Trans-dimerization of JAM-A regulates Rap2 and is mediated by a domain that is distinct from the cis-dimerization interface

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Trans-dimerization of JAM-A regulates Rap2 and is mediated by a domain that is distinct from the cis-dimerization interface

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supported by reports of JAM-A–dependent adhesion between platelets and endothelial cells (Babinski et al., 2002a) and the observation that cells overexpressing JAM-A show accumulation of the protein at contacts between transfected cells (Ebnét et al., 2000; Mandell et al., 2004). However, the molecular basis and functional consequences of trans interactions between JAM-A molecules are not known.

Although definitive molecular evidence for JAM-A trans-homodimerization is lacking, there is structural evidence documenting dimerization in cis that mediates JAM-A function. Mutagenesis studies identified charged amino acids arginine 61 and glutamate 63 in the distalmost Ig-like domain that interact to form a salt bridge mediating cis interactions between two JAM-A molecules (Mandell et al., 2004; Guglielmi et al., 2007). Experiments using JAM-A mutants in which arginine 61 and glutamate 63 are exchanged with alanine (termed 6163) or the entire distal Ig-like domain is deleted (termed DL1) demonstrated important functional roles for cis-dimerization in epithelial cells. It was determined that JAM-A regulates cell migration and does so through association with the Rap exchange factor PDZ-GEF2 and the scaffold protein afadin to activate the small GTPase, Rap1, and subsequently stabilize β1 integrin on the cell surface (Severson et al., 2009). However, overexpression of cis-null JAM-A mutant 6163 or DL1 in cells led to decreased β1 integrin expression and inhibition of cell migration (Severson et al., 2008). Finally, cells expressing a JAM-A dimerization-null mutant lacking the distal Ig-like domain (DL1) exhibited enhanced proliferation compared with cells overexpressing WT JAM-A (Nava et al., 2011).

Whereas previous studies focused on the functional importance of JAM-A cis-dimerization, the observation that exogenously JAM-A accumulates at cell contacts (Ebnét et al., 2000; Mandell et al., 2004) suggests that trans-dimerization may also be functionally important. We reasoned that simultaneous cis- and trans-dimerization of JAM-A could facilitate formation of oligomers that mediate signal transduction through clustering of associated proteins. Indeed, epithelial permeability and cell migration studies suggest that JAM-A dimerization is required to assemble groups of signaling proteins at the apical junctional complex (Bazzoni et al., 2000; Severson et al., 2009; Monteiro et al., 2013). For example, JAM-A–mediated regulation of epithelial permeability was shown to depend on JAM-A interactions with ZO proteins, afadin, and PDZ-GEF1, which activate Rap2c and regulate the dynamics of the apically associated actomyosin belt (Monteiro et al., 2013). However, despite the clear link between JAM-A and regulation of epithelial permeability, the role of dimerization has not been defined.

In this study, we determine whether JAM-A mutants incapable of forming trans- and/or cis-dimers differentially regulate JAM-A effectors implicated in epithelial function. We find that JAM-A trans-dimerization occurs both in cells and between surfaces coated with recombinant JAM-A. Furthermore, we identify putative motifs for trans-dimerization and suggest a functional role for JAM-A trans-dimer formation in epithelial cell signaling.

**RESULTS**

**Recombinant soluble JAM-A forms homodimers in a pH-dependent manner**

To investigate properties of JAM-A homodimerization, we analyzed recombinant soluble JAM-A ectodomains consisting of the two extracellular Ig-like domains by size-exclusion chromatography to determine the Stokes radius. Dimerization was assessed using several different buffer conditions, with pH values ranging from 5 to 8. Size-exclusion chromatography of soluble, full-length segments of extracellular JAM-A resuspended in calcium-free, Tris-based buffers at pH 8 revealed a prominent elution peak at 41 kDa (Figure 1A). Given that soluble JAM-A monomers have a predicted molecular weight of 25 kDa and dimers have a predicted molecular weight of 50 kDa based on SDS–PAGE analysis, the peak observed using size-exclusion chromatography suggests that under native conditions JAM-A is a dimer, as previously reported (Guglielmi et al., 2007).

To confirm the Stokes radius of monomeric JAM-A by size-exclusion chromatography, we analyzed elution peaks for soluble, extracellular segments of a JAM-A mutant lacking the cis-dimerization motif (6163 JAM-A). Size-exclusion chromatography of soluble extracellular segments of 6163 JAM-A in Tris buffer at pH 8 revealed a single peak with a Stokes radius that corresponds to an apparent molecular weight of 29 kDa (Figure 1B). Together these results suggest that monomeric JAM-A has a Stokes radius corresponding to ~29 kDa at pH 8 and that soluble, extracellular segments of JAM-A preferentially form dimers under native conditions.

