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Neutrophils are abundant circulating leukocytes that are rapidly recruited to sites of inflammation in an integrin-dependent fashion. Contrasting with the well-characterized regulation of integrin activation, mechanisms regulating integrin inactivation remain largely obscure. Using mouse neutrophils, we demonstrate in this study that the GTPase activating protein ARAP3 is a critical regulator of integrin inactivation; experiments with Chinese hamster ovary cells indicate that this is not restricted to neutrophils. Specifically, ARAP3 acts in a negative feedback loop downstream of PI3K to regulate integrin inactivation. Integrin ligand binding drives the activation of PI3K and of its effectors, including ARAP3, by outside-in signaling. ARAP3, in turn, promotes localized integrin inactivation by negative inside-out signaling. This negative feedback loop reduces integrin-mediated PI3K activity, with ARAP3 effectively switching off its own activator, while promoting turnover of substrate adhesions. In vitro, ARAP3-deficient neutrophils display defectivePIP3 polarization, adhesion turnover, and transendothelial migration. In vivo, ARAP3-deficient neutrophils are characterized by a neutrophil-autonomous recruitment defect to sites of inflammation.

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Proximally, this involves the adapters talin and kindlin-3, which directly bind to integrin cytoplasmic tails, promoting their activation (6, 7), with Rap and its effectors, Rap1L, RIAM, and Radil, acting upstream. Excessive integrin activity has also been shown to interfere with leukocyte recruitment (8, 9), but mechanisms governing integrin inactivation in this context remain poorly defined.

Class I (agonist-activated) PI3Ks transduce signals through the generation of the lipid second messenger phosphatidylinositol 3,4,5- triphosphate (PIP3) by phosphorylation of PI(4,5)P2 in the plasma membrane. Four class I PI3K isoforms exist and are expressed by the neutrophil: α, β, γ, and δ (10). Class I PI3K isoforms are activated upon receptor ligation by SH2 domain binding to phosphotyrosine motifs in receptors or their adaptors (e.g., in integrin outside-in signaling) and G protein βγ subunits, as well as Ras/Rho family small GTPases. PI3P causes the recruitment to the plasma membrane and activation of numerous PI3K effectors proteins, including several regulators of small GTPases.

ARAP3 is a PI3K- and Rap-regulated GTPase activating protein for RhoA and Arf6 that was identified as a PI3P binding protein from pig neutrophils (11, 12). ARAP3 shares its domain structure with ARAP1/2, which differ in their expression profiles and substrate specificities (11, 13–15). We previously showed ARAP3 promotes integrin inactivation. Despite focusing in this study on integrin activation triggers a negative feedback loop downstream of PI3K by which the data presented in this study identify that integrin activation provides PI3K-dependent recruitment to integrin inactivation. As well, study on ARAP3-deficiency interferes with efficient neutrophil recruitment function of ARAP3 is also broadly applicable elsewhere. As well, study on ARAP3 promotes integrin inactivation. Despite focusing in this study on integrin activation triggers a negative feedback loop downstream of PI3K by which the data presented in this study identify that integrin activation provides PI3K-dependent recruitment to integrin inactivation. As well, study on ARAP3 promotes integrin inactivation. Despite focusing in this study on integrin activation triggers a negative feedback loop downstream of PI3K by which the data presented in this study identify that integrin activation provides PI3K-dependent recruitment to integrin inactivation.

Materials and Methods

Unless indicated otherwise, cell culture reagents were from Life Technologies, cell culture plastics were from Corning, and all other materials were from Sigma. All reagents were of the lowest available endotoxin grade. PI3K inhibitors (Selleck Chemicals) and final concentrations used were as follows: pan-PI3K, wortmannin (50 μM); PI3Kγ, BYL-719 (0.25 μM); PI3Kδ, TOX-221 (40 nM); and PI3Kδ, IC87114 (1 μM).

