig. 1 *D*) (24). Henceforth, we refer to these spatially segregated assemblies as peripheral and central assemblies, respectively. Further, cells seeded on micropatterned supported membrane substrates consisting of small discs of E-cad-ECD-functionalized bilayers (2 μm diameter) showed enrichment and clustering of cellular E-cadherin-GFP as well as cell membrane topography (imaged by reflection interference contrast microscopy (RICM)) in a spatially restricted manner commensurate with the bilayer micropattern (Figs. 1, *E* and *F*, and S1 *H*), indicating that the clusters of E-cadherin observed in the cells represent cellular E-cadherin molecules specifically interacting with E-cadherin molecules on the bilayer (38,39). As reported previously (22), cells extended filopodia on the bilayer, and clusters of E-cadherin that were formed by the retraction of filopodia were subsequently fused to previously assembled clusters, resulting in the formation of an extensive adhesive zone between the cell and bilayer (Fig. 1 *G*).

Spatially differentiated E-cadherin interactions with the cytoskeleton and cadhesome proteins

Having reconstituted E-cadherin adhesions on a synthetic substrate, we then investigated the association of E-cadherin clusters with the actin cytoskeleton. As shown in Fig. 2 *A*, only the peripheral assemblies of E-cadherin clusters were associated with the actin cytoskeleton at the cell-bilayer interface. In contrast to the peripheral assemblies, the central assemblies of E-cadherin did not localize with F-actin filaments. Instead, F-actin filaments were found interspersed between the central assemblies and they appeared to be anti-localized with E-cadherin assemblies at the central part of the adhesion (Fig. 2 *D*). Immunofluorescence staining of cells with anti-β-catenin (Fig. 2, *B* and *D*) and anti-α-catenin (Fig. 2, *C* and *D*) antibodies showed that both types of assemblies were associated with β- and α-catenin. Importantly, such spatial segregation of E-cadherin clusters and their spatially distinct localization with F-actin was not observed when the cells were pretreated with 50 μM Y-27632, suggesting that actomyosin tension is required for the assembly of mature adhesions (Fig. S2 *A*) (40).

Unlike β- and α-catenin, vinculin was found to be associated exclusively with peripheral E-cadherin assemblies (Fig. 3 *A*) (17,41,42). Although vinculin is thought to be recruited to E-cadherin junctions via α-catenin in a force-dependent manner, some immunoprecipitation-based reports have suggested a direct interaction of vinculin with β-catenin in MDA-MB-468 cells (18,43). To understand the mechanism of vinculin recruitment at these hybrid adhesions, we performed a pull-down experiment with purified ICD of E-cadherin and lysates prepared from MKN28-E-cadherin-GFP cells. Although both β- and α-catenin interacted with the ICD of E-cadherin, vinculin could not be detected interacting with the complex (Fig. S2 *B*). This is in agreement with results obtained from immunoprecipitation experiments with A431 and MCF-7 epithelial carcinoma cells (18), and ruled out a force-independent, direct interaction of vinculin with β-catenin in the MKN28 cells. Additionally, the inability of α-catenin to bind vinculin in this solution-based assay, where physical forces cannot be developed, suggests that the interaction between the two proteins is force dependent and requires both of them to be present in the active conformation (11,14,21).

We then investigated the localization of other cadhesome proteins that could potentially play a role in mechanotransduction at the hybrid cell-bilayer junctions. Similar to vinculin, phosphorylated myosin light chain (Fig. 3, *B* and *M*) (40,44), lipoma-preferred partner (LPP) (Fig. S3 *A*) (7), and Zyxin (Fig. S3 *B*) (45,46) localized specifically to the peripheral E-cadherin assemblies. On the other hand, IQGAP1 (47) colocalized with both peripheral and central E-cadherin clusters in a pattern similar to that of α-catenin (Fig. S3 *C*). Thus, recruitment of cadhesome proteins (8) to E-cadherin assemblies is dependent on their association with the actin cytoskeleton, consistent with differential functional roles of E-cadherin clusters at apical versus lateral junctions between epithelial cells (48).