Although soluble JAM-A ectodomains form dimers at pH 8, size-exclusion chromatography experiments using JAM-A ectodomains performed in pH 5 citrate buffer revealed a single elution peak equivalent to 25 kDa (Figure 1C), consistent with JAM-A monomers.

![Image](https://example.com/image.png)
Elution of JAM-A at pH 5.6 revealed a major peak equivalent to 28 kDa (Figure 1C), also consistent with monomeric JAM-A. Size-exclusion chromatography of WT JAM-A incubated in pH 6.9 buffer revealed a single peak equivalent to 50 kDa, suggesting that disruption of JAM-A dimers occurs between pH 5 and 6.9 (Figure 1C), consistent with previous reports that murine JAM-A dimers dissociate in acidic conditions (Bazzoni et al., 2000). To ensure that disruption of JAM-A dimers at low pH was not due to protein denaturation, we performed size-exclusion chromatography of soluble extracellular JAM-A that was first incubated at pH 5 or 5.6, followed by adjustment to pH 8. Elution fractions corresponding to monomeric JAM-A that had been resuspended at pH 5 or 5.6 and adjusted to pH 8 before size-exclusion chromatography revealed a peak of the Stokes radius equivalent to 45 kDa (Figure 1C), consistent with a dimeric form of JAM-A. These observations suggest that pH-dependent disruption of JAM-A dimerization is reversible. It is thus possible that the pH dependence of dimerization may play a role in intracellular trafficking of JAM-A, in which low endosomal/lysosomal pH could potentially inhibit dimerization-dependent signaling events.

Identification of JAM-A trans-dimerization sites by site-directed mutagenesis

The chromatography studies in Figure 1, A–C, indicate that JAM-A ectodomains form pH-sensitive homodimers, which is consistent with previous studies that explored a functional role for JAM-A cis-dimerization (Bazzoni et al., 2000; Severson et al., 2008). However, crystallographic analyses (Kostrewa et al., 2001) raise the possibility that JAM-A forms trans-dimers using sequences distinct from those required for cis-dimerization. These studies identified a potential site for trans-dimerization on the protein surface opposite to that used for cis-dimerization on the distal Ig-like domain of JAM-A.

Because the size-exclusion chromatography studies did not reveal JAM-A complexes larger than dimers, we considered the possibility that trans-dimers interact with low affinity. To investigate whether JAM-A forms homodimers across cells (in trans) and identify sites of JAM-A trans-dimerization, we performed mutagenesis of full-length JAM-A, followed by transfection and determination of distribution between expressing cells. Two new constructs were designed containing alanine substitutions within a putative trans-dimerization region located in the D1 domain on the surface opposite to the cis-dimerization motif (Figure 2A), as predicted from the crystal structure of murine JAM-A and platelet adhesion studies (Kostrewa et al., 2001; Babinska et al., 2002b). The two putative trans-dimerization-null mutants have alterations that correspond to distinct motifs on the D1 domain, which combine to form a three-dimensional groove that might be a favorable site for trans-dimerization mediated by nonionic interaction forces (Figure 2B). The NNP mutant protein has alanine substitutions of glutamine 43, glutamine 44, and proline 45 in the predicted trans-dimerization motif (Figure 2C). The KSV mutant protein has alanine substitutions of lysine 97, serine 98, and valine 99 (Figure 2D).

JAM-A trans-dimerization was assessed by overexpressing full-length WT JAM-A (Figure 3A) and the mutant JAM-A constructs (Figure 3, B–E) in CHO cells, which have no detectable endogenous JAM-A. Using immunofluorescence, we evaluated whether the JAM-A mutant proteins accumulated at intercellular contacts. Surface expression of each mutant was confirmed by flow cytometry. Three independent transient transfections of CHO cells with plasmids encoding WT or mutant JAM-A led to similar levels of JAM-A, followed by transfection and determination of distribution between expressing cells. Two new constructs were designed containing alanine substitutions within a putative trans-dimerization region located in the D1 domain on the surface opposite to the cis-dimerization motif (Figure 2A), as predicted from the crystal structure of murine JAM-A and platelet adhesion studies (Kostrewa et al., 2001; Babinska et al., 2002b). The two putative trans-dimerization-null mutants have alterations that correspond to distinct motifs on the D1 domain, which combine to form a three-dimensional groove that might be a favorable site for trans-dimerization mediated by nonionic interaction forces (Figure 2B). The NNP mutant protein has alanine substitutions of glutamine 43, glutamine 44, and proline 45 in the predicted trans-dimerization motif (Figure 2C). The KSV mutant protein has alanine substitutions of lysine 97, serine 98, and valine 99 (Figure 2D).