Inducible Arap3 mice model

To analyze neutrophils in vitro, 10–12-wk-old sex-matched Arap3fl/fl ERT2Cre mice were induced with a single i.p. injection with 200 mg/kg tamoxifen or vehicle, with experiments performed 10–12 d after induction as described (16). For in vivo experiments, age- and sex-matched Arap3fl/fl ERT2Cre mice and Arap3−/−ERT2Cre controls were subjected to five successive gavages with emulsion containing 1.5 mg of tamoxifen, followed by a rest period of 10 d (Supplemental Fig. 3A for an example). For ease of reading, tamoxifen-induced Arap3fl/fl ERT2Cre mice (or neutrophils) are referred to in the text as ARAP3-deficient and in figures as −/−, whereas vehicle-induced Arap3fl/fl ERT2Cre and tamoxifen-induced Arap3fl/fl ERT2Cre controls are referred to as controls and +/+ with explanations provided in the figure legends. All mice were housed in a specific pathogen-free small animal barrier unit at the University of Edinburgh. All animal work was approved by the University of Edinburgh Animal Welfare Committee and conducted under the control of the U.K. Home Office (PPL 604502 and PFFB 42579).

Neutrophil preparations

Bone marrow–derived mouse neutrophils were prepared on a discontinuous Percoll gradient as previously described (17), using endotoxin reagents throughout, yielding >70% purity as assessed by cytocentrifuge preparations. Unless stated otherwise, experiments were performed in Dulbecco’s PBS supplemented with Ca2+ and Mg2+. 1 g/l glucose, and 4 mM sodium bicarbonate.

Adhesion-induced neutrophil functions

Tissue culture wells were coated overnight at 4°C with fibronectin as indicated. Surfaces were washed three times with PBS, blocked with 10% FBS in PBS, and washed again before addition of prewarmed neutrophils. ROS production was measured indirectly using chemiluminescence produced by 5 × 105 neutrophils per well at 37°C with 150 μM luminol and 18.75 U/ml HRP in the presence or absence of TNF-α (20 ng/ml final concentration) in chemiluminescence-grade 96-well plates (Nunc) using a Cytation plate reader (BioTek) essentially as described (16). Where indicated, neutrophils were preincubated with inhibitors for 10 min at 37°C at the indicated concentrations. Where blocking peptides were employed, neutrophils were plated onto the immobilized stimuli and the competing peptide, such that both were encountered at the same time. Neutrophil adhesion, spreading, and degranulation assays were done as previously described (16). For adhesion to endothelial cells, bEnd5 cells were seeded into 24 wells, allowed to form confluent monolayers for 2 d, and stimulated with 5 nM TNF-α for 16 h. After washing and careful aspiration, 100 μl of HBSS (with Ca2+ and Mg2+) containing 1 × 105 neutrophils were added and allowed to bind to the stimulant immobilized endothelial cells under gentle rocking. After 30 min, nonadherent neutrophils were washed away with HBSS (without Ca2+ and Mg2+). Adherent neutrophils were fixed with PFA, labeled for GR1 (clone RB6-8C5; BioLegend), and counted in randomly taken frames (EVOS imaging system; Advanced Microscopy Group/Thermo Fisher). Transendothelial migration toward the indicated concentrations of MIP2 (R&D Systems) for 1 h in 6.5-mm transwell inserts with 3-μm pore polycarbonate membranes (Corning) was performed essentially as described (17). Transmigrated neutrophils were labeled for GR1, and eight random fields of view were photographed and counted (×20 magnification; EVOS imaging system).

ARAP3 knockdown in αIbb3-bearing Chinese hamster ovary cells

αIbb3-Expressing Chinese hamster ovary (CHO) cells were transduced with lentiviral short hairpin RNAs (shRNAs) directed against mouse ARAP3. shRNA sequences (shRNA1, 5′-CTCGCGCTGAAGGTGTATATG-3′ and 5′-GGAATCCGCAAGAAGTTAAA-3′; shRNA2, 5′-GCAGAAGTGGCGCTCTCTAAAAA-3′ and 5′-TGATGAGACGGCACAGTAGTATG-3′) identified from the Broad Institute RNA interference consortium database (https://portals.broadinstitute.org/gpp/public) were used alongside a non-targeting control (NTC; 5′-GGCGCATAGCGCTAATAATT-3′). Oligonucleotides were synthesized (Sigma-Genosys) and cloned into plKO.1 (18), inserts were sequenced, lentiviral particles were generated, and transduced CHO cell populations were selected with puromycin. Samples were analyzed by Western blot using sheep anti-ARAP3 antisera (11) and anti-human CD41 (MAB7616; R&D Systems), with HPS90 (clone H135C27; BioLegend) serving as loading control.