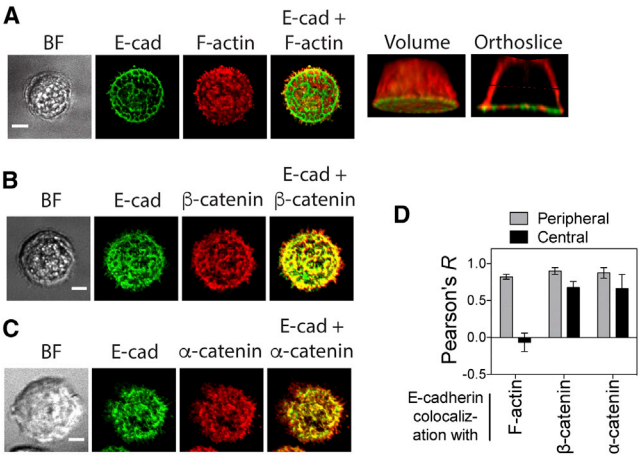

FIGURE 2 Interaction between E-cadherin assemblies and the actin cytoskeleton. (*A*) BF and confocal images of E-cadherin and F-actin (phalloidin) in cells adhering to E-cad-ECD bilayers. A volume and orthoslice reconstruction of the actin cytoskeleton and E-cadherin assemblies, performed using z-scan confocal imaging of the cell, shows a remodeled actin cytoskeleton. Note that the peripheral E-cadherin assemblies associate with F-actin, but the central assemblies do not (although intermittent patches of F-actin are present in the central part of the cell). (*B* and *C*) BF and confocal images of E-cadherin and either β-catenin (*B*) or α-catenin (*C*) in cells adhering to E-cad-ECD bilayers, showing colocalization of E-cadherin with β-catenin (*B*) or α-catenin (*C*), respectively. (*D*) Graph showing Pearson's *R*-value of E-cadherin colocalization with F-actin, β-catenin, and α-catenin in the peripheral and central clusters. Scale bar, 5 μm.

Sustained α-catenin activation in the absence of force

Force-dependent conformational activation of α-catenin has been postulated to be a key mechanotransducing signal at E-cadherin adhesions (11,12,14). We investigated the conformation of α-catenin at the cell-bilayer interface by expressing the vinculin head domain construct without the autoinhibitory tail domain (vinculin head; residues 1–258) (12) and by staining with the α18 antibody (11). Both the vinculin head and α18 bind specifically to activated α-catenin, which is thought to be the conformation that can bind F-actin and vinculin (11,12,14). These experiments revealed that α-catenin associated with both the peripheral and central assemblies of E-cadherin clusters binds the vinculin head and α18 antibody (Fig. 3, *C* and *D*), indicating that α-catenin is present in an open conformation in both types of assemblies. This result is not surprising for peripheral assemblies of E-cadherin, which colocalize with F-actin, vinculin, and phosphorylated myosin light chain (Figs. 2 *A* and 3, *A* and *B*). E-cadherin in central assemblies, on the other hand, exhibits no apparent association with the actin cytoskeleton or phosphorylated myosin light chain, and thus does not appear to be under tensile force. Although