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domain, did not preferentially distribute to contacts between JAM-A–expressing cells but instead was dispersed diffusely at the cell periphery (Figure 3G, arrowheads). This pattern of distribution suggests that motifs located in the D1 domain are required for concentration of JAM-A between cells. Of interest, NNP and KSV JAM-A also distributed diffusely on the surface of CHO cells and was not restricted to cell–cell contacts between transfected cells (Figure 3, H and I, arrowheads). Consistent with previous observations (Mandell et al., 2004), the JAM-A mutant with alteration at residues 61 and 63 required for cis-dimerization (6163 JAM-A) also diffusely localized on the cell surface, even in areas of cell–cell contact between transfected and nontransfected cells (arrowheads; Figure 3J). Quantification of the frequency of JAM-A accumulation at cell–cell contacts, calculated as the percentage of cells expressing JAM-A predominantly at junctions between two transfected cells relative to the total number of cells expressing JAM-A, revealed that 46% (±4% SEM) of cells expressing WT JAM-A displayed localization of JAM-A to contacts between JAM-expressing cells. In contrast, analyses of cells expressing DL1, 6163, NNP, and KSV mutants revealed significantly reduced JAM-A distribution to cell contacts, with frequencies of 16 (±4 SEM), 30 (±5 SEM), 22 (±3 SEM), and 25% (±2% SEM), respectively (Figure 3K). To ascertain that the results in Figure 3 represented loss-of-function effects specific to the altered residues and that mutagenesis itself was not nonspecifically affecting the localization of JAM-A, we also quantified the rate of JAM-A distribution to contacts of cells expressing a JAM-A mutant protein containing cysteine substitutions at residues lysine 72 and serine 112 in the D1 domain. Of importance, this JAM-A mutant accumulated at junctions at a similar frequency as observed for WT JAM-A (Figure 3K and Supplemental Figure S2), indicating that not all mutations affect the accumulation of JAM-A at cell contacts. These findings indicate that the motifs altered by the 6163, NNP, and KSV mutations are important for the interaction of JAM-A across cells.

Although analysis of the subcellular localization of WT and mutant JAM-A in Figure 3 suggested that both cis and trans motifs are required for localization of JAM-A at cell–cell junctions, we performed additional experiments to test whether mutation of the trans-dimerization motifs globally alters the tertiary structure of JAM-A. Recognition by mAb J10.4, which binds a conformational epitope on the D1 domain of JAM-A (Mandell et al., 2004), was retained in all JAM-A mutants with the exception of DLI JAM-A, suggesting that these mutations do not globally alter the tertiary structure.

![FIGURE 3: Subcellular localization of JAM-A mutants. (A–E) JAM-A cis- and predicted trans-dimer mutants were engineered using site-directed mutagenesis. (A) WT JAM-A is composed of two extracellular Ig-like domains, the most distal of which (D1) is involved in JAM-A homodimerization. (B) DL1 JAM-A lacks the entire membrane-distal Ig-like domain, with only the membrane proximal domain (D2) remaining. (C) NNP JAM-A has alanine substitutions of amino acids 43N, 44N, and 45P, which are predicted to be involved in trans-dimerization. (D) KSV JAM-A has alanine substitutions of amino acids 96K, 97S, and 99V, which compose the second motif predicted to be involved in trans-dimerization. (E) 6163 JAM-A has alanine substitutions of amino acids 61E and 63K, previously implicated in JAM-A cis-dimerization. (F–J) CHO cells were transfected with WT and mutant JAM-A and fixed for immunofluorescence staining and confocal microscopy after 48 h. Subcellular localization of exogenously expressed mutants elucidate sites on JAM-A required for trans-dimerization. WT JAM-A accumulated only at cell–cell contacts between JAM-A–expressing cells. All JAM-A mutants diffusely localized on the cell surface, even in areas of cell–cell contact between transfected and nontransfected cells (arrowheads). Distribution of WT and mutant JAM-A expressed in CHO cells was quantified as a ratio of cells with junction-localized JAM-A between JAM-A–positive cells to total number of JAM-A–positive cells per field. (K) A JAM-A isoform with functionally irrelevant cysteine substitutions at Leu-72 and Ser-112 (L72S112) distributed to junctions at a rate statistically similar to WT JAM-A (n > 8 fields, collected for >3 different transfections; mean ± SEM; ***p < 0.001; *p < 0.05; NS, p > 0.2).]
structure of JAM-A. We further assessed the mutant proteins for functional effects on reovirus infection, which is dependent on binding to JAM-A (Guglielmi et al., 2007; Antar et al., 2009). The cis-dimerization interface of JAM-A mediates reovirus attachment and infection (Guglielmi et al., 2007; Kirchner et al., 2008). Having confirmed expression of the mutant JAM-A proteins by immunoblotting (Figure 4A) and flow cytometry (Supplemental Figure S1), we inoculated CHO cells expressing 6163 or DL1 JAM-A with reovirus and observed that infection rates were significantly lower than those observed in CHO cells expressing WT JAM-A (Figure 4B), which is consistent with previous reports (Forrest, 2003; Guglielmi et al., 2007). Of importance, reovirus infection did not differ significantly between CHO cells expressing WT or the trans-null mutants KSV or NNP (Figure 4C). Taken together, these results suggest that alanine substitutions at the trans-dimerization interface do not globally alter the tertiary structure of JAM-A.