Direct analysis of integrin activity status

Activated β1 integrin was detected using an activation epitope–specific Ab (clone 9E97; BD Biosciences) with an AF488-conjugated secondary Ab (Jackson Immunoresearch). Images were acquired with a 63× objective with a Zeiss LSM780 confocal microscope with Zeiss Zen Black software. The corrected total cellular fluorescence was calculated using ImageJ by selecting regions for each cell and nearby regions of background and applying the following formula: corrected total cellular fluorescence = integrated density – (area of selected cell × mean fluorescence of background readings).

Neutrophil binding to an AF647-labeled fibronectin fragment was performed essentially as described (19) using flow cytometry using a 5L LSRFortessa (BD Biosciences). Analysis was performed using FlowJo software (version 10) by gating for singlets, selecting neutrophils based on forward- and side-scatter profile, and measuring the geometric mean fluorescence intensity. Similarly, CHO cell binding to AF568-conjugated phalloidin (Thermo Fisher Scientific); random images were acquired at ×20 magnification (EVOS imaging system). Prior to measuring cell areas with ImageJ, binary images were thresholded, and the watershed feature was applied to define single cells.
Indirect analysis of PI3K activity

Neutrophil lysates were subjected to immunoblotting with a phosphospecific anti-PI3K antibody (clone C25E6; Cell Signaling Technology) essentially as described (17), with Syk used as loading control (clone 5F5; BioLegend).

Analysis of GFP–PH–PKB reporter distribution

Micropipette chemotaxis assays were conducted, and polar plots were derived and overlaid using Anagraph (S. Andrews, The Babraham Institute) and QuimP software (20) (Garching Innovation) as described (21).

Neutrophil adhesion under laminar flow conditions

Purified neutrophils were preincubated for 10 min at 37°C with PI3K inhibitors or vehicle as indicated prior to being perfused through flow chamber slides (Bidi VI) that had been coated with recombinant murine (rm) ICAM-1 (15 μg/ml), rm P-selectin (10 μg/ml; both BioLegend), and rm CXCL1 (10 μg/ml; Biotechne) using a syringe pump (Legato 200; KD Scientific) to deliver a constant shear stress of 1 dyne/cm² at 37°C. Adhesion under flow was captured with >20 magnification by time lapse imaging (2.5 images/s) for 1 min at 1, 5, 10, and 15 min after starting the flow. This was done using a Leica IRB inverted microscope equipped with a temperature-controlled automated stage (Prior), an Orca camera (Hamamatsu), and Micro-Manager image acquisition software (Fiji). Firmly adherent cells were manually counted using ImageJ.

LPS-induced acute lung inflammation

LPS-induced acute lung inflammation (ALI) was performed essentially as described (22). Some mice received 3 μg of allophycocyanin–anti-mouse (30-F11; BioLegend) in 100 μl of sterile PBS i.v. 15 min prior to being sacrificed 4 h after LPS administration, such that in lung digest samples, neutrophils could be stratified by CD45+ and CD45−. BM cells and lung digestes were labeled with FITC–anti-GR1 and allophycocyanin–anti-CD11b (BioLegend) and analyzed by flow cytometry to calculate total neutrophil numbers (GR1high, CD11b−). For imaging, lungs were perfused through the right ventricle with 10 ml of saline, and a portion of the right inferior lobe was collected for single-cell digestion with collage- nase (Roche) and subsequent analysis. Bronchoalveolar lavage (BAL) cells were counted (NucleoCounter; Sartorius). BAL cells and lung digestes were labeled with FITC–anti-GR1 and allophycocyanin–anti-CD11b (BioLegend) and analyzed by flow cytometry to calculate total neutrophil numbers (GR1high, CD11b−). For imaging, lungs were perfused with low-melting-point agarose, allowed to set on ice, dissected, and fixed with formaldehyde. Left lungs were precision-sliced 300 μm thick using a vibratome (5100 MZ; Campden Instruments), permeabilized, blocked, and labeled for PECAM-1 (clone 2H8; Abcam) and S100A9/MRP14 (Hycult Biotech) with DAPI counterstaining. Following brief formaldehyde postfixation, slices were mounted using Mowiol containing 2.5% (w/v) DABCO in gaskets and analyzed using a confocal laser scanning microscope to produce tile-scanned z-stacks (LSM 880 nonlinear optical Airyscan Fast using a 20× plan Apo 1.0 numerical aperture, water immersion objective and 405-, 488-, and 561-nm continuous wave lasers and acquiring in Airyscan Fast mode; Zeiss). Image analysis was performed using IMARIS software (versions 8 and 9, Bitplane; Oxford Instruments). Endothelial surfaces (PECAM-1+) were quantified and overlaid using Anagraph (S. Andrews, The Babraham Institute) and QuimP software (20) (Garching Innovation) as described (21).