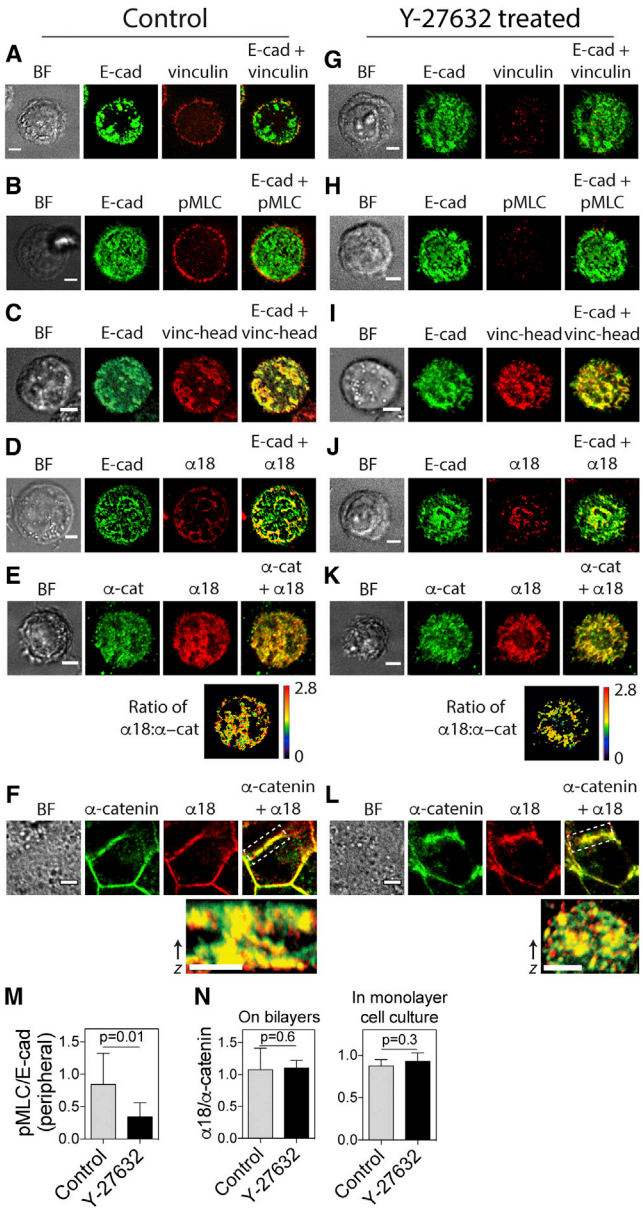

FIGURE 3 α-catenin is sustained in the active conformation irrespective of actomyosin tension. (*A–D*) BF and confocal images of cells forming junctions on E-cad-ECD-functionalized bilayers stained for stained for vinculin (*A*) or phosphorylated myosin light chain (*B*), and cells expressing the vinculin-head domain fused to mCherry (vinc-head) (*C*) and stained for the open conformation of α-catenin using the α18 antibody (11) (*D*). (*G–J*) BF and confocal images of 50 μM Y-27632-treated cells stained for vinculin (*G*) or phosphorylated myosin light chain (*H*), and cells expressing the vinculin-head domain fused to mCherry (vinc-head) (*I*) and stained for the open conformation of α-catenin using the α18 antibody (*J*) (11). (*E* and *K*) BF and confocal images of control (*E*) and Y-27632-treated (*K*) cells adhering to E-cad-ECD bilayers. Cells were stained for total α-catenin using an antibody that binds to the C-terminus of the protein independently of its conformation, and for the conformationally activated α-catenin using the α18 antibody. The lower-right panels in (*E*) and (*K*) are ratiometric images of α18 and total α-catenin staining for the respective cells. (*F* and *L*) BF and confocal images of cells forming junctions in monolayers and stained for total α-catenin and the conformationally activated α-catenin visualized with α18 antibody in control (*F*) or 50 μm Y-27632-treated (*L*) cells. (*M*) Graph showing the ratio of pMLC and E-cadherin-GFP intensities at the cell periphery in control and Y-27632-treated cells, revealing a reduction in the actomyosin tension upon Y-27632 treatment of the cells. (*N*) Graph showing the ratio of α18 and total α-catenin staining intensities in control and Y-27632-treated cells on bilayers or monolayer cultures. No significant differences are observed in the levels of activated α-catenin in control and Y-27632-treated cells. Scale bar, 5 μm.

constitutively active vinculin constructs have been shown to stabilize focal adhesion complexes (49), staining with the α18 antibody after chemical cross-linking-based fixation of the cellular proteins suggests that these results are not experimental artifacts. Further, the absence of vinculin from central E-cadherin assemblies that contain activated α-catenin indicates a more stringent autoinhibition in vinculin caused by its tail domain in the absence of an interaction with F-actin, consistent with the absence of vinculin in the pull-down experiment with the purified ICD of E-cadherin (Fig. S2 *B*) (15,19,21).