Immunofluorescence staining and confocal analysis of CHO cells expressing WT or mutant JAM-A suggested that cis- and trans-dimerization of JAM-A is required for maximal accumulation of JAM-A at cell–cell contacts. To determine whether JAM-A forms dimers independent of other cellular proteins, we used an in vitro bead-based clustering assay. Soluble, C-terminally histidine (His)-tagged extracellular domains of WT and mutant JAM-A with a C-terminal His tag were purified and analyzed by SDS–PAGE. (B) His-binding 1-μm beads conjugated to His-tagged WT JAM-A clustered in solution (arrowhead) compared with unconjugated beads. Bead clustering was quantified by assessing side and forward scatter using flow cytometry, by which frequency of unclustered beads was measured as a function of total beads. Flow cytometry data were pooled, and the frequency of clustering was calculated by subtracting frequency of unclustered beads from 100%. Baseline clustering observed in unconjugated beads (frequency ~30%) was considered to be background and was subtracted from the final readings observed in experiments using WT, NNP, KSV, and 6163 JAM-A–conjugated beads (C; n > 3 per group, mean ± SEM. **p < 0.01 compared with WT).
Atomic force microscopy defines dimerization properties of JAM-A

To define the biophysical profile of JAM-A homodimerization at the single-molecule level, we used atomic force microscopy (AFM). Soluble His-tagged extracellular domains of WT or mutant JAM-A proteins were bound to AFM tips and substrate using amide-linkage reactions. Amide linkage allowed for JAM-A immobilization in parallel and antiparallel conformations that enabled both cis- and trans-dimerization events. Binding-event frequency was assessed by considering the deflection of the cantilever during movement of the tip toward and away from the substrate. Binding-frequency analysis (Figure 6A) revealed that the cis-null mutant (6163 JAM-A) displayed significantly less frequent homodimerization events than WT JAM-A (4.2 vs. 13.3% for all force curves, respectively, \( p < 0.001 \)), NNP JAM-A, which lacks the motif for trans-dimerization as determined by bead-clustering assays (Figure 5), also showed lower binding frequencies than WT JAM-A (11.7 vs. 13.3%, respectively), although these differences were not statistically significant. Pretreatment of WT, 6163, or NNP JAM-A–coated surfaces with J10.4 Fab′ fragments significantly reduced the binding events. WT JAM-A binding was reduced from 13.3 to 3.1% (\( p < 0.001 \)), 6163 binding was reduced from 4.2 to 3.2% (\( p < 0.05 \)), and NNP binding was reduced from 11.7 to 2.1% (\( p < 0.001 \)). These findings suggest that compared with trans-dimerization, JAM-A cis-dimerization occurs at higher detectable frequencies with AFM. In addition, these results suggest that J10.4 Fab′ fragments inhibit both cis- and trans-dimerization events, since binding detected between 6163 mutants (4.2%) was further reduced with addition of J10.4 Fab′.

Owing to the spring characteristics of the AFM cantilevers, unbinding forces were derived from application of Hooke’s law. Force of binding between WT or mutant JAM-A homodimers was deduced by calculating the unbinding force required to disrupt JAM-A interactions observed at different cantilever retraction speeds ranging from 1 to 10 \( \mu \)m/s. Assessment of the average binding force observed for all binding events at a particular retraction speed revealed that WT JAM-A forms homodimers with greater force at higher retraction speeds, as observed for other junction-associated proteins (Baumgartner et al., 2000; Vedula et al., 2008; Spindler et al., 2009; Zhang et al., 2010). At a pulling speed of 5 \( \mu \)m/s, homodimerization of WT JAM-A ectodomains was on average 133 pN (Figure 6B). In comparison, at a speed of 5 \( \mu \)m/s, the force of interacting cis-null 6163 JAM-A was on average 140 pN. Finally, ectodomains of trans-null NNP JAM-A displayed an average interaction force of 116 pN at the same speed. The difference in unbinding force detected between WT, 6163, and NNP mutants was similar with different pulling speeds. However, force differences between 6163 and WT JAM-A were significant only at two speeds (1 and 5 \( \mu \)m/s, but not 2.5 and 10 \( \mu \)m/s), whereas binding forces between JAM-A NNP ectodomains were significantly lower than forces between WT JAM-A at all loading rates. These data suggest that compared with WT JAM-A, trans-null (NNP) JAM-A mutants interact with weaker forces (\( p < 0.001 \)) at the single-molecule level.