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cell integrins. Preincubating the cells with αIbβ3-blocking abciximab significantly reduced the area occupied by control and ARAP3-knockdown CHO cells, suggesting that heterologous αIbβ3 mediated most fibrinogen binding. Interestingly, however, abciximab-preincubated ARAP3-knockdown cells remained more spreadable than controls, suggesting that ARAP3 inactivates not only αIbβ3 but also endogenous hamster integrins that were also capable of binding fibrinogen without being affected by the blocking Ab. Inhibiting PI3K significantly reduced the areas occupied by control and ARAP3-knockdown CHO cells. No significant difference remained between experimental groups after treatment with wortmannin. These observations are in keeping with ARAP3 being a PI3K effector that is able to regulate many integrins, including heterologous human αIIbβ3 in CHO cells.

**ARAP3 acts in a negative feedback loop downstream of integrin and PI3K**

Having established that ARAP3 mediates integrin inactivation, we turned our attention to upstream signaling. In the neutrophil, ARAP3’s master regulator, PI3K, is activated by integrin outside-in signaling downstream of Src family kinases/Syk (27), with PI3Kβ and δ isoforms implicated in mediating integrin-dependent responses (28).

To probe the relationship between integrin, PI3K, and ARAP3, we analyzed ROS production with neutrophils that had been plated onto fibronectin in the presence or absence of TNF-α. Integrin ligation-induced ROS depends on PI3K generation through class I PI3K, in particular PI3Kβ and δ (28), whereas SHIP1 (29) or ARAP3 (Fig. 1) deficiency causes increased adhesion-dependent ROS. Inhibiting individual class IA PI3K isoforms reduced adhesion-induced ROS production observed with control and ARAP3-deficient neutrophils and abrogated significant differences observed between genotypes (Fig. 3A).

ROS production is dependent on PI3K-activated Rac guanine nucleotide exchange factors, inhibition of which could potentially explain the above result. We therefore also analyzed the PI3K dependency of degranulation with control and ARAP3-deficient neutrophils that have been stimulated by being plated onto fibronectin in the presence or absence of TNF-α. Inhibiting class IA PI3Ks also reduced the enhanced degranulation that is characteristic of ARAP3-deficient cells; in particular, following PI3Kδ inhibition, no significant difference remained between genotypes (Fig. 3B).

We analyzed adhesion and spreading of control and ARAP3-deficient neutrophils after PI3K inhibition to fibronectin-coated plastic. Inhibiting PI3Kβδ did not significantly affect the ability of neutrophils to adhere to fibronectin, in keeping with an earlier report that had analyzed neutrophil adhesion to immobilized immune complexes (28), data not shown. However, it resulted in compromised neutrophil spreading in both genotypes, putting an end to significant differences between them (Fig. 3C).

Finally, we compared adhesion of neutrophils under constant flow in parallel-plate flow chambers. As previously reported (16), we noted increased neutrophil adhesion with ARAP3-deficient neutrophils compared with controls. Preincubating the neutrophils with a PI3K-specific inhibitor caused decreased neutrophil adhesion in both genotypes (Fig. 3D). Notably, this abolished the significant difference in adhesion observed between genotypes in the absence of inhibitor treatment. Together, these results show that ARAP3 acts downstream of PI3K in neutrophil adhesion and adhesion-dependent neutrophil functions. Given the heightened responses observed with ARAP3-deficient neutrophils, they also suggest the existence of a negative feedback loop.