The presence of α-catenin in the active conformation in central E-cadherin assemblies, which did not associate with F-actin, was intriguing, especially considering the reversible conformational change in α-catenin observed with the use of a FRET-based sensor (13). To rule out any role of cellular actomyosin contractility in this phenomenon, we observed the effect of a reduction in actomyosin tension by treating adhering cells with pharmacological inhibitors. Treatment of cells with the ROCK inhibitor Y-27632 for 30 min after the formation of stable adhesions (60 min after seeding of cells) resulted in a decrease in the peripheral E-cadherin assemblies and a loss of vinculin and phosphorylated myosin light chain staining (Fig. 3, *G*, *H*, and *M*), thus confirming the effectiveness of the drug in reducing actomyosin contractility in the cells. However, central E-cadherin assemblies were found to be stable under Y-27632 treatment, and α-catenin that associated with these assemblies still bound both the vinculin head (Fig. 3 *I*) and the α18 antibody (Fig. 3 *J*). To obtain a more quantitative comparison of α-catenin activation, we simultaneously stained both control and Y-27632-treated cells with α18 and another antibody that binds α-catenin in a conformation-independent manner and thus reports the levels of total α-catenin (11). Whereas the α18 antibody binds to the central part of α-catenin (11), the epitope for the anti-α-catenin antibody used to stain total α-catenin lies in the C-terminus of α-catenin (residues 890–901), and therefore the two antibodies should not interfere with each other in binding to α-catenin. The relative levels of the open conformation of α-catenin were determined by a ratiometric analysis of the α18- and anti-α-catenin-stained images (11). This analysis revealed that the relative levels of conformationally active α-catenin remained unaltered upon a reduction in cellular actomyosin contractility with Y-27632 treatment (Fig. 3, *E*, *K*, and *N*). Similar results were obtained with the myosin inhibitor blebbistatin (Fig. S4 *A*). Further, these results could be recapitulated in native cell-cell junctions

formed by cells in monolayers (Figs. 3, *F*, *L*, and *N*, and S4, *B* and *C*). Thus, once α-catenin has been activated in the process of adhesion formation, it does not appear to be sensitive to cellular actomyosin contractility and remains in the active conformation in the absence of force.

Nucleation and micron-scale assembly of E-cadherin are required for the activation of α-catenin

We previously reported that E-cadherin adhesion formation involves an active nucleation process in which the retraction of filopodia over distances of a few microns results in the local enrichment of E-cadherin and stable junction formation (22). This process can be physically inhibited by the spatial mutation method, in which a patterned supported membrane is employed to control the movement and geometric assembly of cell-surface receptor proteins (23–26,28,29,50). Here, we fabricated chromium grids ranging from 500 nm to 2 μm on the glass substrate by nano-imprint lithography to physically restrict the movement of E-cadherin. Cells seeded on bilayers containing nanopatterned chromium barriers were allowed to form junctions, and a ratiometric analysis of active and total α-catenin was performed as mentioned in the previous section.

Similar nucleation and micron-scale assembly of E-cadherin clusters were observed in cells adhering to E-cadherin bilayers containing large (2 μm spacing) grids compared with cells adhering to bilayers without any barriers (off grid) (Fig. 4 *A*). This results from the fact that the filopodia retraction and micron-scale movement of E-cadherin is not significantly hindered on the large grids. However, cells that formed junctions on bilayers containing smaller grids (1 μm and 500 nm spacing), where the filopodia-driven nucleation process was impeded, showed a decrease in the micron-scale assembly of cellular E-cadherin (Fig. 4 *A*). In parallel with the reduction of micron-scale E-cadherin assemblies on small grids, we found that the ratio of α18 to total α-catenin staining intensity also decreased (20% and 46% on grid spacings of 1 μm and 500 nm, respectively, compared with the off-grid cells) (Fig. 4, *A* and *B*). Thus, filopodia retraction-mediated nucleation and micron-scale assembly of E-cadherin clusters at the cell-bilayer interface is required for the activation of α-catenin. Additionally, the changes in the relative levels of α18 binding (normalized to total α-catenin) determined in these experiments with chromium barriers of different sizes clearly establish that the α18 antibody is a reliable reporter of the α-catenin conformation.