Finally, by assessing the peak unbinding force at different loading rates in a range from \( 10^4 \) to \( 10^5 \) pN/s, we derived the unstressed interaction force of 116 pN at the speed of 5 \( \mu \)m/s. The difference in unbinding force observed for all binding events at a particular retraction speed revealed that WT JAM-A forms homodimers with greater force at higher retraction speeds, as observed for other junction-associated proteins (Baumgartner et al., 2000; Vedula et al., 2008; Spindler et al., 2009; Zhang et al., 2010). At a pulling speed of 5 \( \mu \)m/s, homodimerization of WT JAM-A ectodomains was on average 133 pN (Figure 6B). In comparison, at a speed of 5 \( \mu \)m/s, the force of interacting cis-null 6163 JAM-A was on average 140 pN. Finally, ectodomains of trans-null NNP JAM-A displayed an average interaction force of 116 pN at the same speed. The difference in unbinding force detected between WT, 6163, and NNP mutants was similar with different pulling speeds. However, force differences between 6163 and WT JAM-A were significant only at two speeds (1 and 5 \( \mu \)m/s, but not 2.5 and 10 \( \mu \)m/s), whereas binding forces between JAM-A NNP ectodomains were significantly lower than forces between WT JAM-A at all loading rates. These data suggest that compared with WT JAM-A, trans-null (NNP) JAM-A mutants interact with weaker forces (\( p < 0.001 \)) at the single-molecule level.

Identification of JAM-A trans-dimerization–dependent cell signaling events

JAM-A interacts with a scaffold protein complex that signals to regulate epithelial permeability through activation of the small GTPase Rap2 (Monteiro et al., 2013). However, it is not known whether this signaling module is dependent on dimerization of JAM-A. Given our results indicating that specific regions in the D1 domain of JAM-A mediate dimerization between cells in trans, we transiently transfected full-length WT and dimerization mutant JAM-A in HEK-293T cells and compared the effects of disrupting cis- and trans-dimerization on JAM-A regulation of signals that trigger barrier function. Because loss of JAM-A in epithelial cells results in enhanced permeability linked to decreased levels of active Rap2 (Monteiro et al., 2013), we assessed whether Rap2 activity was altered by expression of WT or mutant JAM-A. HEK-293T cells overexpressing WT JAM-A demonstrated enhanced activity of Rap2 (Figure 7A), which corroborates previous observations of decreased Rap2 activity in JAM-A-deficient cell lines. HEK-293T cells expressing trans-dimerization–null mutants NNP and KSV JAM-A displayed lower Rap2 levels and activity than WT JAM-A transfected cells. Total Rap2 activity, assessed as the signal of active Rap2 standardized to a tubulin loading control, suggested that trans-dimerization is required for the activation of Rap2 (Figure 7A). In contrast, lysates of HEK-293T cells overexpressing 6163 JAM-A, which exclusively lacks the cis-dimerization motif, did not display reduced activity of Rap2 compared with HEK-293T cells expressing WT JAM-A. These results suggest that JAM-A trans- but not cis-dimerization enhances Rap2

FIGURE 6: Atomic force microscopy reveals that JAM-A can form cis- and trans-dimers. (A) Principle of AFM force spectroscopy. A flexible cantilever (nominal spring constant 0.03 N/m), functionalized with recombinant JAM-A, is lowered onto a mica sheet also coated with the same JAM-A molecules until the cantilever is observed to be bending slightly upward (I). In the event of binding interactions between molecules on the cantilever and mica sheet, as the cantilever is retracted, it bends downward, and the forces are quantified. Forces acting on the cantilever are determined.
Cells were assessed for Rap2 activity by Ral-GDS precipitation. (B) Rap2 activity in confluent or spreading human intestinal epithelial cells (SK-CO15) was assessed with Ral-GDS precipitation.

FIGURE 7: JAM-A trans- but not cis-dimerization is required for Rap2 activity. (A) WT and mutant JAM-A were expressed in HEK-293T cells. Cells were assessed for Rap2 activity by Ral-GDS precipitation. (B) Rap2 activity in confluent or spreading human intestinal epithelial cells (SK-CO15) was assessed with Ral-GDS precipitation.

activity in cells. Because JAM-A trans-dimerization events are, by definition, dependent on contacts between adjacent cells, we tested whether Rap2 activity was increased in confluent epithelial cell monolayers. As seen in Figure 7B, Rap2 activity was almost twofold higher in confluent monolayers of epithelial cells than in subconfluent cultures of spreading cells.