For experimental evidence of this feedback loop, we analyzed PKB/Akt T308 phosphorylation as an indirect readout for PI3K activity with neutrophils that did or did not express ARAP3. PKB T308 phosphorylation was increased more dramatically in ARAP3-deficient than control neutrophils that had been plated onto fibronectin in the presence or absence of TNF-α. Interestingly, however, it resulted in compromised neutrophil spreading in both genotypes, putting an end to significant differences between them (Fig. 3C).

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For experimental evidence of this feedback loop, we analyzed PKB/Akt T308 phosphorylation as an indirect readout for PI3K activity with neutrophils that did or did not express ARAP3. PKB T308 phosphorylation was increased more dramatically in ARAP3-deficient than control neutrophils that had been plated onto fibronectin in the presence or absence of TNF-α. Interestingly, however, it resulted in compromised neutrophil spreading in both genotypes, putting an end to significant differences between them (Fig. 3C).
Integrin–PI3K–ARAP3 negative feedback signaling regulates persistent neutrophil polarization during chemotaxis

Chemotaxing neutrophils are characterized by polarized PIP3 at the pseudopod (30, 31). To analyze whether the negative feedback loop delineated in this study operates to control neutrophil behavior, we analyzed PIP3 generation in the chemotaxing neutrophil in a spatiotemporal fashion. Having crossed inducible ARAP3-knockout mice with mice expressing a PIP3 probe, GFP–PKB–PH (30), we used confocal microscopy to monitor PIP3 production in real time in control and ARAP3-deficient neutrophils that were allowed to chemotax on glass coverslips toward fMLF. Control cells displayed persistent PIP3 polarization toward the chemoattractant. In contrast, ARAP3-deficient cells were unable to polarize PIP3 persistently, with poles observed to move around cells; more than 50% of ARAP3-deficient neutrophils exhibited additional poles (Fig. 4A for an example). We generated polar plots (21, 31), to visualize PIP3 polarization over time in individual neutrophils (data not shown). Overlays of these polar plots confirmed the poor persistency of PIP3 polarization of ARAP3-deficient neutrophils (Fig. 4B).

In the absence of a probe for activated integrins, we were unable to test whether nonpersistent PIP3 polarization of ARAP3-deficient neutrophils coincided with poor turnover of activated integrins. Fixed, adherent fMLF bath-stimulated control, and ARAP3-deficient neutrophils were characterized by polarized activated β1 integrin staining at the pseudopod, where it coincided with F-actin (Supplemental Fig. 2). For efficient forward motion of the neutrophil, these adhesions must be short-lived. Given that ARAP3 is recruited to the plasma membrane by PIP3 (11), it is well placed to be involved in localized integrin inactivation, ensuring persistence of polarization and directionality.

ARAP3 regulates neutrophil transendothelial migration and recruitment to sites of inflammation

We next determined the requirement for ARAP3-dependent integrin inactivation in neutrophil recruitment to inflammatory sites. Whereas interstitial migration is thought to be integrin-independent, barriers
need to be overcome in an integrin-dependent fashion for neutrophil recruitment (e.g., during transendothelial migration). We first addressed whether the increased integrin activity of ARAP3-deficient neutrophils influences interactions with endothelial cells and transendothelial migration efficiency in vitro. As expected, we found that ARAP3-deficient neutrophils adhered more strongly than controls to monolayers of activated endothelial cells (Fig. 5A). Furthermore, ARAP3-deficient neutrophils were characterized by impaired migration to chemoattractant in a model for transendothelial migration, where transwells supported a monolayer of endothelial cells.