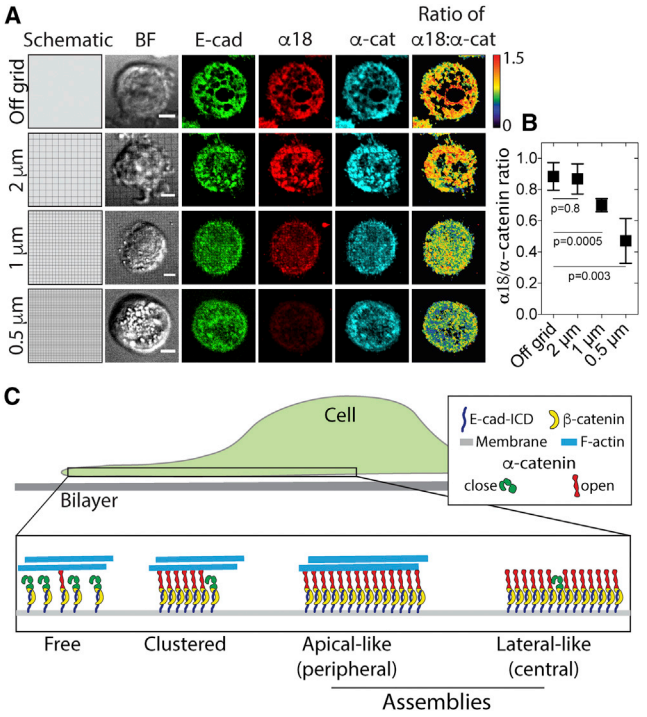

FIGURE 4 Nucleation and micron-scale assembly of E-cadherin clusters regulate the conformational activation of α-catenin. (*A*) Schematic representation of a nanopatterned supported lipid bilayer, BF and confocal images of cells adhering to the respective nanopatterned bilayers stained for total α-catenin, the open conformation of α-catenin using α18 antibody (11), and the ratio of α18 and total α-catenin intensities. Scale bar, 5 μm. (*B*) Graph showing the ratio of α18 and total α-catenin intensities (mean ± SD) obtained from multiple cells from a representative experiment. (*C*) Schematic representation of the conformational regulation of α-catenin by micron-scale assembly of E-cadherin clusters. A cell seeded on an E-cad-ECD-functionalized bilayer clusters E-cadherin by retracting filopodia. α-catenin is activated during the process of assembly of micron-scale E-cadherin clusters due to its association with the actin cytoskeleton. Once activated, α-catenin stays in that form even in the absence of its association with actin cytoskeleton as seen with the central E-cadherin assemblies, or in the absence of mechanical force as seen with the Y-27623 treatment.

DISCUSSION

Here, we report a sustained activation of α-catenin at E-cadherin adhesions in the absence of continued actomyosin tension and interaction with either F-actin or vinculin. Adhesions reconstituted on synthetic, supported lipid bilayer substrates exhibited E-cadherin in spatially and compositionally segregated peripheral and central clusters. Although a variety of junctional structures have been reported in the literature for different members of the cadherin family, studies on E-cadherin junctions formed between polarized epithelial cells, such as the MKN28 cells used here, have described two pools of E-cadherin clusters: one at the apical end of the lateral surface, termed the zonula adherens, and the other on the lateral surface below the zonula adherens (8,51,52). Based on their spatial arrangement and molecular composition, we propose that the peripheral assemblies are reflective of apical clusters that are under cellular actomyosin tension exerted through the peripherally localized actin filaments. On the other hand, the central assemblies are more analogous to lateral clusters that do not appear to be under actomyosin tension. Consistent with this, a reduction in cellular actomyosin tension resulted in a loss of the

peripheral E-cadherin assemblies concomitantly with the loss of phosphorylated myosin light chain and vinculin staining, whereas the central assemblies remained largely intact.