Collectively the findings presented suggest that trans-dimerization occurs at a site distinct from that mediating dimerization in cis, and, despite being a lower-affinity binding event, dimerization in trans mediates specific signaling events that regulate activation of Rap2. As illustrated in the model in Figure 8, we propose that JAM-A on the surface of subconfluent single cells does not activate barrier-inducing signals. However, JAM-A on confluent cells trans-dimerizes to form JAM-A multimers, which are required for inducing signals that regulate barrier function.

DISCUSSION

In this study, we demonstrate that JAM-A forms homodimers in trans at a site distinct from that used to form dimers in cis. Trans-dimerization of JAM-A occurs through lower-probability interactions mediated by the distal Ig-like domain at a site directly opposite to the cis-dimerization interface. Disruption of trans-dimerization by mutagenesis results in alterations in small GTPase signaling implicated in regulation of epithelial barrier function. On the basis of the observations presented in this article, we propose a model in which trans-dimerization–dependent JAM-A multimers initiate signaling events that are distinct from those initiated by cis-dimerization alone (Figure 8).

JAM-A trans-dimerization has been predicted by crystallographic studies of recombinant JAM-A ectodomains and in studies of JAM-A–dependent interactions between human platelets and endothelial cells. Before this study, there had been no investigations using biochemical and cell biological approaches to test whether JAM-A trans-dimerization occurs in nature and is functionally relevant.

By expressing WT and mutant forms of JAM-A in CHO cells, we observed that expression of either cis- or trans-dimerization–deficient mutants exhibited lower rates of JAM-A localization to cell–cell contacts, indicating a role for JAM-A oligomerization at cell junctions (Figure 3). We interpret these findings to suggest that JAM-A cis-dimerization is required to provide the necessary avidity for trans-dimerization to occur. In analogous studies, members of the cadherin family of proteins trans-dimerize and accumulate laterally at junctions to form zipper-like cadherin multimers (Brieher, 1996; Takeda et al., 1999; Baumgartner et al., 2000; Zhang et al., 2010).

Consistent with this idea, JAM-A cis-dimerization is predicted to occur at higher frequency than trans-dimerization, likely due to the charged residues involved in JAM-A cis-dimerization that establish a salt bridge between JAM-A monomers (Mandell et al., 2004), whereas JAM-A trans-dimerization would occur by van der Waals forces between mostly uncharged polar residues. These predictions are supported by the chromatography results shown in Figure 1, which reveal the existence of cis-dimers but no higher-order multimers, which were disrupted by the cis-null (6163) mutation.

AFM analysis of purified JAM-A ectodomains detected both cis- and trans-dimerization events. However, cis-dimerization was detected more frequently, suggesting that it is a higher-probability interaction than trans-dimerization, likely due to ionic interactions between charged residues of the cis-dimerization motif. This supports a model in which higher-probability cis-dimerization events may supply the necessary avidity for JAM-A trans-dimerization and, as oligomers are formed, trans-dimerization events become more favorable and stabilize JAM-A oligomers. On the other hand, assessment of binding forces and off rates (Figure 6, B and C) suggested that when trans-dimerization occurs, as observed in binding events between cis-null...
FIGURE 8: JAM-A dimerization in cis may initiate different signaling modalities than that initiated by JAM-A multimerization, which is dependent on trans-dimerization. Left, subconfluent epithelial cells may engage JAM-A cis-homodimers. In the absence of cell–cell contacts (no barrier), the level of active Rap2 is low. Under these conditions, cell migration and spreading are regulated by JAM-A cis-dimerization and increased Rap1. Right, JAM-A trans-dimerization in confluent monolayers of polarized epithelial cells is associated with a tight barrier. Signaling events downstream of JAM-A trans-dimerization lead to the activation of Rap2, which contributes to barrier regulation.

The results shown in Figure 7 indicate that Rap2 activation is reduced by overexpression of trans- but not cis-dimerization–null mutants. As such, we cannot conclude that trans-dimerization requires the formation of cis-dimers, but that both cis- and trans-dimerization of JAM-A are required for its stabilization at cell–cell contacts, as supported by the data in Figure 3.

Our laboratory recently reported that short interfering RNA (siRNA)–mediated loss of JAM-A decreased the activity of the GTPase Rap2, resulting in enhanced epithelial permeability (Monteiro et al. 2013). Of interest, here we show that trans- but not cis-dimerization results in enhanced Rap2 activity (Figure 7A). The finding that JAM-A trans-dimerization specifically affects the activity of Rap2 independently of cis-dimerization implies that JAM-A cis-or trans-dimerization may act as a molecular switch, as depicted in the model shown in Figure 8. Indeed, we previously reported that JAM-A dimerization in cis results in activation of the GTPase Rap1, which regulates β1 integrin protein levels and cell migration (Severson et al. 2008, 2009). It is tempting to speculate that in populations of subconfluent cells, cis-dimerization of JAM-A alone does not initiate barrier-inducing signals, presumably because spreading cells lack the requisite polarized cell–cell contacts; however, they promote cell migration and spreading through preferential activation Rap1. Trans-dimerization of JAM-A, on the other hand, would require contact between adjacent cells, be more robust in confluent cell
populations, and facilitate recruitment of additional signaling proteins leading to the activation of Rap2. The increased Rap2 activity observed in confluent epithelial cell monolayers (Figure 7B) is consistent with trans-dimerization of JAM-A acting as a molecular switch to regulate epithelial barrier function. These findings support a model in which trans interactions of cis-dimers promote assembly of a multicomponent signaling complex to regulate barrier function.