**FIGURE 3.** A negative feedback loop involving integrin, PI3K, and ARAP3. Neutrophils were prepared from bone marrow of mock (+/+) and tamoxifen-induced (−/−) inducible Arap3-knockout mice and (A–D) preincubated with PI3K inhibitors or vehicle controls as indicated. (A) ROS production and (B) gelatinase release were analyzed with neutrophils that had been plated onto 20 μg/ml fibronectin in the presence or absence of 20 ng/ml TNF-α. Graphs combine results from four separate experiments. (C) Neutrophils were allowed to adhere for 20 min to 5 μg/ml fibronectin-coated tissue culture plastic in the presence or absence of 20 ng/ml TNF-α for analysis of spreading. Results obtained in three separate experiments are integrated in this graph. (D) Neutrophil adhesion under flow. Neutrophils were perfused at constant shear stress through ICAM-1–, P-selectin–, and CXCL1-coated flow chambers as detailed in Materials and Methods. Results obtained in at least five separate experiments are combined in the graph shown. (E and F) Neutrophils were allowed to adhere to tissue culture dishes that had been coated with heat-inactivated FCS (HI-FCS) or a synthetic pan-integrin ligand, poly-RGD (pRGD) for 15 min at 37°C. (G and H) Suspension neutrophils were stimulated with 1 μM fMLF for the indicated length of time. Lysates were subjected to SDS-PAGE and Western blots for probing with a phosphospecific Akt/PKB Ab (T308) as well as a loading control (Syk). Representative blots are shown (D and F), and results obtained from four separately performed experiments are plotted (E and G). All graphs show mean ± SEM. (A)–(C) were analyzed by one-way ANOVA with multiple-comparison post hoc tests; (D) and (H) were analyzed by two-way ANOVA with Bonferroni multiple-comparison tests. Pairwise comparisons (F) were calculated from raw data by unpaired two-tailed Student t tests. (A, F, and H) Analyses were performed on the raw data. Symbols in graphs (A)–(D) refer to differences between control and ARAP3-deficient neutrophils (in the absence of inhibitor treatment). No significant differences between genotypes were identified in (H). *p < 0.05, **p < 0.01, ***p < 0.001.
TNF-α–stimulated endothelial cells (Fig. 5B). In contrast, ARAP3-deficient neutrophils were not defective in transwell chemotaxis (Fig. 5C), in line with our previous findings. Together this suggested that ARAP3-dependent integrin inactivation might be relevant for neutrophil recruitment in vivo.

We therefore analyzed neutrophil recruitment in response to LPS-induced ALI in control and ARAP3-deficient mice. We noted significantly reduced neutrophil numbers in BAL from ARAP3-deficient mice compared with controls (Fig. 6A). This held true with bone marrow chimeras, identifying the recruitment defect as neutrophil-autonomous (Fig. 6B).

To reach the alveolar space, neutrophils have to breach two barriers, the capillary wall and the alveolar epithelium. To differentiate between neutrophils that were firmly adherent to the luminal side of the vessel wall or undergoing transendothelial migration and those that were interstitial (i.e., that had extravasated but not yet breached the epithelial barrier), we generated precision slices of agarose-perfused, inflamed lung tissue, labeling endothelium and neutrophils. Microscopic analysis of such lung slices suggested that larger numbers of ARAP3-deficient neutrophils had adhered to the lung vasculature and/or were in the process of transmigrating in ARAP3-deficient lungs (Fig. 6C, 6D). We also used flow cytometry for a separate, higher-powered quantitative approach to the same question. Mice were administered a fluorescently conjugated anti-CD45 Ab i.v., labeling fully or partially intravascular leukocytes immediately prior to harvesting PBS-perfused, LPS-inflamed lungs for analysis of tissue homogenates. This identified significantly increased numbers of ARAP3-deficient neutrophils (but not macrophages) that had firmly adhered to the vessel wall or were actively transmigrating at the time of perfusion (Fig. 6E, Supplemental FIGURE 4. Integrin–PI3K–ARAP3 negative feedback signaling improves neutrophil polarization. Neutrophils were prepared from bone marrow of mock (+/+)) and tamoxifen-induced (−/−) inducible Arap3-knockout mice expressing a GFP–PKB–PH PIP3 reporter. Cells were allowed to settle on a glass coverslip and then subjected to a point source of chemoattractant (micropipette). Cells were imaged using a Perkin Elmer spinning disk Nikon Eclipse TE2000E confocal microscope using a 100× oil immersion objective. Images were acquired every second for 5 min using a Hamamatsu cooled charge-coupled device camera. (A) Stills taken from a representative control and ARAP3-deficient neutrophil. Yellow asterisk symbols indicate polarization. (B) The distribution of the PIP3 probe along the edge of each frame of the video was analyzed using QuimP software, measuring the image intensity at 100 nodes around the plasma membrane. The signal intensity along the membrane was normalized to that within the cell body. Intensity measurements were plotted using Anagraph, with each frame mapped onto a concentric ring and signal intensity represented by color-coding to generate polar plots. The images shown represent overlays of polar plots generated with 25 control, and 24 ARAP3-deficient neutrophils originating from six individual animals per genotype.