The persistent activation of α-catenin at E-cadherin adhesions that we describe here furthers our understanding of the mechanical regulation of cadherin adhesion. Although this is an unexpected finding, it may not be contradictory to previous studies that suggested swift changes in the levels of activated α-catenin upon an alteration in actomyosin contractility (11,13,41). Investigators have studied this activation of α-catenin both in vitro and in live cells using different tools. In vitro studies have reported either binding of the vinculin-head domain upon mechanical stretching of α-catenin (12) or a change in the α-catenin-F-actin bond lifetime under tension (14). Cell-based assays have utilized either binding of the conformation-specific α18 antibody to α-catenin (11) or a FRET-based α-catenin force sensor (13). Although they generally report a force-induced structural transition, each of these tools reports a distinct conformation or conformational transition in α-catenin (53–56). This is clear from results obtained with the FRET-based α-catenin sensor and a vinculin construct without the autoinhibitory tail (13). Although the FRET-based sensor returned almost instantaneously to the high-FRET, low-tension state upon a reduction in cellular tension, the vinculin construct stayed bound to α-catenin for a much longer period. These results suggest that the FRET-based sensor probably reports a force-dependent partial structural unfolding of α-catenin (12), and not the conformation with exposed cryptic binding sites that allow binding of interacting proteins such as vinculin. It is possible that force-induced partial unfolding of α-catenin follows exposure of the cryptic binding sites. We note that although force is a signaling *input* and is valuable to study where and when forces are applied on α-catenin, exposure of cryptic binding sites is the *response* that is critical for mechanotransduction at E-cadherin adhesions. Therefore, the activation of α-catenin that we observed here using the vinculin-head domain construct and α18 antibody is valuable for understanding α-catenin function. Also, previous cell-based studies were focused on the activation status of α-catenin only at the apical junctions. We, on the other hand, report the α-catenin activation status for both peripheral and central assemblies in parallel. This was possible primarily due to the reconstitution of adhesions on a two-dimensional bilayer surface, thus indicating the usefulness of the hybrid live cell/supported lipid bilayer system for studying cell-cell adhesion. We note that although previous studies utilized other cell lines, results presented here and previously (8,22,52) show that the MKN28 epithelial cells used here display all of the relevant features of E-cadherin adhesion and mechanotransduction. Therefore, the key observations regarding the mechanical regulation of α-catenin reported here should be relevant for other cell lines as well. The mechanism of stabilization of α-catenin in the active conformation may involve a posttranslational modification (57) or an interaction with another protein (5). The final inactivation of α-catenin may require the disengagement of E-cadherin, either by simple unbinding or by proteolytic cleavage (58), at the site of adhesion, and cellular recycling processes such as endocytosis (59).

The use of nanopatterned supported lipid bilayer substrates with physical barriers uncovered a requirement for nucleation and micron-scale assembly of E-cadherin for α-catenin activation (22). Nanopatterned substrates have been successfully employed to elucidate the role of receptor clustering in a number of cellular signaling systems, including T-cell activation, EphA2 receptor tyrosine kinase signaling, and integrin adhesion (24–26,60–66). Unlike genetic or pharmacological perturbations that may have off-target or deleterious effects (e.g., on protein stability), these nanopatterned substrates allow receptor clustering to be perturbed in a purely physical way, thereby avoiding such effects. More importantly, although mutation of the *cis*-interaction interface in E-cadherin results in a change in the dynamics of E-cadherin clusters, cells nevertheless form E-cadherin clusters (22,52,67–69), and thus the *cis*-mutant E-cadherin may not be useful for investigating the role of E-cadherin clustering. Therefore, the use of nanopatterned substrates, as described here, appears to be the only way to definitively manipulate micron-scale E-cadherin assemblies. Further, it is interesting to note that obstruction of the micron-scale clustering of E-cadherin results in a loss of α-catenin activation. This suggests that a decrease in molecular mobility, which is essential for E-cadherin adhesion formation (22), is not sufficient for development of the forces required for α-catenin activation. Instead, a local increase in the concentration of E-cadherin is required for the development of forces and activation of α-catenin. Indeed, the use of these substrates provided insights into E-cadherin adhesion and mechanotransduction that could not have been achieved in a regular cell-based assay.

In conclusion, α-catenin is activated during the initial adhesion assembly process involving filopodia retraction-mediated nucleation and clustering of E-cadherin (Fig. 4 *C*). Activated α-catenin continues to be present in the active conformation while E-cadherin clusters are dynamically organized into peripheral and lateral assemblies. Further, α-catenin does not require continuous force to maintain its open conformation. As such, α-catenin is unable to respond to changes in force in the context of an assembled E-cadherin adhesion, and is unlikely to play a role as a real-time force sensor.

SUPPORTING MATERIAL

Supporting Materials and Methods and four figures are available at http://www.biophysj.org/biophysj/supplemental/S0006-3495(16)30473-8.

AUTHOR CONTRIBUTIONS

K.H.B., R.Z.-B., and J.T.G. conceived the study. K.H.B. and K.L.H. performed experiments and analyzed data. K.H.B., K.L.H., R.Z.-B., and J.T.G. wrote the manuscript.

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