MATERIALS AND METHODS

Cell culture

CHO cells and human embryonic kidney cells (HEK-293T) were grown in DMEM supplemented to contain 10% fetal bovine serum (FBS), 2 mM L-glutamine, 100 IU of penicillin, 100 μg/ml streptomycin, 15 mM 4-(2-hydroxyethyl)-1-piperazineethanesulfonic acid, and 1% nonessential amino acids and were subcultured with 0.05% trypsin (CellGro, Manassas, VA). For reovirus infection studies, CHO cells were cultured in F12 medium supplemented to contain 10% FBS, 2 mM L-glutamine, 100 U/ml penicillin, 100 μg/ml streptomycin, and 25 ng/ml amphotericin B. Cells were transfected with 1–3 μg of plasmid DNA containing WT or mutant JAM-A constructs/10^6 cells using Lipofectamine (Invitrogen, Grand Island, NY) according to the manufacturer’s instructions. Transfected cells were cultured for 48 h before assessment of JAM-A expression, Rap2 activity, immunofluorescence, or infection with reovirus.

Antibodies

The murine monoclonal anti–JAM-A antibodies 1H2A9, J10.4, and JF3.1 were purified as described (Li et al., 2000; Mandell et al., 2004). Other antibodies were purchased as follows: polyclonal affinity-purified rabbit anti–JAM-A (Invitrogen), monoclonal mouse Rap2 and polyclonal affinity-purified rabbit ZO-2 (BD Transduction Laboratories, San Jose, CA), and polyclonal affinity-purified rabbit anti-afadin 02246, monoclonal mouse anti-tubulin, and polyclonal affinity-purified rabbit anti-actin (Sigma-Aldrich, St. Louis, MO). For immunoblots, horseradish peroxidase–conjugated secondary antibodies were used (Jackson ImmunoResearch Laboratories, West Grove, PA). For immunofluorescence studies, fluorescein isothiocyanate– and Alexa-conjugated antibodies (Invitrogen) were used.

Expression and purification of recombinant JAM-A

WT and cis-dimerization–null mutants 6163 and DL1 JAM-A in pCDNA 3.0 were cloned as described previously (Mandell et al., 2004; Severson et al., 2008). The NNP and KSV JAM-A point mutants were engineered using WT JAM-A in pCDNA 3.0 by overlap PCR. Initial amplification of WT JAM-A in pCDNA 3.0 was performed with primer pairs 5′-gctctgacgtcgcacatggqacaagggcgcaaat-3′ and 5′-atatctcagctcacachgqaatgcaggctgctg-3′. NNP and KSV JAM-A point substitutions to alanines were introduced with amplification of plasmid DNA containing WT or mutant JAM-A constructs/10^6 cells using Lipofectamine (Invitrogen, Grand Island, NY) according to the manufacturer’s instructions. Transfected cells were cultured for 48 h before assessment of JAM-A expression, Rap2 activity, immunofluorescence, or infection with reovirus.

Immunoblots

Cells were homogenized in radioimmunoprecipitation assay (RIPA) lysis buffer (20 mM Tris, 50 mM NaCl, 2 mM EDTA, 2 mM ethylene glycol tetraacetic acid, 1% sodium deoxycholate, 1% Triton X-100, and 0.1% SDS, pH 7.4) or 0.1% NP40 or 1% Brij 97 lysis buffer (200 mM Tris, pH 8.0, 150 mM NaCl, 1 mM MgCl_2, 1 mM CaCl_2 supplemented with protease and phosphatase inhibitor cocktails (Sigma-Aldrich). A bicinchoninic acid assay (Pierce) was used to determine lyse protein concentrations. Lysates were cleared by centrifugation and boiled in reducing SDS sample buffer. SDS–PAGE and immunoblots were performed using standard methods. Tubulin was used as a protein loading control.

Immunofluorescence microscopy

Cells were grown on eight-well chambered slides, washed in Hank’s buffered saline solution (HBSS+; CellGro), fixed in 100% ethanol at −20°C for 20 min, and blocked in 5% BSA in HBSS+ for 1 h. Primary antibodies were diluted in blocking buffer and incubated with cells at 4°C overnight. Fluorescently labeled secondary antibodies were diluted in blocking buffer and incubated with cells at room temperature for 45 min. Stained cells were washed in HBSS+ and mounted in Prolong Antifade Agent (Invitrogen). A laser scanning microscope (LSM 510; Carl Zeiss, Thornwood, NY) was used to capture confocal fluorescence images. Images were processed using ImageJ and LSM software.