FIGURE 5. ARAP3-regulated integrin inactivation promotes transendothelial migration in vitro. Neutrophils were prepared from bone marrow of mock (+/+)) and tamoxifen-induced (−/−) inducible Arap3-knockout mice. Neutrophil adhesion (A) to activated mouse endothelial (bEND5) cells. Neutrophil transendothelial migration and chemotaxis (B and C) toward the indicated concentrations of chemoattractant in transwells that did (B) or did not (C) support a monolayer of activated bEND5 cells. Graphs integrate data obtained from three to four separate experiments. All bar graphs show mean ± SEM. Pairwise comparisons were analyzed by unpaired two-tailed Student t tests. *p < 0.05, **p < 0.01.
Fig. 3B, 3C). We concluded that ARAP3-mediated neutrophil integrin inactivation enables efficient transendothelial migration, promoting neutrophil recruitment in vivo (Fig. 7).

**Discussion**

The present work identifies ARAP3 as a regulator of integrin inactivation in the neutrophil and elsewhere. Our findings place ARAP3 downstream of PI3K in a negative feedback loop that promotes integrin inactivation (Fig. 7). This mechanism enables rapid switching-off of integrins following ligand binding–induced outside-in signaling. This feedback loop operates in adherent neutrophils, in which ARAP3-dependent neutrophil activities are entirely dependent upon outside-in signaling-induced upstream PI3K activity. ARAP3 deficiency results in increased integrin activity, which in turn causes increased integrin-induced PI3K activation and downstream events.

We used integrin-dependent neutrophil chemotaxis as an experimental system in which to analyze the integrin–PI3K–ARAP3–integrin negative feedback loop in a spatiotemporal fashion. ARAP3-deficient neutrophils that chemotaxed on glass toward a point source of chemoattractant polarized PIP3 and generated pseudopods, but these were not persistently directed toward the source of chemoattractant; ARAP3-deficient neutrophils frequently displayed two (or more) poles. This is consistent with the poor integrin-dependent chemotactic migration of these cells (16). In chemotaxis on a two-dimensional matrix, class I PI3Ks are activated downstream of chemoattractant-induced GPCR signaling but also by integrin outside-in signaling. Our results suggest that ARAP3 signaling is engaged to regulate integrin inactivation in response to integrin (but not GPCR) stimulation downstream of PI3K. Our observations are consistent with the possibility that ARAP3 might simply be recruited to PIP3 in the polarized neutrophil to inactivate integrin signaling in a spatiotemporally controlled fashion, limiting further integrin-dependent localized activation of PI3K and enabling pseudopod extension. Alternatively, further players, such as PIP3 metabolizing enzymes, might also be recruited to the pseudopod to actively dephosphorylate PI3P. The functions of two PIP3 phosphatases, PTEN and SHIP1, have been analyzed in chemotaxis (29, 30, 32, 33). SHIP1 is activated and functions in adherent neutrophils, in which it regulates neutrophil spreading, chemotaxis, and PIP3 polarization, whereas PTEN is thought to regulate other features.
Physiologically, intracellular neutrophil migration is thought to be integrin-independent, whereas transendothelial migration is integrin-dependent, with some variability depending on capillary bed and stimulus (1, 2, 34). Our work suggests that in these situations, ARAP3-dependent neutrophil integrin inactivation regulates efficient neutrophil recruitment to inflammatory sites by promoting neutrophil extravasation. This identifies that neutrophil extravasation not only requires activation of integrins but, moreover, relies on their subsequent inactivation. The existence of an integrin inactivation step that regulates efficient immune responses had been predicted by an earlier report, in which rendering αLβ2 constitutively active genetically delayed T cell recruitment (9). Similarly, rendering αMβ2 constitutively active using a small molecule interfered with efficient neutrophil recruitment to inflammatory sites (8). Given that ARAP3 is highly expressed in neutrophils but not in lymphocytes (11), we speculate that integrin inactivation in lymphocytes is controlled by alternative mechanisms. ARAP1/2 are already implicated in the control of adhesion-dependent processes elsewhere (35, 36) and are expressed in lymphocytes (37), suggesting that other ARAP family members might be involved in these cells.