Rap2 activity assay

Rap2 activity assays were performed according to the manufacturer’s instructions (Millipore, Billerica, MA, and Cell Biolabs, San Diego, CA). Cells were lysed at 4°C in a Tris and Triton X-100–based lysis buffer. Cell debris was removed by centrifugation, and 40 μl was saved as input to determine total Rap2 levels. Lysates containing equal amounts of protein for each sample (0.5–1.5 mg) were incubated at 4°C for 60 min with Ras-GDS agarase beads to bind active Rap2 (Knaus et al., 2007). Beads were washed three times with lysis buffer, followed by boiling in SDS sample buffer. Samples were analyzed by immunoblotting with detection by Rap2 mAb (BD Laboratories).
Flow cytometry of JAM-A–conjugated beads
His-tagged Dynabeads (5 µl; Invitrogen) measuring 1 µm were incubated with 2 µg of WT, 6163, NNP, and KSV JAM-A in 500 µl of PBS at room temperature for 10 min. Conjugated beads were washed with PBS using magnetic racks before resuspension in 500 µl of PBS. JAM-A–induced bead aggregation was assessed by flow cytometry. Single beads were distinguished from doublets, triplets, and larger aggregates by size, as determined by light scatter. Both forward and side scatter were set to logarithmic scale. To determine whether equivalent levels of JAM-A were bound to beads, conjugated beads were washed once with PBS and incubated with FACS buffer (2% FBS in PBS) containing JAM-A–specific monoclonal antibody J10.4. After incubation at 4°C for 30 min with rotation, cells were pelleted, washed twice with FACS buffer, and incubated with FACS buffer containing an Alexa Fluor–conjugated secondary antibody. For each condition, 10⁶ events were examined. The percentage of single beads detected of the total events counted was determined using FlowJo software (Tree Star, Ashland, OR).

Reovirus infection of transfected CHO cells
WT and mutant forms of JAM-A were expressed in CHO cells and assessed for the capacity to bind reovirus. JAM-A expression in transfected CHO cells was assessed by removing cells from tissue culture plates using CellStripper (Mediatech, Manassas, VA), and pelleting cells at 1000 × g. The mean fluorescence intensity of each sample was determined using flow cytometry.

Reovirus infection of CHO cells transfected with WT and mutant forms of JAM-A was quantified after virus adsorption at a multiplicity of infection (MOI) of 100 plaque-forming units (PFUs)/cell at 37°C for 1 h. Cells were washed twice with PBS, and fresh medium was added to each respective sample. Cells were pelleted, washed once with PBS, and incubated with FACS buffer containing Alexa Fluor–conjugated reovirus–specific antisera. The percentage of reovirus antigen–positive cells was determined using flow cytometry. Cell staining results were quantified using FlowJo software.

Atomic force microscopy
A Nanowizard III AFM (JPK Instruments, Berlin, Germany) mounted on an optical microscope (Axio Observer.D1; Carl Zeiss Microscopy, Jena, Germany) was applied to quantify JAM-A interactions by AFM. Recombinant JAM-A proteins were coupled to flexible Si3N4 AFM cantilevers (MLCT probes, spring constant 0.03 N/m; Bruker, Calle Tecate, CA) and mica sheets (SPI Supplies, West Chester, PA) via flexible polyethylene glycol spacers (acetal-PEG-NHS) as described (Spindler et al., 2009; Wildling et al., 2011). AFM cantilevers and mica were functionalized with amino groups by ethanolamine treatment and coupled to the N-hydroxysuccinimide ester group of the heterobifunctional linker. The acetal function of the linker was converted to an aldehyde group by citric acid treatment to allow reaction with the amino groups of JAM-A.

To measure JAM-A interactions, the AFM tip was lowered onto the mica surface and retracted again, and binding events were detected by continuously measuring the deflection of the cantilever as described (Spindler et al., 2009). The AFM cantilever was moved in constant-force mode with speed of 1 µm/s in a z-range of 300 nm, 0.1-s delay time on the mica, and a retraction set point of 200 pN. At least 500 approach–retract cycles at 25 different positions on the mica were recorded for each cantilever/mica combination. Interactants were maintained in HBSS at 37°C. J10.4 was incubated at a concentration of 15 µg/ml for 30 min.

Statistics
All pooled data figures are representative of at least three independent experiments. Mean values were compared using an unpaired two-tailed Student’s t test between two experimental groups. Error bars indicate SE of the mean. p < 0.05 was considered to be statistically significant.

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