In addition to demonstrating ARAP3-dependent inactivation of neutrophil β1 integrins, our work shows indirectly that ARAP3 also regulates neutrophil integrins that bind to substrates other than fibronectin (e.g., vitronectin, fibrinogen, and ICAM-1; data not shown and Ref. 16). ARAP3, moreover, inactivated heterologous human αIIbβ3 as well as endogenous hamster integrins in CHO cells, again in a PI3K-dependent fashion. Given that ARAP3 is expressed in CHO cells but not in platelets [which express ARAP1; (37)], αIIbβ3 is not a likely bona fide ARAP3 substrate. Rather, these observations suggest a more general function of ARAP3 downstream of PI3K in integrin inactivation. This is of interest given ARAP3’s crucial function in developmentally sprouting angiogenesis and lymphangiogenesis (23, 38), processes that are not only absolutely dependent upon integrins (39) but also integrin-independent, whereas transendothelial migration is integrin-dependent, with some variability depending on capillary site of inflammation: the leukocyte adhesion cascade updated. J. Cell Sci. 17: 678–689.


References


Figure S1. ARAP3-deficient neutrophils are hyperactive in integrin-dependent situations.
Neutrophils were prepared from bone marrows of mock (+/+) and tamoxifen induced (-/-) inducible Arap3 knock-out mice. (A) Analysis of ROS production of neutrophils that were stimulated by being plated onto 20 µg/ml fibronectin (FN) coated plastic in the presence or absence of 20 ng/ml TNFα in a luminol-enhanced chemoluminescence assay. This graph shows the accumulated light emission integrated from 4 separately performed experiments. (B-D) Neutrophil degranulation. Lactoferrin (B) and gelatinase (C, D) release of neutrophils that had been stimulated by being plated onto 20 µg/ml FN in the presence of absence of 20 ng/ml TNFα. A representative Coomassie-stained gel (C) and combined data from 4 separately performed experiments (B, D) are presented. Adhesion and spreading (E-G) were analyzed with neutrophils that had been plated onto 5 µg/ml FN in the presence of absence of 20 ng/ml TNFα. A representative example of adhered neutrophils is presented (E), and averaged numbers of adhered (G) and spread (F) neutrophils per field of view in the indicated conditions obtained in 3 separate experiments. (H, J) Surface expression of β1 integrin was assessed by flow cytometry. A representative example plot shows cell surface β1 integrin in non-stimulated neutrophils (H) and integrated data from 6 separately performed experiments with neutrophils that were or were not stimulated with 20 ng/ml TNFα (J) are plotted. All bar graphs show mean ± SEM. * P<0.05; ** P<0.01. P-values were calculated from raw data by Mann-Whitney U test.
Figure S2. Activated integrins localize to the leading edge of chemoattractant stimulated neutrophils. Neutrophils were prepared from bone marrows of mock (+/+) and tamoxifen induced (-/-) inducible Arap3 knock-out mice. Cells were allowed to settle on a glass coverslip, subjected to a bath stimulation with chemoattractant, fixed, labelled for activated β1 integrin and filamentous actin and analyzed by confocal microscopy using a Zeiss LSM780 confocal microscope and 63x objective. Representative images of control and ARAP3-deficient cells are shown; arrows indicate pseudopods. Scale bar, 5 µm.
Fig S3. Deletion of ARAP3 does not affect monocyte/macrophage localization in LPS ALI. (A) Efficient long-term deletion of Arap3 by repeat gavaging of inducible knock-outs. Arap3\(^{0/0}\) ERT2Cre\(^{+}\) mice were induced by the indicated number of gavages with tamoxifen emulsion. Mice were sacrificed 4 weeks later for analysis of ARAP3 expression (arrow) in purified BMNs and total lung tissue by Western Blot; β-actin as loading control. A representative example is presented. (B, C) Cre was induced by repeat gavaging with tamoxifen of inducible Arap3 knock-out (-/-) or inducible Cre mice (+/+), and LPS ALI induced. Mice were intravenously administered fluorescently coupled anti-CD45 prior to lavaging of perfused lungs. Total and vessel-associated, CD45-labelled monocytes/macrophages obtained from lung digests are plotted. Each symbol represents one mouse; graphs combine data obtained from 2 separate experiments. All bar graphs show mean±SEM.